Evaluation of the Ocean Resources Enhancement and Hatchery Program

Submitted to:
Marine Region
California Department of Fish and Wildlife
Los Alamitos, California
Project no. P1470005

Submitted By:
California Sea Grant Extension Program
Scripps Institution of Oceanography
University of California, San Diego

12 December 2017

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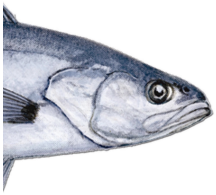

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Submitted by

Theresa Sinicrope Talley, Nina Venuti, Rick Starr
California Sea Grant at Scripps Institution of Oceanography
University of California, San Diego
La Jolla, CA 92093-0232

On behalf of the Science Advisory Committee

Christopher Myrick	Colorado State University, Committee Chair
Kenneth Cain	University of Idaho
Lorenz Hauser	University of Washington
Kenneth Leber	Mote Marine Laboratories
Martha Sutula	Southern California Coastal Water Research Project
Cassidy Teufel	California Coastal Commission
Chuck Valle	California Department of Fish and Wildlife
Robert Vega	Texas Parks and Wildlife
Dallas Weaver	Ocean Resources Enhancement Advisory Panel

Submitted to

Marine Region, California Department of Fish and Wildlife, Los Alamitos, CA

Date: 12 December 2017

With help from our Science Review Panel members John Gold, Kai Lorenzen, Kerry Naish, Penny Swanson, and Robin Waples

Recommended Citation

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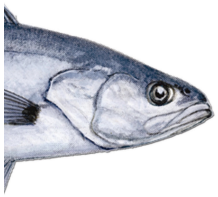

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Background

The Ocean Resources Enhancement and Hatchery Program (OREHP) was established by the California State Legislature in 1983 to conduct a program of basic and applied research on the artificial propagation, rearing and stocking of important marine fish species occurring in ocean waters off southern California (the OREHP's original legislative intent; FGC § 6592). Over the years, the Legislature has modified language describing the intent of the program; current legislative intent provides a focus on determining if hatchery-released fish can enhance stocks of wild species through increased hatchery production of fish, and the monitoring of fisheries to assess hatchery contributions (FGC § 6590). The ultimate goal of the OREHP legislation has been, however, "to enhance populations of marine finfish species important to California for their sport and commercial fishing value."[1]

The legislative intent was used to craft a "primary goal" for the OREHP, which is "to evaluate the economic and ecological feasibility of releasing hatchery-reared fish to restore depleted, native, marine fish populations to a higher, sustainable level."[1,2] The original objectives developed to achieve this OREHP primary goal were to:

1. Develop and implement hatchery operation and growout methods that provide a supply of healthy and vigorous fish.
2. Conduct the replenishment program in a manner that will avoid any significant environmental impacts resulting from operation of either the hatchery or pen rearing facilities.
3. Maintain and assess a broodstock management plan that results in progeny being released that have genotypic diversity very similar to that of the wild population.
4. Quantify contributions to the standing stock in definitive terms by tagging fish prior to release and assessing their survival in the field.
5. Continue to develop, evaluate, and refine hatchery operations to maximize the potential for achieving the goal of the program.
6. Develop quantitative measures of success.

The California Department of Fish and Wildlife (CDFW) administers the OREHP with the assistance of the 10-member Ocean Resources Enhancement Advisory Panel (OREAP). The program is primarily funded by revenue from the federal Sport Fish Restoration Act and sales of California Sport Fishing Ocean Enhancement Stamps. The primary hatchery facility at which OREHP activities take place is the Leon Raymond Hubbard, Jr. Marine Fish Hatchery in Carlsbad, California. Personnel from Hubbs-SeaWorld Research Institute (HSWRI) are contracted to operate the fish hatchery in Carlsbad. As part of their OREHP contractual obligations, HSWRI has developed the culture protocols required to raise White Seabass, and has conducted

[1] California Department of Fish and Game (CDFG). 2010. White Seabass Enhancement Plan. Prepared by: M. Fluharty, V. Frey, K. Johnson, T. Larinto, J. Mello, T. Moore, M. Okihiro, K. Ramey, P. Reilly, and V. Taylor. https://nrm.dfg.ca.gov/FileHandler.ashx?DocumentID=29458

[2] Drawbridge, M., and M. Okihiro. 2007. Comprehensive Hatchery Plan (CHP) for Operation of the Leon Raymond Hubbard, Jr. Marine Fish Hatchery in Carlsbad California (2nd ed.). Hubbs-SeaWorld Research Institute and California Department of Fish and Game. https://nrm.dfg.ca.gov/FileHandler.ashx?DocumentID=55041

research on culture protocols for other species, including California Halibut, Yellowtail, Giant Sea Bass and California Sheephead.

The OREHP is the longest-running marine fish stock enhancement pilot program in the United States. Since its creation in 1983, there have been no formal assessments of the program. In 2015, CDFW requested that California Sea Grant (CASG) coordinate a comprehensive review of the OREHP and its progress in achieving its goals and objectives. With guidance from CDFW, CASG created a 9-member Science Advisory Committee (SAC), comprised of scientists from around the country who were responsible for evaluating the program. The SAC, appointed by the CDFW Director, included members with demonstrated expertise in a wide variety of disciplines, including aquaculture, fish pathology, population dynamics, genetics, and water quality. Comprehensive and rigorous evaluations of marine enhancement programs are, in general, lacking, making this thorough and detailed evaluation one of the first of its kind.

Evaluation of the OREHP

From October 2015 through August 2017, the SAC conducted a review of OREHP hatchery and enhancement operations to assess the hatchery's functionality and efficiency, consider alternative hatchery uses, assess environmental impacts, document scientific accomplishments, assess economic costs and benefits, and evaluate the extent to which the OREHP has succeeded in enhancing wild White Seabass stocks. Details of the review are available in the full evaluation report entitled, "Evaluation of the Ocean Resources Enhancement and Hatchery Program (OREHP)." That full report, summarized here, details the SAC's evaluation process, which included the development of scientific review criteria, the summary and synthesis of all available OREHP data, and the identification of key findings, gaps in information, and recommendations for better meeting the program's objectives and goals.

Fulfillment of the ultimate goal of the OREHP: Enhancement of marine fish populations.

It is clear from the SAC review that the OREHP has met the original intent of the California State Legislature to conduct basic and applied research on the propagation, rearing, stocking, and distribution of an important marine fish, White Seabass (FGC § 6592). In 1983, little was known about the techniques needed to successfully spawn, rear, and release saltwater fishes. Since then, the OREHP has significantly contributed to the world's knowledge about marine enhancement science and techniques. Similarly, the OREHP has been consistent with the modified legislative intent of *determining* if hatchery released fish can enhance wild stocks (FGC § 6590); however it has shown that, at least for White Seabass, enhancement has not been effective to date, thereby falling short of the ultimate legislative goal.

An analysis conducted for this review of tag-recapture data generated by the OREHP between 2000 and 2011 indicated that the program has made a less than 1% contribution to enhancing the California White Seabass population and fishery due to high levels of mortality suffered by hatchery-reared White Seabass following release into the wild. According to the analysis, if mortality rates of released hatchery fish were reduced to equal those of wild White Seabass, then current stocking rates could result in a hatchery contribution of 18% instead of <1% of the total fishery catch. Therefore, in order to achieve the ultimate goal of fisheries enhancement,

the approaches and technologies developed for White Seabass would require further development aimed at reducing post-release mortality, including the related recommendations made throughout the evaluation report.

It should also be noted that, while the White Seabass stock was considered depleted when the OREHP was initiated and White Seabass was chosen as the program's focal species, the stock has since increased, likely due to a combination of high recruitment related to favorable environmental conditions and fisheries management measures (e.g., closure of the coastal gill net fishery).

Fulfillment of the OREHP objectives.
The SAC concluded that several OREHP objectives have been partially or fully met. The biggest achievements of the OREHP have been its contributions to research discoveries surrounding the biology and culture of all life stages of White Seabass (Objective 6). Other notable successes include the development of appropriate hatchery (Objective 1) and tagging methods (Objective 4) for White Seabass, and the constant improvements in hatchery practices that have been made over the years (Objectives 1 and 5). Through its program of releases of tagged fish, and fisheries independent and dependent monitoring of released fish, the OREHP has successfully collected enough data to evaluate the post-release survival of hatchery fish and the contribution of hatchery fish to the White Seabass fishery (Objective 4), both of which have been determined to be low. Substantial outreach regarding White Seabass life history and culture has been conducted to the sportfishing community, K-12 students, and members of the interested public (Objectives 1 and 6). Further, there has been no evidence that the program has caused any adverse environmental impacts at the production levels to date (Objective 2).

Other OREHP objectives, or aspects of objectives, have not been achieved. The analysis of tag-recapture data revealed that hatchery fish suffer high mortality rates within the first few months following release (Objective 1) that likely limit contributions to fishery stock. Low post-release survival and fishery contribution rates likely stem from some combination of fish health and fitness challenges (e.g., effects of unresolved gas supersaturation issues, inconsistency in diagnosis and response to health findings, domestication effects; Objectives 1 and 4), and uncertainty about optimal release strategies (Objectives 1 and 4). While the maintenance of genotypic diversity (Objective 3) has not been sufficiently addressed throughout the program, the lack of significant hatchery contribution to the wild population has prevented any adverse genetic effects to the wild population so far.

Budget conclusions.
The operating budget needed to achieve all aspects of the OREHP objectives exceeds the base funding level of approximately $1.6 million per year that has been available for the program. With inadequate funding, the OREHP objectives suffer. Restricted funding has reduced or limited several OREHP capabilities, including the ability to exchange broodstock at rates needed to ensure adequate genetic diversity in released fish (Objectives 1 and 3), provide stricter oversight of growout facilities (Objective 1), address reoccurring gas supersaturation issues (Objective 1), consistently and extensively perform and address challenges related to the

recapture surveys (Objective 4), and perform fisheries enhancement modeling (Objective 4). Limited resources have also likely prevented the initiation of a genetic monitoring program (Objective 3) and (socio-) economic assessments (Objective 5 and 6). It is important to note that HSWRI has contributed in excess of $400,000 annually to meet operational expenses that are at least in part related to the OREHP and has sought grants and contributions from a mix of private and government sources to make infrastructure repairs and improvements to the hatchery facility; HSWRI has also brought in external funds to cover research and outreach efforts that are related to, but not part of, the OREHP.

Program-level observations and recommendations

Although the SAC did not conduct a comprehensive review of OREHP management processes, it recommended that the organizational structure of those groups overseeing the OREHP be updated to better achieve OREHP goals and objectives. The SAC also noted several program-level weaknesses, and made recommendations for strengthening the OREHP.

Need to strengthen and update organizational structure.

The ultimate authority for many programmatic decisions within the OREHP was unclear. It is necessary to clarify, for example, who has the authority to make decisions relating to research priorities and issues that influence or put hatchery operations and scientific research into conflict with one another. Part of this uncertainty is caused by the OREHP's dual focus on production and research, two activities which can be very different and for which there are limited resources. Additional uncertainty may be due to the change in the internal interpretation and communication of OREHP intent, goals and objectives through time, and in the absence of periodic program reviews.

The SAC noted that the program's advisory panel (OREAP) has not been as effective or valuable as it could be, and that CDFW should reconsider how to best utilize an advisory panel. The current OREAP does not have the representation by the groups detailed in the original legislation, as some of these groups no longer exist or have changed focus. CDFW should restructure and reform the OREAP, and, in addition, form an independent science and technical advisory group with expertise in hatchery science (and associated issues, such as fish health), population dynamics, release and recapture strategy optimization, and genetics to help develop and evaluate quantitative criteria, benchmarks, and timelines to be used in the future evaluation of the program.

Need for external, independent guidance.

Fish health guidance. The SAC was greatly concerned with the differences in opinions between CDFW and HSWRI pathologists regarding the definition of malformed, or deformed fish, and the implications of the range of morphological variability found in hatchery fish on vigor. Currently, these differences in opinions cause a large public relations problem and inhibit smooth operations at the Carlsbad hatchery, thereby resulting in reduced juvenile production due to diversion of resources and delays in decisions about diagnoses and appropriate responses. Further, differences in opinions, and therefore the outcome of diagnoses and actions taken, may ultimately affect release numbers and post-release survival. Although risk of introduction

of disease or unwanted genetic characteristics to the wild fish population is low due to the low likelihood that these factors are linked with malformations, it is critical to have consistent decision-making criteria and set appropriate policy for dealing with malformed fish in order for the program to meet its objective of producing healthy and vigorous fish. The SAC strongly recommended that CDFW and HSWRI engage an independent panel of experts that would be charged with the following:

1. Compare the morphological diversity of wild fish with that of hatchery fish.
2. Determine which unique hatchery morphologies pose a genetic or other biological threat to wild populations.
3. Determine which morphologies cause loss in post-release fitness.
4. Develop a set of criteria and protocols for identifying and responding to fish that have unacceptable types and/or levels of deformity that both CDFW and HSWRI staff agree upon.
5. Develop approaches that minimize frequencies and levels of deformities.

Science and technical advice. The SAC developed assessment topics within each OREHP objective to help in determining the extent to which each objective has successfully been met. Having a more clearly defined set of assessment metrics in place, such as those suggested in Chapter 6 of the full evaluation report, would allow for more efficient, and maybe more frequent, assessments of the program, and would provide clearer guidance to OREHP staff and researchers. Although the assessment topics in the evaluation report can currently be used to guide future assessments, more focused, clear, feasible, and occasionally updated metrics agreed upon by CDFW and OREHP contractors are still needed to identify future successes related to stock enhancement. Again, the SAC strongly recommended that CDFW periodically enlist an independent external group of science and technical experts to work with CDFW and stakeholders to develop (and later help to evaluate) a set of quantitative criteria, benchmarks, and timelines for each of the established OREHP objectives.

Need to strengthen public communication and transparency.
HSWRI has taken the lead on public outreach, stakeholder engagement, and public relations for the OREHP without provision of communications and development professionals, or adequate resources to support this task. This responsibility has taxed HSWRI's already limited resources for the OREHP and added the stress of public scrutiny. The SAC occasionally had to dig deeply to find information needed to assess the status of various aspects of the OREHP and noticed the presence of potentially confusing statistics about various aspects of the program in reports and non-peer reviewed publications (e.g., newsletters). The SAC recommended that HSWRI and CDFW make greater efforts to keep information about the OREHP openly available to each other and to the public, and to improve consistency and transparency of outcomes and incidences, particularly for issues of public interest (e.g., contribution of the program to wild stocks, recapture rates of tagged fish in gill nets, incidences of disease and deformity, occasional accidents or die-offs, costs and benefits of the program, etc.). Improved transparency may include the development of a process that allows communication with a broad range of stakeholders, including those not already associated with the program, to

collect input regarding priorities and development of the program. Further, the SAC recommended that CDFW assist more with this duty, or find and support knowledgeable public communications professionals to help.

The future of the OREHP: Review and reform

The SAC's evaluation of the OREHP objectives, goals, intent and budget revealed that it is timely for the relevant authorities and stakeholders to review the overall focus and strategy for the OREHP in terms of focal species and stocks, and the potential role of enhancement as an additional tool used in the management of those fisheries. The program evaluation has shown that, while the research and technology development objectives of the OREHP have largely been met, the program is not currently in a position to substantially enhance the White Seabass fishery due to a variety of factors.

Post-release survival and therefore contributions to the wild population are low. Further, the California White Seabass stock, which was depleted when the OREHP was established and White Seabass was chosen as its focal species, has since reached a higher level of abundance. These factors, together with changes in the status and management of other California stocks, and increased understanding of the potentials and limitations of stock enhancement to contribute to fisheries management outcomes, suggest that it is timely to reassess the program's utility, and to review and reform the OREHP's priorities and the approaches used to fulfill each of the OREHP objectives.

The SAC recommended the following steps for assessing the future of the OREHP (Fig. 1), noting that these recommendations were made without consideration of cost and thus would need to be evaluated with respect to program priorities and levels of available funding.

A science-based and participatory public process.

The future of the OREHP should be determined through a process that is both science-based and participatory with respect to the program's stakeholders (Fig. 1). Overall guidance for such a process can be found in the Updated Responsible Approach to Marine Fisheries Enhancement[3] and in the Hatchery Reform processes implemented for several salmon hatchery programs in the Pacific Northwest[4]. Scientific methods, such as fisheries models used to assess the potential effectiveness of stock enhancement and other fisheries management measures in achieving desired fisheries management outcomes, enable a systematic approach to the planning of enhancement programs. Stakeholders, principally recreational and commercial fishermen, have played a major role in the operation and funding (through license fees) of the OREHP. It is therefore imperative to involve stakeholders systematically and constructively, and to use current scientific information in making the following decisions about the program's future direction.

[3] Lorenzen, K., K. M. Leber, and H. L. Blankenship. 2010. Responsible approach to marine stock enhancement: An update. 2010. *Reviews in Fisheries Science* 18:189-210.

[4] Northwest Fisheries Science Center. Hatchery Reform Science Program. www.nwfsc.noaa.gov/research/divisions/efs/hatchery/index.cfm

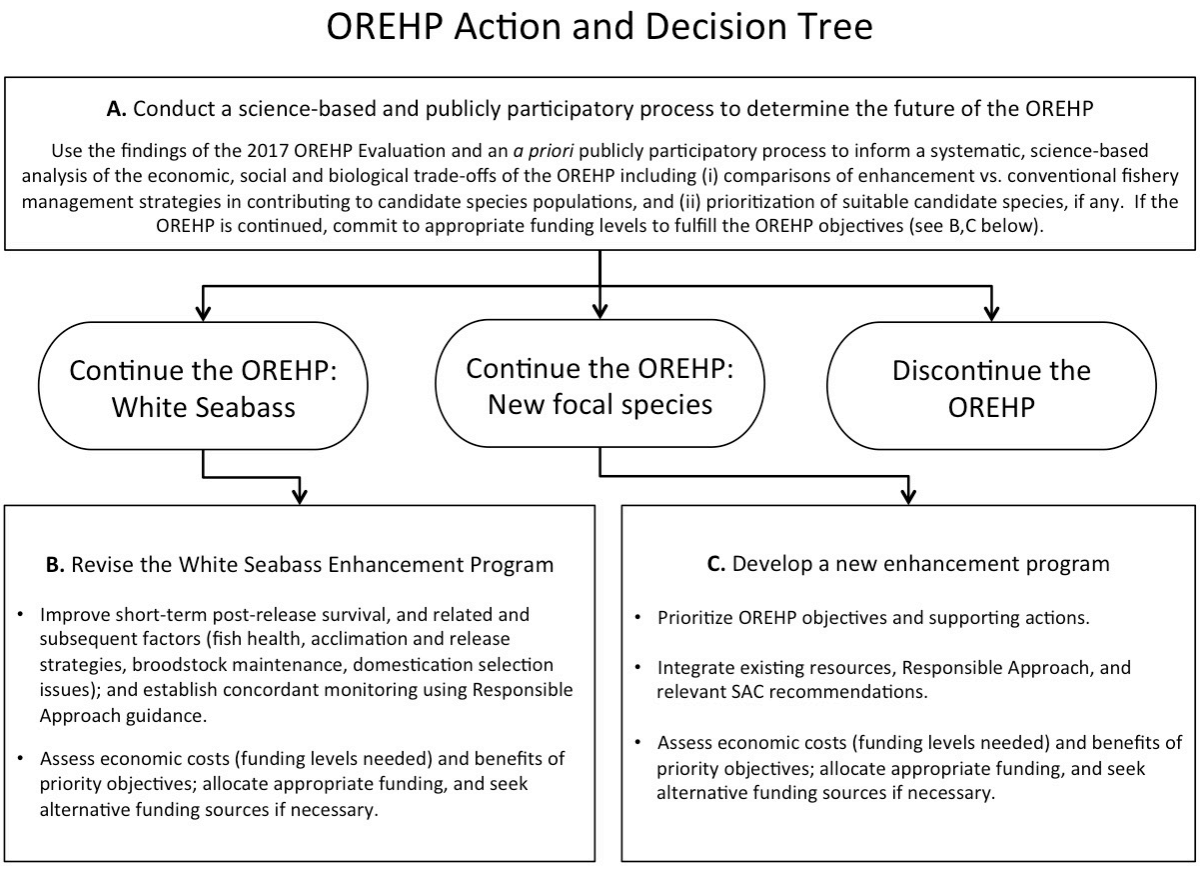

Fig. 1. Flow chart of decisions and actions recommended by the Ocean Resources Enhancement and Hatchery Program (OREHP) Evaluation Science Advisory Committee (SAC) to California Department of Fish and Wildlife (CDFW) to aid in decisions about the future of the OREHP.

Assess the potential role of enhancement in California fisheries management.

The list of candidate species identified by CDFW and HSWRI, including White Seabass, should be honed using analysis of the biological, economic and social costs and benefits of the OREHP as compared to relying solely on (non-OREHP) fishery management strategies (e.g., updating catch quotas and/or size limits) for White Seabass and the other candidate species identified by CDFW and HSWRI (Fig. 1A). If the analysis indicates that conventional fishery management strategies alone may be sufficient for the conservation and management of most candidate species, then discontinuation of the OREHP should be considered as one option, if legislatively feasible. If some stocks are deemed to be extremely low (i.e., severely depleted), and/or if responses to conventional fishery management actions alone are predicted to be ineffective, then further development or modification of the enhancement program should be considered, and funding adjusted to enable the OREHP to meet its objectives. The candidate species lists put forward by CDFW and HSWRI were generally supported by the SAC, with California Halibut of particular interest for inclusion in this initial assessment given the available information on its biology, ecology, and culture practices, its depressed populations, and the high recreational and commercial fishing demand.

Prioritize candidate focal species by enhancement potential.
If the initial assessment of the value of enhancement in relation to other fishery management strategies indicates that the OREHP could likely contribute to some of the candidate species, then the SAC recommended that those species remaining on the candidate list be prioritized. Specifically, the SAC recommended an *a priori* systematic and quantitative assessment of each candidate species (Fig. 1A) similar to the assessment developed by HSWRI[5] but with input from a broader range of stakeholders, inclusion of economic and social costs and benefits, more consideration of fit with fisheries management strategies, and conducted in cooperation with an independent advisory committee. Criteria should include depressed stock numbers (e.g., consistently low enough to offset genetic risks associated with enhancement), ease of culture, life history that is amendable to rearing, tracking and enhancement (e.g., relatively high growth rates, not highly dispersive), geographic range that can be feasibly sampled (e.g., most common in U.S. waters), availability of existing biological information, and high demand and value within commercial and recreational fishing industries and throughout the food supply chain. Clearly, the findings of the economic, social and ecological (e.g., environmental, genetic and population-level) trade-offs analyses used to narrow the candidate species list may be used to inform this process.

The challenges associated with each candidate species should be assessed and applicable recommendations from the OREHP evaluation report should be used. For example, a fish with a range that extends into Mexico will require collaborative efforts for population/fishery assessments, and relatively slow growth rates will still require decisions surrounding size at release trade-offs. New challenges should also be assessed; for example, the demersal California Halibut would require different tank designs than those established for the pelagic White Seabass, and as such would require a significant capital contribution to reconfigure hatchery systems.

If a change of focal species is decided, White Seabass should be phased out by ceasing breeding efforts while completing the rearing and release of existing early life stages. The rate of phasing, however, may depend upon space, resources (including availability of new species broodstock), and other logistical considerations. An independent advisory panel should be used for guidance on planning of the phasing and/or the development and initiation of a new enhancement program (Fig. 1C).

White Seabass Enhancement: Focus on reducing post-release mortality.
The results of the OREHP evaluation stress the importance of minimizing post-release mortality of hatchery White Seabass to increase the potential of the enhancement program. The same need would likely exist for new focal species that might be chosen for enhancement. Greater emphasis should therefore be placed within the OREHP on research of factors that affect post-release mortality, and on husbandry and release strategies that minimize this mortality (Fig. 1B). This focus may require increased funding to the OREHP in order to fulfill a commitment to

[5] MacNamara, R., M. Shane, and M. Drawbridge. 2016b. A species selection framework for marine finfish stock enhancement in Southern California. Hubbs-SeaWorld Research Institute, San Diego, California.

reducing short-term (e.g., 6 month) post-release mortality rates. Increasing production to compensate for high mortality rates is not recommended because of the increased expenses, increased infrastructural and resource needs (e.g., staff time, supplies), and increased risk of fish health issues that would be associated with higher production rates.

In particular, to improve survival and stock contribution rates, greater attention should be given to:

1. Domestication issues (Objective 1).
2. Resolution of fish health challenges (e.g., resolving gas suppersaturation and its health effects, understanding effects of deformity types and severity on fitness, consistency in diagnosis and response to health findings; Objective 1).
3. Continued improvements to placement and oversight of growout facilities (Objective 1).
4. More research focused on optimizing release strategies such as timing, size, location and magnitude of releases (Objectives 1 and 4).
5. More effort on post-release monitoring needed to optimize release strategies and estimate recapture rates (Objective 4).
6. Greater integration with fishery management to understand relationships between enhancement efforts and wild populations/fisheries (Objective 4).

If White Seabass production is increased or if there is a change in focal species, however, potential environmental impacts associated with these changes should be reassessed (Objective 2), and monitoring efforts should be modified appropriately to account for higher production levels and/or different impacts depending upon system changes (all Objectives).

If survival rates increase, improved genetic practices and monitoring should also be implemented in order to address the potential genetic effects associated with enhancement, which to date have not been an issue because of the extremely small possibility that a hatchery fish will survive to spawn with wild fish. If higher survival rates become the focus, then the broodstock management plan should be reassessed and reworked to include more frequent rotation of wild-caught broodstock, more emphasis should be placed on reducing domestication selection and increasing the proportion of spawns that go on to be reared, and monitoring of family contributions throughout the rearing process should take place to maintain the desired levels of genetic diversity and limit domestication selection (Objectives 1 and 3).

Further, a framework for conducting, evaluating and refining the enhancement program (Objectives 5 and 6) should be developed and used, regardless of the focal species selected. For example, the Updated Responsible Approach to Marine Stock Enhancement provides guidance on goal setting and evaluation, research and technology development, and adaptive management strategies (Objectives 5 and 6). In particular, the SAC recommended that an economic analysis be performed for whichever program approaches are selected in order to ensure that the financial benefits of the program outweigh potential costs, and to inform future assessments (Objectives 5 and 6). More attention should also be placed on adaptive

management. The OREHP has many hatchery and growout protocols and plans in place, but data collection, record keeping, and reporting are not currently structured to allow formal assessment of whether protocols are being followed, and how findings and changes are contributing to protocol updates. For example, release strategies need to be optimized, and more formal data collections, record keeping and reporting of results (i.e., adaptive management experiments) can inform the evaluation of model assumptions about survival and the effects of fish size at release, release (micro)habitat, season, acclimation and acclimatization, and release magnitude. Adaptive management would also be useful for addressing many of the other challenges identified.

Address the economics of the program.

Assess the economic benefit of the OREHP. Given that funds for the OREHP are largely public and much of the benefit of the program may be social, an economic (and social) analysis would make program expenditures more defensible, help to indicate social and economic strengths and weaknesses of the program, and may provide insights into stakeholder priorities. Improved economic awareness and efficiency is important because the accomplishment of priority objectives, and the breadth and depth of actions needed to fulfill those objectives, will be dependent upon available funds (Fig. 1B,C). The extent that recommendations made by the SAC through this review can be implemented will also be dependent upon funding levels. For example, if OREHP funding remains static, it may be necessary to narrow the focus of the program to solving the challenges of enhancement that were identified as highest priority by the SAC (e.g., reducing post-release mortality), but if funding is increased, then there may be opportunity to also test and address the challenges of a program that contributes more significantly to wild populations (e.g., developing and initiating genetic monitoring). However, resolution of all identified challenges seems beyond a relatively small increase in funding and may require alternative funding sources, such as private organizations.

Need to expand public-private partnerships. There is a need to expand public-private partnerships such as those established already within the OREHP. HSWRI, the primary contractor for the OREHP, has forged partnerships with private groups, such as recreational fishing groups and private foundations, which have provided a substantial supplement of non-OREHP funds and in-kind resources (e.g., volunteer time, boat time, supplies) to operate the hatchery and growout facilities. Because of the infusion of supplemental funding from HSWRI, the SAC considered the potential for conflict of interest, and concluded that the State has benefited from the private funding, and that all information has been publically shared so that there is no conflict of interest among partners associated with the OREHP. If the OREHP continues, the SAC suggested that CDFW consider expanding the public-private partnership concept to bring in additional partners (and funds), such as other foundations and commercial fishing communities, to expand the capabilities of the OREHP, which may allow for the implementation of recommendations made by the SAC for fulfilling each OREHP objective.

Summary

The OREHP has made groundbreaking progress in developing hatchery rearing and enhancement practices and systems for marine species, and in related scientific discoveries.

The program is not, however, currently in a position to substantially increase the abundance of White Seabass, with hatchery fish released by the OREHP over the last 30 years having contributed less than 1% to the wild White Seabass fishery. Further, the economic (and social) benefits of the program are uncertain because of a lack of assessment in these areas. While decisions about the future direction of the program are ultimately up to CDFW and the California Legislature, this evaluation recommends a science-based and stakeholder participatory decision-making process that will assess the opportunity costs of the program with White Seabass and other candidate species as compared to relying solely on conventional (non-OREHP) fishery management strategies. This evaluation provides objective-specific and program-wide recommendations for use should the program be continued. In general, the breadth and depth of efforts undertaken within the OREHP, as well as success in meeting defined goals, will be dependent upon the following factors: levels of funding; internal organizational cooperation and support; evidence of broader public benefit and support; improved assessment strategies, including stronger adaptive management and more frequent assessments using well defined ecological and economic metrics; and unified, transparent public communications in order to clearly demonstrate the values of the program to commercial and recreational fisheries and society.

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Table of Contents

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Glossary of Acronyms and Abbreviations

Note: this list includes only those acronyms and abbreviations used more than once in the report.

BOD	Biological Oxygen Demand
BMI	Benthic Macroinvertebrate
BMP	Best Management Practice
CASG	California Sea Grant
CCC	California Coastal Commission
CDFG	California Department of Fish and Game
CDFW	California Department of Fish and Wildlife
CICESE	Centro de Investigación Científica y de Educación Superior de Ensanada
CPFV	Commercial Passenger Fishing Vessel
CPUE	Catch Per Unit Effort
CSUN	California State University, Northridge
Cu	Copper
CWT	Coded Wire Tag
DO	Dissolved Oxygen
dph	days post hatch
EPA	Environmental Protection Agency
fe	female equivalent
FMSY	Fishing Mortality at which Maximum Sustainable Yield is achieved
FY	Fiscal Year
GFC	Growout Facility Coordinator
HACCP	Hazard Analysis and Critical Control Points
HSWRI	Hubbs-SeaWorld Research Institute
J1	Juvenile system 1
J2	Juvenile system 2
LARWQCB	Los Angeles Regional Water Quality Control Board
MLLW	Mean Lower Low Water
MSY	Maximum Sustainable Yield
N	adult census population size
N_b	effective number of breeders of a spawning event, season, or year
N_e	effective population size
NOAA	National Oceanic and Atmospheric Administration
NPDES	National Pollution Discharge Elimination System
OREAP	Ocean Resources Enhancement Advisory Panel
OREHP	Ocean Resources Enhancement and Hatchery Program
ORP	Oxidation Reduction Potential
OY	Optimal Yield
PEL	Probable Effects Level
PFMC	Pacific Fishery Management Council
PIER	Pfleger Institute of Environment Research
PIT	Passive Integrated Transponder
Q1	Quarantine tank 1 (for new broodstock)

Q2	Quarantine tank 2 (for new broodstock)
QA/QC	Quality Assessment/Quality Control
RFP	Request for Proposals
SAC	Science Advisory Committee
SDGE	San Diego Gas & Electric Company
SDSU	San Diego State University
SDRWQCB	San Diego Regional Water Quality Control Board
SFRA	Sport Fish Restoration Act
SL	Standard Length
SONGS	San Onofre Nuclear Generating Station
SOP	Standard Operating Procedure
SPI	Sediment Profile Imagery
TEL	Threshold Effects Level
TL	Total Length
TN	Total Nitrogen
TP	Total Phosphorus
TSS	Total Suspended Solids
TVS	Total Volatile Solids
VHSV	Viral Hemorrhagic Septicemia Virus
VNNV	Viral Nervous Necrosis Virus
VRG	Vantuna Research Group
WSFMP	White Seabass Fishery Management Plan
Zn	Zinc

Introduction

Background

The Ocean Resources Enhancement and Hatchery Program (OREHP) was established by the California State Legislature in 1983 to conduct a program of basic and applied research into the artificial propagation, rearing and stocking of important marine fish[6] species occurring in ocean waters off southern California (FGC § 6590, 6592). The program was created because (1) there had been declines in many desirable fish species off of southern California that adversely impacted commercial and recreational fisheries; (2) research and development of the techniques and equipment surrounding propagation, rearing and stocking had been reasonably well developed and there was a need to determine whether these could be applied to enhancing the depressed wild populations; (3) there was a viable funding mechanism supported by those who stood to gain by a resurgence of the depressed fish populations; and (4) such an effort was consistent with CDFW's marine resource management, administrative and policy review responsibilities (FGC § 6590).

Over the years, the Legislature has amended the intent language of the program, with current legislation calling for a focus on determining if hatchery released fish can artificially enhance certain stocks of desirable species through increased hatchery production of fish and increased monitoring of fisheries to assess the hatchery contribution (FGC § 6590). The ultimate legislative intent is "to enhance populations of marine finfish species important to California for their sport and commercial fishing value" (White Seabass Enhancement Plan 2010).

The "primary goal" of the OREHP is "to evaluate the economic and ecological feasibility of releasing hatchery-reared fish to restore depleted, native, marine fish populations to a higher, sustainable level" (Comprehensive Hatchery Plan 2007, White Seabass Enhancement Plan 2010). Six objectives were developed to achieve this goal:

1. Develop and implement hatchery operation and growout methods that provide a supply of healthy and vigorous fish.
2. Conduct the replenishment program in a manner that will avoid any significant environmental impacts resulting from operation of either the hatchery or pen rearing facilities.
3. Maintain and assess a broodstock management plan that results in progeny being released that have genotypic diversity very similar to that of the wild population.
4. Quantify contributions to the standing stock in definitive terms by tagging fish prior to release and assessing their survival in the field.
5. Continue to develop, evaluate, and refine hatchery operations to maximize the potential for achieving the goal of the program.
6. Develop quantitative measures of success.

The California Department of Fish and Wildlife (CDFW) administers the OREHP, with the assistance of the 10-member Ocean Resources Enhancement Advisory Panel (OREAP) consisting

[6]Fish and Game Code defines "fish" as "wild fish, mollusks, crustaceans, invertebrates, or amphibians, including any part, spawn, or ova thereof."

of academic and management agency scientists, representatives of commercial and recreational fishing groups, and members of the aquaculture industry. The OREHP is funded through the sale of State Ocean Enhancement Stamps on sport and commercial fishing licenses, and the Federal Sportfish Restoration Act (SFRA). CDFW's main contractor, Hubbs-SeaWorld Research Institute (HSWRI), operates the Leon Raymond Hubbard, Jr. Marine Fish Hatchery in Carlsbad, California, which raises White Seabass (*Atractoscion nobilis*). As part of their OREHP contractual obligations, HSWRI has developed the culture protocols required for the program, and the assessment techniques to help evaluate the impact of the hatchery-reared fish on recreational and commercial fisheries. Research initially focused on California Halibut (*Paralichthys californicus*) and White Seabass; since 1990 however, the OREHP has focused on White Seabass because of the depressed condition of the stock, its higher value to both the recreational and commercial fisheries, and the availability of aquaculture information on related species.

As of 2015, there had been no formal assessments of the degree to which the goals and objectives of the OREHP and, in particular, the White Seabass Enhancement Plan, have been achieved. The goal of the evaluation was to conduct a comprehensive scientific review to assess the success of the OREHP in meeting its goals and six objectives. The evaluation includes a review of the functionality and efficiency of the hatchery and growout facility operations, consideration of alternative hatchery uses, documentation of OREHP scientific accomplishments, an assessment of available information on economic costs and benefits, and an evaluation of the extent to which the OREHP has succeeded in enhancing the wild White Seabass stock.

California Sea Grant (CASG) coordinated the OREHP evaluation process, including synthesizing and summarizing all available OREHP data, and overseeing the production of the evaluation report. CASG coordinated the establishment of a 9-member Science Advisory Committee (SAC), as described in the White Seabass Enhancement Plan (2010), that was tasked with developing science-based criteria (based on the OREHP's goals and objectives) that were used to assess the progress of the program. The SAC members, appointed by the CDFW Director, included one member with demonstrated expertise in each of these topic areas and/or an affiliation with associated OREHP entities:

1. Fish genetics
2. Fish pathology
3. Marine aquaculture
4. Population biology or dynamics
5. Benthic and/or water quality
6. Croaker (White Seabass) culture research
7. California Coastal Commission
8. Ocean Resources Enhancement Advisory Panel (OREAP; nominated by the OREAP)
9. California Department of Fish and Wildlife

Through the conduit of CASG, the SAC provided CDFW with an interpretation of the degree to which the White Seabass project has met the OREHP goals and objectives, and recommendations

about continuance of the White Seabass enhancement project, if it should be continued, and alternative or expanded uses of the OREHP resources.

White Seabass Enhancement Program Overview

HSWRI has been responsible for conducting, coordinating and/or overseeing the procedures and research related to the hatchery and growout stages of the White Seabass enhancement program, and they lead one of the two teams of contractors (the other led by L. Allen of California State University, Northridge) who have been responsible for the recapture of tagged, released hatchery fish.

Hatchery Stages

The Leon Raymond Hubbard, Jr. Marine Fish Hatchery operated by HSWRI has seven separate (compartmentalized) aquaculture systems: larval food production, broodstock, egg incubation, juvenile 1 (J1), juvenile 2 (J2), raceway culture, and an experimental system used for research trials. Each system uses a dedicated water system to reduce the likelihood of the spread of infection and disease. All of the systems operate on a recirculating water basis, except for the flow-through raceway culture. In the recirculating systems, water flows through a series of filters (bead, floating media, and/or sand), foam fractionators, and UV sterilizers. Filtration regimes differ with each system and are based on the requirements of each life stage. All "make-up" water, or water used to replace losses during the recirculation process, is disinfected using an ozone system. The raceway culture uses a flow-through water system with sand-filtered, oxygenated water.

Broodstock capture and husbandry

White Seabass broodstock are collected by hook and line from Point Conception, California to the U.S. border with Baja California, Mexico using cooperative collecting trips with HSWRI staff and volunteers (White Seabass Enhancement Plan 2010). Broodstock are kept in four separate, temperature and photoperiod-controlled pools (6.1 m diameter X 3.5 m tall tanks) at the Leon Raymond Hubbard, Jr. Hatchery in Carlsbad, California; surplus broodstock are held in HSWRI's Santa Catalina Harbor net pen, or at growout facilities (White Seabass Enhancement Plan 2010, Broodstock Photoperiod Control (Day Length Timers) SOP 2015). Broodfish are quarantined for a minimum of 45 days and are individually identified with PIT tags before entering the hatchery pools (Broodstock Transfer and Tagging SOP 2015, New Fish Acquisition Quarantine SOP 2016, PIT Tagging Procedure for Newly Acquired Broodstock SOP 2016, White Seabass Enhancement Plan 2010).

Egg, Larval, and Juvenile production at the Hatchery

Spawning and incubation. White Seabass are broadcast spawners with external fertilization occurring in spawning aggregations, where the males and females remain in close proximity to each other during spawning. Spawning of male and female broodstock is induced using a temperature and light regime that simulates spring and summer conditions. The goal is to use eggs from 28-32 broodstock spawns per year of at least 2 million eggs. At -2 dph (days post hatch), fertilized eggs are collected using a beaker with a fine mesh bottom, sterilized, and placed

in one of 12 incubation tanks, each of which holds roughly 1,760 liters of water (Egg Collection and Setup SOP 2016, Spawn Harvest and Egg Disinfection SOP 2016, Egg Data Collection SOP 2016, Broodstock Spawn Harvest (Setup or dump) SOP 2015, [Inc] System Components and Mechanical Operation SOP 2016). A small sample of eggs is collected for data purposes, and used to estimate the number of eggs per milliliter (ml) for that crop (Egg Data Collection SOP 2016); it was previously estimated that there were ≈585 eggs per ml (White Seabass Enhancement Plan 2010). Eggs hatch at 48 hours (0 dph), at which point they are transferred to another incubation tank to reduce the amount of bacteria they encounter (Larval Transfers SOP 2016). They continue to be transferred between tanks every three to four days after this, until about 14 dph (Larval Transfers SOP 2016). Larvae begin feeding at 4 days post hatch (Larvae Feeding Schedule (0-21 DPH) SOP 2016). They are fed four times per day with rotifers or 1st instar *Artemia* (brine shrimp) nauplii and nutrient enriched 2nd instar *Artemia* (Feeding Larvae with Live Foods SOP 2015, Pickup and Cold Storage of Rotifers SOP 2015, White Seabass Enhancement Plan 2010).

Juvenile systems. Around 21 dph, larvae are transferred by gravity feed to one of six 7,000 liter pools that make up the first juvenile system (J1) (White Seabass Enhancement Plan 2010, Incubator to J1 Transfers (21 DPH) SOP 2016). At about 30 dph, juveniles begin to be weaned off of live feed and are fed dry pellets (Weaning Larvae SOP 2016), and at about 50 dph, they are transferred to one of four 7,000 liter pools that make up the second juvenile system (J2) (Annual Report 07-08, White Seabass Enhancement Plan 2010). Here, fish are given a dry pellet feed and are slowly acclimated to lower water temperatures (from 23^{o}C to the ambient temperatures of the raceways). During warm months, this can mean holding fish until 80-90 dph (≈20 g), while during colder months fish are held longer (120 dph, ≈40 g) (White Seabass Enhancement Plan 2010).

Raceways. Juveniles are then transferred to one of six 25 m^3 concrete, flow-through raceways, or to the R1_RAS (recirculating aquaculture system) tank, which are separated from the main hatchery and surrounded by chain link fencing and shade cloth. Fish are fed the same dry pellet diet as juveniles in the J2 system, and remain in the raceways until they are 91 to 150 dph. The fish may then be brought to one of the ten currently functioning volunteer-operated growout facilities, or, if more than 20 cm long and/or growout facilities are full, the fish may be released directly from the Hatchery (Growout Procedures Manual 2007). Before direct release or transport to growout facilities, every fish is tagged in the left cheek muscle with a uniquely numbered Coded Wire Tag (CWT) (White Seabass Enhancement Plan 2010).

Transport and Growout

Fish are transported using different transport methods (e.g., vehicles or vessels with tanks) depending upon the number of fish being transported and the final location (release site or growout facility). The most commonly used transport tanks are 1,500 liter marine grade aluminum tanks fitted with independent aeration systems (White Seabass Enhancement Plan 2010).

Throughout the program, there have been between 10 to 16 growout facilities in use, most of which are owned and operated by volunteers associated with angler groups and non-profit

organizations. The exceptions are two growout facilities (Agua Hedionda and the larger of two growout facilities based at Catalina Island) that are owned and operated by HSWRI. The facilities are located throughout Southern California from San Diego Bay to Santa Barbara, and offshore of the mainland at Catalina Island. All facilities except one are located in bays and hold the fish in pens or raceways that are fed by ambient seawater. The one non-waterfront facility (King Harbor) holds fish in above-ground pools. Juveniles are held anywhere from 2-6 months, depending upon growth rates, season, conditions and logistics at time of release. The goal is to release fish at roughly 20 cm in length (White Seabass Enhancement Plan 2010).

Enhancement assessments: Tagged fish surveys

From 1988 to 2008, and again from 2012 to the present, OREHP-contracted researchers from local universities and HSWRI have conducted standardized gill net surveys to capture 1-4 yr-old juveniles in shallow waters to assess the contribution of hatchery-raised fish to the wild population. In 2001, HSWRI began collecting adult White Seabass heads from recreational and commercial fishermen, scanning them to determine whether they bore CWTs indicating that they were raised at the hatchery (Comprehensive Hatchery Plan 2007). The information collected from the juvenile and adult sampling programs is not only valuable to the OREHP, but also to other researchers (e.g., White Seabass Stock Assessment scientists; Valero and Waterhouse 2016) to estimate growth rates, determine patterns of migration, and estimate the contribution of the program to wild populations (White Seabass Enhancement Plan 2010). Among the most valuable and versatile uses of aquaculture in fisheries management is the utilization of hatchery fish to conduct manipulative field investigations of critical ecological uncertainties to gain a better understanding of unresolved questions in fisheries ecology (C.J. Walters, pers. comm.)

Milestones

October 1986	The first experimental release of more than 2,000 juvenile White Seabass took place in Mission Bay (San Diego, California). Fish were propagated and raised at HSWRI's Mission Bay laboratory.
March 1992	The first legal-sized, oxytetracycline-marked, hatchery-raised White Seabass was recaptured.
October 1995	The marine fish hatchery became operational, built on land donated by San Diego Gas & Electric Company (SDGE) on Agua Hedionda Lagoon in Carlsbad, CA; and funded as an environmental mitigation measure for the San Onofre Nuclear Generating Station (SONGS), owners of which include Southern California Edison, SDGE, and the cities of Anaheim and Riverside. Contributions for the construction of the hatchery also came from private, corporate, and foundation donors.
June 1999	The first legal-sized, coded-wire tagged, hatchery-raised White Seabass was recaptured.
2001	The first year more than 100,000 White Seabass were released in southern California waters.
October 2004	The one-millionth White Seabass was released.
June 2007	Oldest adult fish recovery (13.3 yr); The fish was released off Santa Barbara, CA in 1994 and recovered near Ventura, CA.
June 2008	One-hundredth legal-sized hatchery-raised White Seabass recaptured.
September 2010	A tagged fish was recovered from Monterey, CA that had been released at Dana Pt. in August 2000.
August 2013	A total of 2 million fish had been released since the beginning of the OREHP.
December 2016	To date, 199 adult and 1,772 juvenile White Seabass have been recaptured.

Roadmap of the OREHP Evaluation Report

This OREHP evaluation report uses the six original objectives of the OREHP (Table 1) as a framework for evaluation so that each of the first six chapters of this report corresponds with an objective. Within each chapter, specific sub-topics were developed for use as evaluation criteria by the independent Science Advisory Committee, and modified based on usefulness and feasibility by CDFW and HSWRI, to provide a tractable means of assessing each objective. For each evaluation criterion, three areas were addressed: 1. *Key Findings*, which include the main biological, economical and/or regulatory findings relevant to each criterion, 2. *Data and Information Gaps* that are ideally needed to fully assess the criterion, and 3. *Recommendations* for better meeting the program objective. A seventh Chapter presents broad, program-wide findings and recommendations including an assessment of the extent to which the program has fulfilled the legislative intent and stated goal. Recommendations are made without consideration of whether funding is or would be available to support the suggested changes; the budget and funding sources are discussed in Chapter 5. Recommendations assume, but do not advocate for, the continuance of the program.

Table 1. The stated legislative intent, program goal, and six objectives of the OREHP, which were used as a framework for evaluation of the OREHP.

Stated OREHP intent, goal or objective	Description
Ultimate legislative goal[1]	To enhance populations of marine finfish species important to California for their sport and commercial fishing value
Current legislative intent[2]	Determine if hatchery-released fish can enhance stocks of wild species through increased hatchery production of fish, and the monitoring of fisheries to assess hatchery contributions
OREHP primary goal[1,3]	To evaluate the economic and ecological feasibility of releasing hatchery-reared fish to restore depleted, native, marine fish populations to a higher, sustainable level
Objective 1	Develop and implement hatchery operation and growout methods that provide a supply of healthy and vigorous fish.
Objective 2	Conduct the replenishment program in a manner that will avoid any significant environmental impacts resulting from operation of either the hatchery or pen rearing facilities.
Objective 3	Maintain and assess a broodstock management plan that results in progeny being released that have genotypic diversity very similar to that of the wild population.
Objective 4	Quantify contributions to the standing stock in definitive terms by tagging fish prior to release and assessing their survival in the field
Objective 5	Continue to develop, evaluate, and refine hatchery operations to maximize the potential for achieving the goal of the program.
Objective 6	Develop quantitative measures of success.

[1] White Seabass Enhancement Plan 2010; [2] FGC § 6590; [3] Comprehensive Hatchery Plan 2007

Chapter 1

Objective 1. Develop and implement hatchery operation and growout methods that provide a supply of healthy and vigorous fish.

1.1. Development and completeness of hatchery and growout plans in meeting the primary goal and objectives of the OREHP.

The Carlsbad Hatchery program was designed to develop culture techniques for depleted marine fish species and to produce offspring for use in the Ocean Resources Enhancement and Hatchery Program (OREHP). The OREHP goal and objectives, along with a general plan for achieving objectives, are described most fully in the Comprehensive Hatchery Plan (2007). The key issues relevant to evaluating OREHP Objective 1 are determining: (1) the extent to which plans, or strategies, achieve the overarching goal to restore a population to a higher, sustainable level; (2) the extent to which plans, or strategies, have achieved the objectives; and (3) in addition to White Seabass, which other species of depleted, endemic, marine fish could be the subject of OREHP activities.

1.1.1. Key Findings.

1.1.1.1. OREHP goals and focal species.

Interpretation of the OREHP Legislative Intent. The purpose of California Fish and Game Code Article 8 "**Ocean Fishery Research**" (within Chapter 5: Fish Planting and Propagation) is **"…to determine if hatchery-released fish can artificially enhance certain stocks of various desirable species, ... and to assess the contribution of hatchery-released fish…" (FGC § 6590).** In 1983, the California Legislature "… established in state government the **California Ocean Resources Enhancement and Hatchery Program for the purpose of basic and applied research on the artificial propagation, rearing, stocking and distribution of adversely affected marine fish species that are important to sport or commercial fishing [off southern California]" (FGC § 6592**). The statutory emphasis of the program is, therefore, on research related to the development and evaluation of enhancement experiments, with an implicit ultimate goal to actually enhance wild fish populations.

Six objectives. Six objectives were developed to help achieve the primary goal of the OREHP to evaluate the economic and ecological feasibility of using hatchery-raised fish to enhance wild stocks of depleted marine fisheries (Comprehensive Hatchery Plan 2007): (1) Develop and implement hatchery operation and growout methods that provide a supply of healthy and vigorous fish. (2) Conduct the replenishment program in a manner that will avoid any significant environmental impacts resulting from operation of either the hatchery or pen rearing facilities. (3) Maintain and assess a broodstock management plan that results in progeny being released that have genotypic diversity very similar to that of the wild population. (4) Quantify contributions to the standing stock in definitive terms by tagging fish prior to release and assessing their survival in the field. (5) Continue to develop, evaluate, and refine hatchery

operations to maximize the potential for achieving the goal of the program. (6) Develop quantitative measures of success.

OREHP focal species. Initial research under the OREHP involved California Halibut and White Seabass, which were selected as priority species to begin investigations based on several selection criteria (species indigenous to southern California; status as a diminished stock; economic value; both commercial and sport utilization; and potential for success). Research under the OREHP made more rapid hatchery production progress with White Seabass, as did enhancement research on Sciaenids in other parts of the U.S. and Europe; therefore most OREHP research has been focused on that species.

1.1.1.2. Marine enhancement history.

The strategies undertaken to fulfill the original goals and objectives of the OREHP in 1983 (e.g., Comprehensive Hatchery Plan 2007), as well as the choice of focal species, well reflected the 100-year history of enhancement science up to that point. Aspects of the strategies undertaken to address the objectives have since, however, become outdated. The science underlying marine fisheries enhancement was a fairly young, undeveloped field when the OREHP was initiated in 1983. Since then, the field of marine enhancement has progressed in three general ways: (1) improvements in the science and technology related to culture; (2) improvements in the science and theory of enhancement, especially for the family Sciaenidae to which White Seabass belongs; (3) the development of unified approaches to enhancement (e.g., Responsible Approach to Marine Enhancement). The existing goal and objectives are broadly worded so that these new fields and information may be integrated into strategies to update the program without changing the original wording or intent of the objectives.

Culture science and technology. Development of aquaculture technology to rear marine fishes (i.e., fishes that spawn in seawater) has lagged literally centuries behind the technology for rearing freshwater and anadromous species, largely because of significant challenges encountered in rearing the early life stage of marine fish and a reliance on live feed. Some of the first breakthroughs in rearing marine fishes occurred in Europe and Asia in the 1960s and 1970s; thus, the OREHP, which was authorized by the California Legislature in 1983, was tasked with expanding development of a fairly new technology, which had been worked out for very few marine fishes.

Perhaps the marine fish species that is the most well understood in terms of mass-culture for stock enhancement in the United States (U.S.) is Red Drum (*Sciaenops ocellatus*), which also belongs to the family Sciaenidae. As the target species for the Texas marine stock enhancement program, Red Drum enhancement was the first of the modern day enhancement programs in the US that released juveniles into the wild. Given the ease of rearing Sciaenids (e.g., Red Drum and Spotted Seatrout, *Cynoscion nebluosus*, in the U.S.), relative to most other families of marine fishes in the 1980s, the choice of White Seabass (also a Sciaenid) by the OREHP made good sense.

Stocking science and technology. Prior to the last decade of the 20th Century, the history of fisheries enhancement is littered with production-oriented enhancement programs that failed to

take survival of stocked fishes into account. Since the mid 1980's, the science underlying the stocking side (as opposed to the production side) of both salmonid enhancement and marine fish enhancement has been evolving in tandem, and both salmonid and marine fish enhancements share most of the same key issues (discussed in the 'Responsible Approach' papers mentioned below).

Although stocking cultured marine fishes began in the nineteenth century, the technology was limited to stocking only eggs and yolk-sac larvae (Richards and Edwards 1986). Attempts to stock marine fish eggs and 3-day-old yolk-sac larvae fell out of favor in the 1950s for lack of success, seven decades after Spencer Baird initiated U.S. marine stock enhancement at what is now the National Oceanic and Atmospheric Administration National Marine Fisheries Service, NOAA NMFS (Richards and Edwards 1986). There were no published accounts of the fate of released fish until empirical studies of anadromous salmonids began to be published in the mid-1970s (Hager and Noble 1976, Bilton et al. 1982), followed by the first studies (published in English) of stocked marine invertebrates in 1983 (Appeldoorn and Ballentine 1983, Appeldoorn 1985) and the first study of stocked marine finfish in 1989 (Tsukamoto et al. 1989).

Since 1989, the field of marine fisheries enhancement has advanced considerably (Lockwood 1991, WAS 1991, AFS 1993, EAS 1993, Schramm and Piper 1995, Travis et al. 1998, Howell et al. 1999, Nakamura et al. 2003, Leber et al. 2004, Nickum et al. 2004, Bell et al. 2006, Bell et al. 2008, Lorenzen et al. 2013, Sass and Allen 2014; see Leber 2013). Science in this field has rapidly grown, in part because of critical examination and debate about the efficacy of enhancement and the need for quantitative evaluation (e.g., Peterman 1991, Hilborn 1999), and in part because of advances made in aquaculture, genetics, tagging, and fishery modeling technologies, which have enabled quantitative studies and predictions of stocking effects. Much of the progress made in the 1990s was scientific and involved an expansion of field studies to evaluate survival of released fish and improve the effectiveness of release strategies. The earliest studies on effectiveness of stocking marine fishes published in English in the scientific literature were in Japan (Tsukamoto et al. 1989, Kitada et al. 1992, Sudo et al. 1992, Fujita et al. 1993, Yamashita et al. 1994) and Norway (Svåsand et al. 1990, Svåsand and Kristiansen 1990a, Svåsand and Kristiansen 1990b, Nordeide and Salvanes 1991), followed by studies in the U.S. (e.g., Kent et al. 1995, Leber 1995, Leber et al. 1995, McEachron et al. 1995, Willis et al. 1995, Leber et al. 1996, Leber and Arce 1996, Leber et al. 1997, Leber et al. 1998), and Australia (Rimmer and Russell 1998). Progress made with marine invertebrates is well covered by Bell et al. (2005).

1.1.1.3. Responsible Approach.

The potential of stocking marine organisms as an effective addition to fishery management strategies is high, but only when certain conditions are met. For stocking to be productive and economical, and to help ensure sustainability of wild stocks, careful attention must be given to several key factors and stocking must be thoroughly integrated with fisheries management (Blankenship and Leber 1995, Lorenzen et al. 2010). Most marine fisheries enhancement programs fail to do this, including the OREHP. Coupling enhancement with fisheries management and using a comprehensive, unified and careful approach is important, given that stocking can be harmful to wild stocks if not carried out carefully and responsibly (e.g., discussed in Blankenship

and Leber 1995, Tringali and Leber 1999, Leber 2013, Walters and Martell 2004, Lorenzen et al. 2010, Lorenzen et al. 2012).

A unified process, entitled "Responsible Approach to Marine Stock Enhancement," has emerged for developing, evaluating, and using enhancement (Blankenship and Leber 1995, Walters and Martell 2004, Lorenzen et al. 2010, Leber 2013, Sass and Allen 2014). The basic OREHP objectives and Comprehensive Hatchery Plan generally follow some of the original concepts of the Responsible Approach to Marine Stock Enhancement (Blankenship and Leber 1995, Sass and Allen 2014), despite the OREHP having been initiated a decade prior to the first publication of the Responsible Approach. For example, the OREHP has included some adaptive management elements like criteria-based species selection. This is because OREHP-contracted researchers from Hubbs-SeaWorld Research Institute (HSWRI) were among the scientists involved early in the development of marine fisheries enhancement (Kent et al. 1995), and were among the eight scientists who formed the International Working Group on Stock Enhancement in 1993 in Torremolinos, Spain, which initiated the paper presenting the Responsible Approach (Blankenship and Leber 1995). Blankenship and Leber also affiliated this workgroup with the World Aquaculture Society to further promote, develop and update the Responsible Approach (Leber 2013).

A revised version of the Responsible Approach was produced in 2010 (Lorenzen et al. 2010) and remains a unified standard for marine enhancement. The implementation of many elements of the Responsible Approach (Lorenzen et al. 2010) into the strategies used to fulfill the OREHP's objectives would allow for updated approaches that could aid future evaluations and improve the operations of the White Seabass Enhancement Program, as well as provide a framework for the development of future enhancement efforts.

1.1.2. Data and Information Gaps.

The development of strategies (plans, procedures) were based on "best available data" at the time, and updates to those basic strategies have been based on best available data as updates occurred so that there is plenty of information with which to assess the program's progress. The Responsible Approach, a widely-accepted set of guidelines, exists now, however, and should be included into more extensive revisions of the OREHP strategies in order to bring strategies up to date with current approaches. This will allow more accurate evaluation of this, and other programs in the future, and also provides guidance for making decisions about other focal species.

1.1.3. Recommendations.

More than three decades have passed since the inception of the OREHP and many changes have occurred in: (1) the science of rearing and enhancement (e.g., development of culture plans for many fish species, changes in our understanding of diseases and deformities, development of stocking strategies that reduce post-release mortality), (2) genetic approaches and associated analytical tools to assess genetic impacts of enhancement, (3) fishery and ecosystem management (e.g., White Seabass Fishery Management Plan, changes to fishery regulations), (4) technology (e.g., genetic identification of hatchery fish), and (5) approaches to analyzing economic and sociological systems. The goal of the OREHP to **evaluate the economic and ecological feasibility of releasing hatchery-reared fish to restore depleted, endemic, marine fish populations to a higher, sustainable level** remains useful, but the strategies (e.g., Comprehensive Hatchery Plan 2007) used to achieve the program's objectives should be updated to reflect this new knowledge and priorities.

A guide for comprehensively updating strategies for achieving the six OREHP objectives is the revised Responsible Approach (Lorenzen et al. 2010). Lorenzen et al. (2010) point out that "…Not all elements [of the Responsible Approach] are relevant under all circumstances, but most will be. No element should be discounted simply because its implementation is difficult." In particular, the OREHP would benefit from more attention to the following:

Stage I: Initial appraisal and goal setting

Key element 1- Understand the role of enhancement within the fishery system. A stock enhancement model to explore likely outcomes of OREHP fish releases was completed as part of this evaluation, and should be re-run occasionally for White Seabass, or any other chosen focal species, as new information becomes available. Due to recent progress in the development of population dynamics models and assessment methods for enhancements, such evaluations can now be carried out. An assessment tool based on a general population model for enhancements (Lorenzen 2005) is now available in the freeware package *EnhanceFish* (Medley and Lorenzen 2006). This program makes it feasible to perform evaluation of enhancement programs from early planning to full-scale operation (directly informs revision of strategies used to achieve Objectives 4 and 5).

Key element 2- Engage a greater diversity of stakeholders in decision-making. The social and economic implications of the OREHP remain largely unknown, yet are stated as part of the research of the feasibility of marine enhancement in the OREHP's goal and included in the objectives (directly informs revision of strategies used to achieve Objectives 4 and 5).

Key element 3- Emphasize the quantitative assessment of contributions (and costs) of enhancement to fisheries management goals (i.e., quantify economic and ecological costs and benefits; define indicators of success; quantify trade-offs between enhancement, harvest, and habitat management to determine where enhancements add value to other forms of management through population modeling; use quantitative analysis [via population models] early on in the development or reform process). (Directly informs revision of strategies used to achieve all Objectives).

Stage II: Research and technology development including pilot studies

Key Element 6- Establish enhancement system designs suitable for the fishery and management objectives. (Directly informs strategies used to achieve Objectives 1, 3, 4, and 5).

Key Element 8- Use genetic resource management and minimize deleterious effects in wild populations. The original objective of maintaining genetic diversity that has guided much of the research therefore needs to be revised, and genetic monitoring should be implemented. (Directly informs strategies used to achieve Objective 3).

Key Element 11- Use an empirical process for defining optimal release strategies. As discussed in Section 4.4 below, pilot release experiments coupled with substantial and consistent monitoring need to receive much greater attention in order to optimize release strategies and enable adaptive management of stocking effects; this is essential for reducing short-term mortality of stocked hatchery fish and increasing the effectiveness of the stocking program in achieving fishery management goals. (Directly informs strategies used to achieve Objectives 4 and 6).

Stage III: Operational implementation and adaptive management

Key Element 14- Assess and manage ecological impacts on populations and ecosystems; potential ecological impacts should be appraised early on in the development or reform of enhancements. However, because impacts may become apparent only once enhancements are scaled up to fully operational, empirical assessments and remedial management should be conducted in Stage III. (Directly informs revision of strategies used to achieve all Objectives).

1.2. Efficiency of hatchery capacity use.

1.2.1. Key Findings.

In general, the ability of the hatchery to maximize production of high-quality healthy fish relates directly to numbers of fish available for release. The White Seabass program is unique in that it also relies on multiple post-hatchery growout facilities that are essential for producing fish in the desired size class prior to release. Potential capacity for each of the different size classes is dependent on a number of factors and is therefore not completely clear. It seems that realized production is generally at (e.g., eggs, larvae) or below (e.g., juveniles) maximum capacity for the various life stages. HSWRI states that the ideal goal is to release 350,000 hatchery fish per year. This should be achievable while maintaining high-quality conditions at all hatchery production phases and by rearing fish at “modest densities” (Comprehensive Hatchery Plan 2007). However, these target release numbers have not been met due to bottlenecks or limitations, primarily within the hatchery, and decisions about size at release.

1.2.1.1. Bottlenecks.

Potential and realized hatchery bottlenecks are associated with physical system capacity for rearing each life stage; broodstock availability, maturation timing, and spawning frequency; egg production and hatching success; larval survival and growth; live feed production and efficiency of weaning larvae to commercial diets; juvenile growth; and fish health and fitness issues,

including overcrowding, gas supersaturation, periodic disease outbreaks, deformity (also referred to as malformation), and subsequent quarantine and culling as required.

Production limitation is most often linked to disease and deformity concerns, recent genetic constraints that allow only 12,000 fish per spawn to be released, and, to a lesser extent, in response to outcomes resulting from overcrowding in quarantine (e.g., animal welfare, disease, cannibalism, poor water quality) while decisions about diagnoses and responses are made. Strict guidelines exist for identifying pathogens of concern, screening for deformities, and for euthanizing fish in response to the detection of specific pathogens, but decisions associated with the identification of pathogens and deformities, and appropriate responses, can take weeks to several months, during which time procedures are unclear. In some cases, fish are euthanized because of overcrowding before a disease agent has even been identified (OREAP Meeting Minutes, 25 March 2014). In other cases, a disease or deformity is identified yet the causes or triggers become more difficult to identify after the fact. Further, the influence of various external and internal deformities found in hatchery fish on health, growth and long-term survival is largely uncertain, and therefore agreement among OREHP partners on what constitutes a deformity is lacking (see Sections 1.6.1.7 and 1.9 for more detailed discussions on deformity). Better decision-making around initial health diagnoses, and a better understanding of the causes and effects of disease and deformity on fish survival, reproduction and safety, could reduce some of these bottlenecks and allow for exploration into alternative uses for the fish thereby increasing efficiency and capacity of the facilities.

1.2.1.2. Size at release.

In general, the maximum number of individuals that can be reared by the current hatchery is dependent upon size at release, with higher capacity for smaller fish (see Section 4.4.1.5 for more information on size at release and a discussion of release strategies). Smaller juvenile fish may, however, have lower potential survival rates in the wild based on a model of White Seabass, which estimated that a fish released at the size of 200 mm had a 1.5% chance of surviving to minimum legal length (600 mm SL), while a fish released at 400 mm under the best conditions (in the Spring, with prior acclimatization in the net pen) had a 13.8% chance of surviving to the fishery (Hervas et al. 2010). (Note that the minimum legal length for White Seabass is reported as 711 mm TL (28 inches) for both commercial and sport fisheries (FGC § 8383.5, 14 CCR § 28.35)). Even though field tests to validate the results of this model are incomplete, observations from most other hatchery supplementation programs support this finding. No economic assessments of optimal release sizes have been conducted for the White Seabass, but a field test for economically optimal size at release of Striped Mullet (*Mugil cephalus*) was completed (Leber et al. 2005) and may be a reasonable approach to incorporate into the OREHP. This approach revealed that optimal size at release can vary greatly, depending on release habitat and release season (Leber et al. 1998, Leber et al. 2005, Tringali et al. 2008). Earlier release may be preferable to minimize genetic effects on the wild population, because the potential of domestication selection is reduced and that of natural selection is increased. Empirical tests of optimal release season and net pen acclimation, based on the other findings of Hervas et al. (2010), and size are currently being conducted with the data collected from nearshore gill net sampling (Gill Net

Report 14-15). Findings on optimal release size, season and acclimation period may alter hatchery capacity.

Besides using empirical economic and biological (survival) evaluations to determine optimal size(s) at release and, in turn, hatchery capacity, the number of fish per size class needed to achieve a desired effect in the fishery should be considered. Decisions about White Seabass size at release and release magnitude within a size class should include consideration of interactions between hatchery and wild fish. The potential for stocking to increase abundance of the target stock is a function of recruitment, density-dependent mortality, and dispersal characteristics of hatchery and wild fish. Density-dependent mortality should be a key consideration in developing or reforming stock enhancement programs (Hilborn 1999, Levin et al. 2001, Lorenzen 2005), but accounting for it in choices about release strategies can be difficult and expensive (Lorenzen et al. 2010). However, there have been some attempts to quantify this. Population models should be used to explore stocking effect, and field tests are needed to evaluate some of the key model assumptions. For example, the *EnhanceFish* model (Medley and Lorenzen 2006) predicts that stocking hatchery-reared snook results in an overall increase in snook abundance, but at the cost of some displacement of wild fish (Walters and Martell 2004, Lorenzen 2005). Lorenzen (2005) provides thoughtful consideration of density-dependence effects on the outcome of enhancements, unpacking recruitment into a density-independent larval stage, a density-dependent juvenile phase, and pre- and post-release phases according to the size at which juveniles are released. Transition from larval settlement to the juvenile stage coincides with density-dependent mortality (Van der Veer 1986), and Lorenzen (2005) reasons that stage-specific survival puts an upper limit on the potential degree of density-dependence within the juvenile stage. Thus, if survival reflects general allometry of mortality, this implies declining potential for density-dependent mortality with increasing size at release (Lorenzen 2005).

There is a paucity of empirical research to evaluate model predictions about density-dependent interactions between marine hatchery and wild fish, but Leber et al. (1995) examined the effects of stocking juvenile hatchery-reared mullet on abundance of wild mullet recruits in Hawaii and found no density-dependent effect on wild mullet dispersal. Brennan et al. (2008) evaluated density-dependent mortality after stocking juvenile hatchery-reared Common Snook (*Centropomus undecimalis*) into wild snook nursery habitats, attempting to double abundances of snook juveniles at two treatment sites vs. a 5% increase in abundance at their two control sites. Results of that study revealed a sustained doubling effect after snook were stocked at one of the principal treatment (high-stocking-density) release sites, but a large (64 – 85%) loss of stocked hatchery fish at their replicate treatment site, whereas there were no apparent effects of hatchery fish on wild snook densities at either of the two treatment or control sites. These two (mullet and snook) studies both involved the stocking of only relatively large juveniles, six months or more in age. C.J. Walters (pers. comm.) hypothesized that the apparent lack of wild fish displacement in the snook study (Brennan et al. 2008) resulted from stocking hatchery juveniles that had already grown to sizes (8 – 12 month old, 85 – 270 mm Fork Length [FL], mean=177 mm FL) well beyond the density-dependence phase.

Leber (unpublished) hypothesized that biological differences in hatchery and wild juvenile snook in that study resulted in a competitive advantage to wild snook that had recruited after settlement to nursery habitats 8 – 12 months prior to release of the hatchery snook juveniles into those habitats. That study suggests that a key question needing much more research is: do [H1] relatively large juvenile hatchery fish that have never before encountered predators or the diversity of habitats at release sites in the wild have behavioral deficits in competitive abilities at the time of release that result in lack of ability to displace wild fish from microhabitats that afford refuge from predation? A corollary to this is that [H2] hatchery fish stocked at relatively small sizes (post-settlement and early juvenile stages) into wild fish nursery habitats learn competitive skills along with small wild juveniles that recruit to those habitats. Brennan et al. (2008) suggest that overstocking a nursery habitat with 8 – 12 month old juveniles may result in displacement of some or most hatchery fish, but not necessarily displacement of wild fish by hatchery fish. Thus, by choosing to stock quite large juveniles by growing them for several months prior to release, the OREHP's choice of size at release seems conservative, as stocking such large juveniles has likely reduced the potential to cause density-dependent mortality of wild White Seabass.

1.2.2. Data and Information Gaps.

The hatchery capacity has not been fully utilized due to (1) bottlenecks associated with the hatchery and growout facilities and operations, and (2) White Seabass population data gaps as related to enhancement (i.e., more information on size(s) at release).

1.2.2.1. Data gaps associated with bottlenecks.

A brief mention of data gaps associated with common bottlenecks to utilizing full hatchery capacity are mentioned here, but some issues are also treated in extensive detail in other sections of this report, as noted.

1. System upgrades. There is uncertainty about how upgrades may contribute to increased capacity at each life stage.
2. Disease (see a more detailed discussion of disease in Sections 1.6.1.6 and 1.8).
    a. Lack of resolution of gas supersaturation issues in the hatchery and growout facilities.
    b. Lack of streamlined decision-making protocols for the time between disease detection, pathogen identification, and treatment in order to minimize spread of disease and unnecessary euthanasia.
3. Deformity (see a more detailed discussion of deformity in Sections 1.6.1.7 and 1.9).
    a. Lack of information about the specific, often multivariate, causes and extent of morphologic variation in hatchery fish, and the effects that the various types and severity levels of deformity have on growth, reproduction and survival throughout the life of the fish.
    b. Lack of agreement among OREHP affiliates on what constitutes a "deformity."
    c. Lack of information about alternative markets for juvenile fish that are culled from the hatchery because they are in produced in batches that exceed the genetic restriction of 12,000 fish released per spawn, or are not fit for stock enhancement purposes. The

causes of any deformities are not entirely clear so continued screening and culling is likely. Until a cause and solution is found, it would be beneficial if some type of cost recovery could be realized. The fish produced from the program are owned by the State of California, which would allow for funds to cover OREHP budget shortfalls.

1.2.2.2. Population data gaps.

1. Lack of empirical data to determine the relationships between the size of fish at release and survival in the wild, recruitment and dispersal patterns, windows of density-dependent mortality, and interactions with wild populations (See a more detailed discussion in Chapter 4).
2. Lack of empirical data on the extent of domestication selection and its dependence on size at release (See a more detailed discussion in Chapter 3).
3. No empirical economic analysis of optimal size(s) at release to verify earlier economic modeling of this variable (Hervas et al. 2010); in particular release habitat effects on size-dependent survival can conflict with model assumptions (e.g., Leber et al. 1998) (See a more detailed discussion in Section 1.7; also see Section 4.4.3, Recommendations 2 and 3).

1.2.3. Recommendations.

1. System upgrades. Specific needs should be determined and assessed in relation to system upgrades and how such upgrades may contribute to increased capacity at each life stage.
2. Disease. Focus emphasis on control and prevention of known or reoccurring diseases, in particular the effects of gas supersaturation, and streamline triage protocols for the periods of time between disease detection and disease identification and treatment. The strict criteria for euthanizing fish due to detection of specific pathogens is well defined as part of the Release Criteria for White Seabass (CDFW Release Criteria 2015); however, there is need for a more streamlined decision-making process and timely decisions if specific pathogens are not identified in a timely manner. It is recommended that CDFW and HSWRI veterinary personnel develop a mutually agreed-upon comprehensive decision-making process for when questions arise regarding unknown pathology (i.e., what to do if mortality increases but no disease agent is discovered, what to do in the time between when a problem arises and when tests can be completed to diagnose the problem) (See Section 1.6.1.6 for discussion of current disease surveillance and detection protocols). This process should include a detailed and transparent record keeping system to aid in adaptive management.
3. Deformity. Create consistent deformity screening protocols and mandate that a common protocol be accepted by HSWRI and CDFW for screening and culling of fish (See Sections 1.6.1.7 and 1.9 for further information on deformity).
4. Expand capacity. Explore options for utilizing overly successful rearing of individual life stages (e.g., more eggs or larvae than needed) and “culled” fish with various deformities. The causes of deformities are not clear (but may be nutritional) so screening and culling

should continue until solutions are found and changes in screening and culling practices are implemented. In the meantime, some type of cost recovery could be beneficial (e.g., determine if some fish that would be culled out of the program could be sold to commercial aquaculture producers). The fish produced from the program are owned by the state of California, so as long as funds return to the program this may represent a viable revenue stream (see Section 6.6 for considerations regarding potential commercial opportunities).

5. Adaptive management of annual production potential and goals. Use ecological and economic data to determine and maximize production of appropriate size classes to meet current stocking needs and to minimize interactions with wild populations (e.g., enhancing density-dependent mortality). (Also relevant to Section 1.3; see Section 4.4 for more discussion based on analyses conducted for this review).

1.3. Efficiency of growout capacity use.

1.3.1. Key Findings.

1.3.1.1. Growout facility requirements.

Growout site selection is approved by the OREHP Facility Site Selection Committee, whose members include individuals from HSWRI, CDFW, and the White Seabass Committee, which is made up of growout volunteers (Growout Procedures Manual 2007). Selection criteria include ecological and logistical considerations. Growout cages are ideally located in water at least 18 ft deep, although they may be located in water with a mean depth of at least 8 ft, to allow for a minimum cage depth of 3 ft while still maintaining clearance off the bottom at low tide (Growout Procedures Manual 2007). Sites should be located far away from live bait receivers, fish cleaning tables, fueling docks, shipyards, sewage outfalls, and power plants, whenever possible. Consideration of proximity to suitable release areas is not a high priority, because White Seabass range freely along the coast, and in and out of embayments (Growout Procedures Manual 2007). The permitting required to run growout facilities is shown in Table 1.1, but may vary slightly with location (e.g., with local authorities).

Growout facilities are operated by volunteers and are diverse, made up of net pens, land-based pools, or submerged raceways, depending on the location (see Table 1.2). Because they are run by volunteers and researchers, cage facilities are easily accessible by foot or by a short boat trip (Growout Procedures Manual 2007). Accessibility makes maintenance of the facility, and transfer and culture of the fish, much easier. The possibility of expansion of facilities should be kept in mind when choosing a site (Growout Procedures Manual 2007). Startup operations at growout facilities are kept small and simple, but once a growout facility has been tested, expansion may be required to meet annual release goals. It is also important to ensure there are enough volunteers dedicated to culturing fish at each site, which requires a minimum of two hours a day on average, with many more hours required on occasions of receiving, releasing, and treating fish, and cleaning the cage.

Table 1.1. Permits and permissions required to operate an OREHP growout facility. Table from the Growout Procedures Manual (2007).

Regulatory Authority	**Permit or Permission**
Department of Fish and Wildlife	Permission to participate in the OREHP
California Coastal Commission	Coastal Development Permit (CDP)
State Lands Commission	State Lands Lease is required if the tidelands have not been granted to a local authority
State Water Quality Control Board	401 Certification – in the past, this has been waived because the US Army Corps of Engineers has not issued 404 permits.
US Army Corps of Engineers	404 permit (large facility) *or* letter of permission (small facility)
US Coast Guard	Private Aids to Navigation Permit
US Fish and Wildlife Service	Section 7 Endangered Species Act Consultation
NOAA Fisheries	Letter of permission indicating no species of concern will be impacted
Local Authority (City, County, Port Authority)	Requirements vary depending on the authority
Regional Water Quality Control Board	National Pollution Discharge Elimination System (NPDES) permit (large facility) *or* National Pollution Discharge Elimination System (NPDES) waiver (small facility) – may contain monitoring requirements

When there were 13 growout facilities running, it was estimated that almost 1.1 million 200-mm fish (82,000 kg) could be grown out annually (White Seabass Enhancement Plan 2010), but these target release numbers have not been met due to bottlenecks associated with the number of growout facilities, both the potential and realized capacity of each facility, and length of time in growout.

Table 1.2. Growout facility characteristics. Information compiled from Growout Procedure Manual (2007) and HSWRI Annual Reports.

Growout facility	**County**	**Latitude**	**Longitude**	**Start Date**	**Close Date**	**System type**	**Total Culture Vol gal (cubic m)**	**Max Estimated Production lbs (kg)***
Santa Barbara	Santa Barbara	34 24.617	119 41.067	Aug 1993	n.d. 2012?	Net	24,240 (92)	3,028 (1,376)
Channel Islands Harbor	Ventura	34 09.826	119 13.326	Mar 1991		Net	45,960 (174)	5,742 (2,610)
Port Hueneme	Ventura	n.d.	n.d.	n.d. 1999?	n.d. Sept 2004?	Pool	12,152 (46)	1,458 (690)
Marina del Rey	Los Angeles	33 58.764	118 26.730	May 1995		Raceway	7,660 (29)	957 (435)
King Harbor	Los Angeles	33 51.056	118 23.638	Jun 1993		Pool	13,998 (53)	1,926 (875)
Catalina Harbor - Inner Harbor (CSF)	Los Angeles	33 25.549	118 30.624	Jun 1994		Net	68,948 (261)	8,613 (3,915)
Catalina Harbor - Outer Harbor (HSWRI)	Los Angeles	33 25.892	118 30.420	Mar 1998		Net	592,523 (2,243)	74,017 (33,644)
Alamitos Bay	Los Angeles	n.d.	n.d.	n.d. 1999?	n.d. 2000?	Net	7,660 (29)	919 (435)
Huntington Harbor	Orange	33 42.754	118 03.629	Sept 1996		Raceway	3,830 (14)	478 (217)
Newport Bay	Orange	33 36.052	117 53.411	Apr 1993		Raceway	13,407 (51)	1,675 (761)
Dana Point Harbor	Orange	33 27.450	117 41.586	Dec 1994		Net	10,414 (39)	1,301 (592)
Agua Hedionda Lagoon	San Diego	33 08.379	117 20.224	Jul 2003		Net	103,422 (391)	12,919 (5,872)
Mission Bay - Quivera Basin	San Diego	32 45.628	117 14.225	Apr 1997	n.d. 2011?	Net	9,576 (36)	1,196 (544)
Mission Bay - Dana Landing	San Diego	32 46.094	117 14.110	Jul 2001	Oct 2007	Net	2,394 (9)	299 (136)
San Diego Bay - SW Yacht Club	San Diego	32 46.132	117 13.985	Aug 1996		Raceway	3,771 (14)	471 (214)
San Diego Bay - Grape Street	San Diego	32 43.290	117 10.274	Apr 2003		Net	46,055 (174)	5,754 (2,615)

* Maximum production is based on a conservative harvest density of 0.12 lb/gallon (15kg/m^3)

1.3.1.2. Number of facilities.

The OREHP has had 16 distinct growout sites that have been used over the course of the program (see Table 1.2 and Fig. 1.1). The number of growout facilities in operation has fluctuated from year to year, but has remained between 10 and 15 since 1999. There are currently 11 growout sites in use (Table 1.2, Fig. 1.1). Five growout facilities have closed since about 2000 (Santa

Barbara, Port Hueneme, Alamitos Bay, and two in Mission Bay) due to a number of factors, including marine mammal intrusion, shallowness of water at the site, catastrophic fish losses, and loss of volunteer interest (e.g., Annual Reports 04-05, 06-07, 14-15).

Fig. 1.1. Map of active and inactive OREHP growout facilities. Map from HSWRI OREHP Overview Presentation, 20 May 2015.

1.3.1.3. Capacity of facilities.

Fish production efficiency appears linked to a variety of factors at each of the growout locations. These factors include, but are not limited to, training and commitment of the personnel responsible for fish care; water quality at each site (although not always assessed); and, as with hatchery juveniles, disease outbreaks (e.g., gas supersaturation, *Vibrio*) at these sites that result in mortality or culling of infected groups (see Sections 1.6.1.6 and 1.8.1 for more information on disease). Some facilities and groups are better at growout and some areas are less prone to mortality or other problems relating to fish health, which may affect post-release survival (see Table 1.3 for the average percent and range of survivorship of fish at growout facilities within the last three years). The problems seem to be associated with the lack of control once fish are transported to these sites and lack of resources and personnel to properly oversee fish rearing prior to release, and generally result in production of fish under growout capacity (e.g., during the first quarter of 2014, growout facilities were used at 61% of their total capacity (HSWRI OREAP Meeting Presentation, 25 March 2014)).

The operation of growout facilities by volunteers and the lack of HSWRI staff oversight may, at times, lead to problems with monitoring and handling fish. For example, in 2001, hatchery

personnel were unable to assist with the release of fish at the Santa Barbara growout facility, and as a result, the number of fish released was significantly underestimated, and monitoring of daily mortalities was not thoroughly conducted (Annual Report 00-01). Losses have been substantial at some sites in certain years (Fig. 1.2, Table 1.3). These losses are sometimes the result of unforeseeable accidents, such as bleach spills (e.g., a bleach spill nearby King Harbor in 2012 killed about 7,000 fish (Annual Report 11-12, Agostoni 2012)) and power outages (e.g, a power outage at King Harbor in 2005 killed about 3,000 fish (Annual Report 14-15, Mazza 2015)). Other times they are the result of disease outbreaks (e.g., bacterial infection and copepod infestation at Agua Hedionda Lagoon killed 51% of the 31,009 fish stocked there in late 2011), or personnel error (e.g., 100% of the 9,868 fish transported to the San Diego Bay (Grape Street Pier) growout facility died due to unspecified human error during transport of the fish (Annual Report 07-08)).

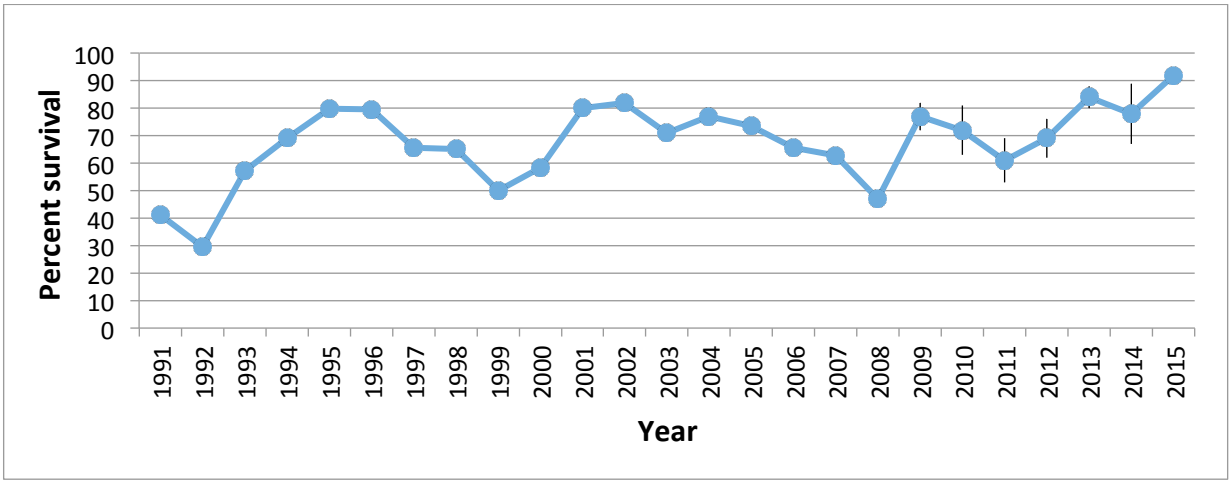

Fig. 1.2. Percent survivorship of White Seabass averaged across volunteer-run growout facilities within each year from 1991 to 2015. Annual survivorship averages from 1991 to 2008 were taken from a figure in the HSWRI OREAP Meeting Presentation, 21 October 2008. Average annual survivorship from 2009 to 2015 was calculated for this evaluation using data included in Annual Reports. For 2009-2015, years shown are the release year where % survivorship was calculated as [100 x (number of individuals in a batch at release / number of individuals in a batch at delivery to the growout facility)]. The percent survivorship of batches were averaged across all facilities for each year; ±1SE bars are shown. Excluded are data from the growout facilities run by HSWRI at Catalina Harbor and Agua Hedionda Lagoon; the figure focuses specifically on volunteer-run facilities.

Although the growout sites can be relatively difficult to monitor or change due to geographic distribution, ambient environmental conditions, and volunteer leadership, the limitation for growout at the hatchery and the need to release fish at a given size make use of volunteer run growout sites the most cost-effective approach to producing fish for release. It is important to note that fish survival has been among its highest since 2013 (Fig. 1.2), and could continue to be improved if oversight was enhanced and/or new and better sites were identified. Management priorities could be established based on the poorest performing and/or most variable performing sites (Table 1.3).

Table 1.3. Average percent survivorship of fish at each growout facility that received fish between 2012 and 2016. Numbers are compiled from Annual Reports, and include batches of fish delivered and released between January 2012 and June 2016.

Growout facility	Average % survivorship (± 1SE)	Range of survivorship
Channel Islands Harbor	81.2 ± 4.9%	63.3 - 93.3%
Marina del Rey	79.4 ± 7.6%	53.9 - 91.4%
King Harbor	78.1 ± 15.7%	0.0 - 97.9%
Catalina Harbor – CSF*	88.5%	88.5%
Catalina Harbor – HSWRI*	60.0%	60.0%
Huntington Harbor	85.6 ± 9.8%	56.5 - 99.0%
Newport Bay	90.9 ± 2.5%	82.2 - 99.3%
Dana Point Harbor	67.5 ± 10.9%	48.0 - 85.6%
Agua Hedionda Lagoon**	83.7 ± 8.3%	0.0 - 99.6%
San Diego Bay - Southwest Yacht Club**	91.2 ± 2.8%	74.7 - 98.6%
San Diego Bay - Grape Street	80.7 ± 3.6%	71.0 - 88.0%

*Between January 2012 and June 2016, both Catalina Harbor growout facilities received and released only one batch of fish each.
**Twelve of the sixteen batches received and released at Agua Hedionda Lagoon during this period were held for short growout cycles of less than two months (usually just a few weeks). One of the six batches received and released at the San Diego Bay - Southwest Yacht Club growout facility was short cycled as well.

1.3.1.4. Length of time in growout.

As with hatchery capacity (Section 1.2), the maximum number of individuals that can be grown out is dependent upon size at release, with higher capacity for smaller fish. The release of smaller juvenile fish may result in lower potential survival rates (Hervas Avila 2007, Hervas et al. 2010, Camp et al. 2014), while longer periods of time in captivity can increase the risk of disease, deformity, and losses due to stochastic events, crowded conditions, pathogens present in the environment, limited nutritional quality, and/or water quality and flow limitations in tanks and pens (e.g., Annual Report 13-14, Annual Report 14-15, Growout Procedures Manual 2007; See also Sections 1.7 and 4.4 for more information on fish release criteria, strategies, and rationale). No economic assessments of optimal growout and release sizes have been conducted for White Seabass, and optimal size at release can vary greatly depending on release habitat and release season (Leber et al. 1998, Leber et al. 2005, Tringali et al. 2008). Further, size at release decisions that incorporate fishery needs and interactions with wild stocks (as described in Sections 1.2.1.2, 1.7.1.1, and 4.4) would further influence length of time in growout and therefore growout facility capacity.

These challenges and the unknowns about triggers and outcomes of disease and deformities, and optimal release size, make this growout phase a distinct bottleneck for the program. The growout facilities run by volunteers and not under the direct control of HSWRI (making consistent data collection and husbandry challenging), are located in diverse, geographically separated areas, yet

are rearing the largest, most valuable fish in terms of resources invested and influence on survival in the wild.

1.3.2. Data and Information Gaps.

The gaps in data and information needed to increase capacity of growout facilities include a better understanding of management challenges (with respect to quality control) associated with multiple, different numbers of growout facilities, the influences on differences between potential and realized capacity of facilities, and the length of time fish spend in growout. Specifically, we see:

1. Lack of audits and updates to the growout plan that reflect issues associated with change in number of facilities, system failures, disease and deformities, and variable survival and growth at the various facilities (i.e., lack of adaptive management).
2. Lack of information on the extent to which growout system upgrades could contribute to increased capacity at growout facilities.
3. Lack of information on sources and triggers of disease at the various facilities (e.g., time in growout, environmental pathogens, abiotic conditions, and fish density).
4. Need for agreement between CDFW and HSWRI to reassess and revise health screening and release criteria based on available historic information and new knowledge about causes and effects of health issues (see Sections 1.6, 1.8, and 1.9 for discussion of health screening, and Section 1.7 for discussion of release criteria).
5. Lack of empirical data to determine the relationships between the length of time in growout and/or release size and survival in the wild, recruitment and dispersal patterns, windows of density-dependent mortality, and interactions with wild populations (see Sections 1.2.1.2, 1.7, 4.4).
6. No empirical economic analysis of optimal size(s) at release (see Sections 1.2.1.2, 1.7, 4.4).

1.3.3. Recommendations.

Growout facilities and the potential problems resulting in loss of these larger fish are clear bottlenecks of the program. We suggest activities that will minimize costs and staff time, as this is mostly an unfunded part of the program, while boosting the training and close oversight of qualified individuals due to the importance of this phase of production and potential impacts on survival of fish in wild. We recommend several programmatic and scientific actions to improve capacity of growout facilities:

1. Future programmatic strategies should include:
    a. Acquisition of specific new sites that can be operated directly by trained HSWRI staff. This is assumed to be one of the most important and critical phases of the program

(highest quality fish are going to these sites and they need to remain healthy up to time of release).

b. Continue to work with volunteer groups to maximize the efficiency of growout operations. This includes changing growout procedures or control measures for the volunteers and/or developing different protocols for the different groups of volunteers assuming different levels of experience, skill and resources to include size of fish (e.g., release fish at smaller size if less experienced growout volunteers to reduce chance of disease and/or mortality); and/or amount or type of scientific research incorporated (e.g., none if inexperienced volunteers). The risk of different procedures is that it becomes more difficult to keep track of which procedures were followed, and what actions led to observed outcomes. Therefore, needed is improved training and guidance to streamline volunteer decision-making, and an adaptive management plan for growout facilities that includes record keeping, data/information submission and regular assessment and revision of plans.

2. Future scientific data collection should include:
   a. Determine relationships among time in growout, incidence of disease, environmental conditions and growout conditions. Much of this information is likely available and should be summarized in a comprehensive review where trends can be determined.
   b. Identify sites where growout can be better controlled by HSWRI staff.
   c. Make informed decisions on when to stock specific shallow water growout facilities in locations known to be prone to warm water and gas supersaturation. Avoid stocking these sites (e.g., Marina del Rey, SW Yacht Club, Huntington Harbor) during El Niño years, and avoid stocking these facilities during the summer in "normal" water temperature years.

1.4. Water supplies: Steps taken to ensure that the hatchery and growout operations have high quality water supplies.

1.4.1. Key Findings.

The key issues related to the maintenance of high water quality supplies that arise for the OREHP are due to hatchery facility infrastructure and water sources, growout facility characteristics and available water quality, and uncertainty about sources of disease and pathogens.

1.4.1.1. Hatchery water quality, protocols, and system design.

Influent monitoring at the hatchery. HSWRI regularly monitors influent, as well as effluent and secondary backwash (covered in Chapter 2), at the Carlsbad hatchery according to the San Diego Regional Water Quality Control Board's (SDRWQCB's) Investigative Order No. R9-2009-0177, and presents the results of both influent and effluent water quality tests in annual reports to the SDRWQCB. These annual water quality summary reports exist for the years 2002 through 2016. Before this, from June 1996 to January 2002, HSWRI submitted monthly reports to the SDRWQCB. Samples collected at influent sample sites are all grab samples (SDRWQCB

Investigative Order No. R9-2009-0177). HSWRI has switched one of the laboratories that processes water quality samples twice, once because of suspected error in analysis (2008), and once because the lab closed (2010) (SDRWQCB Reports 2008, 2010).

HSWRI is required by the SDRWQCB to monitor influent water for pH, temperature, total suspended solids, settleable solids, total nitrogen, total phosphorus, total copper, total zinc, and unionized ammonia (SDRWQCB Investigative Order No. R9-2009-0177). Influent monitoring must be done on the same day, with the same frequency, and for the same parameters as effluent monitoring (SDRWQCB Investigative Order No. R9-2009-0177). Thus, HSWRI monitors pH, temperature, total suspended solids, settleable solids, total nitrogen, and total phosphorus on a monthly basis; and monitors total copper, total zinc, and unionized ammonia on a quarterly basis (in January, April, July, and October) (SDRWQCB Investigative Order No. R9-2009-0177). HSWRI also collects data on the average daily influent flow at the hatchery (SDRWQCB Reports 2010-2015). Before 2010, HSWRI was required to monitor total ammonia, salinity, total Kjedahl nitrogen, organic nitrogen, orthophosphate, nitrate, and nitrite (on top of what it monitors today) in influent waters according to SDRWQCB Order No. R9-2001-0237, NPDES Permit No. CA0109355, which was rescinded in December of 2009 after the SDRWQCB recognized that HSWRI's production levels fall below the aquatic animal production and feeding thresholds detailed in 40 CFR § 122.24 and 40 CFR Appendix C to Part 122 (SDRWQCB Order No. R9-2009-0090).

Hatchery protocols, system design, and water quality within the Hatchery. HSWRI has developed a water quality contingency plan for the Carlsbad hatchery, outlining response protocols in the event of a rapid deterioration of water quality resulting from algal blooms, oil and sewage spills, or urban runoff, among other things (HSWRI Fish Health Management Plan 2016). According to the contingency plan, the severity of the event determines whether the fish should be transferred, released, or euthanized (HSWRI Fish Health Management Plan 2016). Strategies to mitigate water quality and keep hatchery fish alive after one of these events include: turning off make up water if necessary, transferring tagged fish to net pens with cleaner water, reducing feedings or suspending feedings, and closely monitoring dissolved oxygen (HSWRI Fish Health Management Plan 2016).

Throughout the late 1990s and early 2000s, the hatchery's larval and juvenile pools were switched from flow-through to recirculation systems (Annual Reports 98-99, 99-00, 00-01, 02-03); there were plans to change the raceways to recirculation systems as well, but SFRA funding was diverted from the plans to support salmon hatchery programs in 2008 (HSWRI OREAP Meeting Presentation, 3 March 2009). While recirculation of water supplies presents its own challenges for maintaining water quality within the facility, it reduces the amount of discharge from the facility. Operational procedures to maintain higher water quality within the hatchery (and in effluent) include staying within the recognized fish density limits and not overfeeding, either of which will lead to water quality impacts and unnecessary stress associated with culture practices.

While hatchery water quality monitoring protocols exist (Weekly hatchery systems WQ sampling SOP 2016), ensuring the adherence to protocols, especially if production increases, is important

for maintaining the highest water quality. Several pathogen outbreaks since 2009 (e.g., *Miamiensis avidus*; Annual Reports 09-10, 15-16) were attributed by CDFW to biosecurity lapses, such as the likely use of untreated make-up water at the Carlsbad Hatchery and Mission Bay Hatchery, and the transfer of larvae and juveniles from Mission Bay to Carlsbad without quarantine (SFRA Reports 09-10, 15-16, CDFW Pathology Report 2015-059). The hypothesized causes of these outbreaks are uncorroborated by HSWRI documentation. While the specific causes of outbreaks remain uncertain, the development of protocols for movement between hatcheries, even stricter adherence to existing protocols by hatchery staff, and a more collaborative effort between CDFW and HSWRI to address potential causes of outbreaks— including development of mutually agreeable biosecurity protocols, investigations into outbreaks, and unified reporting of such incidences— is needed.

An ozone system to treat influent and make-up water was installed in 2003 primarily to help remove viral nervous necrosis virus (VNNV), but also with the hope of removing organic pollutants. Intake water and recirculated hatchery water, however, have not been monitored for organic pollutants or other contaminants of emerging concern (e.g., many metals, pharmaceuticals, caffeine) because these are not yet part of most water quality monitoring protocols. More extensive monitoring of intake water may be important as pollutants are potential contributors to deformity and fitness. The water source for the Carlsbad system, the Agua Hedionda Lagoon, is known to get extremely warm, and receive organic pollutants from several major upstream uses, including an 18-hole golf course, commercial agriculture, and heavy residential development.

HSWRI staff members have monitored pH levels and alkalinity in broodstock pools since July 2004 (Annual Report 05-06). According to the 2005-2006 Annual Report, “alkalinity and pH measurements taken in brood pools were consistently less than those measured in the incoming ambient seawater.” HSWRI staff suspected that the lower pH and alkalinity levels in broodstock pools were connected to the “bobbing behavior” observed in some broodfish, which may have been associated with the nephrocalcinosis (caused by high CO_2 concentrations) found in some fish (Annual Report 05-06). To mitigate these low levels of pH and alkalinity, HSWRI added sodium bicarbonate first to one, and then to all, broodstock pools (Annual Report 05-06). This effectively raised alkalinity and pH levels, and sodium bicarbonate ($NaHCO_3$) was added to broodstock pools on a weekly basis (Annual Report 05-06). In 2007, $NaHCO_3$ was added to the pools on a daily basis and measured and adjusted on a weekly basis (Annual Report 06-07). Alkalinity is tested weekly (Weekly hatchery systems WQ sampling SOP 2016), and the $NaHCO_3$ addition has helped overall water chemistry, biofilter performance and, although lower pH links to bobbing are not clear, bobbing has not been reported since (M. Drawbridge email to T. S. Talley, 27 March 2017, Annual Reports since 05-06).

Pathogens pose a large threat to any hatchery, and outbreaks may be linked to poor water quality especially for common opportunistic pathogens. Sources of pathogens and triggers of disease outbreaks are often not clear, but most pathogens likely come from influent water or from wild caught broodstock, which may be asymptomatic carriers. With a recycled hatchery water system, such as that used by HSWRI before the flow-through raceways, low water quality

and opportunistic pathogen outbreaks may also be related to a poor recycle system design. The interactions between biosecurity, pathology and system design can be complex. The efficiency of the system at removing pathogens (i.e., bio-filters) is not usually 100% and efficiency may vary with filter type. A system that, for example, allows untreated water into the system may increase the risk of contamination already present. Further, different system designs can result in different water chemistry conditions, which impact pathogens. For example, low pH inhibits and will cure some bacterial and parasitic skin diseases. However only some biofilter designs will continue to nitrify the ammonia to nitrate at pH < 6. Additionally, some biofilters can be adapted to withstand drastic salinity modifications, allowing salinity changes to eliminate or control some pathogens. The hatchery's recycle system design has evolved over the years generally following a unified analytically-based design approach (e.g., a mass balance model for the system), but has also had to be based on shorter-term decisions about the best available equipment for the job, where what is best has been dependent upon many factors, including efficiency, ease of use, mechanical reliability in a salt water environment, and cost (M. Drawbridge email to T. S. Talley, 27 March 2017).

However, the data to assess the specific sources and facilitators of pathogens are lacking, and assessments by hatchery system are needed to know how to change influent and hatchery water quality monitoring protocols and practices. For example, as mentioned above, HSWRI installed an ozone system to treat influent and makeup seawater at the hatchery in 2003, hoping to inactivate the VNNV (Annual Report 02-03). In October of 2008, after mass larval mortalities, HSWRI increased its monitoring of the ozone system to protect against malfunctioning, switching to daily monitoring of oxidation reduction potential (ORP) readings for incoming lagoon water (pre-ozone), post-ozone treatment, and after carbon (post-ozone removal), filter pressure readings, and flow rates (Annual Report 08-09). HSWRI also made modifications to the ozone system, suspecting a buildup of bromate ions in the water, a byproduct of ozonation treatment harmful to larval fish (Annual Report 08-09). The ozone system was changed from a multi-pass, two-speed system to a single pass, single speed system (Annual Report 08-09). Unfortunately, these changes did not improve larval survival (Annual Report 08-09). In 2011, a new filtration system was implemented at the hatchery, which ran water through two high capacity cartridge filter canisters followed by a UV sterilizer (Annual Report 10-11). Filtering ozone treated seawater through this new filtered-UV system greatly reduced the amount of bacteria in the water and dramatically improved larval survival (Annual Report 10-11). In 2013, a new UV-disinfected water filtration system was installed (Annual Report 12-13). Pre-filtered water is run through a high-output UV sterilizer, and then through additional filters and two 150 Watt UV sterilizers to kill bacteria (Annual Report 12-13).

In the case of a disease outbreak, fish are quarantined (see Section 1.10.1.2), and mortalities are collected from tanks and pens frequently in order to minimize impacts on water quality and potential spread of disease (HSWRI Fish Health Management Plan 2016).

1.4.1.2. Water quality at growout facilities.

The water quality at each growout facility is dependent largely upon site location and nearby features that may influence water quality. For instance, if a growout facility is in close proximity

to a pollutant source, such as a fuel dock, a sewage outfall, or a bait receiver, its water will likely be affected and may result in high incidences of fish mortality (Growout Procedures Manual 2007; see Section 1.3.1.1 for more information on growout facility siting). Another factor of site location that contributes to water quality is the flow of water under and around a net pen or cage. Sites located on dead end channels in harbors, with low tidal flow rates, will have poorer water quality than sites located near the entrance of harbors. Greater circulation of clean seawater around and under a pen leads to better water quality for fish (Growout Procedures Manual 2007). Further, the ability to evaluate water quality is limited at the growout sites, which are monitored irregularly due to limitations associated with volunteer run facilities and/or no legal requirements to perform monitoring (see Section 2.1 for more details).

The Growout Procedures Manual (2007) recommends that dissolved oxygen (DO), salinity, ammonia, temperature, pH, turbidity, and hydrogen sulfide be monitored at growout facility sites, especially when growout sites are first established, but given the current operational structure, such monitoring is impractical at growout sites even though it would be useful (see Sections 2.1 and 2.3 for a more detailed discussion of monitoring at growout facilities). Because the growout facilities produce less than 45.3 metric tonnes of biomass, they are not required to have an NPDES permit (Benthic Monitoring Plan 2005). Despite this, HSWRI initiated a 6-year program to evaluate potential benthic environmental impacts produced by the OREHP growout program (Benthic Monitoring Plan 2005).

The Los Angeles Regional Water Quality Control Board (LARWQCB) mandated water quality monitoring in and around four Los Angeles-based growout facilities for dissolved oxygen, temperature, and ammonia (LARWQCB Reports 2008-2014; see Section 2.1.1.1). HSWRI took on the responsibility of executing this monitoring program without any compensation. Reports on water quality for these four growout facilities were submitted to the LARWQCB from 2008-2014, including three reports each for Marina del Rey and Channel Islands growout facilities during this period, which was the goal for all growout facilities (Benthic Monitoring Plan 2005; see Sections 2.1.1.1 and 2.3.1.1).

1.4.2. Data and Information Gaps.

1. Uncertainty about the sources of pathogens contributing to infectious disease in the hatchery.
2. Lack of consistent water quality assessments for all growout sites.
3. Lacking are detailed records of outcomes of water treatment (e.g., bicarbonate), responses of water alkalinity (and fish condition) to treatments, and updates to protocols reflecting changes.

1.4.3. Recommendations.

1. Upgrade the recirculation systems at the Carlsbad hatchery, and the Mission Bay hatchery, if that will continue to be used, to get all water quality specifications within standards, including new regulations on contaminants of emerging concern and organic pollutants.
2. Develop, if needed, and regularly update protocols for each hatchery system with monitoring, detection, and response practices to address recurring and any newly discovered pathogens. Make sure documents are mutually agreed upon by CDFW and HSWRI, and use an independent science panel for input if needed.
3. Select growout sites with minimal water quality concerns or risk of disease occurance based on historical fish health screening data.

1.5. Food supplies and nutrition: Steps taken to ensure that fish in the hatchery and growout operations have high quality, nutritious food supplies.

1.5.1. Key Findings.

It is often difficult to determine appropriate feed formulations for each life stage of a new species for aquaculture, but HSWRI has in general been productive in working on the development of better ways to feed the various life stages of White Seabass, especially the larval stages. Most information on the quality of food supplies is available in HSWRI's Annual Reports, manuals and SOPs (HSWRI Fish Health Management Plan 2016, Comprehensive Hatchery Plan 2007), and publications (e.g., Trushenski et al. 2014, Jirsa et al. 2010, Durazo et al. 2010, Jirsa et al. 2014, Miller and Franklin 2005, Bañuelos-Vargas et al. 2013 (draft)). HSWRI has been conducting externally funded research, often in collaboration with commercial feed companies, to identify needs and requirements for live or commercial diets, and to compare the effects of different diets on growth. For example, Jirsa et al. (2010) examined the potential for a soy-based diet for hatchery White Seabass and found that fish performed better in terms of final weight, percent weight gain, and feed conversion ratio when fed diets that were made up of larger proportions of fish meal than soy. Miller and Franklin (2005) tested the effects of food enhanced with the nonessential amino acid L-Arginine on growth of juvenile White Seabass and found that the group of fish fed L-Arginine had a final mean weight more than twice that of the control group. There have also been unpublished studies conducted to test the effects of various diets with Spirulina and protein concentrate on fish growth and health (e.g., Wrobleski et al. submitted). Assessing the quality of food supplies requires a focus on broodstock, larval and juvenile rearing in the hatchery, juveniles at growout sites, and consideration of interacting environmental factors such as microbial communities in the water and within the fish themselves.

1.5.1.1. Nutritional considerations for broodstock rearing.

Broodfish are fed defrosted frozen sardines Monday, Tuesday, Thursday, and Friday of each week at a ratio of 1% of their fish biomass per day (Broodstock Feeding Schedule SOP 2015, Broodstock: Injecting Premixed Vitamins SOP 2015). As many sardines as possible are injected with a mixture of vitamin premix, ascorbic acid, lecithin, thiamine, and Menhaden oil before being fed to the broodfish; the vitamin loads are reevaluated every 3 months, or in the case of broodfish tank transfer or acquisition (Broodstock Feeding Schedule SOP 2015, Broodstock Food Distribution and Feeding Tips SOP 2015, Broodstock: Injecting Premixed Vitamins SOP 2015, Broodstock Vitamin Update SOP 2015, Comprehensive Hatchery Plan 2007). These vitamin-enriched sardines are supplemented with regular sardines, to achieve the correct feed volume for each pool (Broodstock Feeding Schedule SOP 2015). There does not appear to be a target ratio of vitamin-injected sardines to regular sardines, although on January 19, 2014, the percent of injected sardines fed to broodstock pools ranged from 14.2-26.2% (Broodstock: Injecting Premixed Vitamins SOP 2015). Prior to 2001, the broodfish received their vitamins through a prepared "brownie," the use of which was suspended because of suspected harmful effects (Annual Report 00-01). Since 2006, the broodstock diet has been supplemented with defrosted frozen squid every Wednesday, and broodfish are offered squid three months before the spawning cycle begins until the fish no longer eat it (fish show a preference for squid during spawning) (Broodstock Feeding Schedule SOP 2015, Annual Report 05-06).

1.5.1.2. Nutritional considerations for larval rearing.

HSWRI recognized that minimizing bacterial contamination during early feeding of larvae is important (Annual Reports 06-07, 07-08, 10-11, CDFW Pathology Presentation 2008), and do so by siphoning pools, cleaning larval tank and food production equipment, and sterilizing the water that runs through the feed systems ([J1] Tank Cleaning SOP 2016, Sterilizing *Artemia* Room Containers SOP 2015, Annual Report 10-11, [J1] System Components and Mechanical Operation SOP 2016, Preparing J1 for the First Run of the Season SOP 2016). White Seabass larvae begin to feed at around 4 days post hatch (dph), granted that 80% of their tank's swimbladders inflate (Swim Bladder Inflation (SBI) Rates at 4 DPH SOP 2016, Larvae Feeding Schedule (0-21 DPH) SOP 2016); low rates of swim bladder inflation within a crop or pool warrants sorting out those with uninflated swim bladders, or euthanizing the entire pool (Sorting White Seabass (Swimbladders) SOP 2009, Swim Bladder Inflation (SBI) Rates at 4 DPH SOP 2016). Larvae are kept in incubator tanks until they are about 21 dph, and while in the incubators, are fed a combination of live 1st instar *Artemia* nauplii or rotifers and enriched 2nd instar *Artemia* nauplii (Harvesting 1st Instar *Artemia* and Determine Destination SOP 2015, Larvae Feeding Schedule (0-21 DPH) SOP 2016, *Artemia* Density Calculations and Ration Calculations SOP 2015; See Table 1.4 for feeding regime). Rotifers are currently being tested for efficacy, and are fed to larvae for experimental purposes (M. Drawbridge pers. comm.; see below for further discussion). The managing staff decides which feeding regime to put the larvae on, whether it be rotifer and 2nd instar *Artemia* or 1st and 2nd instar *Artemia* (*Artemia* Density Calculations and Ration Calculations SOP 2015). From 4 dph to 12 dph, the guts of 20 larvae from each incubator tank are checked to ensure larvae are feeding on the *Artemia*; low consumption rates may be an indication of poor food quality or larval health (Gut Checks at 4 to 12 DPH SOP 2016).

Table 1.4. Larval feeding schedule, 0-21 dph. Recreated from Larvae Feeding Schedule (0-21 DPH) SOP 2016. If management decides to feed larvae rotifers, larvae receive rotifers from 2 to 7 or 8 dph, at a volume of 5 rotifers/ml, and begin to eat 2nd instar at 5 dph (*Artemia* Density Calculations and Ration Calculations SOP 2015, Pickup and Cold Storage of Rotifers SOP 2015).

Days post hatch (dph)	Food type and amount
0	No food
1	No food
2	No food
3	No food
4*	3 art/ml (1st instar)
5	3 art/ml (1st instar)
6	2 art/ml 1st instar and 1 art/ml 2nd instar
7	1 art/ml 1st instar and 2 art/ml 2nd instar
8	3 art/ml (2nd instar)
9	3 art/ml (2nd instar)
10	3 art/ml (2nd instar)
11	3 art/ml (2nd instar)
12	3 art/ml (2nd instar)
13	3 art/ml (2nd instar)
14	3 art/ml (2nd instar)
15	3 art/ml (2nd instar)
16	3 art/ml (2nd instar)
17	3 art/ml (2nd instar)
18	3 art/ml (2nd instar)
19	3 art/ml (2nd instar)
20	3 art/ml (2nd instar)
21**	3 art/ml (2nd instar) + B1

*First feeding is given at around 1:00 p.m. given good swimbladder inflation rates. If inflations are low (<80%), management will determine if food should be given at that time.
**Larvae will continue to get 3 *Artemia*/ml (2nd instar) if they need to stay in their incubator past 21 DPH. B1 signifies the smallest size of dry food (Otohime) offered to larvae.

Because *Artemia* lose most of their nutritional value within an hour of hatching, they are batch cultured at the hatchery during this stage of White Seabass larval growth in order to provide the larvae with the most nutritious food possible (Comprehensive Hatchery Plan 2007). 1st instar *Artemia* take 15-20 hours to hatch, and are either fed directly to the larvae (after their shells are removed and they are placed into a cold storage tank), or placed in an enrichment tank to become 2nd instars for the next day (Harvesting 1st Instar *Artemia* and Determine Destination SOP 2015, Prepare a Hatching Cone SOP 2015). *Artemia* are enriched with S.Presso over night for 19 hours (*Artemia* Tasks at a Glance SOP 2015, Harvesting 1st Instar *Artemia* and Determine Destination SOP 2015, Harvesting 2nd Instar *Artemia* SOP 2015). If rotifers are used to feed larvae, they are grown at the Mission Bay facility, and picked up and brought to Carlsbad every morning (Pickup and Cold Storage of Rotifers SOP 2015). The larvae are fed four times a day, at 7:00 a.m., 10:00 a.m., 1:00 p.m., and 4:00 p.m. (Feeding Larvae with Live Foods SOP 2015). *Artemia* nauplii alone are not nutritionally adequate for marine fish larvae, but batch culture and multiple daily feedings, as well as enrichment, are utilized to overcome some of these issues as standard industry practices. It is not clear if nutritional needs are met fully or if malnutrition is responsible

for deformities observed in White Seabass (see below for a description of research HSWRI has conducted on the relationship between feed and deformity). However, average annual larval survival is now consistently high at 25-40% (M. Drawbridge pers. comm.).

Larvae continue to receive 2nd instar *Artemia* for just over a week after they are moved into J1 from their incubators, which occurs around 21 dph (Weaning Larvae SOP 2016, Incubator to J1 Transfers (21 DPH) SOP 2016, Preparing the J1 System to Receive Larvae/Moving Larvae from Incubators SOP 2016). Their final full day of *Artemia* feed is 29 dph (Weaning Larvae SOP 2016, *Artemia* Density Calculations and Ration Calculations SOP 2015). Before 2007, frozen and dry foods were added to the larval tanks starting at 12 dph, and the ratio of frozen food to dry pellets was gradually reduced until no frozen food was offered at approximately 30 dph (Comprehensive Hatchery Plan 2007). The frozen food, freshwater mysis shrimp, *Mysis relicta*, was thawed and shaved into pieces, and fed hourly to the larvae through a drip bucket system (Comprehensive Hatchery Plan 2007). In January 2007, HSWRI discontinued feeding larvae mysis shrimp due to the difficulty the fish had in consuming it, and started to wean larvae off live food using just dry pellets of varying sizes (Otohime) (Annual Report 06-07, Weaning Larvae SOP 2016). The dry food comes in six sizes and is fed to the larvae every hour by hand, and continuously by two 12-hour belt feeders (belt feeders are used mostly at night) (Comprehensive Hatchery Plan 2007, Weaning Larvae SOP 2016). During this stage, the larvae are fed 0.4-2.0 kg/day/pool, according to age, size, and density of fish in each pool (Comprehensive Hatchery Plan 2007). Providing adequate amounts of the right sized food during the larval and juvenile stages of rearing White Seabass helps reduce cannibalism (Comprehensive Hatchery Plan 2007, Annual Report 06-07).

In December of 2010 and January of 2011, HSWRI made modifications to the facilities used to rear *Artemia* in order to reduce bacterial contamination of larval food (Annual Report 10-11). These modifications involved pre-filtering the water used for production of live feed, installing two milk chillers to maintain *Artemia* at a constant temperature of 10° C, incorporating a Filtered-UV Module for higher water quality (see Section 1.4.1), and improving daily disinfection protocols (Annual Report 10-11). *Artemia* cultures are now much cleaner and contribute to greater survival during larval stages (Annual Report 12-13).

It is well documented that nutrition can have a direct effect on deformities in marine fish and research conducted by HSWRI supports this (e.g., Annual Report 00-01, Annual Report 11-12, Annual Report 12-13). For example, in 2011, HSWRI conducted a study to compare the relationship between two different weaning diets, Otohime Hirame (a Japanese-produced diet typically used at the hatchery for fish this age) and Gemma Micro Diamond (a domestically-produced potential replacement), and the extent of deformity in late larval and juvenile fish (Annual Report 11-12). HSWRI found that fish fed Otohime consistently had the lowest malformation levels, 3-4 times lower than fish fed Gemma (Annual Report 11-12). The control fish exhibited malformation rates 2-3 times lower than those fed Gemma (Annual Report 11-12). In 2012, HSWRI, funded by the Western Regional Aquaculture Center, conducted a second study on different weaning diets, and found that the diet in which larvae were fed rotifers enriched with ORI-GREEN (Skretting) at 2 dph, and enriched *Artemia* at 6 dph with S.presso, produced significantly larger White Seabass, as well as significantly lower malformation rates (as compared

to other diet treatments) (Annual Report 12-13). These results have not been consistently reproducible in a production setting, so the effectiveness of this diet is still under investigation (M. Drawbridge pers. comm.). Thus, as mentioned above, the use of rotifers in larval diets at the hatchery is discretionary (*Artemia* Density Calculations and Ration Calculations SOP 2015), and is currently done on an experimental basis (M. Drawbridge pers. comm.).

1.5.1.3. Nutritional considerations for juveniles.

By the time juveniles are transferred to the J2 system (around 40-50 dph and 1-2 grams in size), they should be feeding on dry pellets at least 1.7 mm in size (Feeding in J2 SOP 2016, Preparing J2 for the First Run of the Season SOP 2015, [J2] System Components and Mechanical Operation SOP 2016, Annual Report 07-08). Juveniles at this stage are fed at a ratio of 3-5% of their biomass, determined by batch weights (Feeding in J2 SOP 2016). They are fed primarily by hand (to avoid overfeeding), and supplemented with feed from belt feeders, which are usually run at night after normal work hours (Feeding in J2 SOP 2016, J2 System Feeding SOP 2015). Juveniles in J2 can be fed five different sizes of dry food (Otohime 1.7 mm, Otohime 2.3 mm, EWOS 3 mm, EWOS 4.5 mm, EWOS 6 mm) (Feeding in J2 SOP 2016). The Comprehensive Hatchery Plan (2007) calls for vitamin C supplementation of the juveniles' diet at three times the rate of typical salmon feeds, but it is not added because a marine fish diet is used and not a salmon diet as was used when the plan was written (M. Drawbridge pers. comm.) Ascorbyl palmitate, a fat-soluble vitamin C useful in small fish diets, should be tested as a source since Vitamin C pellets (1 mm) leach out of the system in a few minutes (D. Weaver pers.comm.) After reaching 20-40 g in weight (91 to 150 dph), the juvenile seabass are moved to outdoor raceways, where they are fed the same dry pellets that are fed to the J2 fish by hand four times per day, at a rate of 2-3% of their body weight/day (Comprehensive Hatchery Plan 2007, White Seabass Enhancement Plan 2010). The Comprehensive Hatchery Plan (2007) states that most of the juveniles are fed Skretting pellets 4.0 mm but this information is outdated (M. Drawbridge pers. comm.). Further, there is no SOP for the feeding of juveniles in raceways to document the change in feeding protocols.

1.5.1.4. Nutritional considerations for fish in growout pens.

The Growout Procedures Manual (2007) states that fish in growout are fed an artificial, dry pelleted, high protein diet ("Marine Grower" manufactured by Skretting of Vancouver, Canada), and that three sizes are used (2.5, 4.0 and 6.0 mm) for fish of varying sizes (<10.2 cm receive 2.5 mm pellets, ≥10.2 cm receive larger pellets). However, this information is outdated and needs updating (M. Drawbridge pers. comm.) Normally, if there is a large size variation among fish, then the smaller pellet size is used to prevent starvation of the smallest fish. The automatic feeding system should be checked on a daily basis to verify feed level and to adjust or repair feeders as needed. The amount of food dispensed should be recorded daily in a logbook (Growout Procedures Manual 2007). There is no SOP for the feeding of fish in growout facilities. Therefore, any modification to the details of these procedures cannot be determined.

1.5.1.5. Food storage procedures.

HSWRI has developed detailed SOPs for storing and handling frozen food, pellet food, and vitamins at Mission Bay and the net pen sites (HSWRI Fish Health Management Plan 2016). Frozen food storage and handling protocols include maintaining freezer temperature at or

around -23°C (at Mission Bay) or -28°C (at other facilities), tightly sealing bags and boxes of feed, only purchasing enough food to last two months, and thawing food under cool running water or in a refrigerator (for up to 48 hours) at a temperature of ≤6°C (Frozen Food Storage and Handling SOP 2016, Frozen Feed Thawing SOP 2015, HSWRI Fish Health Management Plan 2016). Pellet food storage and handling protocols include storing feed bags in an air-conditioned feed locker at 15.5°C and ensuring bags of feed are kept for no more than six months (Pellet Feed Storage and Handling SOP 2016, HSWRI Fish Health Management Plan 2016). All food is brought to the hatchery for distribution and shipped to each net pen with the first delivery of fish in two month supplies, depending on biomass and feed rate (HSWRI Fish Health Management Plan 2016). Vitamins (lecithin, vitamin C, thiamin) are stored in 1 gallon bags in cool, dark places, at room temperature (Vitamin Storage SOP 2015).

1.5.2. Data and Information Gaps.

Gaps in knowledge about White Seabass feed include a lack of understanding about the effects of food quality and type, and the interactions among feed, White Seabass and environmental factors. In particular, there is a lack of information about:

1. The extent to which fish quality will be maximized through improvements in early feeding techniques and by maximizing nutritional availability of feed.
2. The extent that deformities are related to feed type, quality and nutrition as compared to genetics or environmental conditions.
3. Updated details on feeding procedures in plans and manuals, and a lack of SOPs for feeding juveniles in raceways and growout facilities.

1.5.3. Recommendations.

Both research and improved protocol and record keeping are recommended to ensure that fish in the hatchery and growout operations have high quality, nutritious food supplies. In particular:

1. Continue nutritional research to improve the understanding of:
    a. Factors contributing to higher quality of live feed, and how feed type, quality and nutrition in turn influences White Seabass health (occurrence and types of deformities), growth and fecundity.
2. Improve record keeping of feed trials and protocols, such as
    a. Continue to keep records of feed trials and nutritional profiles in relation to deformity rates, track links to deformity types, and place a priority on frequently analyzing and publishing the results (e.g., Annual Reports, white paper).
    b. Update the feeding and nutrition protocols in the Comprehensive Hatchery Plan (2007), the Growout Procedures Manual (2007), and associated SOPs, and create SOPs

for feeding juveniles in raceways and growout to ensure that the most current procedures are available, and to update or remove outdated tasks (e.g., no current Vitamin C supplementation, outcome of testing Ascorbyl palmitate as a source of Vitamin C, outdated information on dry pellet diet used at growout sites).

1.6. Growth and health: Steps taken to monitor growth and health of fish.

1.6.1. Key Findings.

The regular sampling of fish according to HSWRI's Fish Health Management Plan and other Standard Operating Procedures is important and required to maintain healthy fish, to assess the outcomes of rearing practices, and to prohibit the spread of disease. HSWRI staff and affiliates, including veterinarians and other fish health professionals, such as CDFW pathologists, are involved with the monitoring of the growth and health of hatchery-reared fish.

The growth of each stage is monitored both regularly and occasionally on designated data collection days, and during other assessments that require handling and/or observation of the fish (e.g., broodstock rotation, quality assessment or control). Similarly, health exams can occur in conjunction with growth measurements, or for the sole purpose of a health checkup. Protocols for growth and health monitoring of broodstock and reared fish are as follows:

1.6.1.1. Growth of broodstock.

Broodstock growth is measured when fish are handled during broodfish rotation among pools, which occurs infrequently, or when the broodstock population is "processed," as seen in 2014 when three broodstock pools were evaluated and some broodfish were culled while others were rotated between pools (Annual Report 13-14). The regular rotation of male broodfish between pools, originally proposed in 1995 (Bartley et al. 1995), was never fully implemented, and was discontinued for logistical reasons (Broodstock Management Plan 2011; see Section 3.2.1.1). Broodfish health issues, including deformities or treatments for pathology, are inventoried monthly (Broodstock Monthly Routine SOP 2015).

1.6.1.2. Growth of larvae.

A number of measurements are taken to mark larval growth and health conditions. At -2 dph, hatching beakers are set up by collecting 100 eggs from a crop and dividing them equally into 10 beakers filled with water from the main incubator sump; at 0 dph, the number of live larvae in these beakers is recorded and used to estimate hatching rates for that crop (Egg Data Collection SOP 2016). At 0 dph, the notochords of 20 larvae from each tank are measured, and 50 larvae from each tank are dry weighed (after being dried in the oven for 24 to 36 hours) (Day Zero Data Collection SOP 2016, Egg Data Collection SOP 2016). Survival beakers are also set up at 0 dph: 100 larvae are collected from the crop and divided equally into 10 beakers filled with water from the main incubator sump; every day thereafter, the number of live larvae in each beaker is recorded, until there are 0 live larvae left (Day Zero Data Collection SOP 2016). Survival beakers are used to

gain insight into the amount of nutrients larvae have stored in their yolk sacs (Day Zero Data Collection SOP 2016). Longer survival times point to lots of nutrient storage, while shorter survival times indicate weaker spawns (Day Zero Data Collection SOP 2016). The method used for collecting a sample of larvae at this stage - using a beaker to take a scoop of water and larvae from the fastest moving area of the tank until the desired number of larvae is collected (Day Zero Data Collection SOP 2016) - is not truly random due to the focused selection of 50 small larva, however it is sufficient. If improvements to the sampling methods are desired, a random sample could be achieved by dividing a truly random net full of fish in half multiple times until roughly 50 larvae remain. The sample size would then be the actual fish count. This would yield a more accurate average than selecting 50 fish from a larger group.

At 1 dph, the above growth measures are repeated, but the methods used to collect a representative sample from the incubator tanks are different. Instead of scooping larvae from a rapidly moving section of the tank, HSWRI lowers a narrow length of pipe into the tank at three different locations, capturing a core of larvae within the pipe and emptying it into a beaker (Day One Data Collection SOP 2015). Once again, the notochord lengths of 20 fish and the dry weights of 50 fish per tank are taken (Day One Data Collection SOP 2015).

Dry weights are also taken for 50 individuals from each tank at 5, 11, and 19 dph (Day Zero Data Collection SOP 2016). At 4 dph, larvae are assessed for swim bladder inflation (Swim Bladder Inflation (SBI) Rates at 4 DPH SOP 2016). If less than 70% of a tank's swim bladders have inflated, that tank may be euthanized (Swim Bladder Inflation (SBI) Rates at 4 DPH SOP 2016). At 18 and 20 dph, 20 larvae per tank are checked for flexion (when the notichord bends upwards and allows for the caudal fin to develop) (Flexion Checks at 18 and 20 DPH SOP 2016). 90% of larvae in each tank should undergo flexion before being transferred to J1 (around 21 dph), as flexion and transport are both stressful for larvae (Flexion Checks at 18 and 20 DPH SOP 2016).

Dry weighing is carried out by collecting and euthanizing 50 larvae, pouring them from a beaker into a suction flask which pulls the water through a pre-dried filter paper and leaves the fish on the paper, placing the paper with the fish onto a pre-dried tin, and putting it into the oven for 24 to 36 hours (Day Zero Data Collection SOP 2016). Taking dry weights in salt water, without rinsing fish in freshwater before drying, may introduce error if there are variable amounts of salt on the fish after drying, however the current protocol for dry weighing is sufficient for now.

1.6.1.3. Growth of juveniles.

While fish are 24 - 40 or 50 dph, or until they reach about 1 g, about 25-50 individuals from each pool are euthanized and weighed wet each week (M. Drawbridge email to T. S. Talley, 15 July 2016). Once fish are 40-50 dph, or about 1 g, three samples of 50 fish are collected from each pool and weighed live each week (M. Drawbridge email to T. S. Talley, 15 July 2016). Quality assessment/quality control (QA/QC) measures are also conducted on fish at 50 and again at 80 dph by selecting 125 individuals per crop, examining them for malformations, and recording total length (TL), standard length (SL) and weight of 50 of the surveyed fish (Quality Assessments for OREHP: 50 & 80 dph SOP 2015). If needed, fish are culled during these checks. See Section 1.6.1 for further discussion of HSWRI's QA/QC protocols.

When fish are 80-100 dph, or about 10-15 g each, 25 individual live weights per pool are measured on a weekly basis (M. Drawbridge email to T. S. Talley, 15 July 2016). Tagging occurs at about this size (90-110 dph), with little culling occurring after tagging, and with transport to growout occurring within 2-3 weeks after tagging.

Pre-transport quality assurance measures. When fish are 110-150 dph, a quality assessment for malformations is conducted on 100 fish per tank according to HSWRI's QA/QC Manual (2011) (Quality Assessments for OREHP: Pre Transport Assessment SOP 2015). Tag retention is measured for all 100 individuals, and TL, SL, and weight are measured for 50 individuals (Quality Assessments for OREHP: Pre Transport Assessment SOP 2015); however, if no malformation-trained personnel are available, then only measurements are taken (M. Drawbridge email to T. S. Talley, 15 July 2016). A health inspection by a CDFW pathologist also occurs at this point, within two weeks prior to fish being transferred to growout.

1.6.1.4. Growth of growout fish.

According to the Growout Procedures Manual (2007), size and weight measures of fish in growout should be collected no more frequently than once per month, due to the stress it causes to the fish (Growout Procedures Manual 2007). This suggests that there is no set schedule for monitoring growth of fish in growout. The Growout Procedures Manual (2007) stipulates that the Growout Facility Coordinator (GFC) must be present at the growout site when fish are measured and weighed, so as to reduce error, ensure proper handling protocol is employed, and administer an anesthetic to fish when necessary. To estimate the growth rate of fish in a growout facility, a subsample of fish should be taken using a crowding device (Growout Procedures Manual 2007). TL is taken for each fish (to the nearest 1.0 mm), and then used to estimate weight if weights cannot be taken directly, according to a length-weight relationship like that shown in Fig. 1.3 (M. Drawbridge pers.comm.). If desired, the GFC can also anesthetize and weigh the fish (Growout Procedures Manual 2007). In general, White Seabass held at growout facilities grow at a rate of 0.2-1.3 mm per day (0.2-1.5 inches per month) (Growout Procedures Manual 2007). Although most of the growout site summaries included in HSWRI's annual reports mention an average TL for fish released, it is unclear how frequently growth measurements are taken at individual growout sites.

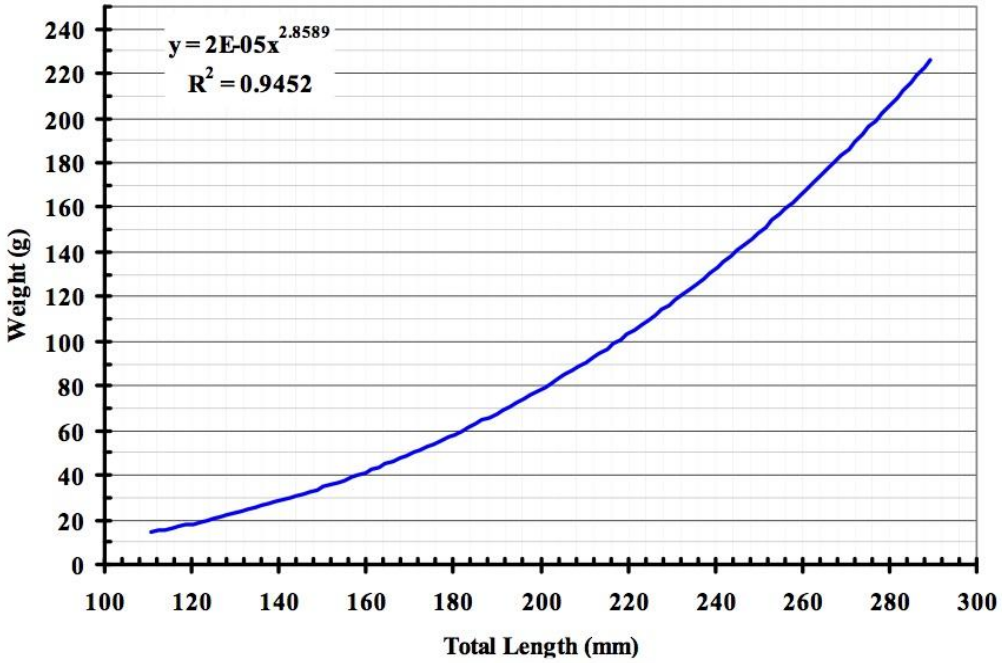

Fig. 1.3. Relationship between length and weight for juvenile White Seabass. One inch = 25.4 mm; One ounce = 28.35 g. Figure from M. Drawbridge email to T. S. Talley, 31 January 2017a. Biomass is calculated by multiplying the number of fish released by the average weight of a subsample of fish taken just prior to release

Pre-release quality assurance measures. One hundred fish per pen are selected for quality assessment before being released from growout facilities, often in conjunction with final count procedures, which occur 1-2 weeks before release (Quality Assessments for OREHP: Pre Release Assessment SOP 2015; see Section 4.1.1 for release procedures). These one hundred individuals are scanned for tag retention and checked for malformations according to HSWRI's QA/QC protocols (2011) (see Section 1.9.1), and 50 individuals are measured for TL, SL, and weight (Quality Assessments for OREHP: Pre Release Assessment SOP 2015). The growout coordinator also takes 96 fin clips at this time (Quality Assessments for OREHP: Pre Release Assessment SOP 2015). A health inspection by a CDFW pathologist also occurs at this point.

1.6.1.5. Growth of released fish.

Growth is measured by comparing size and weight at tagging upon leaving the hatchery, with size and weight when fish are recaptured.

1.6.1.6. Disease surveillance and detection.

Health assessments are conducted to maintain healthy fish and to prevent the spread of lethal or debilitating contagious pathogens both within the hatchery and outside of the hatchery to wild fish stocks after out-planting efforts. There is a set of protocols, *Health Assessment Protocols for Diseases of Concern Associated with the Culture and Release of White Seabass (Atractoscion nobilis) by OREHP* that is being jointly developed by CDFW and HSWRI (draft in review by CDFW). HSWRI staff routinely (daily) performs assessments within the hatchery.

Fish health inspection begins with observation of behavior: whether there are any abnormalities in swimming (spinning, flashing, flared gills), eating, or any obvious lesions (HSWRI Fish Health Evaluation SOP 2016). Fish health exams can either be done in conjunction with other sampling efforts (for size and weight measurements, for example), or for the sole purpose of a health checkup (HSWRI Fish Health Evaluation SOP 2016, Broodstock Disease Screening Protocols SOP 2016). A standard exam includes skin scrapes and gill clips or scrapes (gill clips are easier for smaller fish and either gill clips or scrapes can be done for larger fish) (HSWRI Fish Health Evaluation SOP 2016). Any lesions observed should be sampled, and any grossly visible external parasites (on the skin or in the gills) should be collected and processed according to the Fluke and Copepod Sample/Submission Protocols (HSWRI Fish Health Evaluation SOP 2016, Fluke Sample/Submission SOP 2016, Copepod Sample/Submission SOP 2016). Gills are first assessed visually (bright red gills are healthy, whereas pale gills suggest anemia) and then clipped or scraped (HSWRI Fish Health Evaluation SOP 2016). All samples taken should be processed as quickly as possible, photos should be taken whenever possible, and all results should be passed along to the Fish Health Specialist (HSWRI Fish Health Evaluation SOP 2016).

Health inspections of White Seabass are conducted two times by a CDFW pathologist, once at the hatchery prior to transport to a growout facility and once at the growout facility prior to release (CDFW Release Criteria 2015). Once the pathologist inspects and clears the fish of any disease, the fish may be moved or released within a two-week time frame after passing inspection, unless a new health concern arises (CDFW Release Criteria 2015).

Though broodfish behavior is observed and evaluated daily, broodfish usually undergo physical health examinations on other rare occasions when fish are handled (Broodstock Daily Checklist SOP 2015, Broodstock Disease Screening Protocols SOP 2016, Broodstock Handling and Weight Sample SOP 2016). Broodfish are handled as infrequently as possible, as it is very stressful for fish (Broodstock Handling and Weight Sample SOP 2016). During these examinations, fish are evaluated for all pathogens, especially those with epizootic potential and those that can be transferred vertically to their spawns (diseases that can occur in salmonids through vertical transmission of a pathogen include bacterial kidney disease, infectious pancreatic necrosis (IPN), Coldwater disease caused by *Flavobacterium psychrophilum*, and Piscirickettsiosis) (Broodstock Disease Screening Protocols SOP 2016).

Juvenile White Seabass in the hatchery are inspected by HSWRI staff when there is a sharp increase in mortality (Fish Mortality Classification SOP 2016), and when hatchery personnel note abnormal behavior or high frequency of gross lesions during routine observations of fish (HSWRI Fish Health Evaluation SOP 2016, Comprehensive Hatchery Plan 2007). Inspections involve daily observations of body condition (lesions or other signs of disease), feeding activity and behavior when not feeding (HSWRI Fish Health Management Plan 2016), and in particular include (1) the review of daily mortality logs; (2) the observation of fish in their home pool or raceway; (3) selection of the appropriate fish to examine (e.g., exhibiting reported behavior, with lesions); (4) necropsy (euthanasia, gross external examination, dissection, and gross internal examination); and (5) wet mount cytology, when fish have open skin lesions and/or when gross inspection does not reveal a definitive diagnosis (Comprehensive Hatchery Plan 2007). Exams, necropsies, and

cytology are performed in-house at the hatchery (Comprehensive Hatchery Plan 2007). The Comprehensive Hatchery Plan (2007) also states that initial bacterial and fungal isolations are also performed in-house, but this is no longer the case (M. Drawbridge pers. comm.); identification of bacteria and fungi is conducted at commercial laboratories or the University of California at Davis (UCD).

Necropsy for disease detection. HSWRI's Fish Necropsy Protocol SOP and Necropsy Procedure SOP (2016) are general protocols covering all HSWRI aquaculture endeavors. Necropsies for OREHP fish in particular are performed by HSWRI trained staff in many situations, as part of routine checks on healthy fish (checks as part of the White Seabass in the Classroom are somewhat random and prior to movement). The staff brings a concern or question to HSWRI's clinical veterinarian upon observation of a questionable clinical sign or in the event of a mass mortality (C. Silbernagel, pers. comm. as conveyed in M. Drawbridge email to T. S. Talley, 15 February 2017). The numbers sampled vary depending on each clinician's medical judgment of risk assessment, clinical signs evident, morbidity, if fish are being transported to growout sites, net pens, or release sites, and time needed to make an accurate diagnosis (C. Silbernagel, pers. comm. as conveyed in M. Drawbridge email to T. S. Talley, 15 February 2017). The CDFW fish pathologist performs necropsies on fish before they are transported for growout or release (the HSWRI clinical veterinarian might perform one additional necropsy), and is also notified and invited to sample fish in the event of health concerns that may involve something infectious that may require treatment due to mortality (C. Silbernagel, pers. comm. as conveyed in M. Drawbridge email to T. S. Talley, 15 February 2017). The CDFW pathologist is notified immediately if a reportable, or question of a reportable, infectious disease is identified (C. Silbernagel, pers. comm. as conveyed in M. Drawbridge email to T. S. Talley, 15 February 2017).

HSWRI's course of action in the event of high mortality rates is as follows: If mortality occurs at a rate of 0.3-0.5% per day, the cause of mortality should be investigated; if mortality occurs at a rate of 0.5-1.5% per day, the clinical veterinarian should be called in for consultation; if mortality is ≥1.5% per day, immediate attention is required to solve the problem and save the remaining fish (Fish Necropsy SOP 2016). Juvenile fish are examined externally, dissected, and examined internally to perform health assessments (Fish Necropsy SOP 2016).

During an external exam by trained HSWRI staff, the skin and fins of the euthanized fish are examined for hemorrhage, fraying, erosion, ulceration and/or changes in pigmentation, which are symptoms of bacterial, protozoan and metazoan infections. The head and eyes are examined for cannibalism, deformities, signs of emphysema and lesions, with the lesions classified by type (e.g., EX = exophthalmia; CE = corneal emphysema; GAS = intraocular emphysema) and severity (0-4, "not present" to "massive") (Comprehensive Hatchery Plan 2007). Gills are examined for unusual color, lesions, filament condition; and jaws, gums, oral cavity and tongue are also inspected once the gill operculum is removed. The body of the fish is also measured and weighed, and wet mounts for cytology are made as needed (e.g., cytology is used when open skin lesions are present) (Fish Necropsy SOP 2016).

During an internal exam by HSWRI, the heart, gastro-intestinal (GI) tract, liver, spleen, swimbladder, kidney, urinary bladder are examined. The left lobe of the liver and stomach are used to visualize the right liver lobe, gall bladder, and pancreatic megaislet. If enteritis is suspected, the GI tract is opened and examined for mucosal lesions (Comprehensive Hatchery Plan 2007). When screening for VNNV or Central Nervous System (CNS) disease, the brain and eyes are also examined and otoliths removed. (When larvae are screened for VNNV, the whole fish is used). If a definitive diagnosis is not reached with the necropsy and gross examination of tissues, addition diagnoses such as cytology and histology are used (Comprehensive Hatchery Plan 2007).

While these disease testing protocols are established (and strict), they may not address all pathogens that may affect White Seabass. A lack of information on diseases in wild White Seabass populations and changing climate conditions (e.g., warmer water) may result in new and/or novel pathogens that are difficult to diagnose or detect without the appropriate diagnostic tools (e.g. specific cell lines for viral pathogens). See Section 1.8 for more information on pathogens affecting hatchery-raised White Seabass.

1.6.1.7. Deformity detection.

The first quality assessment for malformation is conducted by HSWRI at about 50 dph (Fig. 1.4) when a compound microscope is required as fish are still quite small (later quality assessments can be conducted visually without a microscope) (HSWRI QA/QC Manual 2011, Quality Assessments for OREHP: 50 & 80 dph SOP 2015). Quality assessments are typically conducted by both HSWRI and CDFW before transporting fish to growout facilities, and before releasing fish into the wild in order to understand how quality might change during growout (Quality Assessments for OREHP: Pre Transport Assessment SOP 2015, Quality Assessments for OREHP: Pre Release Assessment SOP 2015). CDFW and HSWRI have developed and use their own separate sets of protocols to detect, rank, and deal with deformities in hatchery-reared White Seabass. While both of these sets of protocols lack external peer review, HSWRI's protocols were reviewed and approved by CDFW. The discrepancies between these protocols result in inefficiencies in the program. Furthermore, there are few data from the White Seabass program and other stocking programs that justify such rigorous deformity screening protocols. Both of these concerns are elaborated on in Section 1.9.1; here, the two different protocols are simply described.

HSWRI deformity detection protocols. HSWRI relies on the expertise of the HSWRI staff veterinarian, trained staff, and one protocol developed within HSWRI*, Procedures Manual for Quality Assessment and Control of Marine Finfish Cultured for Stock Replenishment* (HSWRI QA/QC Manual 2011), which informs HSWRI's handling of Seabass that do not meet quality standards. This protocol is a self-imposed control measure meant to ensure that high quality fish are released. The manual has been informally peer-reviewed through HSWRI staff presenting protocols to U.S. and international colleagues and incorporating feedback. The HSWRI QA/QC Manual (2011) has also been reviewed by an Adhoc QA/QC Committee made up of CDFW staff members (V. Taylor email to T. S. Talley, 27 March 2017). It deals specifically with malformations that can be identified through external examination, including bony and soft tissue deformity.

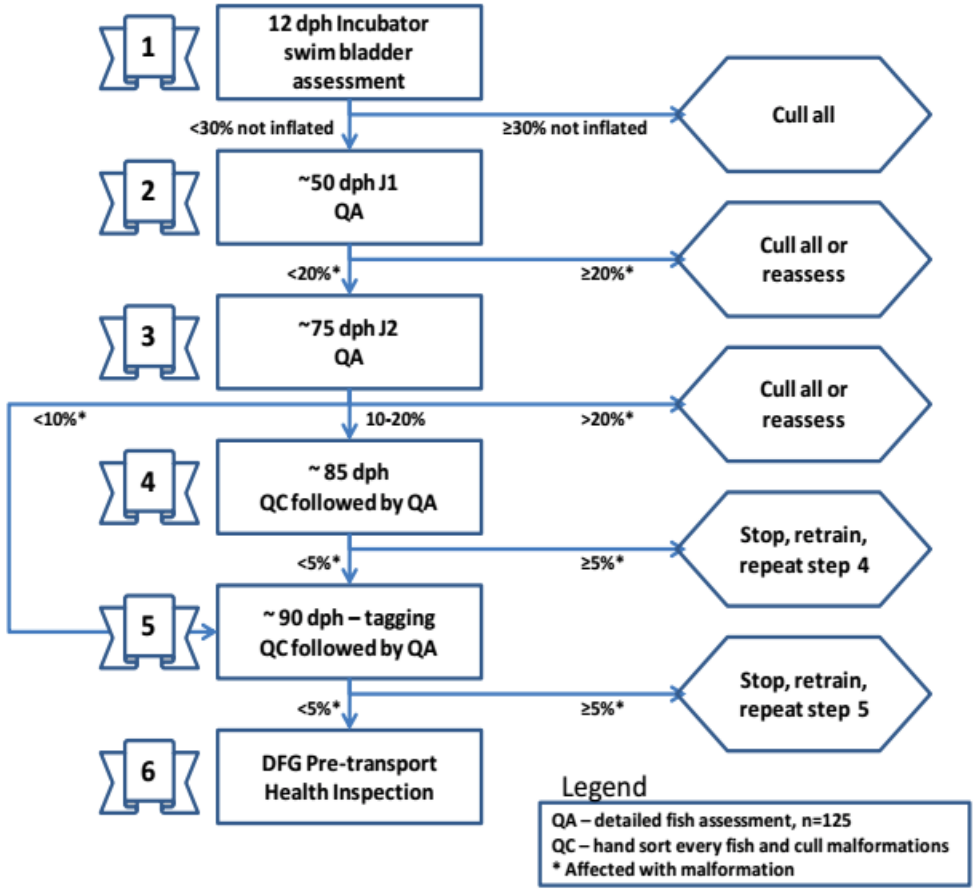

Fig. 1.4. Decision tree for Quality Assessment/Quality Control procedures. Figure included in the HSWRI QA/QC Manual 2011, and taken from Annual Report 11-12.

The stated ultimate goal of HSWRI quality control is to ensure that 95% of the fish leaving the hatchery are in either perfect physical quality or almost perfect physical quality, with minor physical differences that are seen as variations on normal attributes observed in wild White Seabass (HSWRI QA/QC Manual 2011). The criteria for culling are, therefore, based on data collected on the range of variability among wild fish to help distinguish hatchery specific deformities from what may be a part of "natural" variability (HSWRI QA/QC Manual 2011). It calls for the routine assessment of a randomly selected subset of hatchery fish large enough to be representative of the whole population, and quality control procedures, including the sorting and culling of affected fish (HSWRI QA/QC Manual 2011, See Fig. 1.4).

During HSWRI quality examinations, each fish's body, fins, and mouth are examined for 20-60 seconds (HSWRI QA/QC Manual 2011). Each fish is ranked as Grade 0 (normal), 1, 2, or 3. Decisions about whether to sort or cull are made on fish Grades 1-3 or 2-3, depending upon the deformity in question (HSWRI QA/QC Manual 2011). Quality control is an intensive process that involves examining each individual fish, and culling poor quality fish (HSWRI QA/QC Manual 2011, Sorting White Seabass for OREHP SOP 2016). Quality control occurs only when quality assessment

findings indicate that it should, prior to tagging or during the tagging process (HSWRI QA/QC Manual 2011).

The HSWRI QA/QC Manual (2011) also provides details on different malformations affecting hatchery fish, describing the appearance of the malformation, its probable cause, its prevalence among hatchery fish, as well as the life stage at which the malformation first impacts fish, and the severity of the malformation (HSWRI QA/QC Manual 2011). The manual includes images of normal fish, fish with different grades of malformation for reference, and examples of wild fish morphologic variability (HSWRI QA/QC Manual 2011). These images inform the action taken during quality control, whether it be no action, sorting out, or euthanization (HSWRI QA/QC Manual 2011, Sorting White Seabass for OREHP SOP 2016). The manual is supposed to be updated yearly with new information about malformations and treatment strategies (HSWRI QA/QC Manual 2011).

CDFW deformity detection protocols. CDFW relies on the expertise of the CDFW Senior Fish Pathologist and protocols developed within CDFW that have not been externally peer reviewed: (1) *Cultured White Seabass Deformity Report Protocol* (2015), and (2) a series of summary reports entitled "*Deformities in Cultured White Seabass*," each developed upon the discovery of a new deformity, that photographically illustrate the condition and provide diagnostic and severity scoring criteria (e.g., CDFW Deformity Summary Reports 2013, 2016). These protocols address internal and external, soft and hard tissue, malformations of juvenile fish. CDFW's *Cultured White Seabass Deformity Report Protocol* (2015) outlines procedures for sampling and diagnosing juvenile fish, and preparing reports. CDFW assessments are made based on deviations from "normal" hatchery fish anatomy, which is the anatomy of the "best hatchery fish available" (CDFW Cultured White Seabass Deformity Report Protocol 2015). The best hatchery fish resemble typical healthy wild type fish, which are photographically illustrated in the *Necropsy of the Adult White Seabass* Volumes I and II (CDFW Necropsy of the Adult White Seabass 2013a,b), and the deformity summary reports (CDFW Deformity Summary Reports 2013, 2016). The 2015 revision of CDFW's *Cultured White Seabass Deformity Report Protocol* calls for random sampling of fish when possible, which varies from past protocols that called for sampling as many slow or otherwise irregular looking fish as feasible (10 – 100 per tank). Typically, the CDFW Senior Fish Pathologist conducts targeted sampling of fish, removing moribund or weak fish, or fish with grossly visible lesions, from pools in order to diagnose the underlying condition (M. Okihiro email to T. S. Talley, 21 April 2016). If none of the fish are overtly sick or deformed, then the CDFW Senior Fish Pathologist nets out a number of fish that can be reasonably assessed in a given time period, taking up to 100 fish from different spawn groups (M. Okihiro email to T. S. Talley, 21 April 2016).

Fish are assessed for deformity based on size class; if fish are small (less than 12 cm TL), then only the most obvious malformations are noted; if fish are medium-sized (12-20 cm TL), a full evaluation can be performed; if fish are large (greater than 20 cm TL), a full evaluation can be completed, including internal assessments whenever feasible (CDFW Cultured White Seabass Deformity Report Protocol 2015). The largest fish produce the most accurate deformity data (CDFW Cultured White Seabass Deformity Report Protocol 2015). Whenever possible, a Complete

Deformity Assessment should be conducted (CDFW Cultured White Seabass Deformity Report Protocol 2015). A Complete Deformity Assessment includes a thorough investigation of any craniofacial malformations, axial skeletal deformities, appendicular skeletal deformities, eye deformities, swim bladder deformities, and intestinal tract deformities (CDFW Deformity Summary Reports 2013, 2016). Malformations are classified using the following 5-point scale, based on extent of deviation from the norm: not present (0), mild (1+), mild to moderate (1+/2+), moderate (2+), moderate to severe (2+/3+), or severe (3+) (CDFW Deformity Summary Reports 2013, 2016). When possible, specific criteria are used to delinate ranking (e.g., supernumerary pyloric cecae). Information is recorded in a lab book, entered into a spreadsheet, and used to create White Seabass Deformity Reports. New and unusual malformations are also photographed (CDFW Cultured White Seabass Deformity Report Protocol 2015). Data are then summarized including percent of fish examined with one or more deformities, number and types of different deformities documented, total number of deformities, and mean number of deformities per fish.

See Section 1.9 for discussion of the implications of having two different sets of deformity protocols.

1.6.2. Data and Information Gaps.

1. Lack of regularity of growth measurements for each life stage.
2. Lack of consistency in the use of two distinct sets of protocols to detect, rank, and address malformation issues in hatchery-reared White Seabass (the implications of which are discussed in Section 1.9).

1.6.3. Recommendations.

1. Investigate the use of non-invasive protocols (e.g., photographs and imaging) to routinely measure growth of fish at each stage, which can be used to assess the outcomes of routine practices, unforeseen changes, and planned trials.
2. Update Comprehensive Hatchery Plan (2007) to reflect current disease detection protocols.
3. Conduct research and develop protocols to more easily detect deformity issues in younger life stages.

1.7. Fish releases: Quotas and rationale surrounding releases.

1.7.1. Key Findings.

The considerations behind White Seabass release quotas and releases include fish size (i.e., trade-offs among fish post-release survival, caged fish health, and costs), release timing, fish health, broodstock limitations, and limits on allowable hatchery genetic inputs into the wild population.

1.7.1.1. Considerations for release strategies.

Research from the White Seabass program (e.g., Hervas et al. 2010) and other hatchery-based-enhancement, -restoration, and -conservation programs (e.g. striped mullet *Mugil cephalus*, Pacific threadfin *Polydactylus sexfilis*, red drum *Sciaenops occellatus*, white sturgeon *Acipenser transmontanus*) suggests that larger release sizes result in higher survival of hatchery fish. Survival of White Seabass to the size permissible for inclusion in the fishery (71 cm) increased from less than 1.5% to just over 13% when size at release was over 40 cm (Hervas et al. 2010; see Section 4.4.1 for a more detailed discussion of study findings). However, there are trade-offs with growing out fish to larger sizes. Effects of size at release are somewhat confounded with caging effects and may not always relate to survival success. For example, pathogens in the environment and any stress may increase with time in captivity and, therefore, lead to greater susceptibility to a disease outbreak. Further, relationships between size at release and survival are dependent upon the capacity and abilities of the growout facilities. HSWRI researchers have also been evaluating the effects of the timing of releases on survival and growth of stocked fish, with higher survival for fish released in spring, followed by summer and fall (Hervas et al. 2010). The effectiveness of release size and timing on fish survival and contributions to the wild population should be assessed using HSWRI empirical data and a model (K. Lorenzen pers. comm.). As with release size, release strategies are somewhat dependent on volunteer growout programs.

Another trade-off of larger size at release is that larger fish are more costly to grow in terms of food and pen requirements, and expenditures over a longer period of time. The most cost effective size at release in a stock enhancement program is that size at which hatchery-growout-and-release costs are minimal for a stocked fish to recruit to the adult population in the wild. Leber et al. (2005) considered optimal size at release to minimize cost as a function of both (1) hatchery costs to rear various sizes of fish and (2) how release size influences hatchery fish natural mortality, and thus recapture rate, after stocking into the wild; the optimal size at release to minimize cost can be determined when the marginal cost of increasing one unit of recapture rate by increasing release size is equal to the average cost (of hatchery fish) at that size (details of the calculations are given in Leber et al. (2005)).

Natural mortality after stocking is affected by ecological factors, such as predation (Brennan et al. 2006), and by disease and deformity problems at growout sites and the resulting impact those have on post-release mortality. Stocking fish with deformities may contribute to post-release mortality as well, and the extent of deformities may be a function of size at release. Leber et al. (2005) did not distinguish natural mortality from the latter effect (size at release-mediated deformity effects on post-release mortality). If feasible, the effects of particular deformities, or features in question, on post release mortality of White Seabass could be evaluated by establishing separate CWT codes for any stocked fish with the feature in question in order to distinguish them from the “normal” hatchery fish stocked. A similar analysis for White Seabass would reveal optimal size at release points.

The health of the fish, specifically whether the fish are free of pathogens and disease, is a criterion for their release to prevent the spread of disease from hatchery fish to wild populations. Diagnostic criteria for each of six characterized diseases of concern pathogens are listed in *Release Criteria for Cultured White Seabass (Atractoscion nobilis)* (CDFW Release Criteria 2015). The six known pathogens are considered to have potentially serious impacts on White Seabass because they are either presently non-treatable, debilitating, highly contagious, associated with high rates of mortality, or otherwise known to be detrimental to wild fish stocks. The presence of any of these pathogens would disqualify fish from release as discussed in more detail in Section 1.8.

1.7.1.2. Quota and associated rationale.

The annual release quota is determined on a sliding scale, depending on the number of broodstock that are currently kept at the hatchery. This release quota is reassessed every six months (Broodstock Management Plan 2011). A goal of 200 broodfish, with a maximum limit of 350,000 fish released per year, was set at the start of the program (Bartley et al. 1995). At that time, it was assumed for generations that mating ratios were one male to one female, but it was uncertain how many fish actually contributed to a spawning event. It is now known that many males may mate with a single female and that several females may have disproportionate contributions to each spawning event, therefore, limits were more recently set at about 12,000 individuals per female equivalent, *fe* (Broodstock Management Plan 2011). The quota rationale, which persists as a special condition placed on the net pen facilities, is under evaluation. The Broodstock Management Plan (2011) states that there is "no genetic basis for [the] sliding scale," and that the "genetically defensible quota [is] more than 1 million (Gruenthal and Drawbridge 2012)" fish annually, though there was no clear quantitative justification for these claims. Further, even though release size influences post-release survival, there is no consideration of size in quotas (See Section 4.4 for discussion of size at release modeling scenarios).

It is important to note that the maximum annual limit of 350,000 released fish has not been achieved. From July 1996 to June 2015, HSWRI released an average (± 1SE) of about 104,160 ± 14,891 fish per fiscal year (numbers ranged from 15,955 to 278,725 fish per fiscal year within that time period). Release numbers may be lower than the goal of 350,000 juveniles in part because HSWRI has never held 200 broodfish at the hatchery (although, they have had more than 190 broodfish on 6 occasions from 1996 to 2015), and thus have had maximum release quotas lower than 350,000 each fiscal year. If the maximum number of released fish is scaled to the size of the broodstock pool each year, then the number of released juveniles have, on average (± 1SE), amounted to 33.9±4.8% of their annual release quotas between July 1996 and June 2015. Therefore, it is clear that low release numbers are not just due to lower annual release quotas. Shortfalls within any year have been largely due to one or more of the following: the physical system capacity (e.g., tank or raceway space), replenishment of broodstock, the survival and growth of juvenile fish, feed challenges, disease, deformity, or genetic quotas (see Sections 1.2.1.1 and 1.3.1 for a more detailed discussion on bottlenecks in production; Comprehensive Hatchery Plan 2007, Annual Reports). The program currently produces around 1 billion eggs a year, but disposes of hundreds of millions of viable eggs (e.g., Annual Reports 05-06, 07-08); if the

broodstock genetic considerations were addressed (see Chapter 3), and rates of juvenile survival were improved, the hatchery could potentially produce and release many more fish annually. Importantly, an increase in post-release survival would reduce the potential for family-specific survival and domestication selection, and therefore alleviate some of the genetic concerns listed under Objective 3 (See Section 4.4 for post-release survival modeling scenarios).

1.7.2. Data and Information Gaps.

1. Lack of data and information surrounding release quotas, including explanation for release quota shortfalls year to year; rationale for understanding release strategy, site, size, and timing; and updates to release quotas to reflect hatchery and growout facility capacities and abilities.
2. Empirical data on post-release survival are needed, in particular needed are more data on how survival relates to release size, timing, and confounding caging effects.
3. Lack of consideration of fish size in relation to production quotas; i.e. it's cheaper and easier to release more small fish but what is post-release survival, and moreover, what size at release is optimal? Once that is understood, production quotas could be set in terms of numbers of fish.
4. No social and economic cost-benefit data on size at release.

1.7.3. Recommendations.

1. Identify and incorporate ways to better determine target size at release including performing an experimental analysis of the cost effectiveness of size at release to determine optimal size at release (e.g., as in Leber et al. 2005). This involves identifying and comparing rearing costs with recapture rates and relative yields in the fishery from hatchery fish stocked at various size at release increments (see Leber et al. 2005, Section 4.4.3 Recommendation 2).
2. Apply adaptive management, not only to selection of stocking strategies, but to updating plans and SOPs (e.g., growth curve in Growout Procedures Manual 2007), conducting annual assessments of quota shortfalls including reasons behind such shortfalls, seeking solutions for common problems and, if allowable, seeking to revise quotas as conditions change to reflect the capacity and ability of the hatchery and growout facilities.

1.8. Pathology: Effects of pathology on hatchery operations and releases, and how pathology challenges have been addressed.

1.8.1. Key Findings.

1.8.1.1. Pathogens and disease.
Diseases and pathogens are common, well-documented challenges in any aquaculture operation and are often linked to conditions that are stressful for the fish, such as physical conditions (e.g., temperature, oxygen) that are outside the physiological optimum of the fish, and high densities that predispose fish and increase susceptibility to infections. In addition, water quality and other environmental problems can increase disease susceptibility, especially in culture stages that rely on raw (untreated) seawater (i.e., outside of the hatchery). Types and effects of different diseases are well known in other well-established stocking programs (i.e. salmon supplementation programs in the Northwest). Such programs focus primarily on preventing and controlling specific pathogens of concern to limit the impact on the survival of hatchery fish or pathogen transfer to wild stocks. Non-salmonid programs (e.g. red drum, burbot, etc.) also focus on specific pathogen screening prior to release; screening for non-infectious diseases is rarely implemented.

Viral pathogens such as viral nervous necrosis virus (VNNV) and other pathogens have historically impacted the White Seabass program but have in most cases been effectively managed in the hatchery following the installation of ozone treatment of all make-up water in 2003 (Comprehensive Hatchery Plan 2007, Annual Report 02-03). Further, strict disease testing protocols are established (HSWRI Fish Health Management Plan 2016 and SOPs within including HSWRI Fish Health Evaluation SOP 2016, Fish Necropsy SOP 2016, Infectious Disease Emergency SOP 2016, Histopathology Tissue Sample Collection/Submission SOP 2016). Despite this, there continue to be challenges with pathogens that infect White Seabass or other non-infectuous diseases (e.g. gas supersaturation). Prognosis of survival and risk of spread after contraction of an infectious disease depends upon the pathogen's virulence, time until treatment, and the effectiveness of the treatment. Certain diseases are eliminated by euthanasia of infected and other exposed individuals, while other disease problems, such as many forms of dermatitis and external parasites, are treatable (e.g., hydrogen peroxide treatment) (see Section 1.10.1.1).

Occurrence and types of diseases. As with other aquaculture facilities, infectious and non-infectious diseases and pathogens have been a frequent challenge at the hatchery and growout facilities throughout the program. Some of these diseases and pathogens have been novel and/or rare, appearing only occasionally, and resolving after treatment or euthanasia (e.g., fungal infections, unidentified sporozoan parasites; Annual Reports 06-07, 07-08, 10-11, 11-12, 12-13, 13-14). Many of the diseases and pathogens recur to some degree within and between years and can affect one or more stages of White Seabass. Examples of diseases encountered over the past ten years are listed here (as per CDFW Pathology Reports Summaries 2006-2016 and references listed below):

Broodstock. Disease problems have included *Miamiensis avidus* exposure and/or infections that caused meningoencephalitis, olfactory neuritis, and rhinitis, and/or euthanasia (SFRA Report 15-16). Exposure and sometimes infection by herpesvirus has led to gastroenteritis and/or euthanasia (SFRA Report 15-16). Instances of nephrocalcinosis, renal failure, cerebral and ulcerative dermatitis, osmotic shock (e.g., due to loss of scales), and euthanasia due to VNNV SN+ have also occurred, although VNNV has been effectively managed. Deaths from causes other

than disease also occasionally occur, including anoxia/hypoxia during transport and holding in net pens, predation while in net pens (e.g., sea lion attack), trauma from swimming into the side of the tank and tank bolts, or jumping out of tank.

Larvae and juveniles in hatchery. Among diseases known to affect White Seabass, the hatchery has frequently experienced cases of gas supersaturation, which causes eye conditions such as bubble eye or popeye disease, and has occasionally experienced bacterial enteritis (*Vibrio* spp.), fungal infections leading to dermatitis and ulcers, *Uronema* dermatitis, Ichthyobodo parasite infections, panophthalmitis, sporozoan and other protozoan infections, encephalitis, and meningitis. Anemia and cannibalism have also been reported as other causes of death.

Fish in growout. The inability to control water condition at growout facilities has resulted in frequent cases of gas supersaturation, and less frequent cases of panophthalmitis, ulcerative dermatitis due to bacteria, fungi and/or protozoa, Ichthyobodo parasite infections, sporozoan and other protozoan infections, multicellular ectoparasites (e.g., copepods, flukes), herpesvirus enteritis, and hole-in-the-head disease. Other recent mortality events, although not due to infectious diseases, have been caused by stresses due to high water temperatures, hypoxia and/or transport stress (CDFW Pathology Reports 2015-060, 2015-061, and 2015-062), and predators, especially bird strikes. Discovery of VNNV exposure in wild fish (Curtis et al. 2003) made it so that VNNV exposed hatchery or growout fish could be released. This discovery resulted from the work of multiple national and international researchers, including HSWRI and CDFW staff, working in collaboration to solve this problem.

1.8.1.2. Biological effects of disease.

Despite existing protocols to detect and treat disease (HSWRI Fish Health Management Plan 2016), both unexpected and known infectious and non-infectious disease outbreaks have impacted production and operations at the hatchery and growout facilities, ultimately impacting release numbers.

Non-infectious disease. The most common non-infectious disease is gas bubble disease (GBD), an environmental disease caused by supersaturation of seawater, which is exacerbated during warm water conditions and shallow water rearing environments, that is linked to eye conditions (e.g., bubble eye, popeye) in White Seabass at the hatchery and growout facilities (Annual Report 96-97, Dang 1997, Smiley 2004, Smiley et al. 2011). Gas supersaturation was reported in the hatchery and growout facilities (e.g., King Harbor) in the mid-1990s (Annual Report 96-97, Dang 1997) and has been a recurring challenge every year since then (e.g., Annual Report 05-07, Annual Report 14-15, all existing CDFW Pathology Reports Summaries from 2001 to 2016). The ocular lesions caused by gas supersaturation, and subsequent bacterial and fungal infections and blindness, are untreatable, and therefore require preventative action.

The lack of resolution of this disease results in varying levels of fish losses (i.e., culling) and lack of fitness (if not culled) every year. When coupled with equipment or other environmental challenges, however, this disease can have a significant impact on the production and health of the White Seabass. In fall 2014, for example, 90% (18 of 20) of non-randomly sampled fish (i.e.,

likely a sample of slow moving fish) tested from one of the raceways (R1) had ocular emphysema resulting in the euthanization of 2,700 blind or near blind fish (SFRA Report 14-15). A recently installed recirculation system, put in place to reduce gas supersaturation, helped to correct the problem (SFRA Report 14-15). In the 2015-16 year, warm water conditions due to El Niño and summer stocking, especially at shallow water locations, correlated with more than 70% of non-randomly sampled fish in hatchery raceways R4 and R6 having ocular lesions, likely due to gas supersaturation (SFRA Report 15-16). Ocular lesions were also common among the eight coastal growout facilities monitored. Ocular emphysema (intraocular or corneal) was the primary or secondary diagnosis for 17 of the 25 health assessments performed at the eight growout facilities evaluated (SFRA Report 15-16). Again, sampling of fish for examination likely was not random, and other causes of eye lesions were not investigated, although links between ocular emphysema and GSS are well documented (Annual Report 96-97, Dang 1997, Smiley 2004, Smiley et al. 2011).

Infectious diseases. Most of the infectous diseases found in the hatchery and growout facilities are not new or novel to the system. In the 2015-16 year, for example, there were two outbreaks of Ichthyobodo, a flagellated protozoan parasite, that was first recorded in 2004 in four of the growout facilities (CDFW Pathology Reports 2004-065, 071, 081, 084), and in 2006 when it was also identified in the hatchery (CDFW Pathology Reports Summary 2006). The recent infections in the raceways and during transport from HSWRI in Mission Bay to the hatchery resulted in losses of >36,000 fish that died or were euthanized due to the outbreaks (CDFW Pathology Reports Summary 2006). The outbreaks were ultimately controlled with hydrogen peroxide treatments.

There was also a disease outbreak of *Miamiensis avidus*, a cilitated protozoan parasite, at the hatchery during the 2015-16 year. The first outbreak of *Miamiensis avidus* in the hatchery and in growout (e.g., King Harbor) occurred in 2010 (CDFW Pathology Reports Summary 2010). It reoccurred in two growout facilities in 2011 (King Harbor and Catalina Island-HSWRI; CDFW Pathology Reports Summary 2011) and was undetected until the recent large mortality event occurred in the new-broodstock quarantine system (Q tanks) (CDFW Pathology Reports 2016-005 and 2016-006). The parasites were thought to have originated from the wild caught fish. This outbreak resulted in severe meningoencephalitis, and this form of *Miamiensis avidus* is untreatable and lethal. All remaining broodstock White Seabass in both systems (Q1 and Q2) were euthanized as mandated by CDFW (Q1) and out of caution by HSWRI (Q2) due to a lack of information regarding the disease. The 2015 thermal stress event may have contributed to the *Miamiensis* epizootic given the lack chillers in both the Q1 and Q2 tanks, although fish in Q2 did not test positive.

The hatchery protocols include general steps for prevention (e.g., hatchery water sterilization), quarantine, and treatment at the first indication of infection. There is no way to discern the actual training that the staff receives to assure that disease management protocols are adhered to; a lack of training could potentially contribute to recurring infections and a lack of adaptive management (See Section 5.2.1.1). Furthermore, there is minimal instruction for prevention of commonly recurring diseases in culture stages utilizing raw water (e.g., growout sites), and there is little consideration in the protocols for contingencies, such as avoiding environmental conditions that may increase likelihood of disease (e.g., warm water events).

1.8.1.3. Economic and resource dedication effects of disease.

There are costs associated with prevention, detection and response to disease. The addition of more specific preventative measures, in particular for common recurring pathogens and disease and/or for times when environmental conditions may facilitate outbreaks, need to be assessed in relation to the costs associated with response to disease (e.g., treatment, disinfection and fish losses). Preventative measures may include added or updated equipment (e.g., chillers, degassing columns), changes to methods (e.g., timing of transport or raceway use), and/or preventative treatments or future vaccines.

Costs associated with disease detection would remain in place regardless of changes to the prevention protocols and include staff time and supplies to perform routine checks and expert examinations, as well as laboratory analysis fees. Costs associated with disease response (see Section 1.6.1.6 for details of disease response) include the staff time and supplies associated with treatments (Comprehensive Hatchery Plan 2007), quarantine, euthanasia, disposal, and disinfection (HSWRI Fish Health Management Plan 2016 and SOPs within).

For many health and safety plans, prevention proves to be more cost effective than response but this relies upon sufficient knowledge of risks and vulnerabilities, as well as good estimates of potential losses and probability of occurrence of the threats. The frequent and long-term pathology assessments and reports may offer the data needed to perform such an assessment.

1.8.1.4. Miscellaneous: Microbiomes.

There is relatively little clear information in aquaculture about the microbiomes, or communities of bacteria, cyanobacteria, fungi, and/or protists, associated with the internal and external environment of the fish and entire aquaculture systems (e.g., filters, surfaces, etc.) (but see Llewellyn et al. 2014 and references therein). White Seabass culture is also subject to this information gap. These communities may contribute to some of the variability found in the performance of hatcheries such as HSWRI, where individual batches can vary from 25-40% survival (Annual Report 10-11). Bacterial control strategies, for example, have allowed consistently higher larval survival (Annual Report 10-11) than those observed previously to sometimes be as low as 0%. The influences of these communities on fish health may be direct, or indirect through interactions with feed, other microbes present, and other environmental conditions. Variability in survival and growth at all hatchery stages is an indicator of our uncertainty about the important factors; microbiome monitoring procedures and record keeping could result in informative data. HSWRI has submitted several proposals over the years to study microbiomes but has not received funding.

1.8.2. Data and Information Gaps.

1. Limited knowledge of disease dynamics in marine systems, especially in light of changing climatic and associated environmental conditions, rates of disease in wild fish, and

potential susceptibility of White Seabass to the six priority pathogens and other specific pathogens of concern.
2. No compilation and synthesis of data from pathology reports and other sources quantifying the appearance/disappearance and incidence rates of various types of diseases and pathogens, nor relating disease appearance and incidence rates to environmental factors (e.g., water temperature, water quality, occurrences of disease in the wild) to be used in risk assessments.
3. No assessment of the risk of common pathogens and disease that reveals the extent to which preventative measures should be enacted.
4. Little to no data on the microbiomes associated with all parts of the hatchery systems and the different stages of fish (e.g., composition, abundance, spatial and temporal distributions, modes of introduction, relationships with beneficial and detrimental pathogens, relationships with hatchery procedures-cleaning, probiotic use, etc.).

1.8.3. Recommendations.

1. Address the recurring gas supersaturation problems by altering procedures and by updating equipment with existing technologies and supplies.
    a. Avoid growing seabass in shallow water pens during the warm summer months.
    b. During El Niño years, shift the bulk of growout facility stocking to winter and early spring months.
    c. If possible, move the three growout facility pens with the highest incidence of gas supersaturation (Marina del Rey, San Diego Bay-Southwest Yacht Club, and Huntington Harbor) to new locations with higher water quality; in particular place them, and any future pens, in deeper water.
    d. Minimize or avoid hatchery raceway use during warm weather conditions (e.g., summer).
    e. Consult with experts to provide further engineering solutions, including new equipment, or repairs or modifications of existing equipment (e.g., repair or bypass of suction side air leaks, installation of automated control on vacuum degasser) for degassing seawater to levels appropriate for year-round production of seabass at the Carlsbad Hatchery.
2. Have CDFW sample fish for pathogen screening at least 1 month prior to scheduled release to growout sites, and up to 3 months prior to release from growout (so results are obtained in timely manner).
3. Have CDFW and HSWRI collaborate to track and report the incidence rates of common pathogens and disease, and associated conditions, using data from pathology reports and other sources to better understand the rates of infection and potential causes.

1.9. Deformities: Effects of deformities on hatchery operations and releases, and how deformity challenges have been addressed.

1.9.1. Key Findings.

CDFW and HSWRI have developed and use their own respective sets of protocols to detect, rank, and address deformities in hatchery-reared White Seabass (described in 1.6.1.2). The discrepancies between these protocols, and the inefficiencies these discrepancies create, are likely the largest challenges associated with deformites. Furthermore, the effects of the various deformities on fish health and fitness, and on the overall economic impact, are uncertain. Therefore, there are few data from the White Seabass program, and from other stocking programs, that justify such rigorous deformity screening protocols.

1.9.1.1. Different deformity protocols.

What constitutes a deformity. It is a goal of the OREHP to only release fish without deformities (CDFW Release Criteria 2015, HSWRI QA/QC Manual 2011), yet what constitutes a deformity, as compared to a normal fish or one with acceptable amounts of physical variability, differs between CDFW and HSWRI. CDFW protocols define deformity as a variation from an ideal form hatchery fish (CDFW Cultured White Seabass Deformity Report Protocol 2015). HSWRI aims to produce a 'wild-type' fish, so variations in form similar to those found in wild fish are deemed acceptable. HSWRI has sampled, and continues to sample, wild caught fish and wild fish from reference collections to build a database that will document the range of variability in wild fish (HSWRI QA/QC Manual 2011).

Many of the deformities seen in hatchery White Seabass are documented in wild White Seabass (Skogsberg 1939, HSWRI QA/QC Manual 2015) and other wild species. For example, Skogsberg (1939) documented normal wild type Sciaenids to have generally less than 15 pyloric cecae, and wild White Seabass to usually have 4-5 pyloric cecae. Irregular pyloric cecae (4-6) were also documented during necropsies performed on wild White Seabass held in captivity for 5 months (CDFW in 2010) and 16 months (HSWRI in 2016) (HSWRI OREAP Meeting Presentation, 18 April 2016). HSWRI no longer considers variation in number of pyloric cecae to be an 'internal malformation.' Further, bulbus arteriosus dysplasia, which has been observed in the hatchery White Seabass (CDFW Pathology and Deformity Reports), has been observed in sturgeon (Icardo et al. 2002). External irregularities, such as head indentations, lower jaw extension, and maxillary dysplasia have also been observed in wild White Seabass (HSWRI QA/QC Manual 2011).

The actual rates of external and internal deformities in wild White Seabass and other wild fishes are, however, relatively uncertain due to a general paucity of studies and/or the likelihood of mortality (e.g., predation) in the wild if the deformities affect survival ability. Further, rates of *internal* deformities in hatchery fish are not well known because of the impracticality of extensively diagnosing them (i.e., diagnosis requires euthanasia). The often non-random, extensive internal exams performed by CDFW have regularly revealed malformations of the heart, intestines and swim bladder (CDFW Pathology and Deformity Reports). The effects of these deformities on fitness remain untested, and while there is no evidence that minor internal

malformations result in poor survival, more severe deformities of organs critical to movement and respiration (e.g., swim bladder, heart) likely influence post-release survival.

Discrepancy between protocols. The differences in protocols used are creating a conflict between CDFW and HSWRI. Fish that have cleared the HSWRI QA/QC criteria needed for transport to growout sites are deemed unfit for transport by CDFW (CDFW Pathology and Deformity Reports). For example, in September 2016 a group of tagged hatchery fish that had passed through the HSWRI QA/QC protocols, which focus on fish external quality, was examined by the CDFW pathologist who diagnosed a 40% prevalence of swim bladder malformations. The discrepancies in rates and types of deformities diagnosed reflect discrepancies in diagnostic criteria, such as whether or not variability of wild fish are considered during decisions about culling, how the scoring criteria incorporate the biological significance and severity of malformation, and whether or not soft tissue malformation is considered a deformity; and discrepancies in the methods used, such as use of targeted or random sampling methods, whether or not internal organs are routinely assessed, and amounts and control of sampler bias. With extensive internal examinations, it is not surprising that there would be greater number of reports of deformity, however, conducting internal exams as part of HSWRI's routine hatchery protocols was not the original intent of the quality control monitoring and is not feasible from a resource perspective. The question becomes whether the hatchery continues to focus on external quality control, which accepts some level of risk of post release mortality due to internal malformations, or be refocused on comprehensive checks of traits linked to survival, which would require that extensive resources, including fish, be dedicated to screening for internal deformities.

Other deformity protocols. Consultation with various State-level Fish Pathologists and Federal Fish Health Specialists has made it clear that deformity level in hatchery reared fish (e.g., salmonids in the northwest U.S. and California; Red Drum, Seatrout and Southern Flounder in Texas, Red Drum in Florida) is generally of little concern and poses little to no risk to wild fish. To our knowledge, specific deformity protocols do not exist (except for White Seabass), and there is little to no indication that stocking and supplementation programs in the U.S. perform deformity screening prior to release. To our knowledge, no salmon/steelhead stocking and supplementation programs in the northwest perform deformity screening prior to release (M. Blair, US Fish and Wildlife Service; D. Munson, Idaho Fish and Game; pers. comm.). In Florida, raised fish (Snook, Pompano, Red Drum) are visually (externally) graded during the rearing process and culled to reduce cannibalism, then assessed again and, if needed, culled during the tagging process before release (K. Leber, Mote Marine Lab; pers. comm.). As with HSWRI, Mote Marine Lab is also a research center, so such inspections may help to inform the science. It should be noted that hatchery procedures do often include some sort of overall quality criteria (e.g., rearing of fish stages that are similar to form and health of wild fish) where observable changes in behavior or form of fish trigger a diagnosis and response (e.g., changes in diet or water conditions) (Colura et al. 1990, Colura et al. 1991, CDFW 2004).

It is generally assumed that if a deformity is present and affects the health or performance of an animal, it would be quickly culled once released into the wild. For most agencies and programs involved with supplementation or resource enhancement hatcheries, fish are required to meet

disease inspection criteria with requirements for release dictated on the animal's disease/pathogen status. This is typically the case for most fish health programs and ensures that risk of introducing specific pathogens of concern (or carrier fish) into the environment is minimized. A further example involves a more comparable program focused on population recovery and enhancement of a species of freshwater cod (burbot). A recovery program established in the early 2000s set forth to re-establish a nearly extirpated burbot population from the Kootenai river in Idaho and British Columbia, Canada (KVRI Burbot Committee 2005). This progam began releasing fish in 2009 and has been very successful in reviving and enhancing the burbot population in a very short time. This species has a true larval stage similar to White Seabass and fish are reared to various sizes (>5 g) prior to release. This program does not screen for deformities prior to release, but adheres to strict pathogen screening requirements for both the US and for Canada.

The protocol used by HSWRI appears to be among the most rigorous (and one of the only) that could be found among enhancement programs in that it includes regular and thorough external examinations and comparison to wild fish variability. The protocol used by CDFW is even more rigorous with internal exams of soft tissues and comparisons to fish with "normal" features (i.e., narrow range of variability). The rigor and content of the protocol should be developed with the assistance of an independent advisory committee and both agreed upon and followed by both CDFW and HSWRI.

1.9.1.2. Effects of deformity.

The effects of the various deformities on the growth, reproduction and survival of the hatchery fish as well as the effects of deformed fish on wild populations, and overall economic impact, are uncertain. Potential effects may be biological, economic, and social.

Biological. Research on other species reveals that poor food quality (e.g., vitamin, mineral and fatty acid overdoses or deficiencies), poor water quality, flow and/or turbulence, and behavior (e.g., wall-nosing) are the most likely causes of deformity. The causes of most White Seabass deformities are, similarly, likely linked to nutrition, especially nutrition of larvae, and husbandry, where larvae reared in temperatures greater than 18^{o}C are more likely to exhibit cranial protuberance. All White Seabass in this program are progeny of wild broodstock, so it can be assumed that if genetic factors underlie deformities, these genes would already be present in wild White Seabass. The higher survival rates of deformed fish in the hatchery than the wild, however, increases likelihood that phenotypic expression in hatchery-reared juveniles would not be removed by selection thereby potentially increasing occurrence of the detrimental genes in the wild. The risk of heritability of deformities, however, tends to be low in other teleost species (see Chapter 3). A second, more likely scenario is that if environmental factors (e.g., poor nutrition or water quality issues that result in conditions that are the causes of deformity) lead to deformity, then release of deformed fish will have negligible effects on wild populations of White Seabass and other species because released fish are not infected with detrimental pathogens or passing genes to wild fish. Impact would be primarily related to the potential long-term survival of released fish, which would likely be low (e.g., due to predation or inability to compete effectively for food resources).

Economic. Understanding the economic effects of deformities will require assessments and comparisons of the costs and benefits of the different scenarios for addressing deformity. Of particular importance would be comparisons of the cost/benefit analysis of (1) research into the causes of deformity and solutions, (2) more training and more rigorous health exams by staff and subsequent culling, (3) losing deformed but innocuous fish in the wild, and/or (4) selling culled fish for alternative uses.

Social. The social effects of deformity on the program include bad press. Concerned environmental groups and citizens view the release of deformed fish as a waste of money in part because deformities likely lead to mortality once fish are released (e.g., Rivard 2016). The education and stewardship value of this program is great, and as such the perceptions of stakeholders are very important (see Section 6.4.1). Sportfishermen, for example, are major supporters of this program, but there is the potential for negative perceptions of the program to grow with bad press. Furthermore, it is important for anglers (who provide funds for the OREHP through their purchase of Ocean Enhancement Stamps with their licences) to be aware of whether their money is spent efficiently and in ways that they deem important, and that result in positive impacts on the standing stock.

1.9.2. Data and Information Gaps.

1. Lack of consistency between CDFW's and HSWRI's respective protocols for detecting and addressing deformities in hatchery-reared White Seabass.
2. Little quantitative information on rates of various deformities in hatchery fish, and links to potential influential factors.
3. Limited available information on the incidence and rates of deformity in wild fish, especially internal deformity and those internal and external that result in mortality.
4. Lack of information on the effects of deformities on fitness and survival of juveniles during growout and in adults recruiting to the fishery.
5. Lack of comprehensive information on nutritional requirements and specific mechanisms resulting in incidences of malformations in White Seabass.
6. No economic data to assess the costs and benefits of different potential strategies for responding to deformities.

1.9.3. Recommendations.

1. The idea of screening and culling fish with deformities should be brought into question from both a biological and practical point of view. It is generally assumed that a lack of fitness would result in poor fish performance in the hatchery and eventual removal from population upon release. If fish with malformations perform poorly this should be evident in the hatchery. From an economic and practical standpoint, it appears to make little sense to remove deformed fish from a population prior to release. HSWRI and CDFW need

to reach a consensus on the level of culling required. If strict culling procedures due to deformities are preferred, then it is recommended that a comprehensive study be developed that links fitness and deformities. This would serve to establish a ranking of deformity in relation to potential survival in the wild and serve as a guide to developing (or adopting) appropriate deformity screening protocols.

2. It is recommended that HSWRI and CDFW work together to come to an agreement on specific criteria and protocols needed to justify release of hatchery-reared White Seabass. If this criterion is to continue to include extensive deformity screening, then appropriate protocols must be agreed upon between the two groups. It is further recommended that HSWRI and CDFW invite Fish Health pathologists and Fish Culturists who are actively involved with similar programs (i.e. salmon/steelhead supplementation or other marine or freshwater enhancement programs) to participate in a workgroup aimed at developing practical protocols that meet program needs.
3. Studies should be conducted to systematically quantify the rates of deformity in hatchery and wild fish; the effects (if any) of deformities in released hatchery fish on wild populations/communities; the effects that various deformities have on fish growth, survival and reproduction in the wild; and the relative roles of environmental (nutrition, water quality, disease/pathogens) and genetic factors in contributing to deformities. For example, the hatchery has an experimental incubator system and can be used to compare deformity rates among hatchery fish reared in production versus experimental incubators as a means of evaluating the efficacy of activated charcoal.

1.10. Disease control: Steps in place to minimize risk of introduction of identified diseases into the wild.

1.10.1. Key Findings.

The steps in place to minimize risk of introduction of identified diseases into the wild include treatment, quarantine, and/or euthanasia of fish, a strict set of criteria that fish must meet before release, and reporting of fish escapes.

1.10.1.1. Treatment.

HSWRI has protocols in place for the use of hydrogen peroxide and formalin in the treatment of diseased or deformed fish (Hydrogen Peroxide Treatment SOP 2016, Formalin Treatment SOP 2016) (see Section 1.8 for information on disease). Perox-Aid is the only hydrogen peroxide product approved for treatment by the FDA; treatment requires a prescription and can only be carried out under the direct supervision of the clinical veterinarian (Hydrogen Peroxide Treatment SOP 2016). The dosage is determined by the veterinarian, incoming water flow is stopped, and the hydrogen peroxide is added to the designated pool slowly, so as to avoid having fish come into direct contact with high concentrations of the chemical (Hydrogen Peroxide Treatment SOP 2016). Fish are then monitored every 15-30 minutes for signs of distress; dissolved oxygen (DO) is also monitored (Hydrogen Peroxide Treatment SOP 2016). If DO drops

below 4.0 mg/L, or if fish are distressed, treatment is stopped immediately, water flow is resumed and oxygen is supplemented (Hydrogen Peroxide Treatment SOP 2016).

Although the Food and Drug Administartion (FDA) does not require a prescription for formalin treatment, HSWRI only implements formalin when the clinical veterinarian issues a prescription, except in the case of egg disinfection (Formalin Treatment SOP 2016). According to protocols, Formacide-B and Parasite-S are the two formalin products used by HSWRI (Formalin Treatment SOP 2016). The correct dosage is determined by the veterinarian, incoming water flow is turned off, additional oxygen is provided, and the formalin is added to the tank slowly (Formalin Treatment SOP 2016). Fish are then monitored every 15-30 minutes for signs of distress; DO is also monitored (Formalin Treatment SOP 2016). If DO drops below 4.0 mg/L, or if fish are distressed, treatment is stopped immediately, water flow is resumed and oxygen is supplemented (Formalin Treatment SOP 2016). After the treatment is completed, the pH level of the water must be recorded and reported to the Facilities Manager (because of wastewater regulations and permits), and the water must be disposed of by pumping it to the sewer (Formalin Treatment SOP 2016). One hundred fifty percent of the volume of water that was treated with formalin should be flushed through the tank to ensure that the chemical has been sufficiently cleared out (Formalin Treatment SOP 2016).

Two antibiotics, Romet and Oxytetracycline, have been used in an "extra-label manner" for treatment of fish under the direction of a veterinarian (New Fish Acquisition Quarantine SOP 2016). However, starting January 1, 2017, Romet will no longer be permitted for extra-label use, for it will be designated as a Veterinary Feed Directive (VFD) drug (New Fish Acquisition Quarantine SOP 2016). In the future, HSWRI (with prior approval and a veterinarian's oversight) may use other antibiotics under the "Investigational New Animal Drug" (INAD) guidelines (New Fish Acquisition Quarantine SOP 2016).

HSWRI employs the above treatment techniques relatively infrequently. In October 2014, juvenile fish were treated with Perox-Aid to control ectoparasites (flagellates and ciliates) causing ulcerative dermatitis (Annual Report 14-15). In May 2015, juveniles with necrotizing dermatitis were either culled (if dermatitis was severe) or treated with Romet to control the bacteria (presumed to be *Vibrio* and *Flexibacter* spp.) leading to infection (Annual Report 14-15). In FY 2009-2010, the gill fluke *Anchoromicrocotyle guaymmensis* was discovered in some broodfish, treated with Formacide-B, and then again with hydrogen peroxide (Annual Report 09-10). Formacide-B is more effective in killing the parasites, but more difficult to implement because of the required flushing of extra water through the system post-treatment (Annual Report 09-10). Hydrogen peroxide simply provides temporary relief to the fish by causing flukes to drop off the fish (flukes are then siphoned out of the tank) (Annual Report 09-10). It appears that in this case, the infected broodfish were treated with hydrogen peroxide (Annual Report 09-10). It should be noted that limited treatments exist and in some cases hydrogen peroxide has been ineffective in treating certain external pathogens, such as Hexamita and other fungi and bacteria that cause severe skin ulcers, as seen in 2009 and 2010 at the Marina Del Rey growout facility (CDFW Pathology Report 2010-108, M. Okihiro email accompanying Pathology Report 2010-108, 2 February 2011). In this case, fish with severe lesions were culled, and all other fish were treated

with a final peroxide bath before release (CDFW Pathology Report 2010-108, M. Okihiro email accompanying Pathology Report 2010-108, 2 February 2011).

In response to an outbreak of a ciliated protozoan pathogen at the King Harbor growout site in 2011, CDFW pathologist Dr. M. Okihiro suggested that experiments should be conducted to treat fish with freshwater (M. Okihiro email accompanying Pathology Report 2010-112, 16 December 2010, M. Okihiro email to J. Murdick, 17 February 2011). It is unclear whether HSWRI ever undertook these suggested measures, as there is no mention of such experiments in the subsequent annual reports. However, in 1995 and 1996, HSWRI did attempt to treat broodstock infected with *Cryptocaryon iritans* with a combination of freshwater and formalin (Annual Report 95, 95-96). This treatment slowed, but did not eliminate the *C. iritans*; it was not until a combination of copper sulfate and formalin was used that the problem was eradicated (Annual Report 95, 95-96).

1.10.1.2. Quarantine.

Pathogens and infectious disease outbreaks have occurred at the hatchery and at growout sites resulting in the quarantine and treatment of fish or, if widespread, of whole sections of the hatchery or growout facilities (see example below). There are occasional high losses of fish due to disease, including asymptomatic fish lost through euthanasia. The occurrence of abnormal behaviors, external lesions, or high mortality (≥1.5% per day per tank) not caused by handling or mechanical malfunctions triggers the Infectious Disease Emergency SOP (2016) to contain the potential infection. The CDFW pathologist or Fish Health Specialist can recommend a quarantine that remains in effect until "the problem has been diagnosed and/or managed" (HSWRI Fish Health Management Plan 2016). During quarantine at the hatchery, the system affected by the disease outbreak, along with the equipment used within the area, are isolated from other parts of the hatchery (In-Hatchery Quarantine SOP 2016). The movement of fish on and off site stops, no fish are handled, and all visitors and non-essential staff are prohibited from being on site (HSWRI Fish Health Management Plan 2016). Staff abide by strict disinfection protocols, such as using footbaths at all entrances and exits (In-Hatchery Quarantine SOP 2016, Footbath Maintenance SOP 2016). Treatment of the disease may be initiated based on the recommendation of the attending veterinarian or fish health specialist. For example, hydrogen peroxide and formalin are used to treat some external parasites and infections (Hydrogen Peroxide Treatment SOP 2016, Formalin Treatment SOP 2016; see Section 1.10.1.1 above; also see Section 1.8.1.1 for further details on pathogens).

In the hatchery, general procedures involve removal of moribund or dead fish once at the end of each day to minimize impacts on water quality and reduce the risk of spreading disease; necropsies are performed if needed, and fish are stored in a separate quarantine freezer, or designated for disposal (In-Hatchery Quarantine SOP 2016, Mortality Collection and Disposal – Carlsbad SOP 2016). In the growout pens, dead fish are collected frequently, at least once a day (Fish Mortality Classification SOP 2016). At the hatchery, disinfection of personnel, equipment and supplies occurs after the dead fish are collected and disposed (HSWRI Fish Health Management Plan 2016). The Fish Health Specialist or CDFW determines when the quarantine is over depending on the situation, either recommending treatment or euthanasia (In-Hatchery

Quarantine SOP 2016, Euthanasia SOP 2016). Determination of the cause of an outbreak is conducted, and monitoring of fish (behavior, appearance) and water quality continues after diagnosis and treatment to inform further management decisions (HSWRI Fish Health Management Plan 2016).

Quarantine has been implemented periodically at the hatchery and growout facilities. For example, according to HSWRI in December 2013, CDFW issued a quarantine after neurologic behavior was observed in juveniles (Annual Report 13-14). Samples were negative for VNNV, but viral etiology and hepatic encephalopathy were suspected as causes for the abnormal behavior (Annual Report 13-14). In January 2014, with no definitive diagnosis, fish were euthanized due to crowding and continued quarantine (Annual Report 13-14). It appears that generally, HSWRI follows its quarantine protocols, including disinfection protocols, mortality collection and disposal protocols, and its restriction of visitor and staff access to the area.

1.10.1.3. Euthanasia.

As mentioned above, diseased hatchery-reared fish are often culled to prevent the spread of disease to other hatchery fish and their wild counterparts (CDFW Release Criteria 2015) (e.g., Annual Report 09-10, Annual Report 10-11, Annual Report 11-12). HSWRI's Euthanasia Protocol sets out detailed instructions for humane methods of culling fish. For White Seabass, this includes the use of tricaine methane sulfonate, carbon dioxide, sodium pentobarbital, and proper decapitation and pithing (Euthanasia SOP 2016).

1.10.1.4. CDFW release criteria.

Health assessments are performed twice by a CDFW pathologist prior to release; once prior to transport to a growout facility and once prior to release (CDFW Release Criteria 2015; also see Section 1.6.1.6 for a more detailed description of the disease detection process). Release criteria are based on the presence of disease and not deformity. While deformities in fish may be due to a variety of factors (e.g., nutrition, environment, genetics; see Section 1.9.1), the actual links between types of deformities and causes, including pathogens, are uncertain for White Seabass. CDFW's Release Criteria (2015) states that fish are to be euthanized if any one of six pathogens of concern are identified, and the infected facility is to undergo disinfection. Six common pathogens of concern are highlighted due to their potentially serious impacts on White Seabass because they are non-treatable, debilitating, highly contagious, associated with high rates of mortality, and/or are known to harm wild fish stocks (CDFW Release Criteria 2015). Included are a description of the disease and symptoms, and a summary of occurrences in the hatchery and growout of: White Seabass herpesvirus, Viral Nervous Necrosis Virus (VNNV), *Piscirickettsia salmonis*, *Uronema marinum*/*Miamiensis avidus* (ocular and encephalitic variant), a renal "sporozoan" pathogen, and Viral Hermorrhagic Septicemia Virus (VHSV) (CDFW Release Criteria 2015).

1.10.1.5. Reporting of fish escapes.

In the uncommon event of a fish escapement event from the hatchery or growout facility, the facility operator is required to (1) notify the growout facility coordinator and the OREHP Coordinator immediately, (2) estimate the percent of fish loss and be able to provide information

on how and when the fish escaped, and (3) immediately repair the rip or break to prevent further escape (White Seabass Enhancement Plan 2010). Further, fish health records, including relevant diagnoses and treatments, should be made available to the appropriate regulatory authorities upon request (HSWRI Fish Health Management Plan 2016).

1.10.1.6. Wild fish disease surveys.

Wild fish disease assessments should be made in order to understand diseases processes in the wild to better inform hatchery production and release practices. Minimizing risk of disease requires knowledge of which highly contagious, lethal pathogens tend to occur in wild fish. In 2002, there was assessment of wild stocks that were sampled with evaluations dependent on the condition of the fish, where fish are collected (i.e., proximity to laboratory facilities), and the types of pathogens being screened (Comprehensive Hatchery Plan 2007). Species surveyed include White Seabass, California Halibut, California Sheephead, Lingcod, and a variety of *Sebastes* rockfish species, with an emphasis on White Seabass (Comprehensive Hatchery Plan 2007). Most fish are collected dead during the OREHP gill netting efforts and are only suitable for cytology, hematology, and microbiology, not morphologic assessments (e.g., histology). For live fish caught in gill nets or by hook and line, sampling occurs on board the boats, or the fish is transported to HSWRI's Mission Bay research facility or the Carlsbad Hatchery (Comprehensive Hatchery Plan 2007). Some of the wild fish are screened for external parasites using cytology, or have samples fixed for histopathology, but the most important assessments are hematologic and microbiologic assays to determine exposure (antibody levels) or infection to pathogens that are highly contagious and lethal (Comprehensive Hatchery Plan 2007). Wild fish surveys were focused on four major pathogens: viral nervous necrosis virus (VNNV), viral hemorrhagic septicemia virus (VHSV), *Piscirickettsia salmonis*, and a yet uncharacterized enteric virus (possibly a herpesvirus). Three (VNNV, *P. salmonis*, and the unidentified enteric virus) of the four have been isolated from cultured White Seabass. The fourth pathogen, VHSV, has never been isolated from White Seabass, but has been recovered from several baitfish species (sardines and herring) landed in Los Angeles ports (Comprehensive Hatchery Plan 2007). Although such sampling of wild stocks is essential to identifying risks and characterizing susceptibility, such assessments are expensive and have not been ongoing. The success story with VNNV should be noted as work by HSWRI into outbreaks of this disease in White Seabass led to comprehensive control and management strategies.

1.10.2. Data and Information Gaps.

1. Few and/or unclear CDFW criteria for lifting quarantines.
2. Gap in knowledge about possibility of hatchery raised White Seabass transmitting or retransmitting pathogens to wild stocks.

1.10.3. Recommendations.

1. Direct research toward understanding pathogens in wild stocks.

2. Initiate studies to develop better tools and comprehensive disease control strategies during hatchery rearing and prior to transfer to growout sites. This could minimize problems overall.

Chapter 2

Objective 2. Conduct the replenishment program in a manner that will avoid any significant environmental impacts resulting from operation of either the hatchery or pen rearing facilities.

Any hatchery and pen rearing operations have the potential to result in a variety of adverse environmental impacts. These impacts include those to water quality, benthic habitats, submerged aquatic vegetation, and marine wildlife, as well as those associated with the growth and spread of marine invasive species.

In order to evaluate whether the enhancement program has had any significant environmental impacts on the habitats, wildlife, or quality of receiving waters, we first considered the pathways of potential adverse effects, the key indicators representative of those pathways and thresholds that are used to evaluate adverse effects, as explicit criteria for evaluating whether Objective 2 has been meet. We considered what Hubbs-SeaWorld Research Institute (HSWRI) has been required to or volunteered to monitor hatchery and growout pen adverse effects and the evidence of any environmental impact, based on the information available from HSWRI's monitoring and other sources such as CDFW reports and agency reviews. This section includes the findings of this review as well as recommendations to help ensure that any potential significant environmental impacts related to OREHP operations at the Carlsbad hatchery and growout facilities are avoided. For the sake of clarity, "hatchery operations" refers to both the hatchery as well as the net pen located adjacent to the hatchery in Agua Hedionda Lagoon.

2.1. Pathways of impact, key indicators and criteria used to determine significant impacts.

2.1.1. Key Findings.

The environmental impacts of fish hatcheries or mariculture facilities on the water quality or benthic habitats of estuarine and marine environments can be broken down into two general pathways (Gowen and Bradbury 1987): (1) direct, acute or chronic effects of constituents such as nutrients (nitrate, ammonia), oxygen consuming waste, trace and heavy metals, or disease organisms that cause physiological stress and (2) indirect pathways in which excess nutrients and accumulation of organic matter from excess feed or feces can trigger changes in the benthic and pelagic food webs and result in altered sediment or surface water dissolved oxygen and pH. Table 2.1 summarizes key indicators and existing criteria or thresholds that represent a means to characterize environmental effects of hatchery and holding pen operations on the surrounding environment.

2.1.1.1. Water quality.

Direct impacts of hatchery effluent are largely covered by the facility's National Pollution Discharge Elimination System (NPDES) permit or waiver established by the San Diego Regional Water Quality Control Board (SDRWQCB) since 1996; the hatchery had one 5-year permit that was discretionary, due to the low biomass of the facility. In accordance with this permit or waiver, HSWRI has regularly monitored influent, effluent, and secondary backwash at the

Carlsbad hatchery. From 1996-2002, a more limited set of monthly requirements were in place (total and unionized ammonia, turbidity, suspended solids). Since 2002, the requirements were expanded to include acute toxicity, pH, nutrients, and metals (copper and zinc), with the additional constituents removed from the list, depending on the permit cycle.

Table 2.1. Compiled list of reporting parameters measured in hatchery and growout operations, by pathway.

Type	Medium	Indicator	Hatchery	Holding Pen	Criteria Used for Evaluation of Significant Impact
Direct	Hatchery Effluent	Toxicity	Quarterly	N/A	Acute toxicity
		Unionized Ammonia	Monthly		Unionized ammonia
		Turbidity or Suspended Solids	Monthly		
		Settleable Solids	Monthly		
		pH	Monthly		Within pH units of 6-9
		Temperature	Monthly		
		Copper and Zinc	Quarterly		
		Disease organisms	None		Pathology
	Growout Pen Sediment Quality	Sediment Copper	2 surveys per 5 year permit cycle	3 surveys per 5 year permit cycle	197.5 ug/g dry weight threshold effects level (TEL)
		Sediment Zinc			63.4 µg/g probable effects level (PEL)
Indirect	Hatchery Effluent	Total Nitrogen (TN) and Total Phosphorus (TP)	Monthly	N/A	1.0 mg/L total nitrogen and 0.1 mg/L total phosphorus[1]
	Growout facility water quality	Dissolved Oxygen	NS	Twice per year- LA Region Only	7.0 g mg/L DO
		Ammonia	NS		
	Growout facility sediment	Sediment free sulfide	3 surveys per 5- yr permit cycle	3 surveys per 5- yr permit cycle	Significant difference with reference site
		Redox potential			

[1] CRWQCB 2016

The requirement of monitoring copper and zinc is debated by some aquaculturists since fish hatcheries and standard marine constructions like docks and net pens do not use much if any copper, bronze, or brass materials other than for sinks and toilets. Thus, the contributions of

copper and zinc from the hatchery may be relatively small. However, until the practice was discontinued in recent years (OREHP Final Negative Declaration 2012), much of the netting used for the OREHP net pens included copper as an anti-fouling treatment. The use of these nets and their degradation over time introduced a potential source of copper pollution to the enclosed water bodies in which the netpens were located. While fish feeds containing copper and zinc, concentrations can be extremely low (1 µg/g dw) (D. Weaver pers. comm.), their accumultation in sediments around and below grow-out facilities may lead to more significant concentrations (OREHP Final Negative Declaration 2012; Benthic Monitoring Report 2007).

According to available documentation, the OREHP's individual growout facilities produce less than 45.3 metric tonnes (or 100,000 pounds) of biomass, a threshold defined by the EPA (40 CFR Appendix C to Part 122), and therefore are not required to have an NPDES permit or to conduct benthic monitoring. Inspite of this, in 2005 HSWRI undertook a 6-year program at the OREHP growout facilities to determine any impacts from fish cage operations. While this program was voluntary in the sense that HSWRI did not have specific monitoring requirements to follow for the OREHP growout facilities, it fell in line with the guidance set out in the original Coastal Development Permit (Permit No. 183-73), which stipulated that the hatchery and growout facility operations should be closely monitored to ensure that they do not cause significant environmental damage (Coastal Development Permit 183-73, Condition E(3)(h)). In addition, from 2005 until 2015[7] the Los Angeles Regional Water Quality Control Board (LARWQCB) also required that environmental monitoring be carried out at those facilities within its jurisdiction (one each in Channel Islands Harbor and Marina del Rey and two in Catalina Harbor). Environmental monitoring at those four Los Angeles Region growout facilities included biannual water quality monitoring (dissolved oxygen, temperature and ammonia), annual visual benthic surveys, and benthic monitoring every three years when fish were present at the facility. The results are reported in documents submitted to the LARWQCB. In some reporting years, this schedule was deviated from when a growout facility did not host a batch of fish that year, or when a batch of fish experienced high mortality rates, was released early, or was not released at all.

2.1.1.2. Benthic habitats.

In addition to the monitoring required by the LARWQCB at the facilities within its region, indirect impacts were assessed at all the growout facilities by a benthic habitat monitoring plan developed and implemented by HSWRI. Brooks and Drawbridge were commissioned to develop a Benthic Monitoring Plan (2005) to assess the indirect effects of the (then) 13 growout facilities on benthic habitat quality (see Table 2.2 for locations and actual frequency of sampling). This plan called for the measurement of free sulfide, redox potential, total volatile solids (TVS), sediment grain size (SGS), zinc, and copper in the sediment in annual assessments conducted at a minimum of three times over a 5 year period. According to Brooks and Drawbridge (Benthic Monitoring Plan 2005), monitoring at the growout facilities would use "a regression approach to identify trends in sediment free sulfides, redox potential, total volatile solids (TVS), copper, and zinc, as a function of distance from the net pen's perimeter on four orthogonal transects" (Benthic

[7] In a September 22, 2015 letter to CDFW, the Los Angeles Regional Water Quality Control Board concurred with CDFW's request to discontinue environmental monitoring for these growout facilities, citing the results of the previous ten years that demonstrated no adverse impacts to water quality or the benthic community.

Monitoring Plan 2005). Samples would be taken "on the perimeter of each net pen and at a reference location" at each of the thirteen growout facilities within one month before or after a batch of fish was released from the growout facility (Benthic Monitoring Plan 2005). After the six years of monitoring, California Fish and Game would determine whether more intensive monitoring was necessary on a site-by-site basis.

This sampling program was carried out between 2004 and 2012 and included approximately 28 separate sampling events between the 13 in-water net pen sites. Two of these sites (Santa Barbara and Mission Bay - Dana Landing) were abandoned during the course of this effort and the recommended three samples were collected at six of the remaining 11 net pen sites. Approximately half of the sampling events were carried out prior to restocking and after 25 or more days of fallow following the release of fish from a net pen facility (close to or exceeding the monitoring guideline that sampling occur within 30 days of peak fish biomass). Results of these sampling events are provided in the OREHP annual reports.

Table 2.2. Summary of locations where benthic habitat monitoring was conducted and actual frequency of sampling. The abbreviation of a month indicates the month and year that sampling occurred at each facility.

Growout facility	**2004**	**2005**	**2006**	**2007**	**2008**	**2009**	**2010**	**2011**	**2012**	**2013**	**2014**
Santa Barbara		Sept				Sept					
Channel Islands Harbor									Dec		
Marina del Rey		Sept			Sept				Mar		
Catalina Harbor – Inner Harbor CSF			Nov			Aug					
Catalina Harbor – Outer Harbor HSWRI	Sept				Feb						Jan
Huntington Harbor			Sept			Sept			Nov		
Newport Bay			Sept			Sept			Nov		
Dana Point Harbor		Nov			Sept				Mar		
Agua Hedionda Lagoon	Sept					Dec					Jan
Mission Bay – Quivera Basin		Nov			Jul						
Mission Bay – Dana Landing											
San Diego Bay – SW Yacht Club		Oct			Sept				Oct		
San Diego Bay – Grape Street	Sept			Dec			Sept				

Note: Growout operations at King Harbor and Port Hueneme were not monitored because they utilize land-based pool systems, rather than net pens or raceways deployed in marine waters.

2.1.1.3. Marine wildlife.

As discussed in the OREHP Final Negative Declaration (2012),

> *A wide variety of fish, marine mammals, and bird species are attracted to fish farming operations because they are a potential food source for those animals (BCEAO 1997). The*

farmed fish are the main attractant for seals, sea lions, predatory fish, and some birds. Uneaten fish food, fouling plants and animals that grow on farming equipment, and lighting used on fish farms also attract birds, fishes, and other marine life (BCEAO 1997).

In response to this issue of wildlife attraction, the OREHP netpens have all been designed and constructed with fencing and netting that passively excludes predatory wildlife from the White Seabass containment areas. In addition, the program is operated in accordance with a variety of best management practices. These include implementation of passive deterrence measures and prohibitions on active deterrence measures that separate parents and offspring; break the skin or otherwise injure an animal; or are directed at animals located on unimproved property. Non-injury causing active deterrence measures are also in the best management practices, including use of prods, elevated sound levels, water cannons, and similar devices intended to actively disturb and displace wildlife.

Although netpens at two locations (near Stearn's Wharf in Santa Barbara and Channel Islands Harbor) have had three instances of marine mammal entanglement and subsequent mortality (resulting in the loss of nine California sea lions), this situation was thought to have arisen as a result of poor maintenance, management, and siting practices that have since been improved (OREHP Final Negative Declaration 2012). There have been no other reported cases of marine mammal injury or fatality since then. The maintenance of the existing exclusion fencing and netting on the netpens and raceways appears to be an effective strategy, obviating the need to implement active deterrence approaches that would result in marine mammal disturbance or take. Existing authorizations and management protocols require all grow-out facilities to be regularly inspected and maintained to ensure that predator exclusion netting (both for underwater and avian predators) remains intact and in functional condition.

2.1.1.4. Submerged aquatic vegetation.

Several of the net pen facilities are located in shallow embayments in southern California that are known to provide habitat for submerged aquatic vegetation, such as eelgrass. However, net pens in harbors are generally placed in deep water. Only clean, highly flushed sites would receive enough light on the benthos for submerged aquatic vegetation to grow. In the rare instance that placement and long-term use of floating structures occurs above shallow, or deep and clear habitat, it has been shown that these structures could reduce light penetration to the seafloor. The reduced light could result in restricted or reduced growth of species such as eelgrass. In addition, placement and maintenance of mooring devices, anchors, and associated tackle in these shallow or clear, deep habitats may physically displace, disturb, or damage submerged aquatic vegetation.

2.1.1.5. Invasive marine species.

Most of the sites in use for net pen facilities are known to support populations of invasive marine species, including macroalgaes, shellfish, and invertebrates such as colonial tunicates. Many of these species specialize in or are capable of colonizing artificial hard substrates such as those provided by the net pen facilities, docks and other objects in harbors. Activities such as in-water maintenance and cleaning of these facilities (such as the scraping and removal of fouling

organisms from nets and floats) can result in fragments that can disperse and reestablish (e.g., Morris and Carman 2012), thus potentially contributing to their persistence and spread. The relative contribution of the net pens compared to other structures in harbors (e.g., marinas and docks) in contributing to the harboring and spread of invasives is uncertain, but based upon relative surface area clearly not the dominant mechanism.

2.1.2. Data and Information Gaps.

Lack of information about the presence, abundance, and types of invasive marine species on the facilities, especially species of particular concern (e.g., disease vectors, aggressive competitors or predators).

2.1.3. Recommendations.

Remain aware of new invasive species threats in the region, and be watchful of the appearance of new introductions.

2.2. Assessment of impacts of Carlsbad hatchery operations on water quality based on existing criteria and monitoring results.

2.2.1. Key Findings.

2.2.1.1. Direct effects.

Generally, based on a review of HSWRI's annual reports to the SDRWQCB, the quality of hatchery effluent appears to not be significantly different from influent quality, with the possible exception of total nitrogen (TN), which was generally double that of influent TN. Unionized ammonia was generally low or non-detectable and therefore not likely to result in direct effects. However, the impact of nutrient loading on lagoon water quality from the stimulation of algal production is unknown, because this effect is not currently being monitored nor have there been mass balance calculations conducted to estimate effects. The location of the hatchery proximal to the mouth of the Lagoon is advantageous in that the effluent discharged will be diluted by tidal exchange with the ocean, lessening the probability of impact.

Metal concentrations were not found to exceed the TEL and PEL (ie. biologically significant) in any of the Agua Hedionda sediments. However, the 2004-2006 Benthic Monitoring Report (2007) produced by Dr. Brooks for HSWRI notes that the small increases in copper and zinc observed on the Agua Hedionda net pen's perimeter supports the need for effective management practices to minimize copper losses from nets by cleaning them at an upland station and the need for a proteinated form of supplemental zinc. It is unclear whether these management recommendations were ever implemented.

The potential of hatchery effluent as a vector for disease to the Lagoon is uncertain because very little data are available to evaluate the risk of such an impact. There are, however, some procedures in place at the hatchery, including disinfecting tanks that held fish that were euthanized after quarantine, and sterilizing the equipment used on those fish (In-Hatchery Quarantine SOP 2016), that should reduce the likelihood of transmission of disease through effluent.

2.2.1.2. Indirect effects.

Assessments of indirect effects of net pen operations within Agua Hedionda Lagoon were conducted in 2004, 2009, and 2014 (Annual Reports 04-05, 09-10, 13-14), three times over a 10-year period, as opposed to the suggested three times over a 5-year period (Benthic Monitoring Plan 2005). In order to understand these data, it is important to acknowledge that the sediments of many Southern California estuaries and lagoons are already impacted from a number of stressors including contaminants, eutrophication, and hydromodification. Therefore attributing impact to HSWRI operation of hatchery facilities based on a limited monitoring program has recognizable uncertainty.

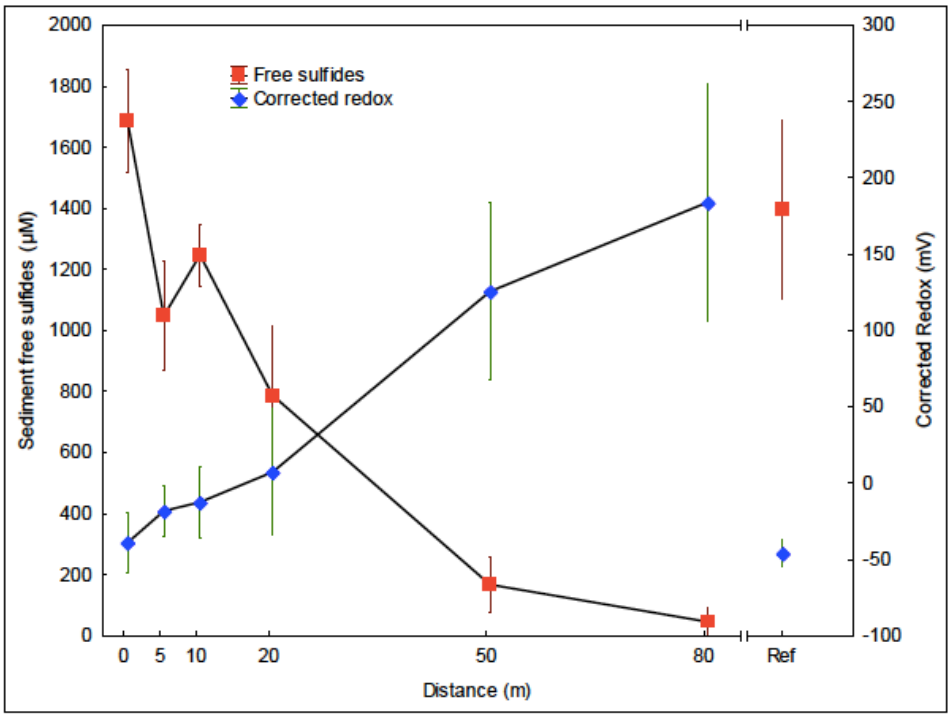

Figure 2.1. Mean sediment free sulfide (left axis) and redox potential (right axis) as a function of distance from the Agua Hedionda Lagoon net pen. From Annual Report 09-10.

That said, according to the 2004-2006 Benthic Monitoring Report (2007) and the OREHP Annual Report (09-10), sulfide concentrations were found to be elevated within close proximity of the net pens, with values up to 1200 µM. According to Brooks (Benthic Monitoring Report 2007), sulfide concentrations of that level can commonly exclude up to 50% of benthic invertebrate taxa

commonly found in marine sediments. However, these effects were found to be restricted within 20 m of the perimeter of the net pen, with no visual accumulation of waste under the pens (Figure 2.1). The author notes that the reference site chosen was also high in free sulfides, but that this site appears not to be a good "reference" site due to the close proximity of a mussel farming operation, and its location within a depositional area created by a gyre (Benthic Monitoring Report 2007). Analysis of collateral data from the Bight 2008 Coastal Ecology Survey confirms that this reference site has moderately impacted sediment habitat quality (Southern California Bight 2008 Regional Marine Monitoring Program Coastal Ecology Committee 2012). This 2008 survey did not sample in close proximity to the net pen, so no Bight 2008 data are available for comparison.

2.2.2. Data and Information Gaps.

1. The impact of nutrient loading on lagoon water quality from the stimulation of algal production is currently unmonitored, and therefore unknown.
2. Very little data are available to evaluate the risk of hatchery effluent as a vector for disease to the Lagoon.
3. Uncertainty about the relative role of hatchery impacts on coastal water quality, especially as compared with other major stressors.

2.2.3. Recommendations.

1. Consider updating benthic monitoring protocols to include new, standardized methods (see Section 2.5.3, Recommendation 2) and to switch the current reference site, which appears to be moderately to highly disturbed benthic habitat, to one at the distal end of the orthogonal transects (≈80 m) (see Section 2.5.3, Recommendation 1d).
2. Consider conducting a limited study of intake and effluent BOD, and net pen *in situ* continuous DO and chlorophyll-a, proximal to and away from the point of discharge plume in order to document receiving water condition at current nutrient and organic matter loading rates (see Section 2.5.3, Recommendation 1b).
3. Consider conducting a simple 1-D or 2-D modeling study to estimate the nutrient and organic matter loading ceiling above which more stringent monitoring should be required (see Section 2.5.3, Recommendation 1c).
4. Consider assessing the efficacy of water treatment and the risk of transmission of disease through effluent especially if hatchery production is increased.

2.3. Assessment of impacts of growout facilities on water quality and benthic habitat based on existing criteria and monitoring results.

2.3.1. Key Findings.

2.3.1.1. Direct effects.

From 2007 to 2014, the waters in and around the four Los Angeles-area growout facilities were monitored for dissolved oxygen, temperature, and ammonia levels. Based on a sample of half of these reports, at no time did DO fall below 7 mg/L, based on grab samples. Ammonium-N was not detected in any samples. No information was shared on methodology to monitor DO, nor detection limit of ammonium.

Direct effects of the growout facilities via sediment toxicity can be inferred from changes in sediment metal concentrations (Table 2.1). Based on assessments of sediment copper (Cu) and zinc (Zn), in general the conclusion drawn across the board subsequent to the implementation of minimization measures such as different feed and discontinued use of copper-treated nets, is that the growout operations are having no significant effects on sediment metal concentrations. In many cases, background concentrations are high, exceeding TEL and/or PEL concentrations in the growout facilities at San Diego Bay (Grape Street and Southwestern Yacht Club), Mission Bay (Quivira Basin), Dana Point Harbor, Huntington Harbor, Newport Harbor, Channel Islands Harbor, and Marina Del Rey, a finding substantiated by collateral sampling from the Bight 2008 Coastal Ecology Studies (Southern California Bight 2008 Regional Marine Monitoring Program Coastal Ecology Committee 2012). Only in Catalina and in Santa Barbara Harbors were Cu and Zn background concentrations low.

2.3.1.2. Indirect effects.

Indirect effects of growout operations on organic matter accumulation were assessed primarily by assessment of sediment free sulfide and redox potential. Redox potential measures largely mirrored trends in sulfide and therefore are not discussed here. In general, effects of growout facility operation on benthic habitat can be grouped into three categories:

1. **Locations with growout facilities that appear to be accumulating organic matter under the pen or within 20 m of pen perimeter, with high free sediment sulfide ranging from 400-3500, that declines rapidly and significantly away from the pen perimeter, relative to reference sites.** This includes San Diego Bay -Grape Street (2004, 2008, 2011) and -Southwestern Yacht Club (2009 and 2013), Mission Bay (Quivira Basin 2008), Catalina Harbor (2004, 2007, 2009), Marina Del Rey (2008), Huntington Harbor (2010), Channel Islands Harbor (2013).
2. **Locations for growout facilities where free sulfide is high, but does not appear to be significantly different away from pen perimeter or reference site.** This includes Huntington Harbor (2010) and Newport Harbor (2010 and 2013).
3. **Locations for growout facilities where free sulfide is below levels of concern, regardless of difference from reference sites.** This includes Marina Del Rey (2012), Santa Barbara Harbor (2010), Dana Point Harbor (2008, 2012).

Authors of the annual reports note that, overall, a great deal of spatial and interannual variability existed across growout pens with respect to basin setting and site-specific factors that can influence the magnitude of indirect effects. For example, in some years, the pen was in a slightly different location. Other times, pen operators failed to apply sound feeding practices (e.g. over reliance on automated feeders) or the standing stock of White Seabass was lower than what was originally anticipated, causing an overfeeding and therefore a higher potential for indirect effects of organic matter accumulation. Such situations could be addressed by improved pen management and operations protocols or greater efforts to ensure that existing protocols are followed.

An evaluation of environmental impacts should consider not only the magnitude of impact, but also the extent and duration of impact. Based on the benthic monitoring program, the extent of impact appears to be somewhat localized to generally within 20 m of the pen perimeter. In addition, because the growout pens were used only intermittently, these pens were often fallowed for long periods of time. Reoccupation of a previous location for growout employed on Catalina Harbor showed that the organic matter accumulation documented as high sulfide in the 2007 annual report had fallen back to reference values by 2009. Thus, it is likely that the duration of the impact from organic matter accumulation is limited and could be expected to be remediated within short time periods of allowing pens to fallow. The exact duration of this period is unknown, however, and should be better investigated.

2.3.2. Data and Information Gaps.

1. Little information on the ways in which fallowing growout pens affects organic matter accumulation and nearby water quality, including the duration of time that growout sites should potentially be fallowed to mitigate environmental impacts.
2. Little data exist on the extent and duration of effects of growout facilities on the surrounding environment.

2.3.3. Recommendations.

1. Consider developing and/or updating SOPs for growout, and electronically linking the SOPs and task checklists to a dynamic, electronic hatchery management system (see Section 5.2.3) to ensure the recording and submission of growout information to HSWRI staff, and to provide growout volunteers with the opportunity to suggest protocol modifications as needed.
2. Investigate the potential of fallowing growout pens strategically, so as to mitigate environmental impact (see Section 2.5.3, Recommendation 2d).
3. Consider changes to the benthic monitoring protocol (see Section 2.5.3, Recommendation 2).

2.4. Assessment of impacts of growout facilities on marine wildlife, aquatic vegetation and marine invasive species.

2.4.1. Key Findings.

2.4.1.1. Impacts on marine wildlife.

Since the first growout pen became operational in 1991 (Annual Report 91), there have been only three incidents that resulted in the lethal take of marine mammals (California sea lions). Two of these incidents occurred at the Santa Barbara net pen while the other death occurred at the Channel Islands Harbor net pen. At the Channel Islands Harbor net pen, one death was reported in 2005 and was likely due to moving the net pen closer to the bait barge temporarily so the dock could be repaired. The situation was corrected, and there have been no other deaths reported at this pen. At the Santa Barbara net pen, one death was reported in 2004, while in 2009 seven malnourished sea lion pups became entangled in the predator net. This was during a period when a high rate of malnourished sea lion pups were reported off the Santa Barbara coast and was considered a rare occurrence (OREHP Final Negative Declaration 2012). To correct this problem, the predator net was temporarily removed. The Santa Barbara net pen was later removed as well and its operation discontinued.

2.4.1.2. Impacts on submerged aquatic vegetation.

Insufficient high resolution, site specific and temporal information is available about the presence, type, and abundance of submerged aquatic vegetation in proximity to the 11 remaining in-water growout facilities. Such information is necessary to adequately assess the likelihood or magnitude of this potential impact.

2.4.1.3. Impacts on marine invasive species.

Insufficient information is available on the specific maintenance and cleaning practices of the 11 remaining in-water growout facilities and the type, presence, and abundance of invasive fouling organisms on the submerged structures of these facilities. Such information is necessary to adequately assess the magnitude and likelihood of this potential impact.

2.4.2. Data and Information Gaps.

1. Lack of information about the type, presence and proximity of submerged aquatic vegetation to the growout facilities. Although the location of eelgrass is taken into consideration when establishing new growout facilities, it is unclear how often or thoroughly surveys of aquatic vegetation are conducted (whether they are presence-absence surveys or more formal taxonomic surveys) or who might be responsible for such surveys (individual facility operators, CDFW).
2. Although HSWRI and CDFW's Growout Procedures Manual (2007) describes the maintenance and cleaning practices that should be carried out at each growout facility,

little information exists about adherence to and implementation of these practices, or the routine maintenance and cleaning practices that are actually employed at the growout facilities. Individual growout facilities keep logbooks and most record cleaning events (especially those with fiberglass raceways), but these daily records are not routinely compiled and evaluated for adherence to practices. HSWRI staff keep only records of when growout facilities change the nets for the net pens they operate (M. Drawbridge email to T. S. Talley, 31 January 2017b).

2.4.3. Recommendations.

1. For each embayment that houses a growout facility, locate sources of maps that show relevant environmental features such as depth, and submerged aquatic vegetation that can be used periodically to demonstrate the proximity of the facility to features such as eelgrass beds.
2. Consider periodic review and update of the Growout Procedures Manual to help ensure minimal impacts of facility presence and operations on wildlife, aquatic plants and facilitation of invasive species (see Section 2.5.3, Recommendations 4 through 8).

2.5. Relation of environmental impacts to current and potential future regulatory compliance.

2.5.1. Key Findings.

HSWRI has complied with regulatory requirements by conducting a program to assess impacts to benthic habitat. HSWRI has carried out this program in the spirit of good environmental stewardship. From available sediment quality monitoring data, it appears that the environmental impact of OREHP operations is not negligible, but is likely low to moderate given the small footprint of the net pen and intermittent operations of the growout facilities over the past decade. That said, this monitoring could be improved to better document the extent and duration of the environmental effects on the surrounding environment. This would be particularly important if the hatchery operations are expanded, as the risk of more extensive or longer duration of impacts would increase as hatchery production is increased.

2.5.1.1. Carlsbad hatchery.

The likelihood of direct toxicity due to effluent discharge is low. Generally, based on data from a sample of influent versus effluent, most of the monitored parameters show no net increase or a slight increase (e.g. nitrogen). However, it is surprising that the required monitoring does not include effluent biological oxygen demand (BOD) and that continuous receiving water monitoring of DO and pH is not required. Other parameters in which HSWRI has demonstrated no significant difference between influent and effluent could be dropped (e.g. Cu and Zn) in order to keep net costs down if additional parameters are added.

Based largely on the available sediment free sulfide data, there is a moderate likelihood of

indirect effects on benthic habitat quality from organic matter deposition due to net pen operation. The magnitude of effect is likely high, given high sulfide concentrations detected, but is spatially limited to the area under and within 20 m of each net pen perimeter. However the duration of the impact has a greater likelihood of being long-term, because the net pens in this location are operated in a more continuous manner. Uncertainty in this determination is high because it is unclear to what extent these sulfide values translate to lasting impacts to the benthic community. Further, this system is fairly disturbed with sand influxes and consequent, occasional dredging by the nearby power plant. Improvements to the monitoring program should be considered in order to document both the types and relative magnitude of benthic and pelagic impacts. In addition, measurement of TN and TP concentrations in effluent do not allow an evaluation of the degree to which effluent nutrients are stimulating primary productivity. Given the fact that the Lagoon is well-flushed, it is not likely that localized increases in phytoplankton biomass and alterations in water column DO are occurring. Nonetheless, it would be reassuring to have improved documentation that no such effects are occurring.

2.5.1.2. Growout operations.

Use of the growout facilities has been somewhat sporadic over the past decade or so of operation. While available data show the likelihood that localized impacts to benthic habitat are occurring, as with the Carlsbad facility, elevated sulfide does not necessarily translate to significant impacts to benthic infauna and there is a lack of documentation of the actual condition of the benthic community. Because of the intermittent operation of these facilities, the likelihood of a significant risk is low. We note, however, that as HSWRI resolves issues with juvenile production, the risk of impacts to benthic habitat could increase because of more sustained operation of these facilities. Therefore, better characterization of the duration of impact of growout pen operation is needed in order to better inform the optimal period for pen fallowing, or pen movement and site fallowing, in order to allow habitat recovery.

2.5.2. Data and Information Gaps.

Information that would contribute to a more complete assessment of the environmental impacts of the hatchery and growout facilities includes:

1. Information about the maintenance and cleaning practices employed at the growout facilities.
2. Information about the presence, abundance, and types of invasive marine species on the facilities, especially species of particular concern (e.g., disease vectors, aggressive competitors or predators).
3. Information about the type, presence, and proximity of submerged aquatic vegetation to the growout facilities.

2.5.3. Recommendations.

Because the Carlsbad hatchery is a long-term operation, we believe this operation has a higher priority than the growout operations to better document potential environmental effects and undertake remediation if such effects are observed. Given this, we recommend:

1. Discuss revisions to the NPDES monitoring requirements with the San Diego Regional Water Quality Control Board to reflect a more streamlined suite of parameters that are more indicative of impacts at the Carlsbad Hatchery. Specific recommendations include:
    a. OREHP monitoring reports have demonstrated that parameters such as effluent suspended and settleable solids, pH, temperature, copper, and zinc are not significantly increased between influent and effluent. Likewise benthic monitoring has shown that unless current practices (including avoiding the use of copper treated nets) change, feed or pen net sources of zinc and copper are not likely to be an issue and these parameters could therefore be dropped.
    b. Based on current data and production rates, risk of contributions to the eutrophication of Agua Hedionda Lagoon is low and therefore requirements for monitoring of TN in effluent are not informative and could be dropped. In lieu of such monitoring, a limited study of intake and effluent BOD, net pen *in situ* continuous DO and chlorophyll-a, proximal to and away from the point of discharge plume could be undertaken in order to document receiving water condition at current nutrient and organic matter loading rates. We note that these requirements are not a recommended priority at this time, when production rates are low.
    c. If production is increased, the risk of eutrophication could rise. To preempt this, the OREHP should consider conducting a simple 1-D or 2-D modeling study to estimate the nutrient and organic matter loading ceiling above which more stringent monitoring should be required for adaptive management purposes. If the organic matter loading at maximum capacity is found to be negligible, much of the existing monitoring could be waived or greatly simplified.
    d. Monitoring at the current reference site should be discontinued. However, as it appears to be moderately to highly disturbed benthic habitat, the distal end of the orthogonal transects (≈ 80 m) can be added as an additional reference site.
2. Changes should be made to the benthic monitoring protocol that can be employed at either the hatchery or growout pens:
    a. At the hatchery, a standardized benthic macroinvertebrate (BMI) monitoring protocol and interpretational framework (statewide index of biological integrity for marine benthic macroinvertebrates (BMI) has been adopted by the State of California) would be recommended for use on a once per year basis to provide a more standardized basis for reporting and comparison to equivalent habitats.
    b. In order to minimize costs associated with benthic monitoring, we recommend partnering with Regional Monitoring Programs or specific agencies which already have a mandate for such monitoring to better leverage scarce monitoring resources. In Agua Hedionda Lagoon, San Diego County and the City of Carlsbad conduct Lagoon BMI sampling to comply with their municipal stormwater permit. For the growout

pens, consider partnering with the Regional Harbors and Ports monitoring program, which conducts benthic monitoring annually.

c. At the hatchery or growout pens, consider the use of sediment profile imagery (SPI) in both Agua Hedionda Lagoon and in the other growout facilities to more thoroughly map the spatial effects and to document recovery over time once the net pen is fallowed. While SPI represents a capital investment in monitoring equipment, the long-term continuous cost of its use is very low (e.g. staff time to process imagery) and could be of great benefit to map benthic habitat adjacent to hatchery net pen and growout facility operations. In addition to SPI, more frequent documentation of sediment %OC and %N should be conducted to indicate organic matter enrichment over time.

d. If benthic monitoring results indicate that operation of one or more growout pens is resulting in adverse impacts to benthic ecology or pen operations are proposed to be expanded or modified beyond those levels evaluated under the benthic monitoring program, opportunities for addressing benthic impacts through a standardized stocking regime should be considered (for example, evenly splitting the production runs between all growout facilities; more heavily stocking those facilities located at greater depths with more flushing potential; changing use patterns so that facilities are not used in consecutive years, etc.).

3. Existing monitoring and regulatory requirements should be enumerated and opportunities for increased coordination and consolidation of monitoring and reporting should be evaluated.

In addition, to help ensure that the presence and operation of the growout facilities continues to be carried out in a manner that minimizes the potential for adverse environmental impacts, periodic review and update of the Growout Procedures Manual (2007) should be considered to incorporate “lessons learned” and address potential new or emerging issues. Incorporating this type of opportunity for implementing adaptive management in the program's operations and management would not only serve to minimize the potential occurrence of adverse environmental impacts, but may also significantly expedite future regulatory permitting by addressing issues that may otherwise trigger permit conditions or a higher level of review. For example, if a new growout facility is proposed in the ocean and operational practices are managed and implemented in such a way as to involve no potential for any adverse effect on coastal resources, the Coastal Commission may issue a *de minimis* waiver for that facility rather than a coastal development permit - a less expensive and more expedient process. Along these lines, the following impact avoidance measures could be considered for possible inclusion in the Growout Procedures Manual:

4. Consistent with vessel maintenance best management practices in areas known to contain populations of invasive marine fouling species, growout facilities operators should consider employing onshore maintenance and cleaning operations that can be feasibly carried out onshore and involve removal of biofouling organisms. Such removed biofouling materials would best be collected and disposed of at an appropriate onshore waste disposal site, although this may be difficult to accomplish with volunteers.

5. To avoid the potential for unintentional disturbance, harassment, or injury to wildlife, operators should consider measures to limit attraction and use of net pen facilities by marine wildlife (primarily sea lions and predatory birds). Such measures can include the use of passive deterrent devices and other approaches (such as design and siting changes) that reduce interactions between wildlife and White Seabass in containment, if such measures have not already been implemented.
6. To help prevent accidental wildlife entanglement, operators or CDFW staff should consider evaluating growout facilities prior to stocking and once per month while stocked to help ensure that nets, lines, and deterrent devices are functioning properly and remain taut and intact. Documentation of wildlife entanglement incidents and relevant circumstances should continue to be included in annual reports to help facilitate adaptive management.
7. Prior to installing or moving net pen facilities, CDFW staff should continue to consider avoiding areas above or adjacent to submerged aquatic vegetation or high use areas for marine wildlife (roosts, haul-out sites, foraging areas, etc.).
8. CDFW should consider annual surveys of the submerged portions of each net pen facility to evaluate the presence of invasive marine species such as *Caulerpa taxifolia* and *Undaria pinnatifida* that may be locally or regionally present.
9. Reduce unnecessary and unintentional release of copper and zinc into the water column and benthic environment, by replacing any remaining copper-treated containment or exclusion netting with netting without copper, and by continuing use of feed with proteinated zinc.

Chapter 3

Objective 3. Maintain and assess a hatchery management plan that minimizes genetic effects on the wild population.

Before reviewing specific genetic issues with the OREHP enhancement program, a brief review of the main genetic risks associated with hatchery supplementation may be useful. Such risks have been reviewed extensively in the scientific literature on Pacific salmonids (Fraser 2008, Naish et al. 2008), where supplementation has been used for almost a century. The genetic risks associated with aquaculture and enhancement programs of marine species specifically have recently been reviewed by a National Oceanic and Atmospheric Administration (NOAA) Technical Memorandum (Waples et al. 2012). Three primary genetic risks were identified: loss of diversity within populations, loss of diversity among populations, and loss of fitness. In addition to the likelihood and magnitude of these risks, the report also discussed the likelihood and speed of reversibility of these losses.

Loss of genetic diversity within populations is primarily caused by extremely high reproductive success of hatchery fish compared to wild fish. Especially in marine species with high fecundity, few hatchery fish may produce a large proportion of the recruitment of the wild population. The effect is an increase in inbreeding, and therefore a large reduction in effective population size (N_e) and thus potentially in genetic diversity, as first described by Ryman and Laikre (1991). The extent of this reduction is relatively easily predictable from N_e of the hatchery stock (N_eh) and of the wild population (N_ew) as well as the contribution of the hatchery stock to the wild population. Recently, the analytical approach to estimating the Ryman-Laikre effect has been extended to marine populations, where the N_e/N ratio (the ratio between N_e and the adult census population size N) may be much smaller in wild populations than in hatchery stocks (Waples et al. 2016). Such reductions in N_e may have significant and serious effects on both short term viability and long term adaptability of populations. A minimum N_e of 50 has been proposed for the avoidance of short term inbreeding effects, while larger N_e of 500 or maybe 5000 may be necessary to maintain long term variability at quantitative traits (Franklin 1980, Lynch and Lande 1998). In marine fishes, these numbers may be misleading, because the number of alleles that can be maintained in a population is an almost linear function of N_e (Ryman et al. 1995, Waples and Naish 2009). A reduction from a very large (e.g. $N_e=10^9$) to a large population (e.g. $N_e=10^4$) may therefore not be particularly worrisome in terms of reaching critical values of N_e, but it may cause a loss of the majority of alleles in that population. Such alleles may be crucial for rapid adaptation in rapidly changing environments, and their loss may considerably impair long term persistence of marine populations (Ryman et al. 1995). The loss of such alleles could never be reversed in completely isolated populations, but may be compensated by mutation over evolutionary time scales. In connected populations, immigration may restore diversity relatively quickly (depending on migration rate), so connectivity among populations is a major aspect in assessing this risk (Duchesne and Bernatchez 2002).

Loss of genetic diversity among populations can occur if widespread contributions from few predominant aquaculture stocks supplant an existing system of locally adapted populations. This

genetic homogenization can reduce the variability among populations in demographic responses to environmental changes and thus the resilience of the species to such changes (portfolio effect, Schindler et al. 2010). For example, declines in population structure were caused by extensive transfers between Coho Salmon (*Oncorhynchus kisutch*) hatcheries in Puget Sound, USA (Eldridge and Naish 2007) as well as by three decades of aquaculture escapes of Atlantic Salmon (*Salmo salar*) in Norway (Glover et al. 2012). In the Snake River system (Oregon), hatchery releases and dam construction led to a reduction in population diversity of threatened Chinook Salmon (*O. tshawytscha*) and concurrently reduced the portfolio effect and increased abundance variance (Moore et al. 2010). In marine species with their high dispersal potential, population structure tends to be naturally less pronounced than in anadromous fishes, though recent research suggests the existence of local adaptation (Hauser and Carvalho 2008, Sanford and Kelly 2011) and portfolio effects (Siple and Francis 2016) even in highly connected populations. Once genetic population structure is lost, its restoration at neutral and weakly selected loci may take a long time to reach previous levels in large populations (Whitlock 1992). Local adaptation, on the other hand, may be restored relatively quickly if it is due to genes under relatively strong selection (Hutchings 2014), though the mechanisms leading to local adaptation in the presence of gene flow are still uncertain (Nielsen et al. 2009, Sanford and Kelly 2011).

Loss of fitness can occur by domestication or by interbreeding between divergent populations due to outbreeding depression or dilution of local adaptation (McClelland et al. 2005, Frankham et al. 2011). In Pacific salmonids, the reproductive success of hatchery-produced fish in the wild appears to be lower than naturally born individuals (Araki et al. 2007, Christie et al. 2012, Christie et al. 2014), possibly because of adaptation to captivity (i.e. domestication, Lorenzen et al. 2012). Such reduction of fitness may occur after a single generation in captivity (Christie et al. 2012) and may thus be relevant to marine supplementation programs, although the relevance of this reduction in fitness to long-term population viability may be dependent on the rearing and release strategies (Baskett and Waples 2013). In a review of 266 studies on the genetic effects of hatchery programs, including salmonids, flatfish, bream, drum, cod and invertebrates, Araki and Schmid (2010) found significant loss of fitness of the cultured species in 23 out of 70 studies comparing wild and hatchery stocks, while none suggested a fitness gain. Predictions become more complicated when hatchery stocks are genetically diverged from wild populations – in such cases, both a fitness gain (heterosis) and fitness loss (outbreeding depression) are possible (McClelland et al. 2005, Hedgecock and Davis 2007, Frankham et al. 2011). In some cases, heterosis observed in the first offspring generation shifts to outbreeding depression in subsequent generations as co-adapted gene complexes become interrupted by recombination. Such a situation may be particularly damaging, as hybrids between hatchery strains and local populations may initially perform better, but may reduce population fitness in later generations. Predicting the interplay between heterosis and outbreeding depression is not possible based on genetic distances among populations estimated from molecular markers (McClelland and Naish 2007), but requires specifically designed experiments (Frankham et al. 2011). Unfortunately, very little is known about the likelihood and timescale of recovery from such loss of fitness due to hatchery supplementation.

Importantly, none of these genetic effects are relevant if hatchery derived individuals do not interbreed with the wild population. If they do interbreed, however, the effects can be severe and difficult to reverse (Waples et al. 2012), but there are conditions under which changes may be small (Mobrand et al. 2005, Baskett and Waples 2013, Waters et al. 2015). It is thus crucial to establish a genetic monitoring program (Schwartz et al. 2007) that quantifies trends in the extent of interbreeding between hatchery derived and wild individuals.

3.1. Genetics of wild White Seabass populations.

3.1.1. Key Findings.

3.1.1.1. Population structure.

The population structure of the supplemented (wild) population is very important for prediction of both the genetic effects of hatchery supplementation and the likelihood and timeline of recovery (see above). Over the course of the OREHP, several research efforts have generally concluded that there is not much evidence for population structure. However, most of these studies had relatively low power, so the true structure of the stock is still unresolved. In particular, no study to date has concentrated on spawning fish - if white seabass home to their spawning location (like many other marine fish, e.g. Atlantic and Pacific cod), genetic structure among spawning populations may be undetectable in samples of mixed feeding aggregations.

An early study using enzyme polymorphisms (allozymes, Bartley and Kent 1990) screened samples from eight locations in the California Bight at 19 polymorphic loci, and detected some significant genetic differentiation, but failed to detect consistent geographic population structure. Similarly, another study did not detect population structure based on mitochondrial DNA D-loop sequences (Medina 2008).

More recent studies have been based on microsatellite DNA. In the first study by Franklin (1997), samples of 12 individuals were collected from ten locations ranging from the Northern Channel Islands to Baja California and the Sea of Cortez, and screened at eight microsatellite loci that were developed specifically for this study. No tests for suitability of the loci were carried out, in particular, tests for Hardy-Weinberg equilibrium, which may detect artifacts like non-amplifying null alleles. The statistical power of tests was quite low, because sample sizes were small and some microsatellites had few alleles. The interpretation of patterns of genetic differentiation was somewhat peculiar: although the final conclusion in Franklin (1997) was that it was “not possible to identify discrete subpopulations” (abstract), the conclusion in the thesis (p. 62) suggests two major stocks, and pairwise tests among sample sites (Table 6 in Franklin 1997) seemed to suggest three populations: one in the northern Channel Islands, one around the Baja peninsula and one in the central California Bay. This interpretation is somewhat different in the White Seabass Enhancement Plan (2010), which also refers to three populations. In any case, similar to the allozyme study above (Bartley and Kent 1990), the study provides some suggestions of population structure, but is certainly not conclusive.

The same microsatellite loci were also used in two other studies. Coykendall (2005) used six microsatellites developed by Franklin (1997), without citing him, and screened by Genetic Identification Services (GIS, Chatsworth, CA) on samples from six locations collected over four years. The sampling scheme was highly unbalanced, with sample sizes of 11, 32, 50, 96, 108 and 297 individuals. Three of the loci showed significant deviations from Hardy-Weinberg equilibrium in the three largest samples, and probably should have been excluded from the analysis, as null alleles may affect estimates of genetic differentiation. The median pairwise F_{ST} was 0.003, and only two of the 15 pairwise comparisons were significant, and so this study showed no evidence of genetic population structure. Buonaccorsi et al. (2001) used the same microsatellites (Franklin 1997) on three samples from Mexico, Los Angeles and Santa Barbara (N=80 each), found similar deviations from Hardy-Weinberg equilibrium and also no evidence for genetic population structure.

Recently, Franklin published the data from his PhD., with much updated statistical analysis, expanded sample set, and using only the five loci that did not show deviations from Hardy-Weinberg equilibrium (Franklin et al 2016). Samples were collected over a wide geographic range, but grouped into four regions: Channel Islands (N=75), central California coast north of San Quintin (N=69), Pacific Baja south of San Quintin (N=16), and northern Gulf of California (N=17). Except for the comparison between Channel Islands and central Californian coast samples, all comparisons between samples were significant, with relatively high levels of microsatellite F_{ST} for marine species (0.033 – 0.126). The study therefore recovered genetic barriers at the San Quintin upwelling area and the Baja California peninsula. Although these barriers have also been found in other species (albeit with much lower F_{ST} values; kelp bass, *Paralabrax clathratus* (Selkoe et al. 2007), barred sand bass, *P. nebulifer* (Paterson et al. 2015)), it should be noted that all significant comparisons in Franklin et al. (2016) involved very small sample sizes, which tend to inflate F_{ST} values.

In summary, the evidence for population structure is equivocal. Early allozyme and microsatellite studies did show some genetic differentiation, which later microsatellite studies (based on somewhat unreliable markers) could not confirm. A recent reanalysis of an earlier study (Franklin et al. 2016) provided some evidence for population differentiation, but was compromised by small sample sizes from differentiated populations. None of the studies carried out so far had the statistical power necessary to detect presumably subtle population structure in a marine fish. A recent study on otoliths found no geographic variation in growth patterns and thus concluded that there was likely a single stock, but also called for additional stock structure studies with phenotypic and genetic markers (Romo-Curiel et al. 2015).

Preliminary data are available from a study employing next generation sequencing techniques, which potentially have much higher resolution power than traditional genetic approaches described above. This ongoing study by researchers at the University of Hawai'i (J.L. Whitney, M. Iacchei and R.J. Toonen) and the University of California Santa Barbara (K.A. Selkoe, H.S. Lenihan) employed ezRAD genetic sequencing techniques (Toonen, et al. 2013) on pooled population samples. According to M. Iacchei and K. Selkoe (pers. comm.), the study included ten samples of 20 individuals each, from eight collection sites spanning from Catalina Island, CA in the north to

San Juanico, Baja California (Sur), Mexico in the south. Fish were collected in spring and summer (March-September), but it is unclear if they were in spawning condition. For two of these sites, two replicate samples with twenty individuals each were used to confirm population allele frequency estimates. Approximately 261 sequences were resolved, with an average of six polymorphic sites in each sequence (1565 total SNPs), with each polymorphic site present in all 8 pools at higher than 40x sequencing coverage. Preliminary results revealed no differentiation among wild samples, indicating a lack of genetic structure in the wild population. Further research employing powerful next generation sequencing techniques, and using samples from spawning aggregations when populations are most likely to be separated, is needed to further test population structure in the species. Results from this preliminary study are in preparation for publication.

3.1.1.2. Effective population size.

Estimates of effective size of the wild population (N_e) would be crucial to predict the effects of hatchery supplementation on genetic diversity. Some estimates are available, but they vary widely and probably should be used with caution. Furthermore, it is unclear which population segment these estimates refer to, as California waters constitute only a small proportion of the total species range.

Coykendall (2005) estimated long term N_e from coalescence of mtDNA sequences, and obtained an estimate of female effective population size of N_{ef}=54,260. However, this estimate assumes a specific and unverified mutation rate, exponential population growth and lack of population structure or selection. Because long-term N_e estimates are very sensitive to even small migration rates, this estimate pertains probably to the entire species rather than just the California component. There are also no confidence limits of this estimate and so this estimate should not be used for extant Seabass populations.

Coykendall (2005) also estimated shorter term N_e from six microsatellite markers. Although these approaches are more suitable for short term estimates of N_e, the loci used in that study had non-amplifying null alleles which may bias estimates. The analysis compared temporal samples by reconstructing nine cohorts (age classes) estimated from head length and van Bertalanffy growth parameters. Membership to these age classes is therefore likely to be fairly inaccurate, and estimates of confidence limits do not include all sources of uncertainty. Nevertheless, Coykendall obtained a moment based estimate (Waples 1989) allowing for overlapping generations (Jorde and Ryman 1995) of N_e=5679 (3977-7678, 95% confidence limits). In addition, they used a likelihood based estimate (Wang 2010) of N_e=6078 (2384-57,310, 95% confidence limits) – however, this estimate does not allow for overlapping generations. The confidence limits of the latter estimate are derived from a x^2 distribution, which was a poor fit to the data, so they are likely inaccurate (Coykendall 2005).

Buonaccorsi et al. (2001) used eight microsatellites, six of which were the same as used by Coykendall, and also suffered from null alleles. They did not estimate N_e, but the data were made available to the review panel. Using linkage disequilibrium to estimate N_e (Waples and Do 2008),

all three samples provided an estimate of approximately N_e= 2000, with an upper confidence limit of infinity, suggesting that N_e is not small.

Estimates of N_e/N from life tables allow some conclusions on the reduction of N_e caused by age structure (Waples et al. 2011). Although this method does not provide an absolute estimate of N_e, we estimated that the N_e/N ratio would be 0.78, if reproductive success was completely random within age classes (Table 3.1). The variance in reproductive success within age classes is likely much higher, but we have no estimate for this parameter.

Table 3.1. Estimates of N_e/N based on the life table information (Table 4.1 in Coykendall 2005) and AgeNe (Waples et al. 2011). Φ = $V_k/\underline{k}$; V_k= variance in reproductive success; $\underline{k}$= mean reproductive success. Φ = 1 denotes random reproductive success in each age class.

φ	Ne/N
1	0.78
5	0.39
10	0.236
100	0.03
1000	0.003

3.1.1.3. Summary.

Population genetic studies to date have provided very limited insight into the size and the structure of the wild population. Small sample sizes, technical artifacts and analytical issues limited the power of these studies. All estimates of wild N_e are problematic, but none suggest very low N_e. True N_e may therefore be at least in the high thousands. The geographic range to which this estimate applies to is unclear because there is uncertainty in population structure. It should be noted that the California White Seabass population is only a small proportion of the total species abundance, which has the center of distribution in Mexico. All these issues complicate predictions of the genetic effects of the hatchery program, and additional studies are urgently required.

3.1.2. Data and Information Gaps.

In order to assess the genetic effects on the wild White Seabass population of the hatchery management plan and subsequent practices, the information and data listed here pertaining to the wild population are needed:

1. Population structure of wild population.
2. Effective size of wild population, especially φ (variance in reproductive success within age classes).
3. Ne/N ratio in wild population.

3.1.3. Recommendations.

We recommend the following two data collections and analyses to fill gaps in information needed from the wild populations, and to improve consistency of involvement of OREHP scientists in scientific research and data collection performed by non-OREHP collaborators:

1. A thorough population genetic investigation should be used to establish (i) population structure and (ii) effective population size in the wild population. Based on this information, the effect of enhancement should be reassessed. The ongoing ezRAD study is a good start, but additional efforts are likely needed.
2. Increase efforts to recapture hatchery-reared adults, collect genetic samples, establish parentage and estimate genetic diversity.

3.2. Hatchery management.

3.2.1. Key Findings.

3.2.1.1. Broodstock Management Plan: Development, assessment and updating process.
The original broodstock plan was developed by Bartley et al. (1995), mainly based on the concept of capturing rare alleles and maintaining heterozygosity as detected by allozymes within the broodstock. They concluded that a minimum broodstock of 74 individuals would have a probability of 95% to contain rare alleles with a frequency of 0.02 and represent 99% of the heterozygosity of the wild population. They also estimated that the effective population size (N_e) would be about half of the census size (N), and so recommended a broodstock size of about 150 fish. Rotation of broodstock among holding pools was recommended to increase the diversity of matings. Pertinent recommendations also included estimating N_e empirically, and physical marking and genetic characterization of larvae and juveniles.

This broodstock management plan was updated in 2008, and again in 2011, mainly based on the work by Gruenthal and Drawbridge (2012), and Gruenthal et al. (2014). Essentially, Gruenthal and Drawbridge (2012) established that reproductive success in captivity was more variable (and thus effective population size lower) in females than in males, and therefore recommended changing the sex ratio of broodstock to 60% females. Furthermore, based on their estimate of effective number of breeders they recommended a total broodstock size of 140-200 fish. In addition, they recommended an increase of the annual broodstock replacement from 20% to 25%, but removed the requirement to rotate broodstock among the four pools mainly because of logistic constraints.

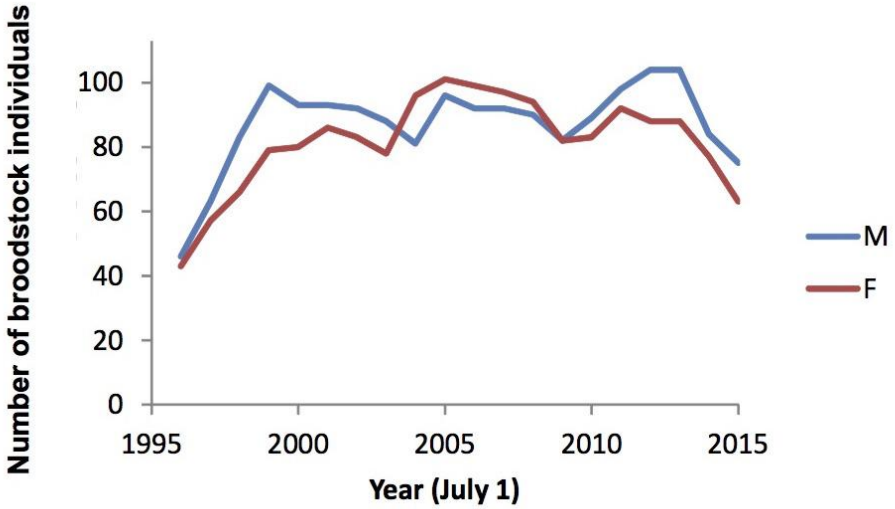

Fig. 3.1. Number of male and female broodstock held at HSWRI.

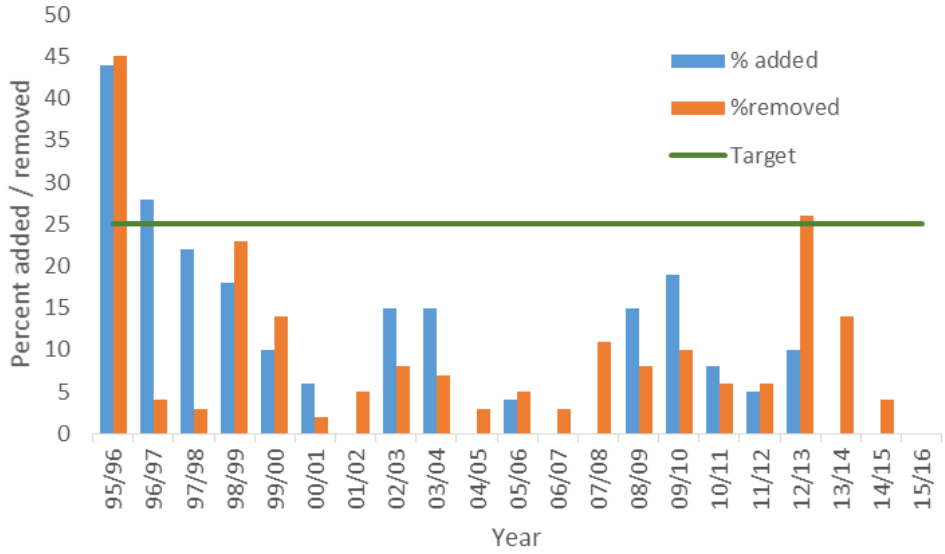

Fig. 3.2. Percentage of broodstock that were added and removed each year. The green horizontal line represents the 25% target exchange rate.

These recommendations were followed only to some degree: the broodstock size has been above 160 fish since 1999. Since 2010, the broodstock was slightly male biased (Fig. 3.1), and the desired sex ratio of 60% females was never fully achieved. However, the rotation of broodstock in and out of the system fell far short of the 25%/year suggested by Gruenthal et al. in the Broodstock Management Plan (2011), possibly because of funding and logistic constraints (Fig 3.2).

Instead of an average residency in the broodstock of about four years, the average time in the system was 8.2 ± 5.2 (1 SD) years for males and 7.8 ± 5.9 years for females, with a wide range of residence times between 0+ and 19+ (Fig. 3.3). About 40% of the broodstock was in the system for more than 10 years. Of the 132 fish currently in the system, 11 (seven males and four females) have 'served' since 1996, after the hatchery facility was constructed, and 59 (30 males, 29 females) have been in the system for more than 10 years. Interestingly, the rationale for the recommended 25% broodstock rotation by Gruenthal et al. was based on their estimate of a four-year generation time (Broodstock Management Plan 2011). More empirical estimates based on life tables give longer generation times of 7.88 years (MacCall et al. 1976, in Coykendall 2005) and 7.4 years from the life table in Coykendall (2005, Table 4.1). These estimates of generation times are similar to the residence time in the broodstock. However, this similarity is misleading for two reasons: first, to maximize N_e in the hatchery, the optimal strategy would be to exchange broodstock every year, although it is recognized that this may not be realistic every year due to the difficulty in capturing sufficient numbers of broodstock annually. Second, the empirical generation time in the hatchery is about 14 years, because generation time is defined as the mean age of parents, and broodstock entered the hatchery at several years of age.

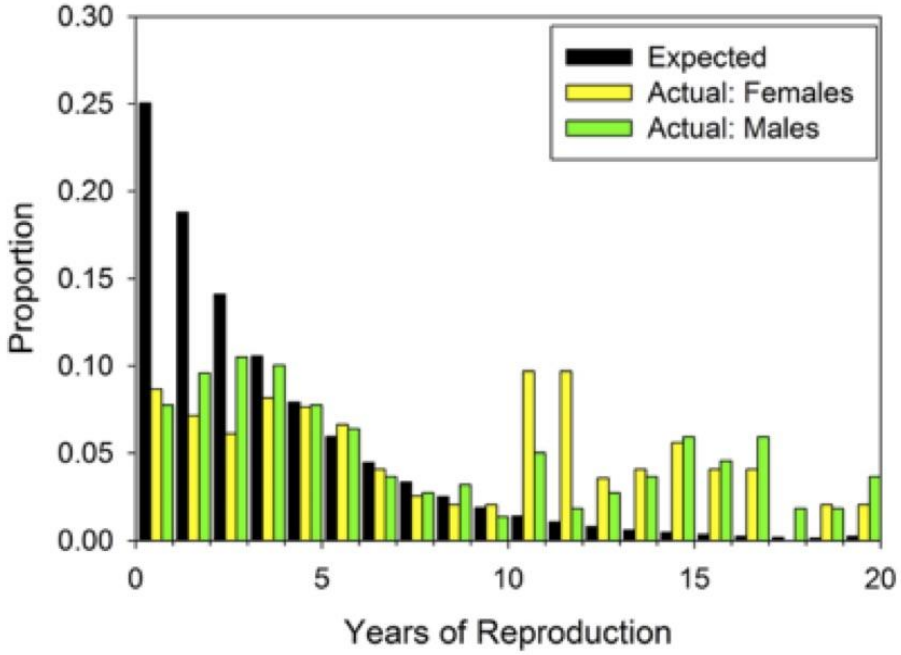

Fig. 3.3. Comparison of expected and actual distributions of number of years of reproductive lifetime in the White Seabass captive program. The expected distribution assumes random replacement of 25% of the broodstock each year. The empirical distributions reflect the number of years individual males and females spent in the broodstock.

The primary effect of this long residency and especially the high variance in residence time is: (1) a smaller number of broodstock used in total, and (2) a high variance in reproductive success and consequent low effective population size in the hatchery.

1. The average number of broodstock was 177 fish since 1996 – consider 160 fish for simplicity. If 25% of that broodstock (40 fish) had been exchanged every year, 920 fish would have been through the system over the 19 years of operation (160+40*19=920). Instead, less than half that number was used (416 fish).
2. If every fish in the broodstock had been replaced systematically, the reproductive span of each fish would have been exactly four years, and not considering other factors, reproductive success would have been random (N_e=416). If 25% fish had been replaced randomly, a relatively wide distribution of residence times would have been expected (Fig. 3.3). In that case, the N_e/N ratio would have been about 0.57 (see Table 3.1 for approach), even if reproductive success was random among fish of a given age (i.e. total N_e=237). Compared to this expectation, a much larger fraction of males and females reproduced over more than 10 years, and a much smaller fraction of both sexes reproduced only for 1-3 years. This means that effective population size was likely much smaller in the captive population than if the broodstock plan was completely adhered to, though exact calculations are difficult because fish entered the broodstock at varying ages and fecundity. In addition, N_e may be further reduced by variation in weight dependent fecundity among fish (i.e. older larger fish are more fecund and may produce offspring with better survival, Berkeley et al. 2004) and acclimatization to the system (i.e. reproductive success may be lower in the first year). Using genetic parentage assignment, these effects could be, but have not been, quantified.

An additional concern is whether broodstock fish represent a random sample of the population, rather than fish that are either particularly suited to hatchery conditions, or biased in some other way. *A priori* there is no reason to assume such a bias, though the long residence time and potentially targeted exchange of less productive fish may result in broodstock that differs from wild fish. Somewhat concerningly, a recent (yet unpublished) study employing 261 ezRAD loci on pooled sampled of 20-25 fish (J.L. Whitney, M. Iacchei, R.J. Toonen, University of Hawai'I, and K.A. Selkoe, H.S. Lenihan, University of California Santa Barbara, discussed in Section 3.1.1.1) detected significant differentiation between HSWRI broodstock and wild samples. No differentiation was found among ten samples of wild fish, or between the two samples of HSWRI broodstock, but the two broodstock samples were significantly differentiated from wild samples (F_{ST}~0.05). This result is difficult to explain, because broodstock were caught from the wild and thus should represent a random sample of the wild population. Temporal differentiation, biased sex ratios, and inclusion of related individuals appeared unlikely causes. Inadvertent selection on broodstock fish in the hatchery also appears unlikely, as mortality (and exchange rates) were low (see Section 3.2.1.1), and the detected genotypic differentiation was not restricted to few loci (which would have suggested selection at nearby genes). Methodological issues related to the pooling of individual fish for sampling are possible, though the fact the differentiation was only found between broodstock and wild, and not within these two groups, is puzzling. Validating and explaining this surprising differentiation between wild and captive fish would require genetic analysis of individual fish and larger sample sizes.

3.2.1.2. Photothermal regime in broodstock.
Most temperate zone teleost fishes, such as White Seabass, reproduce seasonally with release of gametes programmed so that progeny are produced when environmental conditions are favorable for survival. Thus, the entrainment of reproductive cycles to favor offspring survival is paramount for reproductive success in nature. A range of environmental (photoperiod, lunar cycles, temperature, salinity, rainfall), nutritional, and social factors play important roles in controlling the timing of spawning, though for most temperate zone species photoperiod and temperature are the dominant factors (Migaud et al. 2010). Therefore, photothermal regimes are commonly used in commercial finfish aquaculture to synchronize or alter spawn timing to maximize year-round production of larval fish for growout (Bromage et al. 2001; See Section 3.2.1.6 for more discussion of photothermal regimes and domestication).

3.2.1.3. Effective population size.
Some estimates of effective population size for the captive White Seabass population are available. However, it should be noted at the outset that all these estimates were derived from the variance in the number of juvenile offspring among spawners, whereas N_e estimates of wild populations refer to the adult to adult variance in reproductive success. Given relatively high post-release and juvenile mortality, these estimates are likely to be overly optimistic, and true N_e may be much lower. In that way, the explicit aim of the OREHP to "Maintain and assess a broodstock management plan that results in progeny being released that have genotypic diversity very similar to that of the wild population" (White Seabass Enhancement Plan 2010) is already inherently flawed because it does not consider adult to adult survival. Nevertheless, given clear logistic constraints to estimate adult-adult reproductive success of individual hatchery fish and this explicit aim of the program, it is crucial to establish a genetic monitoring plan that monitors family contribution at each stage of the rearing process. According to HSWRI, such research was in progress, but funding ran out.

As in the wild population, estimates of N_e are also complicated by the iteroparity and longevity of Seabass. To avoid these complications, all available estimates refer to N_b, the effective number of breeders of a spawning event, a spawning season or a year. N_b is generally smaller than N_e, because N_b represents parental contributions in one year instead of an entire generation, though the exact relationship depends on specific life history characteristics (Waples et al. 2011). Table 3.1 shows the reduction in N_e/N based on age structure in the wild population – estimating this for the captive broodstock is difficult because the approach assumes a stable age structure. Empirical estimates below should therefore be seen as an indication of some factors reducing the N_e/N ratio rather than as absolute estimates of effective broodstock size.

Empirical estimates from molecular genetics. Coykendall (2005) estimated N_b in 10 spawning events in two years (5 spawning event in 1998/1999, five from 2001) with 254 possible parents and 6-7 microsatellites developed by Franklin (1997). She estimated inbreeding and variance N_b per spawning event from the mean and the variance of reproductive success estimated from parentage assignment from 7 microsatellites using methods in Lande and Barrowclough (1987) and Crow and Denniston (1988). The success of parentage assignment was quite low, and only 83% of juveniles could be assigned to at least one parent (range 23%-100%). Both estimates of N_b

were very low, ranging from 2 to 8.1 individuals and giving an N_b/N ratio ranging from 0.03-0.08. Coykendall (2005) also estimated N_b from allele rarefaction (Hedgecock et al. 2007), which is less direct but has the advantage that all juveniles can be used, not only those which can be assigned to parents. Using this method, she obtained an estimate for the 2001 'spawn population' of N_b = 34.6 individuals (95% confidence limits, 20.6-76.5) and of N_b/N of 0.14.

Coykendall's (2005) estimates and approach were severely criticized in an appendix of the White Seabass Enhancement Plan (Gruenthal and Drawbridge 2008). There were several criticisms of the N_b estimates from parentage analyses. First, the broodstock size used by Coykendall (2005) appears to be a cumulative number over three years in all four tanks, whereas only broodstock in two tanks and two years contributed to the analyzed juveniles. Second, juveniles over several spawning events were combined, and there is confusion about the number of spawn events analyzed. Probably more importantly, few spawn events were analyzed, probably only representing 5% of the total output for the year. Not mentioned by Gruenthal and Drawbridge (2008) are potential problems with the parentage assignment. Given a relatively small broodstock and known mating groups, the assignment success was surprisingly low, and no probabilities for parental assignments were provided. In such circumstances, parents with rare alleles may be assigned false offspring (Wang 2010), thus increasing variance in reproductive success and reducing N_e. Gruenthal and Drawbridge (2008) also criticized estimates from the allele rarefaction method, mainly because of unrepresentative sampling of released fish and, again, comparison to the entire hatchery broodstock rather than the specific groups producing the offspring. All these criticisms appear to be valid, and so these estimates have to be enjoyed with caution.

The most reliable estimate of N_b (albeit based on a very early life stage) comes from Gruenthal and Drawbridge (2012) who assigned parentage to yolksac larvae in one broodstock tank over a breeding season. Probabilities of parentage assignments are also not provided, but simulations suggested that all offspring could be assigned to both parents with more than 95% confidence. Gruenthal and Drawbridge (2012) used five of the same microsatellites as Coykendall (2005), but parentage assignment was more powerful because only one broodstock tank with 25 males and 25 females was used. Gruenthal and Drawbridge (2012) screened 4249 larvae from 71 spawn events over a single breeding season and used demographic estimates of N_b from parentage assignment as well as indirect estimates from linkage disequilibrium. More than half of all spawn events (36) had a single female parent of more than 95% of larvae, and multiple females contributed relatively equally only in a third of the spawns. Consequently, the effective number of breeders was low per spawn event (mean N_b=5.3 ± 3.6), but relatively high over the season (N_b=31.1, N_b/N=0.62).

The implication of the results by Gruenthal and Drawbridge (2012) is that single spawn event may well represent single families (half-sib families produced by one female mated with several males), but that different females are active at different spawn events (see also discussion of female equivalents below). Very similar pattern were seen in a Red Drum hatchery supplementation program, where females also had higher variance in reproductive success, and where N_b was small in single spawn events but larger over the season (Gold et al. 2008). In reality, the OREHP used a fairly large number of spawn events for releases: according to recently

released data (HSWRI Releases Dataset 2016), over the last 10 years offspring from an average of 32 spawn events were released annually. However, on average over the last 10 years, juveniles were derived from only 6-10 spawns per broodstock tank annually (Table 3.2): N_b/N may therefore be much lower than the N_b/N=0.62 estimated from 71 spawns by Gruenthal and Drawbridge (2012).

Table 3.2. Number of spawn events contributing to annual releases of juvenile White Seabass. Note that B5 was a tank that was used as replacement of B4. Tank B3 was out of service in 2015-2016. Average number of spawn events are provided for the last 10 years and for all years since 1996. Note the Tank B3 was out of service in 2016.

Year	B1	B2	B3	B4	B5	Total
1996	0	10	0	0	0	10
1997	0	2	29	0	0	31
1998	14	10	2	0	0	26
1999	14	0	1	9	0	24
2000	5	1	13	17	0	36
2001	18	25	20	29	0	92
2002	28	25	11	23	0	87
2003	13	64	28	30	0	135
2004	10	27	25	75	0	137
2005	4	24	9	18	0	55
2006	3	23	3	9	0	38
2007	11	13	16	19	0	59
2008	4	6	11	14	0	35
2009	12	2	8	2	0	24
2010	0	6	5	1	6	18
2011	3	4	13	0	11	31
2012	3	4	13	4	2	26
2013	9	3	15	1	10	38
2014	0	9	19	1	1	30
2015	16	5	0	3	0	24
2016	0	4	0	0	0	4
Total	**167**	**267**	**241**	**255**	**30**	**960**
Average over last 10 years	**6.1**	**7.5**	**10.3**	**5.4**	**3**	**32.3**
All	**8.0**	**12.7**	**11.5**	**12.1**	**1.4**	**45.7**

Other estimates of N_b: Female equivalent. Gruenthal and Drawbridge (2012) discovered an interesting proxy for the estimation of the number of effective females in a spawn event, the female equivalent (*fe*). Based on genetic parentage, they found a tight correlation between the total volume of eggs produced and the effective number of females involved in the spawn (r^2=0.16) and that approximately 3 liters of eggs corresponded to one effective female (or female equivalent). Reanalysis of their data showed that the correlation was even tighter with the effective number of breeders N_b (r^2=0.34; Fig. 3.4). There were outliers in both relationships, but most involved spawns with more effective breeders producing relatively few eggs, indicating that the relationship is quite conservative. Gruenthal and Drawbridge (2012) recommended 28-32 female equivalents per year, which would give a N_b =57-120, if Fig. 3.4 can be simply extrapolated. However, it probably cannot because breeders may be involved in several events, and so the relationship likely reaches a plateau. Their method is a very innovative way to estimate N_b from very basic data. The guideline of the number of female equivalents per year seems to have been heeded in terms of the number of spawns per year (Table 3.2), but data were not reported in annual reports, even in years when the method was specifically mentioned (Annual Reports 09-10, 10-11)).

Contribution of different broodstock tanks. Reductions in N_e compared to census size can be due to variance in reproductive success beyond random expectations, but also due to variation in productivity among demes (or groups of individuals). In fact, the extremely low N_e/N ratio (N_e/N = 10^{-3}) in Red Drum in the Gulf of Mexico was primarily explained by variance in productivity among subpopulations in different estuaries (Turner et al. 2002). This may be a consideration at the hatchery where batches of juveniles from each broodstock may be exposed to different environmental conditions that can influence their survival, such as in the J2 system where fish are exposed to different temperature ranges and acclimated for varying amounts of time before release depending upon ambient water temperatures (e.g., 80-90 days to 120 days in colder months) (White Seabass Enhancement Plan 2010). Such effects are particularly relevant if there is genetic differentiation among and inbreeding within demes (Nunney 1999), but even differences in release numbers among the four breeding stocks could seriously decrease N_e, down to the N_e of a single broodstock if juveniles of only that stock were released. However, even though numbers released from each group varied (Table 3.3), release numbers from the four groups were reasonably even, and applying equation 19 in Nunney (1999) to total release numbers in each group (combining B4 and B5) suggests that the N_e/N ratio caused by unequal release number was 0.96, assuming random reproductive success within broodstock groups. These calculations are not entirely correct, because Nunney (1999) did not consider overlapping generations. Nevertheless, they suggest that the difference in release numbers between broodstock tanks does not cause an excessive reduction in N_e. Note, however, that it may be possible to increase N_e above N by completely equalizing output from the four broodstocks.

Table 3.3. Number of juveniles of each broodstock group released per year. Note that B5 was a tank that was used as replacement of B4, and tank B3 was not in service 2015-2016.

Year	B1	B2	B3	B4	B5	Total
1996	0	14,013	0	0	0	42,061
1997	0	7,610	50,214	0	0	57,824
1998	13,995	16,160	1,885	0	0	32,040
1999	16,382	0	715	8,018	0	25,115
2000	6,290	821	5,788	14,946	0	27,845
2001	25,783	20,912	23,399	31,826	0	101,920
2002	21,266	29,214	11,049	62,509	0	124,038
2003	10,397	69,673	22,385	39,536	0	141,992
2004	25,476	35,098	82,352	127,988	0	270,913
2005	7,160	46,868	13,947	32,937	0	100,911
2006	7,419	72,313	2,594	27,936	0	110,261
2007	25,219	28,221	52,664	93,579	0	199,682
2008	1,627	16,681	20,630	19,546	0	58,484
2009	69,457	27,013	52,196	3,992	0	152,658
2010	0	25,355	35,546	1,115	26,986	89,002
2011	1,932	23,333	63,259	0	9,272	97,796
2012	5,227	24,535	31,266	16,375	31,391	108,794
2013	27,231	3,276	94,266	6,911	37,801	169,484
2014	0	8,956	53,678	11,252	7,003	80,889
2015	94,424	15,746	0	4,764	0	114,933
2016	0	12,178	0	0	0	12,178
Total	**359,284**	**497,974**	**617,832**	**503,229**	**112,453**	**2,118,820**

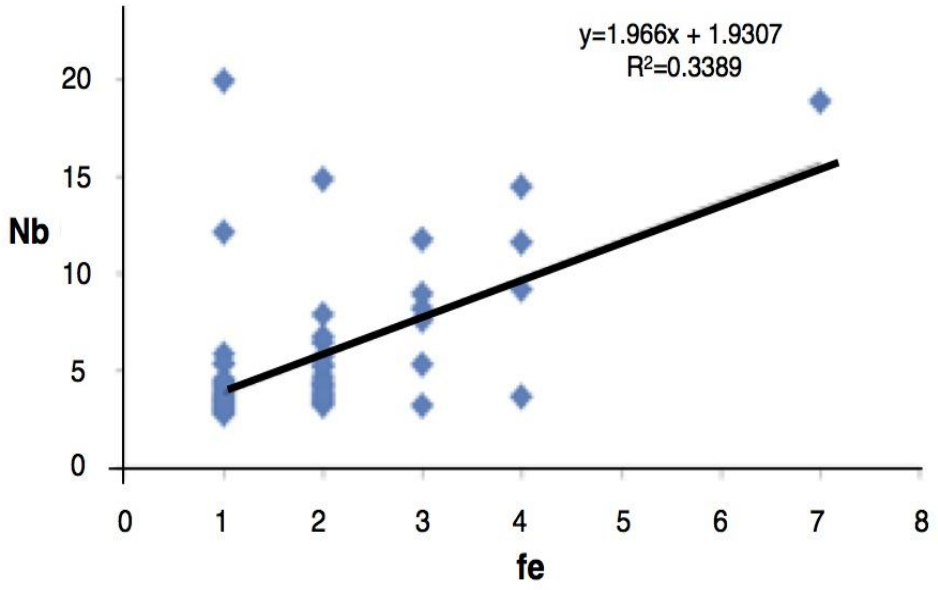

Fig. 3.4. Effective number of breeders (individuals) as a function of female equivalents (*fe*; egg volume). Data from Gruenthal and Drawbridge 2012.

3.2.1.4. Summary of broodstock management.

In summary, the success of the broodstock management program to maintain and assess a broodstock management plan that "results in progeny being released that have genotypic diversity very similar to that of the wild population" is difficult to assess because of difficulties in estimating the effective size of the broodstock. However, it is likely that the current practices fall well short of stated targets, for the following reasons:

1. Average residence time in the hatchery of adult broodstock was 8 years instead of the planned four years, with some fish residing for 20 years in the hatchery. This resulted in a 50% reduction of total number of adult fish used for spawning (N=416 instead of 920 fish) and a reduction in the N_e/N ratio from 0.57 to a much smaller, but unknown proportion.
2. Despite plans for a female biased broodstock, there were consistently more males than females, although the effect of this bias is likely small.
3. Empirical estimates of N_b/N range from 0.03-0.62 (Coykendall 2005, Gruenthal and Drawbridge 2012). This wide range can be explained by the variety of juveniles and adults used in the analyses, though larger estimates appear more reliable.
4. Some variation in the number of juveniles released from each broodstock tank, age structure and limited number of spawn events may further reduce the effective population size of the hatchery broodstock.
5. An interesting method of estimating N_b from egg volume was developed and remains in use (M. Drawbridge, pers. comm.), but is generally not reported.

6. Most importantly, all estimates only consider reductions in N_e/N caused by broodstock management, and family specific mortality during rearing and after release may further reduce this ratio. All the estimates are therefore likely to be overly optimistic for an adult-to-adult N_e/N ratio comparable to N_e/N in the wild population. Differential survival studies could help to reduce this discrepancy.

3.2.1.5. Egg, larval and juvenile rearing.

A further reduction in effective population size may be caused throughout the egg, larval and juvenile rearing phase if any of the mortality occurring is specific to families or genotypes. Although no specific information about such family specific mortality is available, hatchery practices allow some predictions based on the literature.

In the first instance, the small proportions of spawns retained for rearing may reduce the effective number of breeders considerably. HSWRI produces about 300 spawns per year (Annual Reports 00-01 to 15-16; Average: 331.2, range: 224-397). Only about 10% of these spawning events are used for release (≈32 events, Table 3.2), so potentially, 90% of the potential diversity is discarded. Eggs are collected several times throughout the spawning season, because single spawns are often dominated by a single female (Gruenthal and Drawbridge 2012). Nevertheless, this small subset of spawns retained suggests that some of the broodstock do not contribute to spawns ultimately used for release. Ideally, the best strategy would be to retain some eggs from each spawn event during the season, as such a strategy would provide the highest number of effective breeders and come close to the N_b/N = 0.62 from 71 spawns in Gruenthal and Drawbridge (2012). However, this strategy could present logistical challenges of rearing eggs and larvae of very different ages, ranging from increased competition to potential cannibalism. It may be worth investigating the possibility of changing the hatchery system to smaller units (tanks) at each stage to accommodate larvae and juveniles from different families and at different ages. These tanks could be run on several different recirculation systems that adjust temperature to control growth rates, thus producing approximately equal sizes to be combined after weaning onto dry diets. Given several hundred spawns per year, spawns from consecutive days (possibly induced by temperature spiking, though HSWRI reports mixed results for this technique) could be so combined. Overall, retaining more spawns for rearing would potentially have more impact on the genetic diversity of released juveniles than adding more broodstock. Clearly, if more spawns are retained, some larvae and juveniles will have to be euthanized to avoid overloading the rest of the system although overall culling rates may not change much compared to the current culling rates aimed at equalizing family sizes. If one of the mass euthanization points is post weaning, the excess fish, which should be in the tens of millions, could be a sellable product for commercial hatcheries as they are past the high mortality stages while still small enough to economically ship via air freight.

In addition, egg quality may differ between females as well as between different spawn events of a female within a single spawning season (Gruenthal et al. 2014). Egg volume, oil volume, percent oil volume and number of oil globules declined significantly throughout the spawn season. Some of these characteristics, especially oil volume, were correlated with larval and juvenile growth,

stress resistance and survival (Gauger 2010). If eggs of different quality from different females are incubated together, it may well be that larvae hatching from the best eggs survive better than others, thereby causing family specific mortality and a reduction in genetic diversity. A study of differential survival among spawning events, including the larval, J2, and release-ready juveniles, was started but not finished because funding ran out (M. Drawbridge, pers. comm.). Family specific survival may also be caused by infectious and non-infectious disease and xenobiotics. HSWRI encountered several disease issues throughout the program, often resulting in high mortalities (White Seabass Enhancement Plan 2010). Disease resistance has a relatively high heritability in many fish species (e.g. salmon, Carlson and Seamons 2008) and there are successful programs selecting for disease resistance in aquacultured species (Odegard et al. 2011, Gjedrem 2012). These data suggest that mortality from disease exposure may vary across families, and thus may reduce genetic diversity in the hatchery. The same is true for mortality caused by exposure to xenobiotics, as tolerance to pollutants may have a strong genetic basis (e.g. estrogen mimics, Brazzola et al. 2014) and has been correlated to genetic variation at specific genes (e.g. dioxin in killifish, Proestou et al. 2014). The estimated average larval survival rate of 25-40% (M. Drawbridge pers. comm.), which may be suitable for fish production, may therefore cause considerable bias in family size and thus high variance in reproductive success of individual broodstock fish.

The potential for family specific mortality is less clear in the case of euthanization because of malformation. A sizeable proportion of larvae and juveniles (up to 60% in some 2007 and 2008 spawns) developed deformities primarily of the skull and jaw, and were euthanized (White Seabass Enhancement Plan 2010). Such malformations are fairly common in fish aquaculture, but their exact reason is not known. There are some estimates of the heritability of such malformations from other species, i.e. the proportion of phenotypic variation that is due to genetic factors. These heritabilities are commonly low, but sometimes moderate: for example, heritability of skeletal abnormalities was zero (h^2=0) in *Sparus aurata* (Castro et al. 2008), low for mouth and fin deformities (h^2=0.03-0.07) in Common Carp *Cyprinus carpio* (Kocour et al. 2006), highly variable among year classes for vertebral deformities (h^2=0-0.36) in Atlantic Salmon (Gjerde et al. 2005), but high for spine deformities in *Dicentrarchus labrax* (h^2=0.21) (Bardon et al. 2009). The heritability of deformities, and thus differences between families in its occurrence (and thus euthanization) is unknown, but is an issue that should be addressed soon.

3.2.1.6. Domestication selection.

Domestication selection is best known from terrestrial agricultural species, where selection has provided striking phenotypic changes (e.g. teosinte - maize). This domestication was mostly, but not entirely, intentional. However, domestication selection in enhancement programs is largely unintentional. In salmonid programs, behavior seems to be a trait that is most affected by hatchery rearing, with hatchery reared juveniles being more aggressive and less predator aware than wild fish (e.g. Swain and Riddel 1990). It is unclear, however, whether this difference is due to phenotypic plasticity or heritable genetic variation. In addition, rapid growth induced by high food availability in the hatchery may select for high metabolism and growth in the early life history and so affect later growth and maturation schedules and other correlated traits (Berejikian et al. 2016). There is relatively little information about adaptation to the hatchery

environment in marine finfishes such as White Seabass (see Section 4.4.1.6 for discussion of the potential role of domestication in contributing to high mortality of released hatchery fish). Therefore, we briefly review underlying mechanisms, the available evidence for domestication selection, and possible management approaches that may reduce the effects of selection.

Adaptation to domestic environments results from two primary causes: (1) the hatchery environment is inherently very different from the wild, and (2) the success of a hatchery in producing large number of offspring may permit the survival of juveniles that would otherwise die in the wild; that is, there is a relaxation of selection in the culture environment. The accumulation of deleterious mutations in hatchery populations with high survival because of the relatively beneficial environment has been termed 'supplementation load' (Lynch and O'Hely 2001). This supplementation load may accumulate relatively rapidly, even if broodstock are wild caught, and may potentially lead to deleterious fitness effects, and in the extreme, to the extinction of the population. In the wild, these mutations would be removed by selection in every generation. For White Seabass, it may be pertinent to investigate whether malformations or disease susceptibility can be attributed to supplementation load. In any case, Lynch and O'Hely (2001) conclude that gene flow from domesticated populations may render "long-term supplementation programs…incompatible with the permanent maintenance of self-sustaining wild populations…" (p. 377). Given long generation times in White Seabass, 'long-term' may mean many decades, but the general trend may still hold.

The effect of domestication selection on fitness losses in the wild population depends on several factors (Ford 2002, Baskett and Waples 2013). Adaptation to the hatchery environment depends on the strength of selection, the N_e of the population, the number of generations that the broodstock is held in captivity, and the magnitude of genetic variation underlying the fitness trait under selection. The divergence between wild and captive-reared individuals is largest when the latter are cultured throughout their lives for many generations. The impact on the wild population depends on the degree of differentiation between hatchery and wild fish, and the frequency of interbreeding between wild and differentiated hatchery stocks.

Examples of differences between hatchery and wild populations have been extensively reviewed (Leber et al. 2005, Fraser 2008, Fraser et al. 2008, Naish et al. 2008), but in most cases rely on comparisons between deliberately domesticated populations or involve hatchery species that have been maintained separately over many generations (Garlock et al. 2014, Segovia-Viadero et al. 2016). In hatchery environments, there are several pedigree-based studies in salmon that point towards reduced fitness of hatchery fish mating in the wild (Christie et al. 2014), whereas other studies have shown no significant difference between the two (Berejikian et al. 2009, Ford et al. 2012, Hess et al. 2012, Schroder et al. 2012). This variation in results might be attributed to a number of factors, such as short term environmental variation affecting relative fitness, the species cultured, or the number of generations reared in the program. Where fitness is reduced, the causes have been investigated in very few studies. One study attributed fitness differences to rearing environment rather than genetic divergence (Chittenden et al. 2010). On the other hand, rearing density (Thompson and Blouin 2015), changes in the growth and maturation schedule (Larsen et al. 2013, Spangenberg et al. 2014), and release habitat (Ford et al. 2015) have all been

implicated in genetic change in salmon populations. There are few pedigree-based or mechanistic studies in marine enhancement programs. However, in Red Drum enhancement programs in Florida, there is evidence that size at release influences survivorship (Tringali et al. 2008), that hatchery fish are subject to density dependent mortality at release (Camp et al. 2014) and that habitat usage by hatchery fish can differ from wild fish (Carson et al. 2014). It is possible, therefore, that the selective regimes for hatchery fish post release may differ from wild fish, emphasizing the importance of long term monitoring and tracking.

While there are few concerted studies on domestication selection in marine stock enhancement programs, its potential effects on fitness is widely recognized (Lorenzen et al. 2012). It is worth remembering that an increase in the effective population size in the hatchery provides greater opportunities for adaptation to the domestic environment. Therefore, we recommend investigating "best practices" based on theory developed in the management of salmon hatchery programs (Mobrand et al. 2005, Paquet et al. 2011). Here, the "integration" of hatchery and wild fish has been widely implemented; that is, wild-born fish are used as a significant contributor to hatchery broodstock. While all broodstock in White Seabass are currently wild born fish, hatchery-born fish may be caught once hatchery supplementation provides a sizeable proportion of the wild population. At that point, precautions to avoid using hatchery born fish as broodstock, such as checking for CWTs and/or performing genetic parentage analysis, will become crucial. Although this is currently not an issue because hatchery born fish are rare in the wild, new broodstock fish are scanned for coded wired tags, and one fish was already rejected for that reason (M. Drawbridge, pers. comm.). Once hatchery fish become common in the wild, genetic methods could be used to also exclude offspring of hatchery born fish. The aim of this "integrated" management approach is to prevent the hatchery fish from diverging from their wild counterparts through gene flow, and relies on processes in the natural environment to drive the ongoing evolution of the population as a whole. The genetic guidelines provided by the Hatchery Scientific Review Group (Mobrand et al. 2005, Paquet et al. 2011) are based on theoretical modeling that shows that if gene flow from the wild to the hatchery is greater than the reverse, then divergence between the two can be reduced. This approach is being empirically tested in a number of studies. One survey of changes in genome-wide diversity has shown that in the short term (four generations of culture), genetic divergence from the founding broodstock has been reduced in an "integrated" population compared to one that relies solely on hatchery origin broodstock (Waters et al. 2015). While this study shows that genetic change has been small overall, it does not directly measure fitness changes in the wild population. There are a number of related pedigree-based studies that are currently underway that aim to provide this information. We also caution that it is still not known whether "integrated" programs maintain genetic diversity over the long term.

Special consideration is warranted of the current practice of maintaining four different broodstock groups with different photothermal regimes that may not only affect survival but may also cause adaptation to a specific regimen. White Seabass, like most temperate zone teleost fishes, reproduce seasonally with release of gametes programmed so that progeny are produced when environmental conditions are favorable for survival. Thus, the entrainment of reproductive cycles to favor offspring survival is paramount for reproductive success in nature. A range of

environmental (photoperiod, lunar cycles, temperature, salinity, rainfall), nutritional, and social factors play important roles in controlling the timing of spawning, though for most temperate zone species photoperiod and temperature are the dominant factors (Migaud et al. 2010). Therefore, photothermal regimes are commonly used in commercial finfish aquaculture to synchronize or alter spawn timing to maximize year-round production of larval fish for growout (Bromage et al. 2001).

Stock enhancement programs, such as the OREHP, that aim to integrate hatchery fish into the natural population would ideally be designed to produce and release juveniles close to the natural timing (and size) for the species to optimize survival. However, there are practical reasons to adopt photothermal regimes for out of season larval production in stock enhancement programs, such as limitations in facilities for larval rearing and production of live feeds, and growout of juveniles. Maximal juvenile production is often achieved by staggering spawning time in groups of broodstock using photothermal regimes, such as that used by HSWRI for the White Seabass.

The question is whether the photothermal regimes that induce out of season spawning increase the risk of hatchery-induced genetic selection or epigenetic changes that result in reduced offspring fitness. Unfortunately, there are no studies that have directly tested this, and it would be challenging to do this type of study, especially with wild-caught broodstock such as that used for the White Seabass. However, differential selection due to photothermal regimes that produce offspring out of season, compress spawning periods, or reduce gamete quality cannot be completely ruled out. For example, reduced survival of juveniles has been observed with out of season releases of Red Drum (Sherwood et al. 2004), thus some selection could be occurring in offspring post release. While numerous studies have demonstrated successful alteration of spawn timing in marine species using photoperiod or photothermal regimes (Carrillo et al. 1989, Blythe et al. 1994, Bromage et al. 2001, Hansen et al. 2001, Norberg et al. 2004, Penney et al. 2006, Watanabe et al. 2006, Stieglitz et al. 2012), not all have followed effects of the photoperiod manipulation on fecundity and gamete/embryo quality, and none monitored family variation in reproductive success, particularly in programs that rely on volitional spawning of adults in tanks where such evaluations are difficult to do. When light cycles and temperature are strictly controlled and appropriately applied, minimal or no effect of photoperiod regimes that alter spawn time on gamete quality have been observed when compared to natural photoperiod regimes unless out of season spawning occurred in suboptimal water temperatures (Brooks et al. 1997, Bobe and Labbé 2010, Hansen et al. 2012). Advancing and/or compressing the interval between spawning periods using photoperiod induced a more protracted spawning period and reduced total volume and size of eggs, without affecting fertilization rates or incidence of developmental abnormalities in embryos compared to natural photoperiod control groups (Carrillo et al. 1989, Penney et al. 2006). The reduction in egg size was attributed to the shortened period of time for incorporation of yolk into developing oocytes, and effects of egg size on larval size and development are well documented (Knutsen and Tilseth 1985). Developmental abnormalities have been documented when temperature at spawning is suboptimal or the timing of egg collection/deposition is delayed (termed post ovulatory aging of the egg (Bobe and Labbé

2010)). This emphasizes the importance of strict temperature control systems on broodstock tanks.

In conclusion, we support using wild origin broodstock as much as possible, as outlined in the original hatchery plan. This goal converges with our earlier recommendation to rotate the White Seabass broodstock more frequently. Such a step requires excluding hatchery origin fish based on CWT (as currently done) or genetic tagging, so that they are not used in future breeding programs. Further, a monitoring program aimed at estimating effective size of the hatchery fish, recommended earlier, also has the advantage of providing information on family-specific survivorship in the hatchery and post release. In addition, we recommend exploring the logistic and financial feasibility of avenues to reduce domestication selection, such as moderating growth rates, reducing densities, maintaining natural photothermal regimes and releasing juveniles early.

Finally, while not aimed explicitly at examining the effects of domestication selection, we also recommend investigating genetic approaches that allow the tracking of hatchery origin individuals in the wild (Anderson and Garza 2006, Tringali et al. 2008, Abadía-Cardoso et al. 2013, Steele et al. 2013). In the medium term, it will be possible to examine the effects of different release strategies (age at release, numbers of individuals released, location of release) on survivorship and estimate long term contribution of hatchery fish to enhancement goals. The ongoing ezRAD study (J.L. Whitney, M. Iacchei, R.J. Toonen, K.A. Selkoe, H.S. Lenihan) should provide genetic markers suitable for this purpose.

3.2.2. Data and Information Gaps.

1. Lack of documentation of female equivalents per year, at least in the annual reports.
2. Lack of consistency and/or control by HSWRI scientists over collaborators' scientific efforts related to the hatchery.
3. Lack of information on effective number of breeders.
4. Lack of information on hatchery-induced family-specific survival, i.e. by selecting spawns, euthanizing deformed fish, disease, competition, cannibalism.
5. Lack of information on selective effects of photothermal regimes.

3.2.3. Recommendations.

1. In future collaborations with academic institutions, especially student projects, HSWRI should retain greater control over scientific procedures to ensure quality control and publication. It is unfortunate if dissertation findings need to be criticized in later management plans. For example, members of HSWRI could require inclusion on students' committees if a substantial part of the dissertation uses hatchery samples.
2. Female equivalents should routinely be recorded, and used as a routine low-tech approach to estimate $N_{b.}$ The approach should be confirmed with parentage testing.

3. Systems and operating procedures should be modified to take advantage of more spawning events to maximize the diversity of the final stocked fish.
4. Initiate a genetic monitoring study by developing genetic markers for large scale parentage based tagging (Anderson and Garza 2006). Using next generation sequencing, both discovery of markers and application on a large scale would be relatively affordable. Some marker may already be available for an ongoing ezRAD study (M. Iacchei et al., pers. comm.). This study could be used to:
    a. estimate the effective number of breeders.
    b. estimate the heritability of traits such as growth, disease susceptibility and incidence of deformity, which in turn may allow prediction of domestication selection.
    c. quantify family specific survival at each stage of the rearing process and in different treatments (e.g. temperature, density, hatchery incidences).
    d. estimate batch specific mortality potentially caused by seasonally varying conditions and acclimatization conditions.
    e. estimate the genetic diversity of juvenile fish released into the wild.
    f. identify hatchery born individuals and their offspring in the wild to avoid their inclusion in the broodstock.

3.3. Genetic effects of hatchery supplementation.

3.3.1. Key Findings.

3.3.1.1. Reduction of genetic diversity in the wild population.

One of the primary objectives of the OREHP was to "Maintain and assess a broodstock management plan that results in progeny being released that have genotypic diversity very similar to that of the wild population" (White Seabass Enhancement Plan 2010). It is interesting to note that very few attempts have been made to assess this objective empirically throughout the lifetime of the program. The only viable attempt was the study by Coykendall (2005), which was later heavily criticized (in part for valid reasons) in the White Seabass Enhancement Plan (2010) (Gruenthal and Drawbridge 2008).

Serious efforts should be made to establish a genetic monitoring plan that assigns parentage, quantifies family contributions and estimates genetic diversity throughout the rearing process, but most importantly upon release of juveniles. Some marker may already be available from an ongoing ezRAD study (M. Iacchei, pers. comm.). In any case, some predictions on the effects of supplementation can be made even in the absence of genetic screening data.

The reduction in effective population size (and thus genetic diversity) in a wild population due to hatchery supplementation is known as the Ryman-Laikre effect. Because of the high fecundity of marine fishes, including White Seabass, relatively few hatchery fish may produce a large proportion of juveniles in the wild, thus increasing relatedness and reducing genetic diversity in the wild populations. This effect has been originally described by Ryman and Laikre (1991), who also provided a simple analytical way to quantify this effect. The approach has been recently

extended by Waples et al. (2016) who also considered (1) the census size of captive and wild populations, (2) N_e/N ratios in captive and wild populations, and (3) the adult to adult replacement rates of wild and captive populations, which determine the proportion of hatchery derived fish in the wild.

Three scenarios were compared using this approach (Waples et al. 2016) (Fig. 3.5): one under the current conditions (i.e. 416 broodstock over the lifetime of the project (16 years), i.e. 208 fish/8 years, N_e/N=0.25), one under the scenario envisaged in the broodstock management plan (25% replacement every year, i.e. 920 fish total, N_e/N=0.57), and finally a scenario, where new broodstock is used every year, i.e. 1600 fish over eight years, with an N_e/N=0.78 (Table 1). The results suggest that hatchery supplementation would reduce effective size of the wild population quite considerably in almost all realistic scenarios, though the size of the hatchery broodstock has a considerable influence on expected effects. For example, if about 10% of the wild population are hatchery bred, and the wild population has an N_e/N ratio of about 0.01, the current management regime would cause a reduction of wild N_e to 21%, and the broodstock management plan to 62%, while the ideal (but possibly unrealistic) scenario requiring annual replacement of broodstock would cause no reduction in N_e.

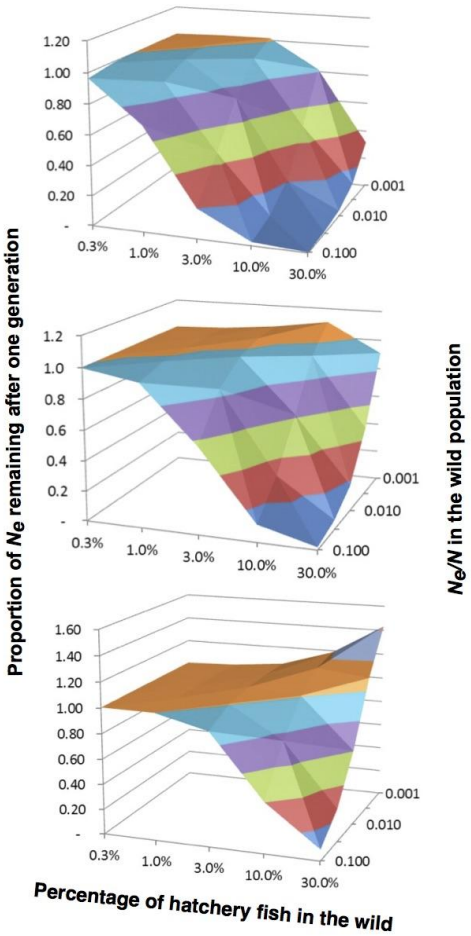

Fig. 3.5. Potential reduction of N_e in the wild population because of hatchery supplementation (Ryman-Laikre effect) in White Seabass. All three simulations assume a wild census population size of 2.5 million adults, and a hatchery broodstock size over 8 years (≈ 1 generation). Note the logarithmic scale of x and z axis.

Top panel: current broodstock management (N=208, N_e/N=0.25), i.e. half the fish that came through the system.

Middle panel: broodstock management plan, 25% fish exchanged per year (N=460 over 8 years, N_e/N = 0.57).

Bottom panel: ideal conditions. Broodstock exchanged every year (N=1600 over 8 years, N_e/N=0.78).

A higher hatchery contribution to the wild and higher N_e/N ratios in the wild would lead to quite considerable reduction in wild N_e. Note that these estimates pertain to a single generation and assume that hatchery fish in the wild have the same reproductive success as wild fish. Over several generations the effect could be more severe, while low reproductive success of hatchery fish in the wild could reduce the Ryman-Laikre effect. For example, in Red Drum, low genetic diversity (and thus low N_e) of hatchery released fish was found (Karlsson et al. 2008), indicating considerable potential for a Ryman-Laikre effect (Gold et al. 2008), while there was no evidence of a loss of diversity and reduction of N_e (Ryman-Laikre effect) in the wild population (Carson et al. 2009). A similar result was found in a later study of Red Drum, in which a small pool of broodstock (3-11 of each sex in each of eight brood years), low genetic diversity of hatchery fish and high sub-adult enhancement rates (as high as 50%) resulted in no observed decline of genetic diversity in the wild, though estimates of N_e demonstrated a potential Ryman Laikre effect (Katalinas et al. 2017). As White Seabass, Red Drum populations are buffered from adverse genetic effects by their longevity and the large number of breeding year classes in the population. Nevertheless, hatchery supplementation may lead to virtually irreversible loss of genetic diversity that is difficult to predict. It is therefore crucial to monitor the reproductive success of hatchery fish in the wild, most easily by assigning wild-born juveniles back to their grandparents in the hatchery.

3.3.1.2. Domestication selection.

The effect of domestication selection on the wild population is much more difficult to predict, and almost impossible to quantify. However, under the assumption that domestication selection causes some deviation from the 'optimal' wild genotype, two extremes can be identified (Waples et al. 2012), which are related to the 'integrated' and 'segregated' strategy identified above (Fig. 3.6). If hatchery influence is sufficiently weak and short to cause virtually no domestication selection (integrated), hatchery fish would interbreed freely with wild fish and there would be no effect. On the other extreme, the 'segregated' scenario, domestication selection is sufficiently strong to prevent reproduction of hatchery fish in the wild, again resulting in no genetic effect on the wild population. Between those extremes, interbreeding will be rarer if hatchery and wild populations are more dissimilar, but the genetic effect of such rare events will be more detrimental to the wild population. However, the exact effects of such interbreeding are not known because they depend on the similarity between captive and wild populations, the shape of the curve (Fig. 3.6) (Waples et al. 2012), timing of release, selection, density-dependent mortality and reproduction in the wild, and the nature of release (pulsed vs continuous) (Baskett et al. 2013, Baskett and Waples 2013).

There is little empirical information of the effects of domestication selection in White Seabass, or indeed any marine species. However, the potential for such effects exists. For example, the widely varying residence time of broodstock in the hatchery (Fig. 3.3) suggests that some fish fare better in the hatchery than others, either by surviving or by being selected for being prolific spawners. This suggests that such surviving fish already represent a non-typical selection of wild fish, even though they all originate from the wild population. Similar processes are likely to occur during larval and juvenile rearing – for example, hatchery practices may select for more disease resistant, faster growing fish that are less prone to malformation. Such

effects could be investigated in the hatchery by assigning parentage throughout the rearing process and ascertaining any non-random mortality within or between families. Again, a study was started to address this, but was ended because funding ran out (M. Drawbridge pers. comm.) Next generations sequencing technologies could also be used to identify specific genome regions that are under selection in the hatchery.

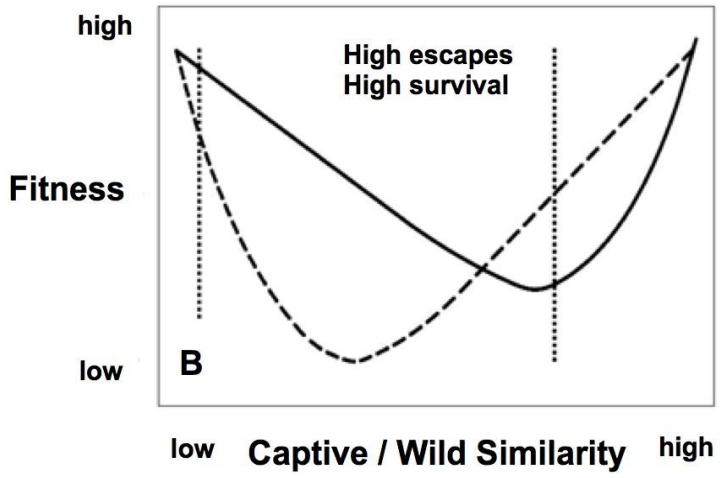

Fig. 3.6. Fitness effects of escapees/enhancement on wild populations depending on the degree of genetic similarity between captive and wild population. The two curves represent possible shapes of this function, the vertical lines indicate practical limits of similarity that can be achieved. From Waples et al. (2012).

One way to reduce domestication selection would be to release juveniles at a very early stage. This would not remove selection during the broodstock rearing and early rearing, but it may reduce overall effects. However, modeling and empirical data suggest that supplementation programs are only effective if juveniles are reared over the period of density dependent selection, which appears to be during the early juvenile phase for most species (Lorenzen 2005). There is therefore a clear trade-off between domestication selection and demographic effects of the supplementation, which should be more explicitly quantified by computer models based on empirical data for White Seabass specifically.

3.3.1.3. Enhancement and genetics: A summation.

Enhancement programs may have some benefits, but there are genetic risks under most realistic management scenarios. In well studied systems such as salmon hatcheries, it is widely accepted that enhancement programs are one of several key tools to maintain populations that are at risk of extinction. In self-sustaining populations, however, the genetic risks of enhancement likely outweigh the benefits, unless considerable effort is spent on broodstock management, rearing procedures and monitoring. White Seabass in California is not at risk of extinction, supports both commercial and recreational fisheries, and appears to respond well to favorable environmental factors and fishery management actions. Furthermore, longevity, high fecundity and high juvenile mortality of White Seabass aggravate many genetic risks known from salmonids. To achieve the aim of improving recreational and commercial catches with

relatively limited resources, efforts therefore may be better spent on improving fisheries management rather than on the possibly damaging attempts of enhancement.

3.3.2. Data and Information Gaps.

In order to assess the genetic effects on the wild White Seabass population of the hatchery management plan and subsequent practices, the following information is needed:

1. Effective size of broodstock:
   a. Individual reproductive success within years and overall.
   b. Estimates from parentage assignment of released juveniles.
2. Reproductive success of hatchery fish in the wild (by applying parentage based tagging).
3. Population structure of the wild population.
4. Census population size of wild population (this should be available from the stock assessment, but an unsolved question is the geographic extent, and thus the size, of the wild population).
5. Effective population size of the wild population (from genetic data, though also dependent on the population structure of the wild population).

3.3.3. Recommendations.

We recommend the following to fill gaps in information needed for the estimation of genetic effects of hatchery supplementation on the wild population

1. A genetic survey of the wild population to resolve the size and structure of the wild population. This survey also could discover markers for the parentage based study.
2. A parentage-based genetic monitoring system needs to be established as soon as possible. Samples could be screened very cost effectively and on a large scale using approaches described in Campbell et al. (2015). This would allow to estimate
   a. the effective population size in the hatchery from released offspring.
   b. the contribution of hatchery produced fish to the wild population.
   c. the reproductive success of hatchery fish in the wild (this would also provide insights into domestication selection).
3. With lower priority, studies to detect domestication selection (See also Section 4.4.3 Recommendation 1) by
   a. Comparison of phenotypic characters of hatchery fish in the wild and wild fish.
   b. A genome scan study to identify genes under selection in the hatchery.
4. Assess methods of improving recreational and commercial catches that can be enacted with relatively limited resources, such as modifications to fisheries management (e.g. catch restrictions), before investing more effort in the potentially damaging genetic effects of enhancement.

Chapter 4

Objective 4. Quantify contributions to the standing stock by tagging fish prior to release and assessing their survival in the field.

4.1. Procedures used to tag and release fish.

4.1.1. Key Findings.

Tagging is an essential methodology for the study and refinement of stock enhancement effectiveness. Several methods are available for tagging fish (Brennan et al. 2005, Pine et al. 2003, Leber and Blankenship 2011), including coded wire tags (CWT), visible implant elastomer (VIE), visible implant alpha (VIA), integrated transponder (PIT) tags, acoustic tags, and genetic fingerprinting. PIT tags are used by HSWRI to identify broodfish at the hatchery (Broodstock Transfer and Tagging SOP 2015, PIT Tagging Procedure for Newly Acquired Broodstock SOP 2016), while CWTs are used to tag fish before release. The CWT is the most suitable tagging method for this project because use of CWT facilitates high capacity tagging of large numbers of small fish, high information control, low impact to fish health, low effects on behavior or survival, reasonable cost considerations, and because CWTs enable coding and identifying an unlimited number of experimental-treatment variables, -controls, and -replicates. The latter (unlimited number of tag codes) is a critical requirement in selecting a tagging system for fisheries enhancement research and evaluation, given that a wide range of release-recapture experiments are required in order to optimize release strategies to maximize survival of released fish (Blankenship and Leber 1995, Lorenzen et al. 2010, Leber et al. 2016).

Coded wire tags are non-transmitting and must be extracted and viewed under a low-powered microscope to retrieve the code. The CWT is ideal for identifying White Seabass in release-recapture experiments to evaluate the effectiveness and efficiencies of stocking strategies (e.g., fish size at release, release habitat, release season, and stocking magnitude). The CWT is not visible, though, and therefore, fishers cannot visually distinguish tagged hatchery White Seabass from wild White Seabass. HSWRI staff use a MagniViewer device or a dissecting microscope to read the codes on the tags after extracting them from the fish (Reading Sequential Decimal Coded Wire Tags SOP 2016). Release, tagging, and recapture data are compiled in a database so that when a fish is recaptured, all of its history at the hatchery and growout facilities can be accessed (Comprehensive Hatchery Plan 2007).

When used appropriately, CWTs typically have little impact on tagged fish (Blankenship and Thompson 2003, Davis et al. 2004, Brennan et al. 2005, Vander Haegen et al. 2005). Because of their small size, they can be used in species or life stages that are too small for other tagging methods (i.e. there is no other tag with high information content that can be used to identify multiple batches of small fish in typical stocking events, which require multiple, often many dozens, of different tag codes. Since their invention, CWTs have become the most extensively used tags for fisheries management. The management of Pacific salmon by the United States and Canada relies on the use of CWTs, releasing about 50 million tagged salmonids annually (Nandor et al. 2010) to collect information about such things as stock distribution, survival, and

catch. In spite of having been invented nearly 45 years ago, no other method has been developed that can provide the code capacity, ease of use, cost effectiveness, and unambiguous data that are the signature characteristics of the CWT. Around the world, the use of CWTs continues to expand as researchers and managers take advantage of these features and the continual improvements to the CWT system initially targeted at the Pacific salmonid program.

4.1.1.1. Tests of alternative tagging methods.

Within the first few years of the OREHP, Hubbs-SeaWorld Research Institute (HSWRI) experimented with freeze branding and tetracycline marking as methods for tagging its fish (Annual Reports 87-89). HSWRI also experimented with Floy tags in the late 1990s and early 2000s (White Seabass Enhancement Plan 2010, CDFW Pathology Report 2003-035). Since 2000, HSWRI has been hosting (Stutzer 2004) and undertaking experiments (Annual Reports 02-03, 03-04, 04-05, 05-06, 11-12) on the viability of implanting acoustic tracking devices into White Seabass and California Halibut (see Section 4.2.1). In 2001, a study was started to acoustically track hatchery-raised juvenile White Seabass movements. In 2003, 10 juveniles with sonic tags were released into Mission Bay; 5 of those were from the hatchery and 5 spent several months at the growout facility in Mission Bay. A dozen hydrophones were placed around Mission Bay and adjacent coastal waters. Both sets of fish had individuals that emigrated from the Bay within a few days, and had individuals that suffered mortality in the Bay (Annual Report 03-04). In 2004, two more releases of acoustically tagged White Seabass were conducted, one in Mission Bay and the other in Agua Hedionda. Nearly half of the tagged juvenile White Seabass emigrated from each bay within one week of release, and all emigrated at night on an ebbing tide (Annual Report 05-06).

Studies were suspended pending investigation of whether the pinging sounds attracted marine mammal predators. Bowles et al. (2010) found that harbor seals could potentially detect VEMCO 69 kHz Ultrasonic Coded Transmitters (UCTs) at ranges between 19 and >200 m, while odontocetes could potentially detect them at ranges >1 km, and California sea lions were not expected to detect them at all except perhaps at very close ranges. Acoustic tracking trials have not continued, largely because of expense and lack of funding. This method could get expensive if a large tagging effort was undertaken; battery life is somewhat limiting, there is relatively high loss rate of acoustic tracking devices (Annual Reports 02-03, 03-04, 04-05, 05-06), and there is the potential for pinging to attract marine mammal predators (Bowles et al. 2010). However, this method can potentially greatly improve knowledge of White Seabass movement, dispersal patterns and life history. Visible Implant elastomer tags have also been used effectively by HSWRI for short-term experiments, as these tags can become obscured by pigments as fish age, but are very suitable for short-term studies.

4.1.1.2. Genetic parentage.

The Red Drum enhancement program in Florida has used a parentage based (familyprinting) Bayesian approach (Tringali 2006, Tringali et al. 2008) with some success. Tringali et al. (2008) employed genetic fingerprinting technology to definitively identify fish too small at stocking to effectively mark with CWTs (phase-1 Red Drum), along with 2 additional sizes of larger fish.

With the help of genetic testing, Tringali et al. (2008) identified 3,000 hatchery fish out of 20,000 that were surveyed (through fishery-dependent and fishery-independent methods) (63 were identified via CWT detection, the rest via genetic testing). For evaluating principal treatment effects, the focus was narrowed to strictly Red Drum that were ≥200 mm SL at capture; it was assumed that released fish reaching this size had survived long enough to overcome short-term release effects and had grown large enough to recruit into the recreational fishery (Tringali et al. 2008). Nearly 10,000 of these "recruitment-sized" fish were examined at time of publishing; among these, 282 hatchery fish were identified (42 via CWT detection; 239 via genetic testing). Further, using genetic testing, researchers were able to assess the effects on recapture rate of the following treatments: release site (river, and different habitats within river), size at release (phase-1, -2 and -3), and release season (the latter was examined here only with phase-1 fish) (Tringali et al. 2008).

4.1.1.3. Coded wire tags.

Since late 1990, HSWRI has been using coded wire tags to identify and track hatchery-reared White Seabass (Annual Reports 90). After initially using borrowed CWT equipment, HSWRI staff purchased their own CWT injector, quality control device, and field sampling CWT detector in June 1990. By 1998, HSWRI had 3 complete sets of this equipment. In 2009, HSWRI built a 5 person tagging station, which includes two holding tanks, one with ozonated water, and another dosed with MS-222 which serves to anesthetize fish before they are tagged (Annual Report 08-09, White Seabass Enhancement Plan 2010, Marine Finfish Anesthesia SOP 2015).

Tagging entails implanting a single CWT beneath the skin and parallel to the muscle fibers in the right cheek muscle of each juvenile White Seabass using Mark IV tagging machines made by Northwest Marine Technology (Proper Tag Placement and Technique: Coded Wire Tagging SOP 2016, Comprehensive Hatchery Plan 2007). Full-length tags are 1 mm long x 0.25 mm diameter (Comprehensive Hatchery Plan 2007). Fish must be at least 10 grams to be tagged ([J2] System Components and Mechanical Operation SOP 2016). If fish are abnormally small in comparison to their cohort and cannot retain a tag, they are euthanized (Proper Tag Placement and Technique: Coded Wire Tagging SOP 2016).

The CWT is a small, magnetized stainless steel wire, marked with rows of numbers denoting codes of batches of fish and individuals. Each tag is printed with numeric data used to identify fish by release group upon recapture (Reading Sequential Decimal Coded Wire Tags SOP 2016, Comprehensive Hatchery Plan 2007). The size of each batch of fish depends on the capacity of the growout facility that will hold that batch (White Seabass Enhancement Plan 2010). Tags are sequential, and the first and last tags used in a batch are recorded in order to identify individual batches (White Seabass Enhancement Plan 2010, Comprehensive Hatchery Plan 2007). According to the Comprehensive Hatchery Plan (2007), the CWT process is highly efficient, and "as many as 800 fish can be tagged per hour by an experienced operator" (p. 85).

Fish are tagged at an age of three to four months (about 80 dph) (HSWRI OREHP Overview Presentation, 20 May 2015). After the tags are inserted, the fish are either put through a quality control device or passed under a handheld wand detector to ensure that all of them have been

tagged (Proper Tag Placement and Technique: Coded Wire Tagging SOP 2016, Quality Control Device (QCD) Operation and Maintenance Protocol SOP 2016, Handheld Wand Detector SOP 2016, Comprehensive Hatchery Plan 2007). A subsample of fish is tested one to two weeks later for tag retention, and another subsample (of 100 fish) is tested right before release (Comprehensive Hatchery Plan 2007) (these 100 fish are also weighed and measured (Growout Procedures Manual 2007)). The proportion of the 100 subsampled fish that have retained their tags prior to release is applied to the total number of fish in that release batch and used to estimate how many White Seabass will be identifiable in the future (Comprehensive Hatchery Plan 2007). If less than 90% of fish have retained their tags, fish may have to be sorted and re-tagged (Growout Procedures Manual 2007), which occurred in 2008 at the King Harbor growout site (Annual Report 07-08); see Section 4.3.1 for more information on tag loss).

White Seabass are released either directly from the hatchery, or from growout pens. The White Seabass are transported to growout facilities in a number of ways, the most common of which is through three 1,500 L independently-aerated tanks made of marine-grade aluminum (Comprehensive Hatchery Plan 2007). Two of these tanks fit on a trailer, and one fits in a truck bed (Comprehensive Hatchery Plan 2007). The bottoms of all three tanks are sloped towards gate valves to allow everything to empty out of the tanks easily (Comprehensive Hatchery Plan 2007). Before being transported, the fish are deprived of food for 24 hours (Comprehensive Hatchery Plan 2007). Each tank is filled with fish at a maximum density of 40 kg/m^3 (Comprehensive Hatchery Plan 2007). The water in the tanks is "static," and compressed oxygen is employed to oxygenate it (Comprehensive Hatchery Plan 2007). The water is treated with Fritzguard "to protect the ectodermal mucous layer and to maintain an appropriate electrolyte balance" (Comprehensive Hatchery Plan 2007, p. 89-90). If the fish are transported directly from the hatchery to the release site, the temperatures of the tank water and the body of water into which the fish will be released are taken (Comprehensive Hatchery Plan 2007). If there is a discrepancy of more than 2 degrees Celsius, water from the release site is pumped into the transport tanks (Comprehensive Hatchery Plan 2007). Fish are transferred from their transport tanks to the growout pens through a 10.1 cm diameter flexible hose (Growout Procedures Manual 2007). The fish held at growout facilities are already acclimated to the temperature of their release sites, as they are typically held for 2-6 months within these waters before release (CDFW Release Criteria 2015).

The wide range of holding times in growout (2-6 months) may be due to differences in the sizes of fish when delivered (if the fish are large, they are held for shorter amounts of time before release, M. Drawbridge pers. comm.), differences in water temperature (water temperature drives growth rates), or seasonality (waiting until a specific season can increase survival, Hervas et al. 2010). For example, fish may be overwintered at growout sites (Annual Report 10-11), with release times dependent upon changes in water quality and other environmental conditions (e.g., early releases due to sudden water quality decrease as per the Net Pen Water Quality Contingency Plan SOP (2016) or Health Assessment for Fish Release SOP (2016)). Also, release time can be delayed because of a need to treat fish for disease before release (e.g., peroxide treatment of fish for *Hexamita* at Marina del Rey growout facility (Annual Report 08-

09)), a need to retag fish (e.g., at King Harbor in 2008 (Annual Report 07-08)), or personnel logistics (e.g., developing a release plan, organizing volunteers).

Since April 2011, HSWRI has measured and counted all fish within 3-4 weeks before release, allowing the fish to recover from the stress of handling before being released (Annual Report 10-11). HSWRI then removes the end grates or lowers the nets of each growout pen, enabling fish to swim out of pens on their own, without being handled (Annual Report 10-11). Fish are ideally released at night or in the afternoon (Annual Report 10-11) to reduce predation (M. Drawbridge pers. comm.), although it is unclear how strictly this is followed. Also, it is unclear how release time-of-day affects survival and how this was tested.

CDFW has described release protocols in the document, *Release Criteria for Cultured White Seabass (Atractoscion nobilis)* (CDFW Release Criteria 2015). Prior to being released, a small batch of White Seabass is assessed for malformations, once before the fish are transferred to growout facilities, and once before they are released (see Section 1.6 for a description of the quality assessments conducted by hatchery personnel). Additionally, White Seabass are inspected for disease twice by a CDFW pathologist, once at the hatchery (before being transported), and once at the growout facility immediately prior to release, with the goal of preventing the transmission of pathogens to wild stock. The fish may be transported or released within two weeks of being cleared by the pathologist. The pathologist may do more tests if other health concerns come up, if there is a sudden increase in mortality, or if HSWRI requests further tests. Pathogens that are of particular concern, and which trigger specific CDFW release criteria include the White Seabass herpesvirus, Viral Nervous Necrosis Virus (VNNV), *Piscirickettsia salmonis*, *Uronema marinum I Miamiensis avidus* (ocular and encephalitic variant), a renal “sporozoan” pathogen, and Viral Hemorrhagic Septicemia Virus (VHSV). See sections 1.8.1 and 1.10.1 for more information on diseases in White Seabass.

The growout facility operators and the Growout Facility Coordinator (GFC) work together to create a “release plan” a few weeks before the White Seabass are actually released (Growout Procedures Manual 2007). This ensures that there is enough time for fish to be weighed, measured, checked for tags, and cleared of pathogens, and that a sufficient number of volunteers have been contacted to help during the release process (Growout Procedures Manual 2007). The GFC instructs all volunteers on how to properly handle fish on the day of release (Growout Procedures Manual 2007). Each batch of fish should be released within one to two days, at the same location (Growout Procedures Manual 2007).

Hatchery-reared fish are released all year round, but since 2010 have mostly been released in the spring through fall (Comprehensive Hatchery Plan 2007, Annual Reports 07-15, HSWRI Releases Dataset 2016), when survival rates are highest, according to a recommendation set forth by Hervas et al. (2010). For the most part, HSWRI follows this recommendation, but occasionally still releases fish during the winter (HSWRI Releases Dataset 2016). All White Seabass should be greater than 20 cm in length at time of release to improve likelihood of survival (Comprehensive Hatchery Plan 2007), and in actuality ranged from 9.2 to 30.6 cm total

length (TL), and averaged (±1SE) 20.2 ± 0.3 cm TL, between January 2007 and June 2015 (Annual Reports 07-15).

4.1.2. Data and Information Gaps.

Regarding the procedures used to release fish, there is not yet enough information available to enable selection of optimal release habitat, optimal growout holding times, size at release, optimal timing of releases, and optimal release magnitude. One of the requirements of establishing a meaningful test of stock enhancement effectiveness is to first gain an understanding of whether the fish release strategies used to conduct that test will enable evaluation of the full potential of hatchery fish to augmenting the target stock (i.e., assessing the similarity in post-release mortality of hatchery and wild fish; and whether survival of hatchery fish after release is at or near its full potential, or whether some combination of release strategies is causing higher mortality of hatchery fish than could be realized when optimal release strategies are identified and used to conduct the field test of enhancement effectiveness).

4.1.3. Recommendations.

1. Take a more systematic approach to testing release strategies, including continuing trials to identify optimal release conditons, and developing and running models to assess the effectiveness of release strategies, and reassess occasionally to inform adaptive management.
2. Continue use of CWT system as the primary identifier of stocked hatchery fish. This is needed to continue adaptive management of stocking.
3. Continue and expand the use of multiple tagging systems as needed for routine operations and for evaluating critical uncertainties about the effectiveness of stocking, e.g., sonic tags are a great addition to the tools used for understanding the relative contributions of the principal mediators of abundance over time at release sites – mortality and dispersal.
4. Explore feasibility of incorporating genetic parentage assignment (i.e., using genetic fingerprinting; Tringali 2006, Tringali et al. 2008) to “tag” fish, which is coupled with an angler assisted fin-clip program to collect tissue samples from White Seabass taken in the fishery. This effort could identify the location, number and proportion of hatchery-derived fin clips among the total fin clips sampled by the commercial and recreational fisheries. Since the recovery of hatchery fish in the fisheries is currently extremely low (0.26%) (see Section 4.4.1 for further discussion), this effort would be more useful if juvenile White Seabass were clipped for genetic analysis and released in large numbers, but would enable assessment of hatchery contribution to the catch-and-release component in the fishery (e.g., undersized White Seabass caught in the fishery and released). This method could involve more fishermen, including commercial and recreational fishermen, Mexican fishermen via CICESE and Pfleger Institute of

Environment Research (PIER), and other stakeholders (e.g., volunteers) in sampling the catch and constructing fin-clip kits (volunteers). Florida used this method effectively to track hatchery Red Drum in the fishery and also to resolve some critical experimental questions about release strategies (size at release, release season, release habitat) in Southwest Florida (Tringali 2006, Tringali et al. 2008). This effort should be weighed against the costs and benefits of the head return program. Genetic tagging would also provide estimates of individual reproductive success of broodstock fish (Anderson and Garza 2006). Although a very useful addition for boosting angler involvement and increasing data recovery to identify hatchery fish contribution to the fishery, genetic tags are no substitute for the benign, high-information CWT that HSWRI already uses, which is needed for adaptive management.

4.2. Procedures used to recover tags.

4.2.1. Key Findings.

4.2.1.1. Lack of a specific tag recovery plan.

Existing management plans cover all stages of the program from broodstock management through the release of White Seabass from hatchery or growout facilities; however, there is no dedicated plan for monitoring the released fish. A tag recovery monitoring protocol is outlined in the White Seabass Enhancement Plan (2010) under "Current Research and Future Needs," and is reviewed in each of the Annual Gill Net Reports and summarized in associated publications (e.g., Allen et al. 2007).

4.2.1.2. Insufficient funding for tag recovery field surveys.

After the hatchery was built, it was realized that funding was insufficient to run a large hatchery, and also to perform field sampling to recover tagged fish. When mitigation funds from San Onofre Nuclear Generating Station (SONGS) and British Petroleum Oil Spill were given to HSWRI to supplement funding, the money was used to supplement hatchery operations and for the release of fish. HSWRI received an annual average (±1SE) of $1,509,521 ± $73,487 over the last five fiscal years (fiscal years 2012-13 to 2016-17) to run the hatchery and to capture tagged adult White Seabass (OREHP Budgets Summary 2002-2015, OREHP Budget 16-17). In addition, San Diego State University (SDSU) has received an annual average (±1SE) of $90,141.80 ± $7,493.05 over the last five fiscal years (fiscal years 2012-13 to 2016-17) to conduct juvenile gill net surveys in the Southern California Bight with HSWRI (OREHP Budgets Summary 2002-2015, OREHP Budget 16-17). SDSU receives 20% of the money designated for gill net surveys, and contracts out to HSWRI, which receives the remainder of the funds to complete the surveys (K. Johnson email to T. S. Talley, 23 January 2017). Given the difficulty in capturing tagged White Seabass and the low contribution rate of hatchery fish to wild populations, a substantial boost in the monitoring component would be needed to sufficiently sample the wild population for tagged hatchery fish; with costs likely exceeding the available budget.

4.2.1.3. Approaches to tag recovery.

A nearshore gill net survey program to capture wild and hatchery White Seabass has been conducted by HSWRI and SDSU for 25 years. While these efforts do not sample the entire standing stock, they do provide estimates of contribution rates of hatchery White Seabass to local portions of the wild stock (generally portions in proximity to release locations. In the late 1990s, this program was coupled with an angler-assisted adult White Seabass head return program to increase the range and sample size of White Seabass collected (Annual Report 98-99). HSWRI's choice of the coded wire tag to identify hatchery White Seabass has been a critical element of the success of efforts to identify hatchery contribution rates.

4.2.1.4. Juvenile Gill Net Surveys.

Recapture of hatchery-reared and released White Seabass by OREHP-contracted researchers and staff is accomplished using experimental gill nets. Experimental gill nets have been the most effective gear because (1) they recover a wide size range of White Seabass (20-60 cm standard length), (2) they can be deployed in a diversity of habitats (e.g., kelp beds, embayments, rocky reefs), and (3) they have relatively high catch rates relative to other gear. The downsides of gill nets are that they do not catch White Seabass that are 20 cm in length or shorter as effectively as larger size classes (Fig. 4.1), and bycatch rates are high (M. Drawbridge email with attachment to T. S. Talley, 29 August 2017).

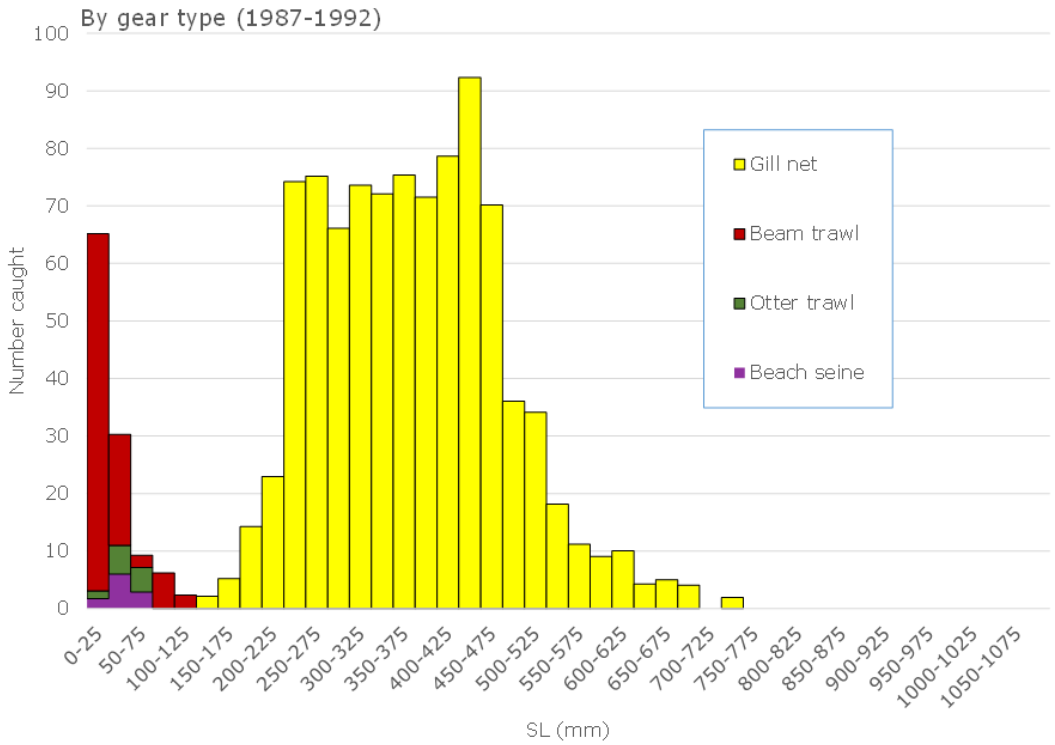

Fig. 4.1. Number of White Seabass of different size classes caught using various gear types between 1987-1992 by HSWRI. Number of fish caught is not adjusted for fishing effort but is meant to reveal general trends the effectiveness of gear types for capturing fish of different sizes. Hatchery released fish are generally about 20 cm (8 inches). Graph provided by HSWRI. (M. Drawbridge email with attachment to T. S. Talley, 29 August 2017).

From 1988 to 2008 and again from 2012 to 2015, OREHP-contracted researchers conducted standardized gill net sampling surveys in shallow waters from Santa Barbara south to San Diego's border with Mexico. Initially, from 1988 to 1994, the surveys focused on the recovery of tagged fish, and gathering information about the wild population. From 1995 through the spring of 1996, surveys focused on determining the distribution of 1-4 year old White Seabass. Starting in the fall of 1996 efforts focused on recruitment of 1-year old fish and recovery of tagged fish (White Seabass Enhancement Plan 2010). Between 1988-1994, SDSU and HSWRI were contracted by the OREHP to perform field surveys for wild and hatchery reared White Seabass. During this time, many of the procedures for the gill net sampling program were established, including gear type, and spatial and temporal definitions, which aimed to maximize the catch of White Seabass (White Seabass Enhancement Plan 2010, Gill Net Reports 91-93, Allen et al. 2007).

Table 4.1. Juvenile gill net sampling sites, FY 1995-96 to 2007-08, and 2012-13 to 2014-15. Gill net sampling did not occur between 2008 and 2012 due to budgetary restrictions. Table adapted from Table 11-1, White Seabass Enhancement Plan 2010.

Coastal Sites	CSUN/VRG	SDSU/HSWRI
Santa Barbara	X	
Ventura	X	
Malibu	X	
Catalina Island – West	X	
Catalina Island – East[1]	X	
Palos Verdes	X	X[a]
Seal Beach	X	
Newport Beach	X	
Oceanside		X
Carlsbad		X
La Jolla		X
Point Loma		X
Silver Strand/Imperial Beach		X
Embayment Sites		
Marina del Rey	X	
Catalina Harbor	X	
Newport Bay		X
Agua Hedionda Lagoon		X
Mission Bay		X
San Diego Bay		X

[1]Catalina Island – East station was dropped in FY 2004-05 due to budget constraints.

[a]SDSU/HSWRI sampled at Palos Verdes in FY 2012-13, 2013-14, and 2014-15. It appears that the CSUN sampling team stopped participating in gill net surveys in 2008.

Note: in 2001, four additional sites were sampled: Santa Cruz Island, Santa Barbara Island, San Nicolas Island, San Clemente Island (Gill Net Report 00-01).

From 1995 through the spring of 1996, sampling was conducted by researchers from California State University, Northridge (CSUN) and the Vantuna Research Group (VRG) of Occidental College. Beginning in the fall of 1996, sampling duties were split between two teams: (1) researchers from SDSU and HSWRI sampled the southern portion of the Southern California Bight; and (2) researchers from CSUN and VRG sampled the northern portion of the Bight (see Table 4.1). Beginning in FY 2005-06, only CSUN researchers conducted sampling in the northern area. In FY 2006-07, sampling in the southern area was conducted by HSWRI researchers only. There was no sampling conducted from July 2008 to September 2012 due to budgetary constraints. FY 2007-08 appears to be the last year the CSUN sampling team participated in gill net surveys, after which SDSU and HSWRI continued to sample the Southern-most sites, along with Palos Verdes (see Table 4.1; Gill Net Reports 12-13, 13-14, 14-15).

The sampling program has utilized various types of gill nets over the years, but primarily two types of gill nets have been used. Type 1 includes monofilament gill nets that have been used in OREHP surveys since 1992; they are 45.7 m in length and 2.4 m in depth, and consist of six 7.6 m panels of three different mesh sizes: two each of 25.4, 38.2, and 50.8 mm square mesh. Type 2 gill nets, first used in FY 1996-97, have the same dimensions as the Type 1 nets but have three panels each of 25.4 and 38.2 mm square mesh, sizes that were most effective at catching juvenile White Seabass in past years.

Coastal site sampling. Coastal sites have been sampled since 1988 with the exception of the years in which sampling was not funded, as mentioned above. Beginning in 1995, six replicate Type 1 gill nets, and two replicate Type 2 gill nets were set within each coastal site. Substrates at these sites included sand/rock, and reef/kelp habitat. All nets were set perpendicular to the shore (or the kelp line) in water depth of 5 to 14 m below Mean Lower Low Water (MLLW) where prior sampling established that juvenile White Seabass were most abundant.

Embayment site sampling. Embayments have been sampled since 1988 with the exception of the years in which sampling was not funded, as mentioned above. Recent surveys have deployed six Type 1 nets in a minimum water depth of 2.5 m (below MLLW), and distributed within the outer, middle and inner areas, resulting in sampling of the different habitats. Pairwise comparisons of embayment and coastal sites were made using only Type 1 net catches.

Sampling dates. Sampling has generally been conducted in April, June, August, and October. In recent years, lack of funding forced HSWRI to reduce sampling to two months each fiscal year. Table 4.2 shows the sample coverage over time (White Seabass Enhancement Plan 2010).

Data collections. At each net set, date and time, geographic coordinates, and surface and bottom temperatures are recorded upon retrieval. For all fish caught, species identity and total length were recorded along with net number, mesh size and replicate panel number. Captured white Seabass were also assigned a unique ID number, measured for standard length, weighed, and necropsied to determine sex. The stomach content of each Seabass was identified and the

otoliths were removed. Saggital otoliths were used for a while during a study to determine the age and growth of White Seabass but not all fish were aged and this effort was discontinued. Each White Seabass caught has been scanned for CWTs to determine if it was hatchery raised. If a CWT was found, the fish was frozen and given intact to HSWRI for processing. White Seabass marked with Floy tags (1996-1998) were turned over to CDFW following CWT extraction and a post-mortem exam was conducted by HSWRI (White Seabass Enhancement Plan 2010).

Table 4.2. Juvenile gill net sampling schedule FY 1995-96 to 2007-08, and 2012-13 to 2014-15. No gill net sampling occurred 2008 – 2012 due to budgetary restrictions. Table adapted from Table 11-2, White Seabass Enhancement Plan 2010.

	North (CSUN/VRG)					**South (SDSU/HSWRI)**			
Year	Aug	Oct	Apr	Jun		Aug	Oct	Apr	Jun
1995/96	x[1]	x	x	x		x	x	x	x
1996/97	x	x	x	x		x	x	x	x
1997/98	x	x	x	x		x	x	x	x
1998/99	x	x	x	x		x	x	x	x
1999/00	x	x	x	x		x	x	x	x
2000/01	x	x	x	x		x	x	x	x
2001/02	x	x	x	x		x	x	x	x
2002/03	x	x	x	x		x	x	x	x
2003/04	x	x	x	x		x	x	x	x
2004/05[2]	x	x	x			x	x	x	x
2005/06[3]		x		x		x	x		
2006/07[3]		x		x		p[4]	x		x
2007/08	x	x	x	x		x	x	x	x
2012/13							x[5]		x[5]
2013/14						x[5]	x[5]		x[5]
2014/15							x[5]		x[5]

1. "x" indicates all stations were sampled.
2. To stay within their budget, VRG contractors had to drop one month (June) of sampling.
3. Sampling was reduced to 2 months due to budget constraints.
4. "p" indicates that only partial sampling (La Jolla and Mission Bay) was conducted.
5. SDSU/HSWRI also sampled one Northern site, Palos Verdes.

4.2.1.5. Commercially caught White Seabass surveys.

An adult head-collection program was started in 1998 (Annual Report 98-99). The targets of the program were commercial fish markets that buy White Seabass and that would allow the scanning of heads. The head collection program entails measuring head length (to create a

head-total length conversion), scanning for CWTs, removing otoliths for aging, and recording information on where and when a fish was caught. Since the start of the program, HSWRI researchers have opportunistically scanned commercially caught White Seabass at commercial markets. In June 2008, the California Department of Fish and Wildlife (CDFW) began its commercial White Seabass sampling program to assist in gathering data for both the OREHP and for the White Seabass Fishery Management Plan (WSFMP). Targeted markets were selected based on information from commercial landing receipt data from previous years. Depending upon staff availability, as many of the major ports as possible were covered by samplers when the season opened in June each year. Staff members were redirected to specific ports as necessary in accordance with changing market conditions. Staff members were in frequent contact with commercial fishermen as well as major fish dealers at the major Ports in Southern California.

A Microsoft Access database was created for data entry of CDFW's commercial White Seabass samples. Information collected during sampling included biological information (e.g., sex, length) as well as catch information (e.g., CDFW block, port of landing, total pounds landed). Prior to 2008, White Seabass were sampled opportunistically (and infrequently) by staff in San Pedro that were sampling other species. Data from these samples are available in a separate database.

Because fish were most often scanned by CDFW staff directly from commercial gill net boats as they were offloading fish, the source and location of catch were verifiable. If fish were being scanned at a market rather than directly from a vessel, the dealer would have transportation receipts or landing receipts indicating location of catch. If fish were identified as being transported from Mexico, they were scanned for the presence of a coded wire tag and noted as such in CDFW records.

4.2.1.6. Recreationally caught White Seabass surveys.

Fishing tournaments. Efforts to collect data from recreational fishing tournaments began in 1998, at a tournament where 61 adult White Seabass were scanned for CWTs, and another 339 heads were turned in by recreational fishermen. One (the first) CWT-tagged adult was recovered in June 1999 (Annual Report 98-99, White Seabass Enhancement Plan 2010). Collaborating groups (Marina del Rey Anglers and San Diego Oceans Foundation) have sponsored incentive-driven tournaments aimed at collecting White Seabass heads from recreational fishermen.

"Save your White Seabass head" program. The program, started in 2004, targets Commercial Passenger Fishing Vessels (CPFVs) and asks passengers to save and freeze White Seabass heads, and to drop them off at any of 31 freezer locations in Southern California. By 2008, roughly 40% of all CPFV-caught White Seabass were estimated to have been turned in through this program (White Seabass Enhancement Plan 2010). In 2008, five hatchery-raised White Seabass were detected from the 1,835 heads saved by the CPFV fleet (White Seabass Enhancement Plan 2010).

California Recreational Fisheries Survey (CRFS). This program, which replaced the Marine Recreational Fisheries Statistical Survey (MRFSS) in 2004, began scanning White Seabass in June 2008. The CRFS places samplers at public access points (e.g., piers, jetties, beaches, boat launches, and on CPFVs) who scan and measure White Seabass. This program also includes telephone surveys of licensed anglers and CPFV operators (White Seabass Enhancement Plan 2010), although it is unclear how often these occur and how participants are chosen.

4.2.1.7. Tag surveys in Mexico.

Oscar Sousa and colleagues at Centro de Investigación Científica y de Educación Superior de Ensenada (CICESE) have been collecting data from fish camps down the coast of Baja California to get information on catch of White Seabass; results are pending. This recent effort will lead to improved White Seabass data; however, catch records have grouped White Seabass with corvina, making it difficult to know real numbers of White Seabass caught throughout Baja California through time. While HSWRI and CDFW have never conducted tag surveys in Mexico, they have scanned White Seabass that were landed in Mexican waters (HSWRI OREHP Overview Presentation, 20 May 2015; e.g., 3,034 White Seabass landed in Baja California were scanned in FY 2007-2008; Annual Report 07-08). Out of the more than 21,600 fish landed in Mexican waters that were scanned between 1998 and 2011, none carried OREHP tags (M. Drawbridge pers. comm.).

4.2.2. Data and Information Gaps.

1. There is no comprehensive and stable plan for retrieving tagged White Seabass.
2. There is little information on how the changing recapture protocols, especially for juveniles (e.g., efforts vary year to year with season, site), influence recapture rates.

4.2.3. Recommendations.

1. Develop a more comprehensive tag recovery plan that includes a strong quantitative, regular-interval, fishery-independent monitoring plan targeting juveniles and subadults.
2. Develop a plan for expanding fishery-dependent monitoring of hatchery fish contribution rates to the fishery including incorporation of an angler assisted fin-clip program to recover tissue samples from White Seabass taken in the fishery, coupled with the use of genetic fingerprinting– See Section 4.1.3 Recommendation 4.
3. If recapture rates of tagged fish increase, establish adaptive-management stocking experiments as part of each year's stocking plan to enable steady improvement and refinements to stocking strategies and to field test critical model assumptions. Every hatchery-fish release event is a missed opportunity if a question about stocking effectiveness is not posed and tested.
4. Involve more stakeholders to help financially or logistically support tag retrieval efforts (e.g., U.S. commercial fishermen, Mexican fishermen).

4.3. Estimation of tag loss.

4.3.1. Key Findings.

HSWRI has done work to evaluate tag retention, as tag loss can mask actual recovery rates and release information. As mentioned in section 4.1.1.3, a quality control device manufactured by Northwest Marine Technologies is used to test for tag retention after the White Seabass have been tagged at the hatchery (Comprehensive Hatchery Plan 2007). A subsample of fish is also scanned for tag retention one to two weeks after tags are initially inserted, and another subsample (of 100 fish) is scanned right before release to estimate an average rate of tag retention for the entire release batch (Comprehensive Hatchery Plan 2007, Growout Procedures Manual 2007).

Protocols exist to postpone releases if tag retention is less than 90%. For example, in 2008 fish from the King Harbor net pen were sampled on release day, and found to have retained only 78% of their tags (Annual Report 07-08). Release was aborted, untagged fish were tagged again, and the fish were eventually released 5 months later (Annual Report 07-08).

CWT retention studies began in 1990 with the assistance of Dr. Ray Buckley, then a biologist with the Washington Department of Fish and Wildlife. The initial CWT retention tests at HSWRI conducted with Buckley's assistance showed that cheek tissue was the most appropriate target tissue in White Seabass for injecting CWTs (Annual Report 90 – Interim Report 1). The experiments with Buckley revealed CWT tag retention ranged from 77.4% in small White Seabass (39.1 mm standard length) to 94.6% in medium size White Seabass (56.3 mm SL) to 99.9% retention in large White Seabass (92.5 mm SL) (Annual Report 90 – Interim Report 1). CWT tag loss is most likely to occur within the first one to two weeks after initial tag insertion, and is usually caused by improper needle penetration (and expulsion of the tag as the fish heals) (Comprehensive Hatchery Plan 2007, Annual Report 90 – Interim Report 1). HSWRI experiments found that long-term tag retention is high, with more than 90% of White Seabass keeping their tags for more than 300 days (Annual Report 90 – Interim Report 1, Drawbridge et al. 1995, Comprehensive Hatchery Plan 2007; See Table 1 and 2 (p. 4) in the first interim report from 1990 for a summary of the results of tag retention experiments). By 1998, when tag retention was reviewed, HSWRI staff (a team of three) could tag 7,000-8,000 White Seabass per day with retention rates >90%.

4.3.2. Data and Information Gaps.

There is no recent information on long-term tag loss rates (most recent tag retention rates are from Annual Report 90 – Interim Report #1).

4.3.3. Recommendations.

1. Continue to monitor tag retention on the day of release.
2. It has been a long time (ca. 27 yrs) since Ray Buckley assisted in performing a systematic tag loss evaluation verifying the low tag loss rates. HSWRI should consider assessing whether the current routine tag loss checks are sufficient enough for verifying that there is still, as in 1990, low tag loss under current operators, holding and tagging conditions; this is needed to update the shortest period needed for identifying tag loss in fish that have been tagged prior to release, and to make sure that there isn't some substantial unrealized tag loss. Consider a formal six-month long study, either in tanks or in net pens, to quantify current tag loss over time, with periodic checks (day 1, day 3, day 7, then weekly checks for 6 weeks post tagging to determine when tag loss stabilizes, followed by monthly checks of tag loss until 6 months post-tagging). Involve all of the people who routinely tag the fish by having them tag in the same way as they routinely do (you would not want them to tag more carefully or more slowly for this experiment than they routinely do).

4.4. Estimation of post-release survival of tagged hatchery fish.

4.4.1. Key Findings.

Estimation of the post-release survival of hatchery-reared fish provides crucial information for two purposes: (1) optimization of release strategies, and (2) assessment of the contribution of releases to fisheries management goals. Optimization of release strategies through empirical pilot release-recapture experiments enables enhancement programs to elevate short-term survival of released fish by controlling stocking variables such as fish size at release, release habitat, acclimation and acclimatization, timing of releases, and release magnitude in informed ways that minimize post-release mortality (Blankenship and Leber 1995, Lorenzen et al. 2010, Leber et al. 2016). Although some of these factors have been investigated by HSWRI (e.g., acclimatization in net pens and release season impacts), little empirical data exists for optimizing the other release variables for the OREHP. Further, the potential inefficiency of gill nets for catching juvenile hatchery fish of the sizes that are generally smaller than those released (<20 cm) makes assessment of release strategies challenging (Fig. 4.1; although increasingly low recapture rates of larger, effectively-captured size classes, ≥20 cm, suggest that high post-release mortality and dispersal are the main reasons for low recapture rates). Thus, assessment of the contribution of the OREHP's White Seabass releases to fisheries management goals is disadvantaged by release strategies that may not afford short-term post-release survival results at the levels that the program could otherwise achieve through systematic release strategy optimization. This somewhat compromises assessment of the OREHP's potential.

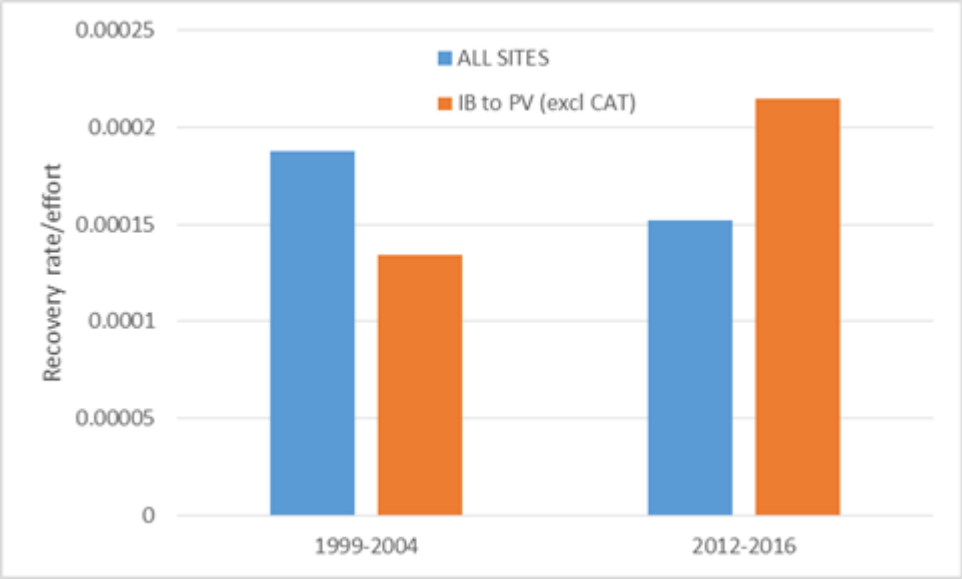

Fig. 4.2. Recovery rate standardized by fishing effort of hatchery raised juvenile White Seabass using gill nets. In the period 1999-2004, gill nets were set throughout the range of White Seabass releases (All Sites) to measure recovery rate of tagged fish. In the period 2012-2016, gill nets for recapture were only set south of Palos Verdes in Los Angeles to Imperial Beach at the U.S.-Mexico Border (excluding Catalina Island; IB to PV (excl CAT)). Therefore, changes in recapture rate between the time periods should be assessed by comparing recapture rate of fish released and caught again from either set of sites during 1999-2004, with the recapture rate of fish released and caught between IB to PV in 2012-2016 (in order to account for the low likelihood of recapturing fish released north of Palos Verdes in the southern area). Graph provided by HSWRI. (M. Drawbridge email with attachment to T. S. Talley, 29 August 2017).

A quantitative assessment of post-release survival of White Seabass was conducted in 2008-2009, using recapture data obtained from the juvenile gill net studies between 1999 and 2004 when this sampling program was most comprehensive and consistent (Hervas et al. 2010). It is acknowledged that survival estimates obtained in the Hervas et al. (2010) study may not be fully representative of post-release survival in the most recent years of the program. Efforts have been made since about 2009 to enhance survival by modifying release strategies in the light of results from Hervas et al. (2010) and other considerations. Moreover, changing environmental conditions may have influenced survival. Survival has not been quantitatively re-assessed since the Hervas et al. (2010) study, mostly due to a relative paucity of data and combined with changes in sampling regime that that would require re-estimation of a large number of parameters from limited data. Gill net studies were discontinued between 2008 and 2012 and have subsequently been re-started in a modified design and at only about 25% of the sampling intensity achieved in 1999-2004. While a full quantitative assessment has not been conducted, overall recoveries of hatchery juveniles in the gill net studies since 2012 (0.022% of stocked fish) are similar or slightly greater than recoveries achieved from 1999-2004 (0.019% of fish stocked at all sites, 0.013% of fish stocked south of Los Angeles, between IB and PV; Fig. 4.2). While there is no indication of dramatic changes in post-release survival between these periods, the increase in recapture rates holds promise for the modified release strategies that have been enacted since 2010.

Table 4.3. Adult White Seabass scanned for tags, and tagged hatchery fish recovered from the commercial and recreational fisheries. Scanning was conducted by HSWRI, CDFW, and California Recreational Fisheries Survey (CRFS). 2008-2016 numbers obtained from CDFW and are totals of commercial, recreational and CRFS data. Data from Jan-Jun of both 2008 and 2009, and 1999-2007 were obtained from CCC reports.

Year	# adult fish scanned	# adult recaptures	% hatchery fish
2016*	1819	2	0.11%
2015	1,903	4	0.21%
2014	2,324	5	0.22%
2013	1,909	5	0.26%
2012	3,189	4	0.13%
2011	6,257	6	0.10%
2010	5,679	15	0.26%
2009	7,341	30	0.41%
2008	10,140	29	0.29%
2007	9,592	24	0.25%
2006	10,850	25	0.23%
2005	4,430	14	0.32%
2004	3,441	12	0.35%
2003	8,171	6	0.07%
2001/2002	1,847	2	0.11%
2000/2001	1,368	2	0.15%
1999/2000	920	3	0.33%

* Numbers from Jan – Nov 2016 provided by CDFW at the time this table was made do not include CRFS efforts to scan fish (usually only 10-20 per year).

4.4.1.1. Recapture of tagged fish.

Reported recaptures of tagged fish in the commercial and recreational fisheries were low in absolute terms: between 1999 and 2016 only 197 tagged adult White Seabass (legal size) were recovered (Table 4.3). This included several older hatchery-raised White Seabass (10 to 13 years old), but most were between the ages of three and nine (see Figures 11-2 and 11-3 in White Seabass Enhancement Plan 2010). It should be noted that the number of White Seabass scanned for tags varied greatly between years (from 920 to 10,850, i.e. by more than an order of magnitude) and so did the numbers of tagged fish recovered. However, the proportion of hatchery fish in the sample has been fairly constant at 0.24% on average (range 0.09-0.41%).

Recoveries of hatchery fish in the juvenile gill net studies were far higher than in the commercial and recreational fisheries. Between 1988-2008 and 2012-2014, nearly 1,500 hatchery-raised juvenile White Seabass were recovered in the gill net studies within the Southern California Bight (White Seabass Enhancement Plan 2010) (see Section 4.2.1.2). An example of recoveries in juvenile gill net sampling between 2012 and 2015 is given in Table 4.4.

Note that annual recoveries of White Seabass are still low in absolute terms (100-200 fish) and are associated with substantial bycatch of non-target species (thousands). The proportion of hatchery-released fish among the overall number of juvenile White Seabass captured in gill net studies can be substantial, up to 27.7%. The majority of recaptures of hatchery fish in juvenile gill nets occur shortly after release, when fish have been at liberty for between 7 to 1,084 days (mean 139 days).

The large difference in proportional contribution of hatchery fish between commercial and recreational fisheries for adults (0.09-0.41%) and gill net surveys for juveniles (7.5-27.7%) is due to two factors: dispersal and mortality. Many gill net sampling stations are associated with release locations, and therefore high recapture numbers and proportional contributions of hatchery fish are observed in the months following releases (when hatchery fish remain concentrated near the release locations and are numerically abundant even if high mortality rates reduce abundance rapidly over time). The difference between recaptures of the juveniles and sub-adults in the gill net sampling and the capture of older adults in the fishery is, in part, caused by dispersal of juveniles and sub-adults (i.e., up to age 3) away from the study area and by mortality. There's a short term localized abundance of hatchery fish in the study area, followed by predation and dilution of abundance once they disperse, and eventually enter the fishery, which samples a much broader area.

Table 4.4. Recoveries from HSWRI/SDSU juvenile gill net sampling in the Southern portion of the Southern California Bight, fiscal years 2012-2015.

Fiscal year	total # White Seabass recovered	# hatchery White Seabass recovered	% hatchery fish	Range of days at liberty	Avg days at liberty ±1SE	# fish from other species recovered[1]
2012-2013	100	16	16.0	13-731	81.7 ± 45.6	4,144
2013-2014	191	53	27.7	2-392	31.5 ± 9.4	5,634
2014-2015	113	10	8.8	27-405	134.9 ± 50.3	3,675
2015-2016	254	19	7.5	3-760	140.1 ± 53.9	5,466

[1] Other species commonly caught in gill nets include: Yellowfin Croaker (*Umbrina roncador*), Spotfin Croaker (*Roncador stearnsii*), Pacific Chub Mackerel (*Scomber japonicus*), and Salema (*Xenistius californiensis*) (Gill Net Reports 12-13, 13-14, and 14-15).

4.4.1.2. Analysis and modeling approach.

Estimating post-release survival of hatchery fish requires a mark recapture model, such as the one developed by Hervas et al. (2010) to estimate dispersal and mortality rates for stocked White Seabass.

Preliminary analyses by Hervas et al. (2010) indicated that the number of reported recaptures of legal-sized fish in the fisheries was too small to allow meaningful mark-recapture modeling.

Instead, Hervas et al. (2010) focused their analysis of survival on data from 720 tagged hatchery fish recovered in the juvenile gill net fishery between 1999 and 2004, a period during which the gill net studies were most intensive and consistently implemented.

The analysis used all available data on releases and recaptures in nearshore habitats along the continental coast of Southern California, but excluded releases and recaptures from Catalina Island. Release and recapture sites along the continental coast are connected by contiguous nearshore habitat preferred by juvenile White Seabass, allowing for free distance-based dispersal that could be estimated using a diffusion model (see below). By contrast, preferred nearshore habitat on Catalina Island is restricted to the island's coast and possibly, smaller areas along this coast, thereby restricting dispersal in ways that could not be assessed using the available data. Since the dispersal model estimated for the continental coast could not be applied to the island conditions and a more appropriate model could not be estimated, it was not possible to account for the effect of dispersal on recapture probabilities at Catalina and therefore, it was not possible to reliably estimate post-release mortality rates for this site. (A higher proportion of released fish was recaptured at Catalina compared to releases along the continental coast, and this difference is consistent with the effect of lower dispersal at Catalina, keeping fish more concentrated and therefore more catchable.)

4.4.1.3. Estimation of dispersal and mortality rates.

Hervas et al. (2010) developed a mark-recapture model to estimate post-release mortality of White Seabass while accounting for the effects of fish dispersal and the spatial and temporal distribution of gill net fishing effort and selectivity on observed recaptures. The overall pattern of observed and predicted gill net recaptures after release, aggregated over all releases that were monitored between 1999 and 2004, is shown in Fig. 4.3. The rapid decline in recaptures over the first 1.5 years reflects a combination of dispersal and mortality. Since many experimental fishing sites are co-located with release sites, high recaptures are obtained shortly after releases have taken place, but recaptures decline rapidly due to a combination of dispersal (reducing aggregation of fish near release/sampling sites) and natural mortality (reducing overall numbers). Regardless, the model used to estimate mortality accounts for the dispersal of fish and the temporal patterns of fishing relative to the time of release, eliminating a need to exclude recapture data taken within 2 weeks of a release. Note that very few fish have been recaptured at more than 1.5 years since release, even though the fish remain vulnerable to capture by research gill nets to about age 3 and the sampling program covers much of the coastline where the fish can be expected to be located, given the dispersal characteristics. Movement into Mexico or out of the northern boundary of the sampled area is accounted for by the dispersal model. The model assumes no movement of juveniles into deeper water (away from the coastal strip covered by gill net sampling). This assumption is consistent with life history information (and the assumptions underlying the design of the gill net survey). Furthermore, the mortality rates estimated are consistent with observed contributions to the adult stock. If juveniles emigrated into deeper waters and survived to contribute to the adult stock, the hatchery contribution at that stage would be expected to be larger than observed.

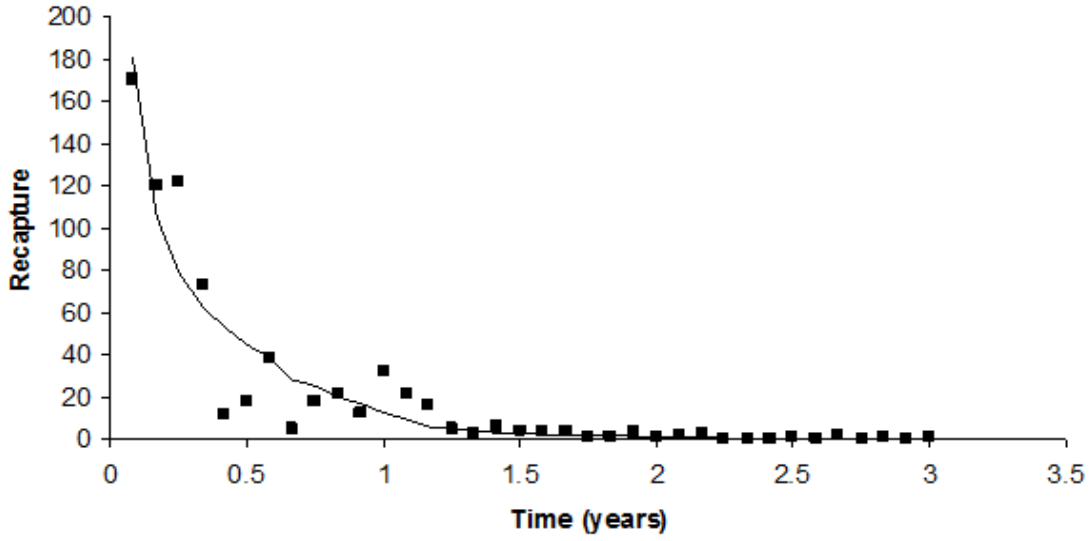

Fig. 4.3. Observed and predicted recaptures in the juvenile gill net research fishery as a function of time after release (data from the analysis of Hervas et al. 2010).

In the first step of the work that Hervas et al. (2010) conducted, a dispersal model was developed and fitted to observed spatio-temporal patterns in recaptures of released hatchery-reared White Seabass. The dispersal model describes movement of released juveniles along the coastline (where preferred juvenile habitat is concentrated in a narrow band) using a two-dimensional diffusion model. Model results showed that fish disperse from release sites rapidly after release, but that 50% of fish remain within 47 km and 95% within 135 km of the release site at the end their third year at large (Fig. 4.4).

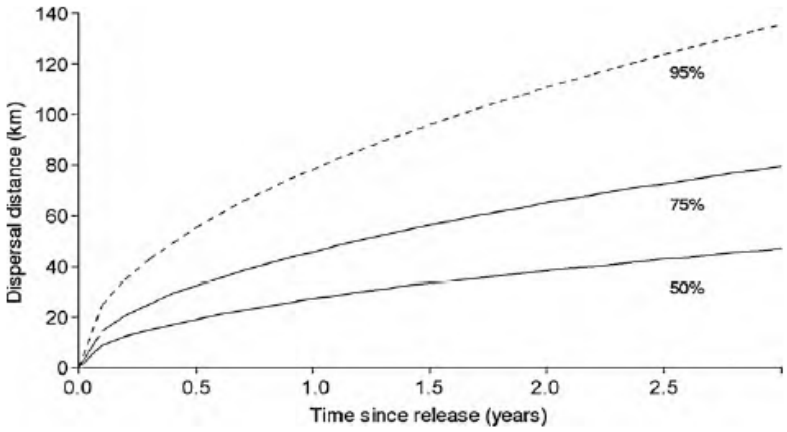

Fig. 4.4. Estimated dispersal of stocked White Seabass from the release sites (from Hervas et al. 2010).

Using information on fish dispersal and growth, locations and times of gill net sampling, and the size selectivity of gill nets (e.g., Fig. 4.1), Hervas et al. (2010) constructed a temporal pattern of gill net fishing (=sampling) effort for each release event. In line with common practice in

fisheries science, natural mortality was expressed as an exponential rate M, which is related to proportional survival over a period of time t by the equation $S = e^{-Mt}$ (or, rearranged: $M=-\ln(S)\, t^{-1}$). An annual mortality rate M of 0.2 implies a survival S of 0.82 (82%) per year, M=1 implies S=0.37 (37%) and M=5 implies S= 0.007 (0.7%). Note that M can be greater than 1, sometimes considerably so.

Natural mortality rates in fish are well known to be size-dependent, and this is an important consideration for hatchery programs that typically release juvenile fish at a much smaller size than that at which fish from the same stock are harvested in fisheries (typically as adults). Mortality rates were therefore modeled using a size-dependent mortality function. Natural mortality rates within natural fish populations are strongly size-dependent with an allometric weight exponent of around –0.33 (range –0.29 to –0.37 in different studies) (Lorenzen 1996). In other words, natural mortality is approximately inversely proportional to length:

$$M(L)=M_1 L^{-1}$$

where M(L) is the natural mortality rate at length L, and M_1 is the natural mortality rate at unit length. (When length is measured in centimeters, M_1 can be interpreted as the mortality rate of a fish at 1 cm length, the 'unit length'. Note that this is simply a model parameterization chosen for mathematical convenience, it does not imply that 1 cm long fish actually exist in the study population or elsewhere). The resulting relationship between M(L) and L is illustrated in Fig. 4.5 (note the logarithmic scaling on both axes, which means that the length-inverse relationship is shown as a straight line of slope -1). The length-inverse model has been shown to provide good predictions of survival in relation to release size in fish stocking experiments (Lorenzen 2000). Moreover, by expressing size-dependent mortality in terms of a single parameter (M_1), the length-inverse mortality model facilitates comparative analyses of data from experiments in which mortality rates have been measured for different fish sizes.

Four alternative natural mortality models were tested for White Seabass:

Model (1) Length-inverse mortality model applied to all releases without allowing for seasonal or release method effects,

Model (2) Length-inverse mortality model allowing for seasonal and release size effects by estimating separate M_1 parameters by season and an additional M_1 for direct (non-pen) releases,

Model (3) Length-inverse mortality model with an additional short-term mortality term applied only to the first month after release, without allowing for seasonal or release method effects (this is essentially Model (1) with an added short-term mortality effect), and

Model (4) Length-inverse mortality model with additional short-term mortality terms applied only to the first month after release and accounting for seasonal and release method effects by estimating separate short-term mortality terms by season and release method.

Table 4.5. Results of the model selection and parameter values. L is the negative log likelihood, m the number of parameters estimated, QAIC the quasi Akaike Information Criterion, Δ the difference in model QAIC to the lowest QAIC in the set, and W the Akaike weight (an approximate probability of the model being the best model in the set).

Model	Parameter	Value	95% CI	L	m	QAIC	Δ	W
Model 1	M_r	66.3	[55.8, 77.3]	1724.9	2	1728.9	30.7	0.00
	F	0.013	[0.011, 0.016]					
Model 2	$M_{r,Winter}$	85.1	[62.8, 112.7]	1701.7	6	1713.7	15.5	0.00
	$M_{r,Spring}$	24.7	[5.2, 46.9]					
	$M_{r,Summer}$	48.2	[33.6, 65.8]					
	$M_{r,Autumn}$	41.5	[28.9, 56,1]					
	$M_{r,direct}$	41.7	[24.7, 60.1]					
	F	0.013	[0.011, 0.016]					
Model 3	M_r	36.9	[25.2, 49.5]	1701	3	1707	8.8	0.01
	M_s	576	[418, 732]					
	F	0.051	[0.035, 0.072]					
Model 4	M_r	34.1	[21.6, 46.8]	1684.2	7	1698.2	0	0.99
	$M_{s,Winter}$	737	[487, 984]					
	$M_{s,Spring}$	295	[86, 493]					
	$M_{s,Summer}$	421	[208, 636]					
	$M_{s,Autumn}$	410	[202, 621]					
	$M_{s,direct}$	166	[53, 276]					
	F	0.037	[0.022, 0.053]					

Model (4) provided the best fit to the mark-recapture data overall (Table 4.5). This implies that separating short-term post-release mortality from the longer-term pattern, and accounting for season and release method effects on short-term post-release mortality provides the best description of the observed recapture patterns.

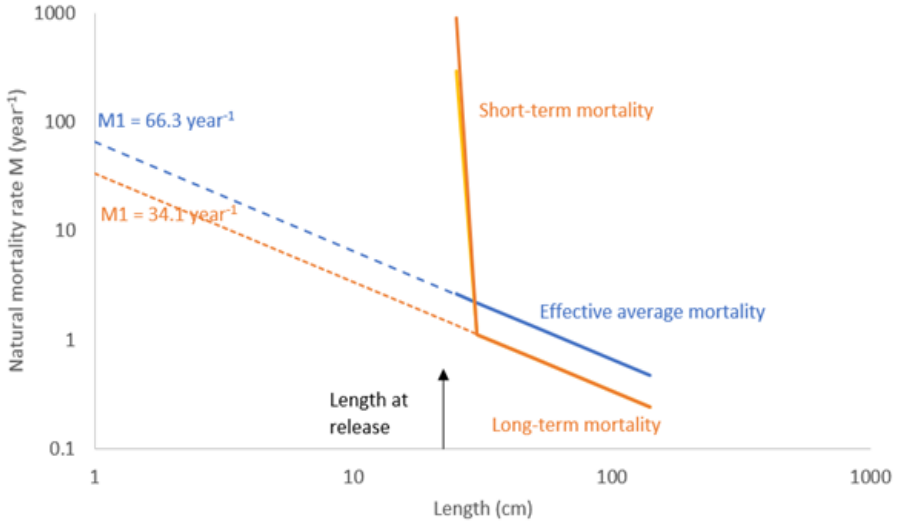

Fig. 4.5. Size-dependent mortality patterns for stocked hatchery White Seabass as estimated in the mark-recapture modeling. Model (4) incorporating short-term and long-term mortality rates (orange) provided the best fit, while Model (1) which provides an 'effective average' lifetime mortality pattern (blue). The dashed and dotted lines extrapolate the size-dependent mortality patterns to unit length (1 cm) and illustrate the meaning of the M_1 parameter.

Model (1), while the worst-fitting of the four candidate models, still provides useful information on the effective average mortality rate M_1 for all releases combined (regardless of season and release method), in a way that is directly comparable to size-dependent mortality rates estimated wild fish. The appropriateness of the Model (1) M_1 as an estimate of effective average mortality is also borne out by the fact that it provides accurate predictions of the proportional contribution of hatchery fish to the overall adult population (see Section 4.6). Mortality models (1) and (4) are further illustrated in Fig. 4.5.

4.4.1.4. Benchmarking of mortality.

Comparative information on the size-dependent mortality rate M_1 (natural mortality rate at 1 cm) in wild and released hatchery fish has been compiled in Lorenzen (2006). Data for wild fish were compiled from a large meta-data set covering 308 marine and freshwater fish (Lorenzen 1996). Data for released hatchery fish were compiled from a smaller sample of 53 stocking events involving seven populations of freshwater fish (Lorenzen 2000). Mortality data for released hatchery fish were derived from both stocking experiments purely for research purposes and experiments conducted as part of operational fisheries enhancement programs. The data are indicative of the range of post-release mortalities suffered by hatchery-reared fish in the wild, and do not represent post-release mortality rates that are achieved only in hatchery programs that are demonstrably effective at enhancing fisheries.

Frequency distributions of mortality rates in wild released hatchery fish are shown in Fig. 4.6. The distribution of M_1 for wild fish is skewed (approximately log-normal) with a median of 16.5 $year^{-1}$. The cumulative distribution shows that 75% of M_1 values in wild fish are below 30 $year^{-1}$, while 90% are below 55 $year^{-1}$. The distribution of M_1 in released hatchery fish extends far to the right of that for wild fish, with a median of 66.5 $year^{-1}$ and 25% of estimates greater than 175 $year^{-1}$. Thus post-release performance of hatchery fish may be similar to that of wild fish, but is often much lower. As further estimates of M_1 for released hatchery fish become available, the distribution shown here could become increasingly informative and may allow quantifying the benefits of measures aimed at improving post-release mortality such as habitat enrichment, life skills training or artificial selection (Jonasson et al. 1997, Olla et al. 1998, Brown and Day 2002, Beamish et al. 2004) and optimization of release strategies (e.g., Leber et al. 2016).

The mark-recapture modeling provides two estimates of M_1 that can be directly compared to the distributions in Fig. 4.6. The Model (1) estimate of M_1 =66.3 $year^{-1}$ represents an effective average M_1 for all releases monitored in 1999-2004. The Model (4) estimate of M_1 =34.1 $year^{-1}$ represents a long-term mortality pattern after the initial, additional short-term mortality has subsided. It represents an ideal case that could be attained if the additional short-term mortality was eliminated through improvements in release strategies. Comparing these values to the distributions in Fig. 4.6, the effective average mortality of stocked White Seabass in 1999-2004 was close to the median for other hatchery fish released into the wild (66.5 $year^{-1}$) while the long-term mortality component alone would be substantially lower. Both the effective average (66.3 $year^{-1}$) and long-term mortality rates (34.1 $year^{-1}$) were substantially higher than would be expected for wild fish populations (median: 16.5 $year^{-1}$). The range of M_1 values bounded by the 1999-2004 effective average (66.3 $year^{-1}$) and long-term mortality component (34.1 $year^{-1}$) may be regarded as the 'space of opportunity': the range of post-release mortality rates that can realistically be achieved through improvements in husbandry and release strategies. The 'best' release strategies (spring, summer and fall releases following acclimation in net pens, see below) already performed in this space in 1999-2004. Conducting all releases using these strategies should increase the overall average from 66.3 $year^{-1}$ but not attain 34.1 $year^{-1}$ since even the best strategies were associated with substantial short-term post-release mortalities additional to the long-term component. Further reductions in mortality

could be achieved only through changes in husbandry and/or release strategies that were not tested in 1999-2004.

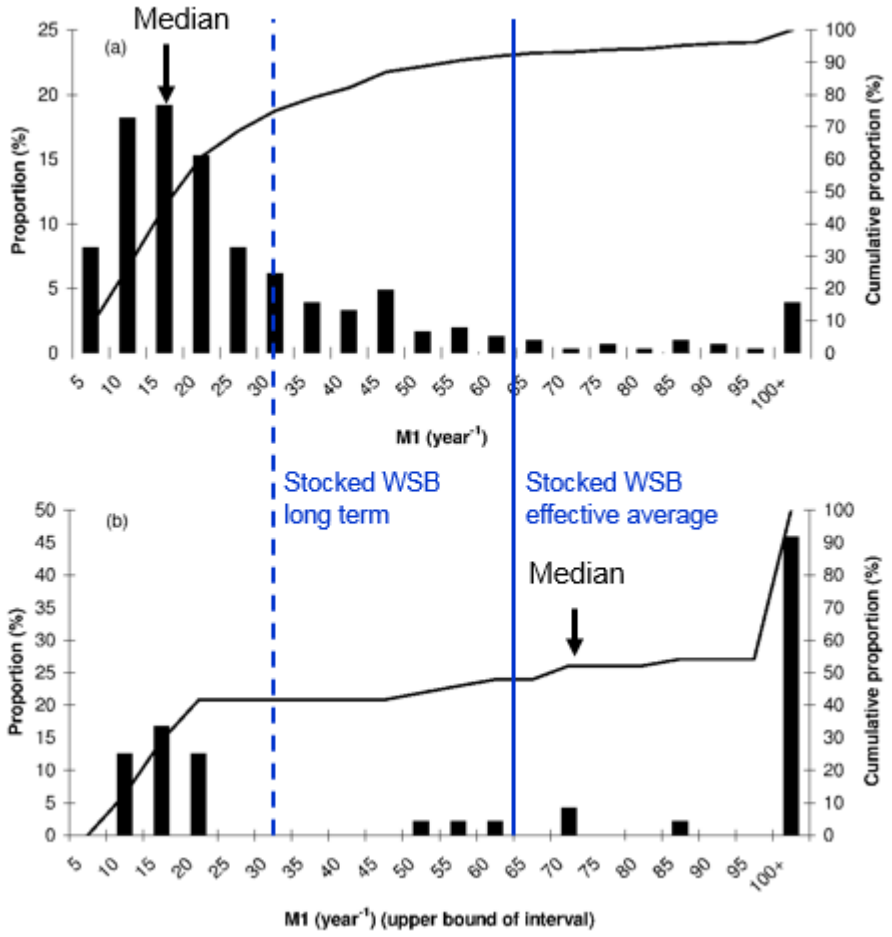

Fig. 4.6. Comparison of stocked White Seabass mortality rates with (a) distributions of mortality M_1 (at a reference length of 1 cm) for natural populations, and (b) released hatchery fish. Distributions from Lorenzen (2006), based on data from Lorenzen (1996) and Lorenzen (2000).

The high natural mortality rates estimated for released hatchery fish from mark-recapture modeling of juvenile gill net sampling data are corroborated by recapture data from the commercial and recreational fisheries, and by the relative contributions hatchery fish make to the stock at different stages of the life cycle. The White Seabass population, set up using the post-release mortality rates estimated from the gill net study, accurately predicts the proportional contribution and recapture ratios of hatchery fish in the commercial and recreational fisheries (Section 4.6; Figs. 4.12 and 4.13). Furthermore, with the stock assessment estimating recruitment (abundance of juveniles at around 28 cm TL) to be in the range of 100,000-500,000, with an average of 300,000 per year in 1995-2004, the average of 135,000 hatchery juveniles released per year would contribute in the order of 30% to the abundance of

juveniles at the size at release. In the gill net samples, which cover juveniles of a size range attained with three years of release, the contribution of hatchery fish was about 5% in 1999-2004 (15% in 2012-2016, Table 4.4, when natural recruitment was lower than in 1999-2004, Fig. 4.8). In the commercial and recreational fisheries harvest, which covers mature fish aged around five years and older, the contribution of hatchery fish is only 0.26% on average. The decline in contribution of hatchery fish to the stock and to the catch with increasing time (or age) after release is due, in small part, to hatchery fish dispersal and associated dilution effect, and in large part to higher mortality rates of hatchery fish than wild fish.

4.4.1.5. Evaluation of release strategies.

The mark-recapture model further indicated that survival of released hatchery fish was highest in Spring, moderately lower in Summer and Autumn, but much lower in Winter releases. Acclimation in net pens had a substantial, positive effect on survival relative to direct releases. (This applies to survival upon release into the wild, not release into the net pen. Survival of fish while in net pen facilities can be highly variable as discussed in Section 1.3.1). Predicted survival to the legal minimum length is shown in Fig. 4.7, as a function of length at release and for different release seasons and methods. Survival of hatchery fish to 600 mm SL in the fishery was estimated at 1.5% for a release size of 200 mm, rising to 13.8% for a release size of 400 mm, under optimal conditions (Spring releases with net pen acclimation) (Hervas et al. 2010). Also shown in Fig. 4.7 is the expected survival of wild fish from the same initial 'length at release'. The expected survival of wild fish is based on a value of $M_1 = 15$ year^{-1}, consistent with the empirical distribution shown in Fig. 4.7 and with a natural mortality at the length at maturity (700 mm) of 0.21 year^{-1}, the same value as used in the White Seabass stock assessment (Valero and Waterhouse 2016). Clearly, the survival of hatchery White Seabass is much lower than would be expected for wild fish, and that holds for all release strategies tested in 1999-2004.

Modifications of release strategies since 2009. Since about 2009, HSWRI has implemented changes to its release strategies for White Seabass. Some of the changes were informed by the Hervas et al. (2010) study: (1) minimum 200 mm size (8") (a standard since 1996), (2) releases in spring, summer and fall (reduction/discontinuation of winter releases), and (3) reduction/discontinuation of direct releases without net pen acclimation of at least two weeks (Fig. 4.7). Two other changes were made because HSWRI staff considered them likely beneficial: (4) pre-release assessment (count and handling) was carried out at least 21 days prior to release (BMP to reduce stress at release), and (5) releases from the net pens were carried out at dusk (BMP to reduce predation at release).

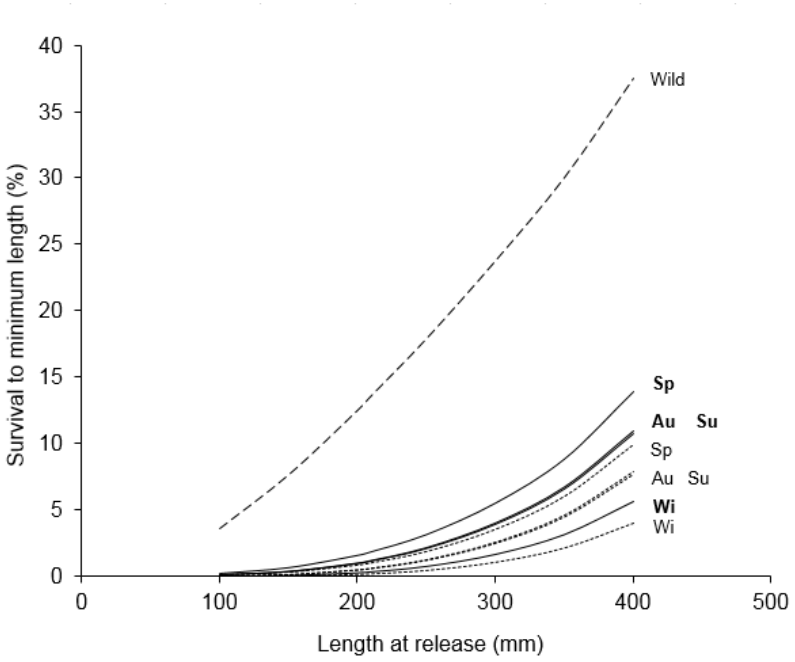

Fig. 4.7. Predicted proportion of released hatchery white seabass surviving to the legal minimum length as a function of release size, for Spring (Sp), Summer (Su), Autumn (Au) and Winter (Wi) releases and net pen acclimation (solid lines) or direct release (dashed lines). Also shown for comparison is the expected survival of wild fish from the same initial length (Modified from Hervas et al. (2010)).

Again, survival has not been quantitatively re-assessed since the modified release strategies that followed Hervas et al. (2010), but there may well have been incremental improvements (Fig. 4.2) that could be better quantified by a new mark-recapture analysis once sufficient data are available. Incremental improvements are the most realistic expectation, given the magnitude of differences in survival associated with different release strategies and the fact that the mix of release strategies employed during the Hervas et al. (2010) study already included many 'above average' releases, while even in the most recent years some releases had to be carried out under sub-optimal conditions.

It should also be borne in mind that mortality rates may vary in response to environmental conditions. The stock assessment shows very high levels of natural recruitment of White Seabass (100,000-500,000 recruits per year) between 1995 and 2004, with much lower levels (50,000-200,000 recruits per year) in the preceding and following years (Fig. 4.8). If strong recruitment pulse in 1995-2004 is indicative of unusually good environmental conditions for juvenile White Seabass, it is possible that mortality rates at the time were in fact lower than in subsequent years (for the same release strategy).

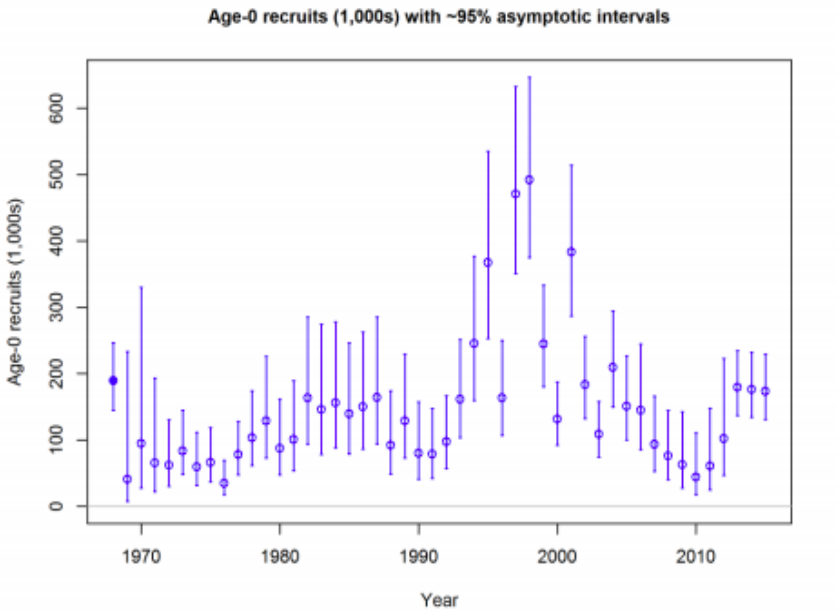

Fig. 4.8. Figure e in the White Seabass Stock Assessment Report (Valero and Waterhouse 2016) showing time series of estimated age-0 recruits with 95% asymptotic confidence intervals. The solid blue dot before the start of the time series is the estimated equilibrium unfished average recruitment with 95% asymptotic confidence interval.

4.4.1.6. Reasons for high mortality of released hatchery fish and the role of domestication. Survival of hatchery fish released into the wild is dependent on a number of factors including life skills training, fish fitness, and release strategies. Release strategies include the number, size, timing and place (habitat) of release as well as whether or not fish are acclimated to the release site (e.g. in net pens) prior to full release into the wild. HSWRI has experimentally explored certain aspects of release strategies, namely: release size, season, and the effects of acclimation or outgrowing in net pens. These factors are likely to have the largest impact on mortality rates right after release. Considerations to try to improve survival (increase tag returns) discussed in the White Seabass Enhancement Plan (2010) include altering release strategies. In the mark-recapture modeling of Hervas et al. (2010), such effects were analyzed as influences on the short-term component of mortality. Impacts of hatchery rearing on fitness in the wild, on the other hand, are likely to have effects on both short-term and long-term components of mortality.

Rearing in fish culture facilities subjects fish to domestication: a process of change in the cultured organism that involves genetic changes occurring within and over generations and developmental effects (phenotypic plasticity) recurring during each generation (Price 2002, Section 3.2.1.6). Exposure to the environment of culture facilities (characterized by confinement in small spaces, high densities, low habitat complexity, regular supply of feed in quantities meeting or exceeding the organism's needs, low predation risk, etc) alone is sufficient to alter selection regimes and developmental pathways of fish substantially (Lorenzen et al. 2012). Hence strong domestication effects can occur within the culture of first-generation

hatchery juveniles even if, as in the case of White Seabass, only wild fish are used as broodstock in order to avoid inter-generational domestication selection.

Domestication effects are manifested in a suite of biological changes in the cultured organism including a general acceleration of the lifecycle, reduced behavioral complexity including reduction in foraging and predator avoidance behavior, and increased activity and movement (which uses energy and increases exposure to predators) (Lorenzen et al. 2012, Garlock et al. 2014). Domestication effects have strong and almost always negative impacts on the capacity of hatchery fish to survive, grow, and reproduce in the wild (Lorenzen et al. 2012), and such effects are likely to contribute to the high mortality rates measured for stocked White Seabass. It should be noted here that domestication effects are related to husbandry practices commonly employed in aquaculture but mostly reflect inadvertent effects of good conventional husbandry rather than "husbandry problems." Indeed, many domestication effects improve the performance of fish within aquaculture systems and are problematic only once fish are released to face the challenges of natural environments.

A variety of measures, such as rearing in near-natural environments, environmental enrichment, life-skills training and soft release strategies, can counteract domestication effects (Olla et al. 1998, Brown and Day 2002), but the effectiveness of these measures in actually improving post-release survival is variable and often unknown. Aquaculture production for release into natural ecosystems may thus benefit from culture practices that differ from those normally employed in facilities producing organisms for on-growing in aquaculture facilities and may also require different genetic management (Lorenzen et al. 2012). Domestication effects in hatchery-reared fish and their management became a vibrant research area in the 2000s, long after the original design of the OREHP, and has not yet been taken up in the program. More attention to this area is indicated as the program moves forward, but it must be appreciated that progress in reducing domestication effects and mortality in the wild is likely to be slow and will require substantial research investment.

At present, it is not known what husbandry or release strategy changes might be beneficial to post-release survival. A thorough review of the literature and the current hatchery operation would be required to identify promising interventions, and ideally these would be tested in smaller-scale experiments prior to implementation on an operational scale and true 'field testing' of effects on survival.

Experimental work with White Seabass is constrained by the very low effectiveness of sampling released fish. Even with the high level of gill net sampling effort used in 1999-2004, only 0.2% of released hatchery fish were recovered. Since the precision of mortality estimates depends on the number of fish recaptured, very high numbers of fish must be released for every experimental treatment in order to measure treatment effects with good precision. If a higher sampling efficiency could be achieved, for example by using different sampling approaches or conducting experiments in enclosures under simulated habitat and predation conditions, that would radically improve the scope for experimental work to improve post-release survival. It is

unclear, however, whether more effective sampling/experimental approaches can be developed for this species.

4.4.2. Data and Information Gaps.

There is uncertainty about why there are low recapture rates, including:

1. Uncertainty about the contribution of low catch rates in gill nets as compared to short-term post-release mortality of the smaller size class juvenile fish (≤20 cm SL).
2. The relative contributions of domestication effects, physical robustness, and release strategies to the high natural mortality rates experienced by released hatchery White Seabass (see also Sections 1.9.1.2, 3.2.1.6, and 3.3.1.2).
3. Husbandry and release approaches that may increase post-release fitness/reduce mortality rates of hatchery fish in the wild.

4.4.3. Recommendations.

1. Develop a research program to assess domestication effects during hatchery rearing of White Seabass and options for reducing such effects or counteracting their impact on post-release survival. Such a program would entail a major and long-term effort but is likely required if enhancement is to make an effective contribution to stock enhancement or rebuilding (See also 3.3.3 Recommendation 3).
2. Evaluate optimal size(s) at release for White Seabass, based on integrating survival effects of size at release with the costs to rear White Seabass to various stocking sizes (after Leber et al. (2005)) (See Section 1.7.3 Recommendation 1).
3. Identify optimal release habitat by monitoring stocked fish released in a variety of sites, that have been identified as White Seabass juvenile nursery habitats, and determining whether some stocking sites result in disproportionately higher post-release survival rates than others (See Section 1.2.2.2 Data gap 3).
4. Conduct more consistent market surveys to collect more data on adult tagged fish (i.e., contributions to the fishery).
5. If tagged fish recapture rates can be increased (improved recapture methods for small fish, and improved health for all fish), develop and run a model to evaluate the effectiveness of release strategies using mark recapture data (a model proposed by M. Drawbridge (HSWRI) and Dr. Kai Lorenzen). Occasionally update model to inform adaptive management.
6. Evaluate effects on short-term post-release survival of various pre-release acclimation strategies (exposure in the hatchery to natural substrate, natural prey and predators).
7. Evaluate effects of release magnitude on survival and dispersal at various release sites.
8. Concentrate a large proportion of monitoring efforts on evaluating short-term mortality over a period of ≈ 6 months after stocking. This will more rapidly inform adaptive management needs and provide rapid results from pilot release experiments designed

to evaluate effectiveness of release strategies and allow optimization of those strategies.

9. Place a higher priority on (i.e., significantly increase the budget for) monitoring post-release survival and the contribution of hatchery releases to fishery management goals.
10. We do not recommend increasing hatchery output at the expense of increasing short-term post release survival. The latter is a more powerful way to achieve more success from hatchery releases. Optimizing release strategies can result in increases to recapture rates and short-term survival by as much as double, triple or even an order of magnitude (Leber et al. 1998, Leber et al. 2016).

4.5. Estimates and uncertainties of stock size.

4.5.1. Key Findings.

4.5.1.1. White Seabass stocks.

The White Seabass Stock Assessment (Valero and Waterhouse 2016) and historical data show that stock abundance and fisheries catches had declined to historically low levels in the late 1970s, at the time when the idea of the OREHP was conceived. The stock recovered naturally throughout the 1980s and increased dramatically in the 1990s as a result of very strong recruitment. Recruitment returned to much lower levels in the 2000s and as a result, spawning stock biomass has been declining over the past nine years. Hence recovery is thought to have been achieved naturally, due to a combination of more stringent fishing restrictions and favorable environmental conditions.

The White Seabass stock dynamics and fishery appear to be very strongly driven by climatic factors and therefore, substantial variation is expected to continue into the future. This may include further episodes of low stock abundance and catches. Whether stocking can augment catches over such periods will depend, at least in part, on the mechanisms through which climatic factors influence stock dynamics.

White Seabass stock structure and dynamics*.* White Seabass is a sciaenid (croaker) with a full geographic distribution from Magdalena Bay, Baja California, Mexico to Juneau, Alaska (Thomas 1968), but most commonly occurring in coastal waters off of California and Baja California, and to a lesser extent off of Oregon and Washington (Valero and Waterhouse 2016). As discussed in Section 3.1.1, the wild population structure has been studied, but is unclear, as some studies (Franklin 1997) suggest genetic differentiation, and others (Coykendall 2005, Buonaccorsi et al. 2001) provide no evidence for genetic population structure. As reviewed by Hervas et al. (2010), White Seabass is thought to form a single breeding population, with a center off central Baja California, Mexico (Moser et al*.* 1983, Vojkovich and Reed 1983). White Seabass moves actively throughout the Southern California Bight and beyond (Aalbers and Sepulveda 2015), and is usually found in nearshore habitats associated with rocky headlands, sandy areas, and in and around kelp forests. Spawning occurs near shore during the spring and summer months

peaking in June (Donohoe 1997). The dynamics and possibly, distribution of the stock have long been regarded as strongly influenced by climate-driven environmental variation (Young 1973).

The California White Seabass fishery. White Seabass are fished commercially and recreationally. The commercial fishery started in the late 19th century and has dominated catches until very recently. The recreational fishery developed in the mid-20th century and has expanded to the point where its harvest exceeded that taken in the commercial fishery during the late 1990's and early 2000's (WSFMP 2002, CDFG 2008). However, recent recreational landings have not exceeded more than about a third of total (commercial and recreational) landings. The White Seabass population is believed to have declined substantially since the onset of the fishery, most severely during the 1920s-1930s and again during the 1960s-1970s, although it is unclear whether these low points in abundance were actually due to low biomass in California waters, or to changes in the availability of fish off California (if they migrate elsewhere, for example) (Thomas 1968, Vojkovich and Reed 1983, Valero and Waterhouse 2016). Two studies on historical fisheries and population trends have concluded that the population was moderately exploited but not depleted (MacCall et al. 1976, Dayton and MacCall 1992). Fishing restrictions were first introduced in 1931 and currently comprise a minimum landing size (71 cm TL, equivalent to 60 cm SL), closure during the spawning season (mid-March to mid-June), bag limits, and gear restrictions.

Although the data for White Seabass are classified as “data moderate”, an Optimal Yield (OY) measure was estimated in the White Seabass Fishery Management Plan for use as an interim managment measure until more comprehensive data could be collected and integrated into the plan (WSFMP 2002). Several “triggers” were also defined included taking management action if the total annual commercial catch of White Seabass, in pounds landed, declines each for two consecutive years by 20% or more from the prior five-year average of landings; and if recruitment of juvenile White Seabass declines each year by 30% or more from the prior five-year average of recruitment as determined from the best available data (WSFMP 2002). These actions outlined in the FMP were expected to allow recovery of the fishery. The 2016 stock assessment revealed that biomass had been decreasing over the last 9 yrs (Valero and Waterhouse 2016) indicating that an assessment of the “triggers” may be warranted.

4.5.1.2. White Seabass Stock Assessment 2016.

A first full stock assessment of the White Seabass wild stock was conducted in 2015-2016. The stock assessment was conducted using an integrated, statistical, age-structured model. Growth was estimated separately for males and females, but the same mortality and selectivity estimates were used due to the lack of age data and sex specific size data for White Seabass. Different model runs were conducted for population dynamics between 1870-2014, 1889-2014, and 1969-2014, with 1889 being the first catch records available for White Seabass, and catch calculations (starting points) differing before and after 1969. The models were fit using several different datasets of abundance and length data.

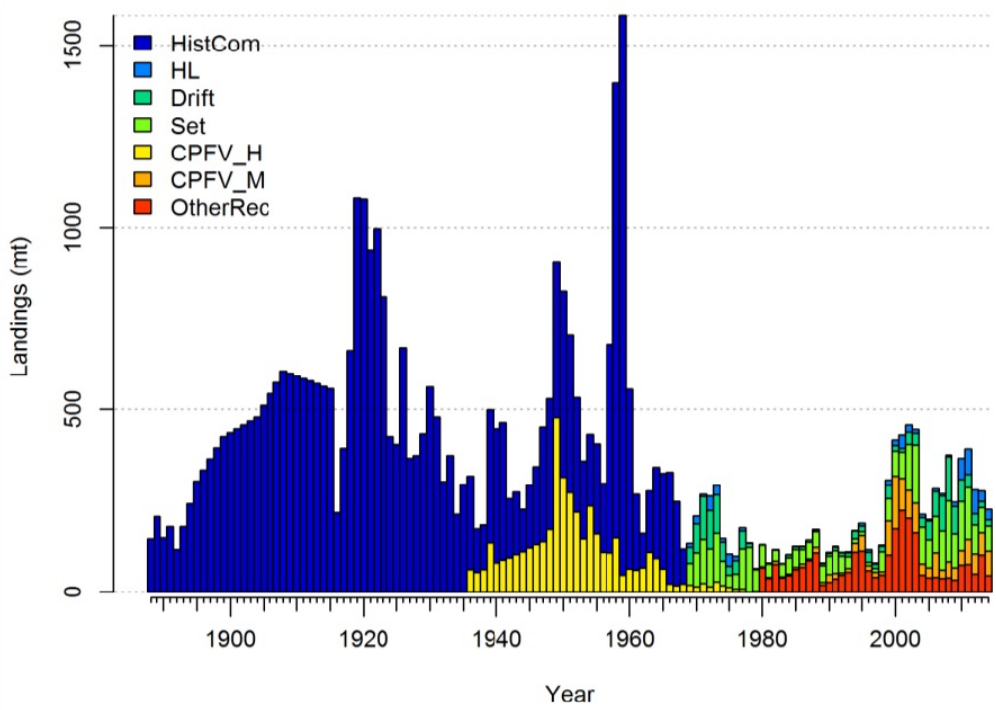

Fig. 4.9. Fig. 6-3 in the White Seabass Stock Assessment Report (Valero and Waterhouse 2016) showing total landings in metric tons (mt) from 1889 to 2014 by fleets as defined in the stock assessment model: Commercial historical (HistCom), Hook and Line (HL), Drift Gill net (Drift), Set Gill net (Set), Historic Commercial Passenger Fishing Vessel (CPFV_H), Modern Commercial Passenger Fishing Vessel (CPFV_M), Other Recreational (OtherRec).

The abundance datasets included landings from Commercial Passenger Fishing Vessels (CPFV) historically (pre-1980), CPFV modern times (post-1980), drift gill net logbook catch per unit effort (CPUE), and set gill net logbook CPUE (e.g., Fig. 4.9), as well as HSWRI gill net CPUE, and Power Plants Heat Treatment CPUE. The length datasets included hook and line, drift gill net, set gill net commercial fisheries, HSWRI gill net surveys, Power Plants Heat Treatment, CPFV observers (modern and historic) and/or a combined "other recreational" group. The HSWRI also provided conditional age to length data for the modeling effort.

Maximum Sustainable Yield. The interim management measure adopted with the WSFMP set the optimum yield (OY) for White Seabass at 544.3 metric tons, which is the limit of total take in the recreational and commercial fisheries (WSFMP 2002, based on estimates of pre-exploitation biomass and natural mortality rate in Dayton and MacCall 1992, and MacCall et al. 1976). This OY was established by making a conservative adjustment to a maximum sustainable yield (MSY) proxy that was calculated from an estimate of the pre-exploitation biomass of White Seabass (following protocol outlined in Restrepo et al. 1998). The 2016 model, however, estimated a MSY of 307 mt (95% asymptotic CI: 238 – 376 mt), much lower than the previously estimated OY, and that corresponds to a female spawning biomass (B_{MSY}) of 447 mt (CV = 0.14; 340-554 mt) and a depletion of 0.24 (Valero and Waterhouse 2016). Estimated MSY depends on the size of fish caught, natural mortality, growth and the steepness (productivity) of the spawning stock curve. There was, however, uncertainty surrounding many of the biological and

fishing processes including the stock-recruitment relationship, natural mortality, growth, maturity, survival rates and numbers of discarded fish (Valero and Waterhouse 2016).

Population depletion and recovery in the context of the OREHP. The White Seabass Stock Assessment and historical data show that stock abundance and fisheries catches had declined to historically low levels in the late 1970s, at the time when the idea of the OREHP was conceived. The stock is thought to have recovered naturally throughout the 1980s and increased dramatically in the 1990s as a result of very strong recruitment. Recruitment returned to much lower levels in the 2000s and as a result, spawning stock biomass has been declining over the past nine years. The recovery of the stock is posited to have occurred naturally, due to a combination of more stringent fishing restrictions and favorable environmental conditions. While changes in environmental conditions are essentially unpredictable, the effects of alternative fishing regulations can be predicted using fisheries assessment models and such an evaluation could have been carried out in the early planning stages of the OREHP in order to consider the scope for natural recovery.

4.5.2. Data and Information Gaps.

Gaps in information needed to perform more accurate stock assessments are listed in the White Seabass Stock Assessment (Valero and Waterhouse 2016); those that overlap with needs of enhancement program assessments include:

1. Lack of non-CPFV and CPFV data, including information on catch and on trips that catch nothing, catch information where White Seabass is caught as bycatch or is otherwise not targeted, and spatial information on fishing effort and catch.
2. Lack of morphological and life history data, including data on fork length needed to convert between total and standard length, gender-specific age data, and maturity data in particular maturity size and/or age.
3. No data on discarded fish (e.g., sex, size, survival).
4. Lack of information on seasonal and inter-annual movements throughout range (transboundary across the U.S. and Mexico border), as well as data on life history, catch history, and effects of oceanographic conditions that influence distributional changes.

4.5.3. Recommendations.

Increase collaboration with U.S. commercial fishermen and CPFV businesses, and Mexican scientists and fishermen to better understand White Seabass dynamics throughout more of its full range.

4.6. Contribution of hatchery fish to the standing stock: Approach, current results, and population-model predictions.

4.6.1. Key Findings.

A population dynamics model for the enhanced White Seabass fishery was developed by Dr. Kai Lorenzen for this review. The model was based on information from the recent White Seabass Stock Assessment (Valero and Waterhouse 2016) and from the mark recapture study of stocked hatchery White Seabass (Hervas et al. 2010). The model indicated that, over the period of releases from 2000-2011, stocked fish contributed on average 0.26% to California White Seabass catches. The overall observed recapture ratio (proportion of stocked fish recaptured in the commercial and recreational fisheries) was 0.036% indicating that the model, which has been developed using information from the White Seabass Stock Assessment and the research fishery only, provides reasonable predictions of enhancement contributions to the fishery. Further, the model revealed that the stocking program to date has made a negligible contribution to the White Seabass population and fishery presumably due to the high natural mortality rates suffered by released hatchery White Seabass. *According to the model, if mortality rates of released hatchery fish equaled those of wild Seabass, stocking of 100,000 juveniles per year (the approximate average release rate of recent years) would increase catches by 18%.* This illustrates the profound effect of high post-release mortality on the effectiveness of the stocking program.

The recapture rate and proportional contribution for the OREHP are not unusual for enhancement programs, particularly in the marine realm. Indeed, as pointed out in Section 4.4.1.4, the post-release mortality rates of White Seabass are slightly below the median for a set of comparative data for other species, yet substantially higher than expected for wild fish. In 2008, Tringali et al. conducted a study that used release-recapture experiments to begin to identify optimal release methods for Red Drum in Tampa Bay, Florida; their release-recapture experiments yielded a recapture rate of 0.00086 for Phase-3 Red Drum (≈8 months old, 130-180 mm SL), and 0.000036 for Phase-1 fish (≈1 month old, 25-45 mm SL), (Tringali et al. 2008). Proportional contributions of 2-3 yr old Cobia based on genetic analysis in South Carolina also began at under 1% in 2007 and 2008, but then rose to 2.7% in 2009, 7.3% in 2010 and 4.7% in 2011 as the fish started to recruit (SCDNR 2015). Although migratory, Cobia returns to the same estuary to spawn where 78% of recaptures occurred; 22% were captured offshore (SCDNR 2015). Proportional contributions of hatchery fish to Red Drum in Texas have ranged from 0% to 30% depending upon place (bay), year, and/or timing of release and recapture (Vega et al. 2011). Red Drum generally stay in the same area and within 5 km of the release site for the first three years after release (TPWD 2017). After that, they move from bays into the Gulf of Mexico, but occasionally return to bays (TPWD 2017). These are in contrast to White Seabass, which disperse within the first few months from release sites up to 135 km away.

4.6.1.1. Population dynamics model of White Seabass enhancement.

Age-structured population dynamics models such as the one used in the White Seabass Stock Assessment are widely used in the assessment of fisheries and evaluation of management

options (Walters and Martell 2004, Edwards et al. 2011). Lorenzen (2005) introduced several extensions to conventional fisheries models with a view to enabling the assessment of enhanced fisheries in the same general framework. The first key extension is the differentiation of the population into components according to genotype and origin (Fig. 4.10). The three components of the total stock considered are wild (wild genotype, naturally recruited), hatchery (hatchery genotype, naturally recruited) and stocked (hatchery genotype, stocked). This differentiation allows us to address a range of different questions, including the contributions of stocking and natural recruitment to yield, and the implications of releasing genetically maladapted fish. Sub-stocks may differ in life history traits such as survival. Interactions between wild, stocked, and hatchery fish occur as a result of density-dependent survival in the pre-recruit stages of the life cycle and may also occur as a result of density-dependent growth in the recruited stage (not considered in the White Seabass model). In the model, all population components are impacted symmetrically (equally) by density-dependent processes. Once released, stocked hatchery fish and their offspring are subject to natural selection which can be expected to result in the fitness of hatchery-type fish increasing over generations in the wild and eventually approaching the fitness of wild fish. The model mimics this effect of natural selection by allowing hatchery-type fish to transition into the wild-type component at a rate equivalent to the heritability of fitness traits (Lorenzen 2005).

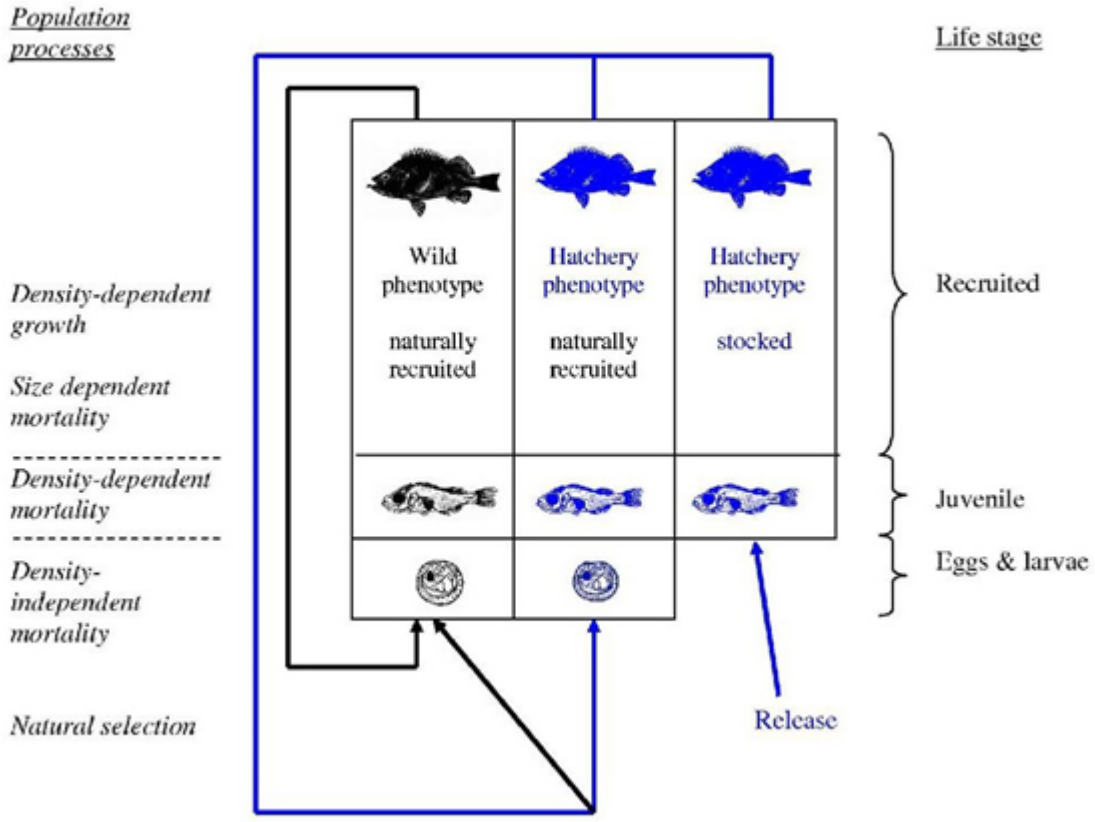

Fig. 4.10. Schematic of the population model for enhanced fisheries (Lorenzen 2005, Medley and Lorenzen 2006).

In line with the normal convention used in fisheries models, recruitment is defined as the number of late juveniles entering the fishable stock following a period of highly density-dependent (compensatory) mortality. Explicit consideration of the processes that impact mortality rates during the juvenile (pre-recruit) stages are critically important to outcomes of enhanced fisheries, since most fish are stocked at a life stage and size when survival is density dependent but also size dependent (Lorenzen 1996, Lorenzen 2000, Lorenzen 2005, Hazlerigg et al. 2012). The model accounts for both density and size dependence in survival of stocked fish by 'unpacking' early life, "pre-recruitment" mortality into multiple stages (Lorenzen 2005). This allows us to represent stocked fish experiencing some density dependence in survival during the pre-recruit period following release (such that the amount of density dependent survival depended on the size of stocking). Methods for accounting for specific components of juvenile mortality prior to recruitment to post-density dependent mortality, sub-adult stages are described in detail in Lorenzen (2005). The lifetime pattern of natural mortality is described by the size dependent mortality function (Lorenzen 1996, Lorenzen 2000) already introduced in the analysis of White Seabass tag recaptures above.

The White Seabass enhancement model used here is structurally identical to the Lorenzen (2005) model. It was set up in the EnhanceFish software package (Medley and Lorenzen 2006), but could also be run using equivalent R code or Excel VBA modules (Lorenzen 2005, Camp et al. 2014). The model was 'tuned' to resemble key attributes and dynamics of the White Seabass stock assessment model (Valero and Waterhouse 2016). Certain structural differences such as the use of a constant (as opposed to size-dependent) natural mortality rate and multiple fishing fleets with different selectivity patterns in the stock assessment made it impossible to emulate the stock assessment exactly in the enhancement model, but this does not matter for the exploratory analyses conducted here. The parameter values used in the stock enhancement model are shown in Table 4.3. The size-dependent natural mortality rate of wild fish was set to $M_{1,W}$=15 year^{-1}, the average for wild fish in general (Lorenzen 1996, Lorenzen 2006) and consistent with the constant M= 0.225 year^{-1} for recruited fish assumed in the stock assessment ($M_{1,W}$=15 year^{-1}, which implies M(70cm)= 0.214 year^{-1} at the length of maturity (70 cm).

The model was used here for an exploratory, equilibrium analysis of the impacts of hatchery releases on the fishery. Model predictions were generated of the impact of stocking on equilibrium yield from different population components while fishing at the fishing mortality rate at which MSY is achieved from the wild stock (F_{MSY}) (Fig. 4.11). Predictions were generated for two alternative assumptions about the biological basis of reduced fitness (increased natural mortality) of hatchery-reared vs. wild fish: heritable genetic effects or phenotypic plasticity that affects directly stocked fish but is not passed on to offspring. Clearly, effective average (combined short and long-term mortality rates) are too high for stocking to have any noticeable impact on yields, regardless of the biological basis of the high mortality rates. Even if short-term post-release mortality rates could be reduced to the extent that survival is well described by just the long-term component of mortality, stocked contributions to yield would be very small. Only if short and long-term mortality components were reduced to 'near-wild' levels would stocking result in substantial net increases in total yield.

Table 4.3. Parameter values used in the White Seabass fisheries enhancement model.

Parameter	Baseline value (range)	Definition
Growth		
$L_{\infty L}$	140 cm	Asymptotic length at B-> 0
K	0.16 $year^{-1}$	Growth rate
g	0.0 cm kg^{-1}	Competition coefficient
α	$4.75x10^{-6}$ kg cm^{-3}	Coefficient of length-weight relationship
β	3	Exponent of length-weight relationship
Natural mortality		
$M_{1,W}$	15 $year^{-1}$	Mortality of wild genotype at L=1 cm
$M_{1,H}$	15 (15-120) $year^{-1}$	Mortality of hatchery genotype at L=1 cm
Reproduction		
L_m	70 cm	Length at maturity
p	-1 cm^{-1}	Steepness of maturity function
r	1, 0	Relative reproductive performance of stocked fish
Recruitment		
a	1.913 kg^{-1}	Initial slope of stock-recruitment relationship
b	351,847 $year^{-1}$	Maximum recruitment
Lr	28 cm	Length at recruitment
Fishing		
F_∞	0.2 $year^{-1}$	Fishing mortality asymptote
L_c	70 cm	Gear selection length
q	-0.2 cm^{-1}	Steepness of selectivity curve
Stocking		
L_s	22 cm	Length at release
N_s	135,000 $year^{-1}$	Numbers stocked
Evolution		
h^2	0.2	Heritability of life history traits

Note that, if fitness loss was heritable, reproduction of stocked fish in the wild would give rise to a naturally recruited 'hatchery type' population component which would partially displace the truly wild population component. The displacement is a result of two factors: the contribution of stocked hatchery-type fish to the overall spawning stock, and the fact that pre-recruit density-dependence acts on the combined abundance of offspring (and is assumed here to affect both components equally). Since the naturally recruited hatchery type component would still suffer elevated natural mortality, the combined naturally recruited components

(wild and hatchery type fish) suffer a slight overall reduction at intermediate levels of fitness loss. This effect is, however, predicted to be fairly moderate. (Note that 'very unfit' stocked hatchery fish pose little threat to the wild stock because they simply do not survive well enough to reproduce).

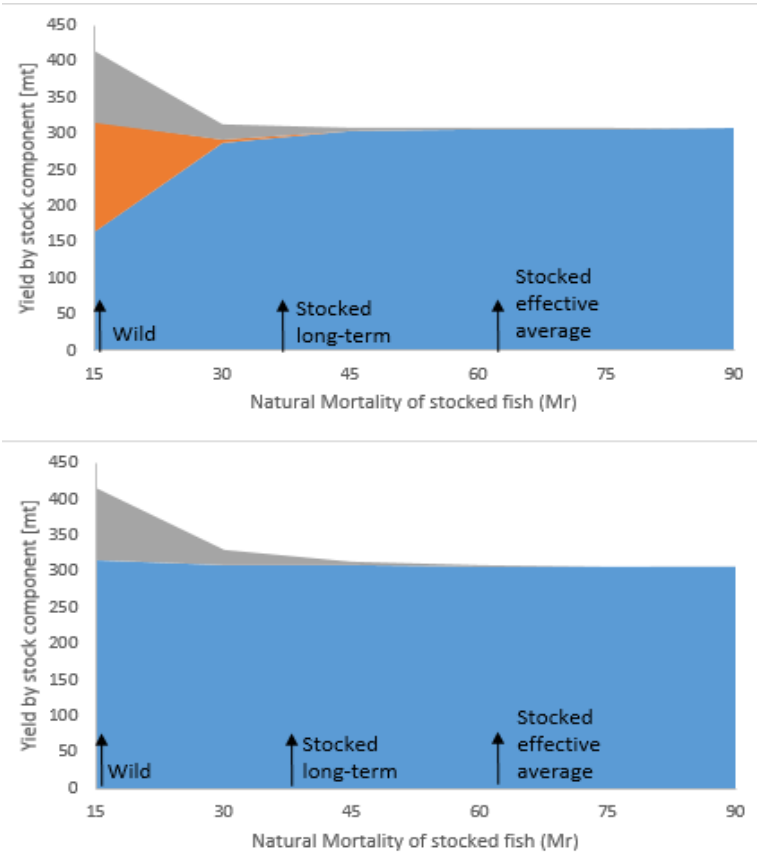

Fig. 4.11. Predicted impact of stocking on equilibrium yield from different population components while fishing at F_{MSY}, assuming that fitness (natural mortality) differences between stocked and wild fish are heritable (top) or based exclusively on phenotypic plasticity (bottom). The population components are wild (blue), directly stocked (grey), and naturally recruited hatchery-type fish (orange).

Quantitative predictions of the proportional contribution of stocked fish to the total catch for different levels of natural mortality are shown in Fig. 4.12, along with the observed average for the White Seabass stocking program. Model predictions for the effective average mortality are very similar to observed average values. For releases occurring between 2000-2011, stocked fish contributed on average 0.26% to California White Seabass catches (Fig. 4.12). The model results reveal the strong effect that high post-release mortality has on the contribution of hatchery fish to the stock. If released hatchery fish mortality rates can be lowered to similar levels as for wild White Seabass, stocking of 135,000 juveniles per year (the approximate average release rate of recent years) would increase the hatchery fish contribution to catches to 18%. The prospect of this level of contribution to wild stocks appears promising, but also

suggests a potential for substantial ecological and genetic interactions between hatchery reared and wild White Seabass (See Objectives 2 and 3).

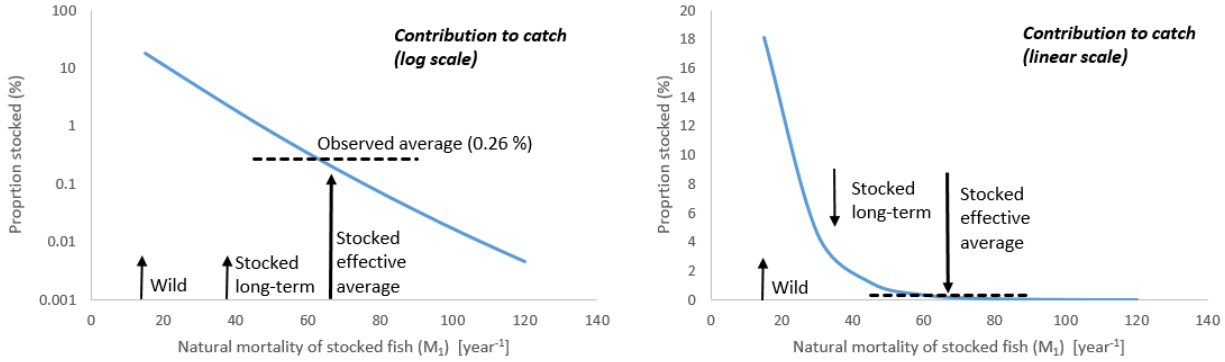

Fig. 4.12. Proportional contribution of stocked fish to the total catch White Seabass fisheries model for different levels of natural mortality and observed averages for the White Seabass stocking program.

The recapture ratio is a quantitative prediction of the proportion of stocked fish recaptured for different levels of natural mortality are shown in Fig. 4.13, along with observed averages for the White Seabass stocking program. Model predictions for the effective average mortality are very similar to observed average values. For releases occurring between 2000-2011, the proportion of stocked fish recaptured in the commercial and recreational fisheries was 0.036%.

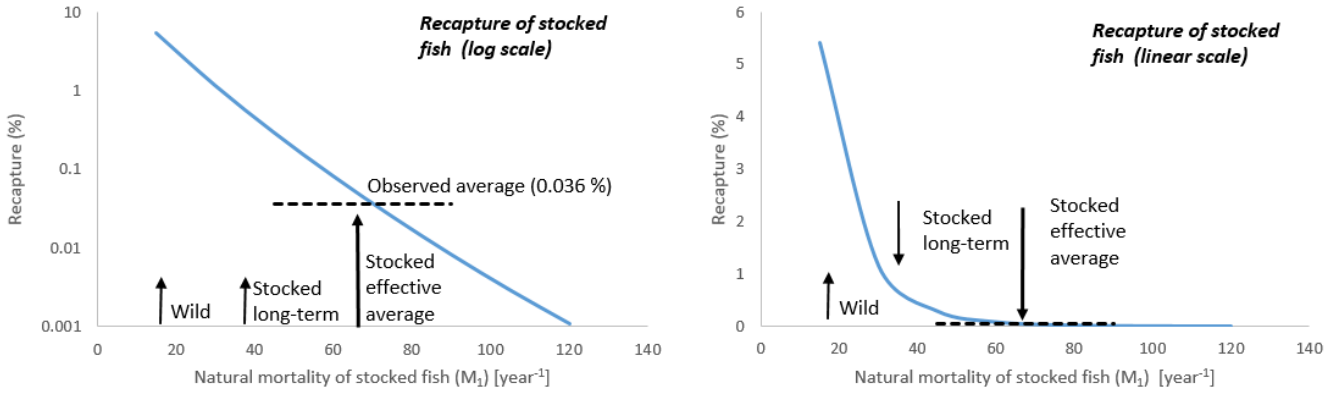

Fig. 4.13. Recapture ratio (proportion of stocked fish recaptured) as predicted by the White Seabass fisheries model for different levels of natural mortality and observed averages for the White Seabass stocking program.

Are released fish adding to the stock or replacing wild fish? An important question in any stock enhancement program is to what extent stocked fish add to the total stock or replace wild fish due to biological interactions such as competition or predation? Population dynamics theory and empirical evidence hold that in fish populations, density-dependent mortality is strongest within the early juvenile (pre-recruit) stage of the life cycle (Lorenzen 2005). Stocking of fish in stages where density-dependence is occurring should elicit compensatory mortality that may

affect wild as well as stocked fish and thereby result in a replacement (or displacement) or wild by hatchery fish.

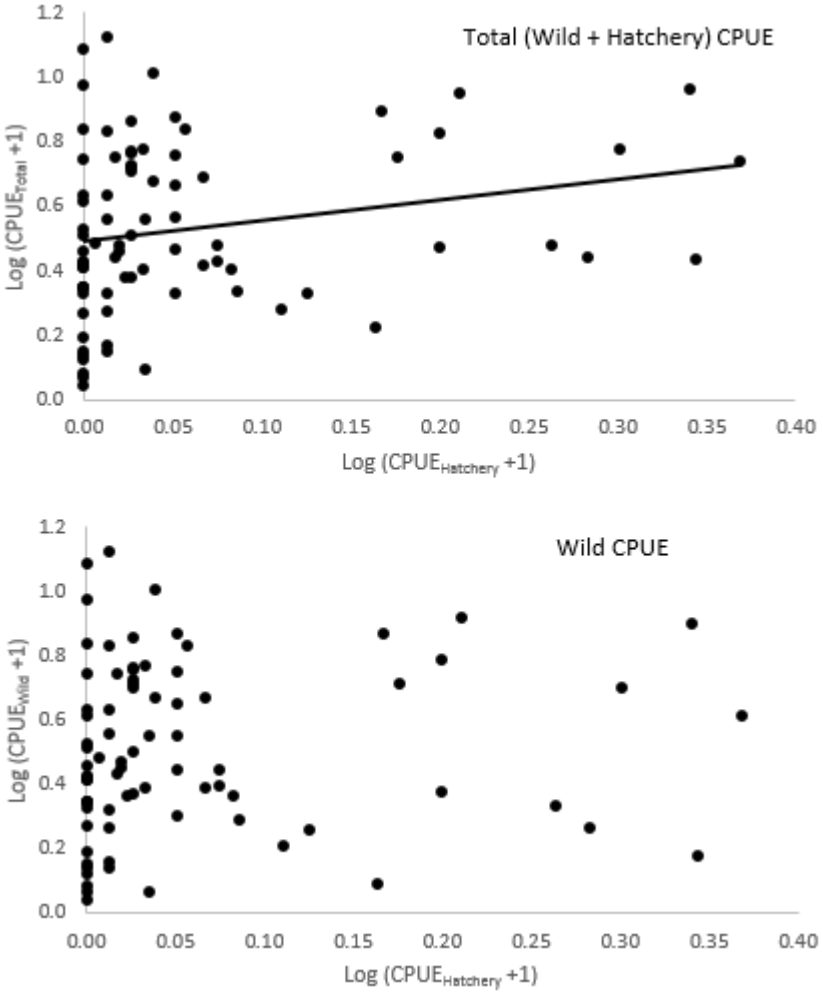

Fig. 4.14. Relationships between total (top panel) and wild (bottom panel) juvenile White Seabass catch per unit of effort (CPUE) and hatchery juvenile CPUE in the same sampling location. Data from the gill net sampling program 1999-2004. Data points represent aggregated annual CPUE for each location and year.

White seabass are stocked as advanced juveniles, at a stage that can be expected to sidestep the early juvenile stage characterized by strong density-dependence in mortality rates. Nonetheless, the high level of releases relative to natural recruitment implies that even a moderate level of density dependence at the advanced juvenile stage could adversely affect wild recruits. Therefore, juvenile gill net sampling data for 1999-2004 were analyzed to test whether a high abundance of hatchery fish in the samples was associated with reduced abundance of wild fish. Research gill net catch per unit of effort (CPUE) was used as a measure of relative abundance, comparable across sites and over time. As shown in Fig. 4.14, total (wild and hatchery White Seabass) juvenile abundance was positively related to hatchery juvenile abundance. Abundance of wild juveniles was unrelated to the abundance of hatchery juveniles at the same location and time. It may therefore be concluded that the stocking of hatchery

juveniles had an additive effect on total juvenile abundance and did not negative affect the abundance of wild fish.

4.6.1.2. Exploratory bio-economic analysis.

While it was not possible as part of this review to conduct a full bio-economic analysis of the enhancement program (See Section 6.4.3 Recommendation 1), it is useful to at least explore economic implications of the recapture ratios achieved. The estimated recapture ratios for different levels of post-release mortality of stocked White Seabass as shown in Fig. 4.13 provide the biological/technical information needed for this analysis. Ballpark estimates for the costs of hatchery-reared juveniles and the value of fish recaptured in the commercial and recreational fisheries were constructed as follows. The dockside (ex-vessel) value of commercially caught White Seabass was estimated at $3.60 per pound (CDFW 2013b). The marginal value of a fish caught in the recreational fishery has not been estimated for White Seabass, but may be substantially higher than that of a fish caught in the commercial fishery. In the California halibut fishery, the value of a recreationally caught fish has been estimate to be around 10 times that of a commercially caught fish. Using the same ratio for recreational vs. commercial value (10x) and assuming a 20-pound fish, the value of an additional White Seabass would be $72 on the commercial, and $720 in the recreational fishery. A rough estimate of the cost of hatchery-reared juvenile White Seabass was derived from dividing the rounded average operating costs ($1.6 million per year) by a rounded average the number of fish released (136,000 per year), which yields a cost of $11.80 per fish. The precise costs per fish are difficult to ascertain (for example, the operating costs include monitoring and research but exclude voluntary contributions by anglers during the pen-rearing stage) and therefore the analysis was conducted using a range of values from $5 to $15 per released juvenile fish (see Section 5.1.1.3 for more accurate and comparative cost estimates of different sized hatchery White Seabass.)

The resulting costs and value per recaptured fish are shown in Fig. 4.15. At the effective average post-release mortality (for 1999-2004), the costs of producing and releasing hatchery-reared juveniles greatly exceed the value of the fish recaptured for all assumptions about per-unit costs and values. Costs exceed the value generated for all assumptions regarding per-unit cost and value and for all release mortality levels achieved in the past (even the best-case scenarios between the long-term mortality component and the effective average). Only if post-release mortality was reduced to below the long-term mortality component would costs drop below the value generated.

As illustrated in Fig. 4.15, the economic performance of the White Seabass enhancement could be improved by reducing mortality, by reducing hatchery production costs, or by increasing the value of harvested fish. However, it is clear from Fig. 4.15 that any realistic reduction in production costs would be insufficient to close the gap between costs and value generated over the range of post-release mortality rates achieved in the past. *Reducing post-release mortality must therefore be a key goal, even though this may necessitate changes in husbandry and release techniques that could increase production costs (e.g., reduced densities in culture, habitat enrichment in culture facilities, conditioning on live feeds, etc.).*

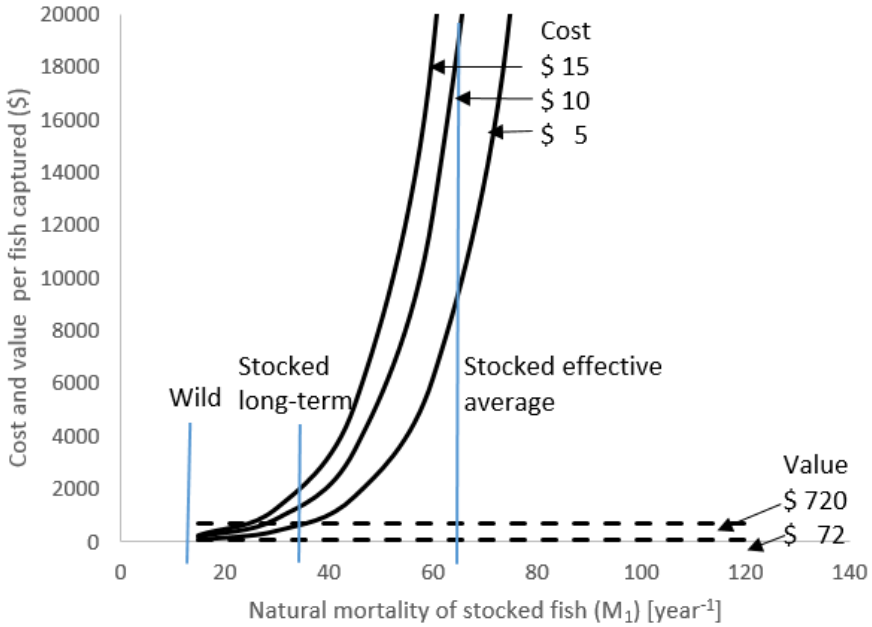

Fig. 4.15. Approximate cost and value of a hatchery-reared White Seabass recaptured in the commercial and recreational fisheries, in relation to post-release mortality. Values of $72 and $720 per recaptured fish are rough estimates of the value of a 20-pound fish in the commercial and recreational fisheries, respectively. Cost estimates are given for a range of costs per released juvenile of $5 to $15. The Overall cost of a recaptured fish is calculated as the cost per released fish divided by the recapture ratio. See text for details and note that the value of cost estimates are very approximate 'ballpark' figures.

4.6.1.3. Synthesis of current contribution to the fishery and options for improvement.
The current contribution of the hatchery program to the White Seabass fishery can be summarized as follows:

1. The proportional contribution of hatchery fish to recreational and commercial fisheries catches is very low at about 0.26%.
2. The contribution of hatchery fish appears to be additive, i.e., the hatchery fish add to the overall stock rather than replacing wild fish through ecological interactions such as competition or predation.
3. The low, additive contribution of hatchery fish to the stock is achieved at relatively high levels of input (stocking of about 135,000 juveniles per year, long-term average). This implies a very low technical efficiency: only 0.036% of stocked juveniles are recaptured upon entering the fishery, which implies that about 2800 juveniles must be stocked to add one fish to the catch.
4. Although no direct estimates are available of stocking costs or economic benefits generated by hatchery contributions to the fishery, exploratory analyses using reasonable 'guesstimates' indicate that costs likely exceed benefits by a substantial margin.
5. The low contribution, technical and economic efficiency of the program is due to primarily the high mortality rates suffered by hatchery fish after their release into the wild.

It is clear from the above considerations that multiple indicators must be considered when judging the performance of the fisheries enhancement program. Likewise, management goals and targets should be formulated involving multiple criteria including measures of overall contribution to the fishery, effects on the wild stock, and efficiency (output relative to input). The proportional contribution of hatchery fish to the catch appears to be the most commonly used indicator of OREHP performance and increasing this contribution a frequently stated goal. Proportional contribution is easily measured (as long as hatchery fish can be identified) and indeed provides some indication of performance but it is insufficient and can be misleading. Proportional contribution of hatchery fish is influenced by variation in the recruitment of wild fish (e.g., the proportional hatchery contribution is reduced when natural recruitment increases), it does not indicate whether hatchery fish have added to the overall stock or replaced wild fish through ecological interactions, and it does not directly relate to technical or economic efficiency (there is no consideration of the inputs required to achieve this contribution). The recapture ratio (proportion of stocked fish that are recaptured) is a more informative performance measure that is not conflated with variation in wild fish abundance and provides a measure of efficiency (fish recaptured as a proportion of fish stocked). The recapture ratio still does not specify to what extent stocked fish have added to the overall population or replace wild fish. To establish additivity, it is necessary to compare abundance of wild fish or total (wild and hatchery fish) abundance under different levels of stocking. As shown in Fig. 4.13, this can be done using consistent estimates of relative abundance (e.g., CPUE). Identification of informative and measurable performance indicators and targets should be pursued as part of the review of management procedures (Chapter 5).

With respect to future management of the program, it is clear that reducing post-release mortality of hatchery fish is essential to improving overall performance. Although proportional contribution to the fishery could be increased by simply stocking more fish, this would not address the low technical and economic efficiency of the program (the low returns per fish stocked, and fact that costs are likely to exceed benefits).

Unfortunately, it is currently unknown whether or how substantial reductions in post-release mortality of White Seabass could be realized. A focused research program on this issue would be indicated but the feasibility of such a program would hinge on establishing more efficient experimental approaches to testing post-release survival of stocked fish than the current procedures which suffer from insufficient sampling effectiveness (only a very small proportion of stocked fish can be sampled).

4.6.2. Data and Information Gaps.

1. Contributions of the enhancement program to the White Seabass stock and fishery have so far been very limited due to the high mortality of released hatchery fish. This also limits the need for more in-depth modeling and assessment and the opportunities for resolving remaining uncertainties. However, if steps are taken to reduce post-release

mortality (See Chapter 1), the current magnitude of releases could have very substantial population and fisheries contributions and call for more advanced modeling.
2. Key uncertainties that need to be resolved once the enhancement contribution to the stock and fishery increases include assessment of environmental impacts of releases (e.g., potential for disease amplification (See Sections 1.7, 1.8, Chapter 2)), environmental forcing on mortality and growth of wild and stocked White Seabass, contribution of natural selection vs. phenotypic plasticity to the low apparent fitness of released hatchery fish, and fitness of naturally recruited offspring of released hatchery fish (See Chapter 3).
3. Population and fisheries management objectives need to be better defined in order to allow evaluation of the potential for the hatchery program to support enhancement or restoration objectives.

4.6.3. Recommendations.

1. Population dynamics modeling is a key tool for exploring the potential for releases of hatchery fish to enhance or rebuild the White Seabass fishery or other candidate stocks for enhancement (see Garlock et al. 2017). Population dynamics modeling should be used routinely to inform strategic planning and management decision-making in the future development of the OREHP.
2. Management objectives for the fishery and the enhancement program should be defined more specifically and quantitatively with the help of population dynamics and fisheries system modeling (Lorenzen 2005, Camp et al. 2014, Camp et al. 2017. The 'Updated Responsible Approach' to stock enhancement provides broad guidance in this respect (Lorenzen et al. 2010).

Chapter 5

Objective 5. Continue to develop, evaluate, and refine hatchery operations to maximize the potential for achieving the goal of the program.

5.1. Budget considerations.

5.1.1. Key Findings.

5.1.1.1. Putting the OREHP in context.

The White Seabass portion of the OREHP has made much progress in a relatively short amount of time (~30 yrs), as compared to other enhancement programs, such as those for Pacific salmon (*Oncorhynchus spp.*) on the west coast of North America. Various hatcheries and enhancement programs for Pacific salmon were started as early as the 1870s (e.g., the Baird Hatchery on the McCloud River in Northern California) and these have continued, in some form, to the present day. Despite the decades of money (and effort) spent on the research, development, and implementation of Pacific salmon enhancement programs (e.g., on the Columbia and Sacramento River systems), the enhancement efforts are still not entirely effective, with lingering questions, including ones about the genetics of the stocks being used, the distribution and behavior of the fish in the ocean, and how to improve hatchery techniques to better prepare the fish for release into the wild. The programs in the Columbia River, for example, undergo periodic reviews that then recommend changes to the enhancement program, including changes in broodstock selection, hatchery production levels, and research and monitoring efforts to better understand the interactions between hatchery-produced fish and wild stocks. Interestingly, these are some of the same challenges being addressed within the OREHP, except the resources available to the OREHP are roughly 8-14 times lower than those available to the hatcheries supported by the Mitchell Act (16 USC § 755-757) ($12 - $22 million per year versus ~$1.6 million per year for the OREHP, see next paragraph).

5.1.1.2. OREHP funding sources and adequacy.

The OREHP is supported by revenues from the sale of fisheries enhancement stamps on commercial and recreational licenses south of Point Arguello and by matching federal Sport Fish Restoration Act (SFRA) Funds (16 USC § 777 et seq. (Dingell-Johnson Act)) and, from 2002-2005, by some funds from the San Onofre Nuclear Generation Station (SONGS) mitigation and the British Petroleum (BP) oil spill settlement (OREHP Budgets Summary 2002-2015). These funds total approximately $1.65 million annually (OREHP Budgets Summary 2002-2015) and have trended at that same funding level since 2002 while basic rates of economic inflation have increased during this time period. The OREHP FY 2016-17 base budget is $1,443,234 (OREHP Budget 16-17), which reflects a roughly $100,000 reduction in funding each year over the previous two years. In short, the OREHP's hatchery budget is currently underfunded, as the base operating budget needed to achieve all aspects of the OREHP objectives exceeds the available program funds. Over the years, HSWRI has obtained several federal grants from the National Marine Fisheries Service to help support shortfalls, but these funds are not secure over time and the amount available fluctuates dramatically from year to year. HSWRI has contributed in excess of $400,000 annually to meet operational expenses as well as sought

grants and contributions to make infrastructure repairs and improvements to the hatchery facility. The sources of external funds vary and include a mix of private and government sources.

5.1.1.3. OREHP budget proportions.

From FY 2002-03 to FY 2015-16, an average of 89.9% of the money allocated for OREHP contracts was apportioned to HSWRI's contract (covering hatchery operations and adult fish sampling), while an average of 7.7% of the budget was allocated to gill net survey contracts, an average of 1.5% was allocated for pathology contracts, and an average of 0.1% was allocated for miscellaneous contracts (OREHP Budgets Summary 2002-2015; see Fig. 5.1). In addition, an average of 6.6% of the budget was set aside for the Evaluation of the OREHP during the last two fiscal years of this period (see Fig. 5.1).

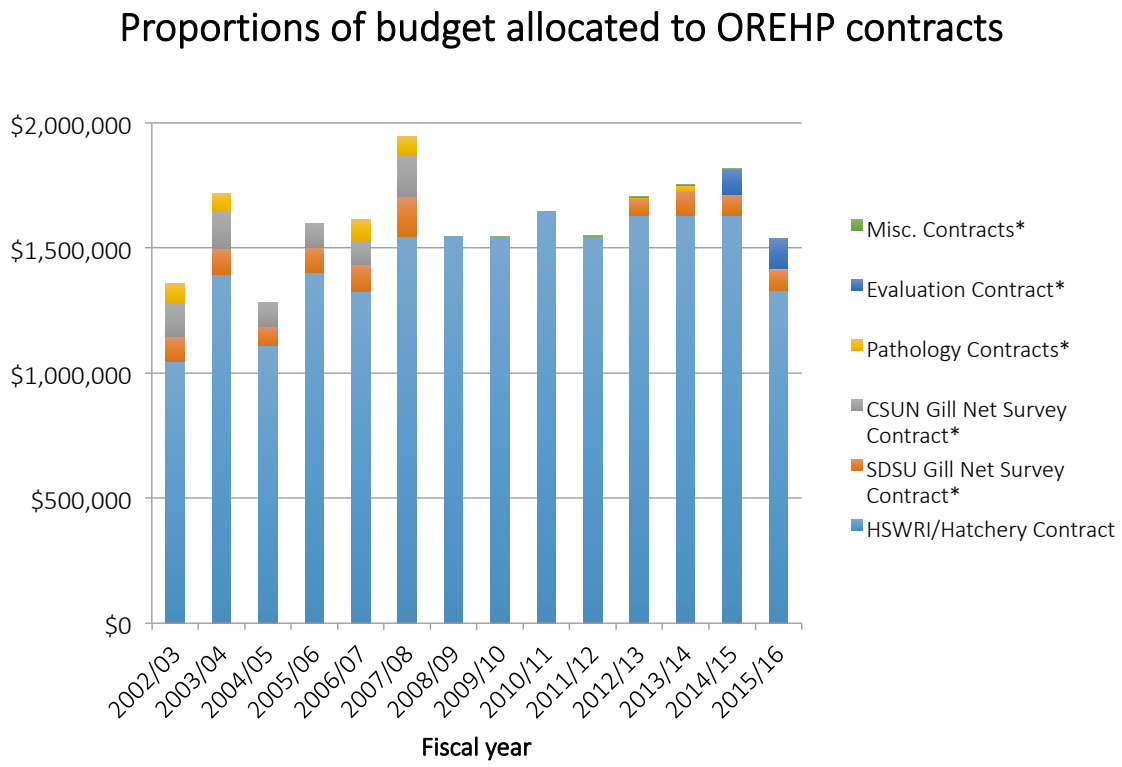

Fig. 5.1. Proportions of the budget allocated to OREHP contracts from FY 2002-03 to FY 2015-16. Asterisks mark those contracts that were funded by the OREHP Enhancement Stamp Account only. The figure does not reflect actual expenditures, but rather money apportioned for each contract.

The average (±1SE) hatchery operations budget awarded to HSWRI for the last four fiscal years (2012-2013 to 2015-2016) was $1,554,522.50 ± $75,002.83 (a range of $1,329,514 to $1,629,531). During these fiscal years, HSWRI released an average (±1SE) of 106,487 ± 23,905 8-inch/20-cm fish per fiscal year (a range of 55,902 to 169,440 fish). This is a biomass equivalent of 432,046 ± 96,989 3.5-cm fish (the size of "fingerlings" released in Texas' Red Drum enhancement program, see next paragraph); or 8,352,308 ± 1,874,988 Phase 1 (1g) fish per

fiscal year (based on a total length vs. weight correlation of 2016 HSWRI White Seabass data, M. Drawbridge email to T. S. Talley, 31 January 2017a).

HSWRI's average cost (±1SE) of producing and releasing an 8-in/20-cm White Seabass over the past four fiscal years was $14.60 ± $3.14 per fish (a range of $9.17 to $27.81 per fish per fiscal year calculated by dividing the total OREHP hatchery budget by number of hatchery fish released that year). This cost is higher than estimates available from other programs. A 28-cm California hatchery raised trout, by rough comparison is estimated to cost $2.40 (M. Clifford email to V. Taylor, 1 May 2017). Other cost per White Seabass individual equivalents include an average cost of $3.60 ± $0.77 per 3.5-cm fish, as compared to $0.134 for a Texas hatchery raised Red Drum fingerling; and $0.186 ± $0.040 per Phase 1 (1g) fish based on biomass equivalents.

A White Seabass of minimum legal size is worth $36 if commercially landed (7.5 lbs average weight of minimum legal size X $4.80 per pound ex-vessel value averaged between 2011-12 to 2015-16; CDFW 2013a, CDFW 2016). This value would require that 43,181 hatchery fish survive in the wild to the state legal lower size limit of 711 mm TL (28 inches) in order to reach a 1:1 break-even point of OREHP annual funding (calculated as OREHP average annual budget/cost per legal fish). However, no recreational restitution value was available for legal-sized White Seabass as was used to calculate the Red Drum break-even point presented in the next paragraph. In the California Halibut fishery, a recreationally caught fish is estimated to be 10x the value of a commercially caught fish (Section 4.6.1.2). Using the same ratio for recreational vs. commercial value (10x), a minimally legal-sized White Seabass would be worth $360. This value would require that 4,318 hatchery fish survive to minimum legal size to reach the break-even point at the recreational value level. The number of fish between 4,318 and 43,181 needed to reach minimum legal size would, therefore, depend upon the proportion of fish caught recreationally compared with commercially. Between FY2011-12 to 2015-16, an average of 37% of White Seabass were recreationally landed while 63% were commercially landed, making *the break-even point roughly 28,801 hatchery released fish needing to reach minimum legal size of 28 inches*.

By comparison, Texas Parks & Wildlife Department's base budget cost of operating three marine fish hatcheries for purposes of stock enhancement (FY 2013-14 and FY 2014-15) averaged $1,144,204 per year. Staff salaries for 31 employees to operate the three hatcheries averaged $1,535,880 per year. The base budget plus salaries totaled $2,680,084 per year. During FY 2013-14 and FY 2014-15, an average of 20 million (Table 5.1) hatchery-reared Red Drum fingerlings (3 - 4 cm TL) were released into the wild. Each Red Drum captured (entering the recreational fishery at 20 inches) is worth $256 (TPWD Restitution Value FY 2016-17; FY 2016-17 Restitution Value was used because FY 2014-15 Restitution Value could not be obtained. The two values should be very close). A general estimate [(Base Budget + Staff Salaries)/Fish Value] of the Break-Even 1:1 Benefits would require that approximately 12,000 hatchery-reared Red Drum survive in the wild to the state recreational regulation lower-limit size of 20 inches. The general cost estimate of each Red Drum hatchery-reared fingerling [$2,680,084/20,000,000 fingerlings] is $0.134 per fingerling. The general cost of production

when considering all three species of hatchery-reared fishes [$2,680,084/30,000,000 fingerlings] is $0.09/fingerling.

The labor cost of the hatchery contract portion of the OREHP, including indirect (15%) costs and fringe cost, is about 50% of the total hatchery budget (OREHP Expenses Summary 2009-2015). This cost is the largest, but its magnitude often depends upon the details and scope of the enhancement hatchery operations, which in this case includes coordination and support for the volunteer-run growout pens and the HSWRI-run growout facility. Hatcheries with more research components would be expected to have higher labor percentages and very routine hatcheries working with very well known species can operate at lower staffing levels. There is also a big difference in hatcheries producing large numbers of small fish or hatcheries producing large tonnages of large fish. The production of large numbers of small fish requires more labor and much less feed on a relative basis.

Table 5.1. Texas Parks & Wildlife Department marine fish hatcheries fingerling stocking totals for FY2014 & FY2015. Hatchery facilities are CCA Marine Development Center (MDC), Sea Center Texas (SCT), and the Perry R. Bass Marine Fisheries Research Station (PRB). Fish species cultured and stocked for purposes of coastal stock enhancement are: Red Drum, Spotted Seatrout, and Southern Flounder.

	FY15 Fingerling Stocking Totals (9/1/14-8/31/15)		
	Red Drum	Southern Flounder	Spotted Seatrout
MDC	3,595,400	114,031	2,478,303
SCT	4,783,598	298	7,837,743
PRB	7,502,506	0	3,873,300
Total	15,881,504	114,329	14,189,346
	FY14 Fingerling Stocking Totals (9/1/13-8/31/14)		
	Red Drum	Southern Flounder	Spotted Seatrout
MDC	3,687,154	46,011	824,177
SCT	11,832,861	5,015	10,662,683
PRB	8,945,891	0	167,065
Total	24,465,906	51,026	11,653,925

For purposes of general comparison of non-labor costs, two large-scale state operated marine fish hatcheries in Texas dedicated to enhancing gamefish populations have base operating budgets that include the following categories (budget %'s, excluding labor): Electricity/utilities = 25%, Maintenance = 20%, Supplies = 22%, Non-capitalized equipment = 16%, Fuels and lubricants = 4%, and Miscellaneous = 13%. The major Texas operating expenditures over the years have been Electricity/utilities and Supplies. In past years, Electricity expenditures consumed up to 40-50% of the hatcheries' operating budgets. Electricity costs have been reduced through statewide agency contracting/agreements instead of individual facility electricity contracts. In addition, solar panel systems are being installed (installation and equipment costs covered by Green Mountain Energy Company grant) at one of the hatcheries to evaluate whether further reductions in electricity expenditures can be achieved.

Commercial private hatcheries using recycle technology producing large numbers (about 20 million/yr) of small fish have similar cost distributions, with labor being the largest percentage, closely followed by electricity, then feed, and depreciation of capital cost. In this case, water/sewer costs was the lowest line item cost.

A summary review of the OREHP hatchery base operating budget (FY's 12-15) shows similar expenditure patterns as compared to the Texas hatcheries (budget %'s, excluding labor): Electricity/utilities = 36%, Maintenance = 7%, Supplies = 48%, Non-capitalized equipment = 5%, and Miscellaneous = 4% (OREHP Expenses Summary 2009-2015). Major operating expenditures include Supplies and Electricity/utilities categories. Breaking down these categories, growout fish feeds and facility electrical costs are the major operating budget expenditures as should be expected for a large-scale marine stock enhancement program.

The budget information provided does not have any category for depreciation, but a depreciation value of $83,015 for FY2016 (M. Drawbridge pers. comm.), which is about 5% of the total budget and is included in the General and Administrative costs. (This is a reasonable value; if one figures an initial capital cost in the $3.5 million category and a 15-year linear depreciation [special purposes agricultural structure – IRS], the result would be $230,000 additional cost or about 14% of the total budget.)

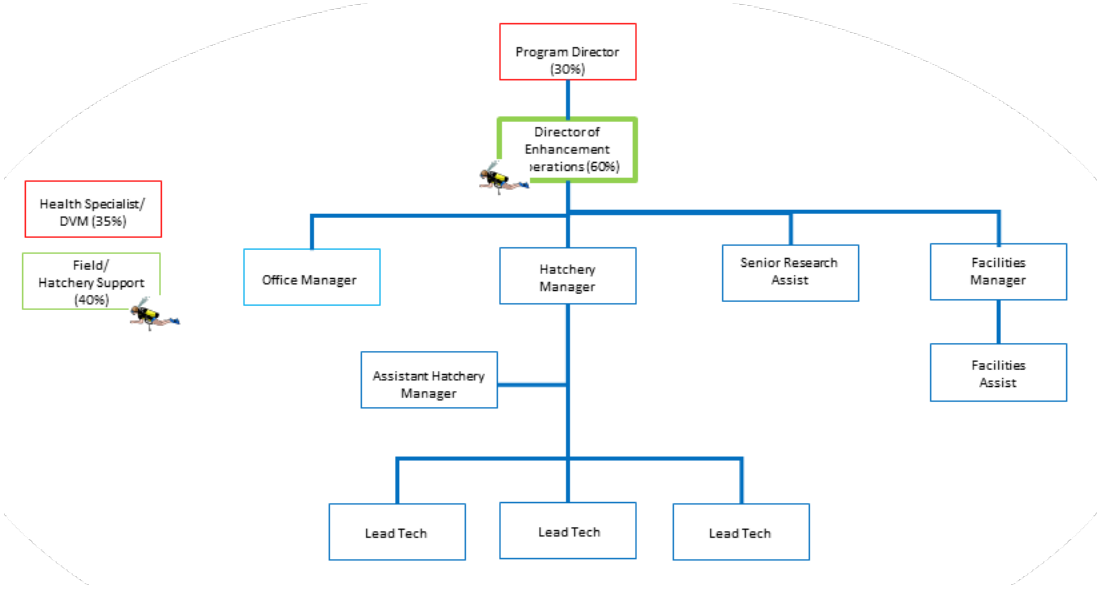

Fig. 5.2. Organizational chart for HSWRI's OREHP operations as of March 2017. Percentages within boxes denote the amount of those employees' time that is paid for by the OREHP; boxes without percentages represent 100% time paid for by the OREHP. Chart provided by M. Drawbridge of HSWRI.

When funding is reduced, staffing levels may not be sufficient to perform some enhancement operations. In recent years, cuts in funding have impacted several OREHP areas, in particular, loss of applied research, such as post-release survival assessments, disease research (e.g., resolution of GSS issues), and fisheries modeling. The HSWRI (OREHP and non-OREHP supported) staffing level as of 2015 was about 22 but recent budget cuts have reduced staffing levels. The current level of HSWRI staff paid by OREHP funds (Fig. 5.2) is 9 full-time staff

members, and 4 staff members who are partially paid with OREHP funds. Assistance is also received by staff who are not supported by OREHP funds (Fig. 5.2). This previous and especially current staff size, even after funding supplementation, may not be sufficient to perform all current and expected duties, including conducting hatchery operations (from collection of broodstock through all hatchery stages), overseeing growout facilities and completing surveys for tagged fish, coordinating and engaging in outreach and education efforts, regularly updating protocols, developing and ensuring adherence to adaptive management plans and ensuring compliance to all regulations, and performing research.

In addition, hatchery and research programs may be impacted by insufficient operating funds. In recent years, cuts in funding have impacted several OREHP areas, in particular, loss of applied research, such as post-release survival assessments, disease research (e.g., resolution of GSS issues), and fisheries modeling. Oftentimes, government sponsored fish hatcheries that experience budget constraints respond by reducing outreach, research, fish production (broodfish maintenance), and infrastructure and maintenance components of operations. Besides operational costs, funds are inevitably also expended on mandatory obligations (e.g., to satisfy the facility's compliance with environmental and aquaculture regulatory permit requirements) and used to cover an IDC rate shortfall (OREHP funds an indirect cost rate of 15%, but HSWRI's IDC rate is 44% so that $400,000 annually is fundraised for project related overhead). Furthermore, given the fact that the OREHP annual operating budget has remained static for more than a decade, and the annual rate of inflation and operating expenses have continued to increase, the program is persistently underfunded.

Besides externally funded research, not included in the official budget figures are the in-kind contributions of the volunteer run growout facilities. Between June 16, 2015 and June 15, 2016, contributions to the OREHP from the eight volunteer-owned and -operated pens had an estimated total value of $233,118 (Marshall and Shedd 2016). These contributions came from 4,830 hours per year of volunteer time spent on all aspects of operating and maintaining the facility, caring for the fish, recruiting and managing other volunteers, fundraising, and assisting HSWRI with other aspects of the program. Volunteer time was estimated to be worth $220,890 ($72,450 of pen manager time, $120,720 of regular volunteer time, and $27,720 of occasional volunteer time); and pen related spending and costs covered by pen operators (e.g., air pumps, compressors, pen repair supplies) totaled $12,228 (Marshall and Shedd 2016).

5.1.2. Data and Information Gaps.

1. Lacking is a detailed analysis of the budget, which would require a far finer resolution of budget categories and information on how funds are allocated within each category, especially more detail on maintenance and depreciation issues given that this is a saltwater hatchery in a steel building. A finer grained, internal cost analysis is of primary value to the managers and their decision-making.
2. Lacking of data on the actual costs and benefits of ancillary research to the OREHP.

3. An annual accounting of volunteer contributions and time would benefit OREHP public outreach efforts, and be an incentive/motivation to encourage volunteer recruitment.
4. Estimates of costs of the recommendations listed throughout this evaluation report.
5. An explicit list of high priority goals and operations, and what would be added/enhanced with additional funds, including genetics analyses, post release survey protocols, development and modification plan for SOPs, etc.

5.1.3. Recommendations.

1. Investigate whether the hatchery can obtain a state agency or HSWRI bulk group rate for the facility's electricity as dictated by the facility's property lease agreement.
2. As the facility is almost two decades old and electricity costs have changed significantly during that time period, a detailed energy audit is recommended to improve energy efficiency. Specific suggestions include adding insulation materials to the hatchery broodfish spawning tanks, adding an enthalpy wheel heat recovery system combined with upgrading the CO_2 stripping system, and evaluating whether more efficient air blowers (high speed centrifugal blowers) and/or other hatchery seawater pumping systems are due for upgrades.
3. Assemble CDFW, the OREAP, HSWRI and/or independent experts to review and prioritize the execution of recommendations listed throughout this review document based on agreed upon program goals, priorities and budget constraints. The program should fit within available funding levels, make operational adjustments and prioritize research. If feasible, consider expanding resources if funds have not kept up with inflation and/or requirements of research, especially as efforts get closer to influencing wild populations (then research is more critical).
4. Compile a list of and explore more sources of external funding, including non-profits and other private foundations (e.g., Coastal Conservation Association, a national non-profit organization comprised of recreational anglers, that has funded buildings and equipment for the enhancement program in Texas), including seeking donor funding to establish a fund that could provide interest that could help fund program.

5.2. Decision-making protocol: The process by which operational, budgetary, emergency, and research decisions are made and followed by HSWRI for the OREHP.

5.2.1. Key Findings.

Decision-making processes are key both to making a complex system function at high performance levels and to increasing understanding of hatchery and growout operations. HSWRI staff have done an impressive job over the years at developing Best Management Practices (BMPs) including the Comprehensive Hatchery Plan (2007), Broodstock Management Plan (2011), Growout Procedures Manual (2007), Fish Health Management Plan (2016), Benthic

Monitoring Plan (2005), and associated SOPs. The challenge for HSWRI is to maintain and regularly update the associated Best Management Practices documents, and to ensure that staff who are performing day to day duties are following protocols. The long-term decision-making at HSWRI is centralized at the upper management level, and often complicated because of all of the entities involved with the OREHP (see Section 5.3). The short-term operational and emergency decision processes are generally practical and functional as defined. Details about short- and long-term decision-making processes for operational, budgetary, emergency, and research decisions are discussed here. Many of the following details on decision-making processes were summarized from supplementary information provided by M. Drawbridge of HSWRI (M. Drawbridge email with attachment to T. S. Talley, 17 February 2017).

5.2.1.1. Operational decisions.

General management structure. HSWRI generally follows a top-down decision-making structure, with management staff making program-wide decisions according to their expertise and areas of focus. While it generally makes sense for management level staff to make OREHP decisions, this top-down approach may at times hinder quick decision-making. Daily oral and written (email) communication between technical staff and managers that occurs at HSWRI, however, may help reduce this risk, inform managers decisions and ensure that SOPs are being followed (M. Drawbridge email with attachment to T. S. Talley, 17 February 2017).

Budgetary decisions. The cost of a purchase determines who at HSWRI signs off on the decision to spend money; in general, the more expensive a purchase, the higher the authority needed to sign off on it, with decisions sometimes requiring the approval of HSWRI's CEO or CFO. Priorities for spending on capital purchases are determined with the help of management staff, including the Hatchery Manager and the Facility Manager. Because funds are often tight, money is typically spent on critical life support systems and infrastructure. The Hatchery Management staff has authority to purchase supplies for day-to-day needs, including feeds, oxygen, and lab supplies, among other things. On an annual basis, the Operations Manager and project PI's set and adjust budget allocations according to the program's needs (M. Drawbridge email with attachment to T. S. Talley, 17 February 2017).

Other operational decisions*.* Other decisions relevant to the OREHP include decisions regarding personnel, fish health, and production. Supervisors and HSWRI Human Resources staff are responsible for decisions pertaining to personnel. HSWRI's staff veterinarians, HSWRI's Fish Health Management team, and CDFW's pathologist (when appropriate) are responsible for fish health decisions (see Section 1.6.1 for further detail). HSWRI's Institutional Animal Care and Use Committee (IACUC) reviews animal welfare considerations semi-annually. HSWRI's Senior Hatchery Management team, made up of the Program Director, the Director of Operations, the Hatchery Manager, and the Assistant Manager, develops the program's annual production plans, while day-to-day production decisions are made by the Hatchery or Assistant Manager (M. Drawbridge email with attachment to T. S. Talley, 17 February 2017).

Internal communications practices that support decision-making*.* HSWRI's management staff hold a weekly "Manager's Meeting" on Tuesday mornings, during which participating staff

members report on the happenings within their respective focus areas and discuss program priorities and scheduling (among other things). The following staff members participate in Manager's Meetings: (1) the Program Director, (2) the Operations Director, (3) the Hatchery Manager, (4) the Assistant Hatchery Manager/Growout Coordinator, (5) the Office and Data Manager, (6) the Fish Health Specialist, and (7) the Facilities Manager. Previously, the Research Coordinator and the Tagging Coordinator also participated in this meeting, but these positions were recently (in 2016 and 2017, respectively) eliminated due to lack of funding. These meetings help inform operational decisions (M. Drawbridge email with attachment to T. S. Talley, 17 February 2017).

On Tuesday afternoons, the Hatchery Manager holds an all-staff meeting to relay information from the Manager's Meeting to the rest of the staff, and the staff report on their focal areas (e.g., live feeds, larval production). Additional meetings are held as needed to discuss more specific program issues (such as health, research, personnel management, or infrastructure). Furthermore, a member of the hatchery staff is responsible for sending daily update emails for each stage of the following six sectors of fish production: (1) *Artemia* production, (2) Egg-larval production – incubation system, (3) Larval-juvenile production – J1 System, (4) Juvenile production pre-tagging – J2 system, (5) Juvenile production post-tagging 1 – raceways, (6) Juvenile production post-tagging – Agua Hedionda Lagoon cages. The email includes information on water quality, fish behavior, mortality, and feed levels for each sector, and is sent to all project staff. In addition, the Broodstock Manager sends out a weekly email summarizing spawning and egg production for that week. These emails keep management staff abreast of daily hatchery operations (and at HSWRI's growout operations in Agua Hedionda), indicate that procedures are generally followed, and help managers make informed daily decisions (M. Drawbridge email with attachment to T. S. Talley, 17 February 2017). However, this information is not formally cataloged, synthesized or analyzed, making it difficult to perform adaptive management. These individual units of data and information could be compiled to help detect signs of an impending problem, and to ensure procedures are not being overlooked.

Training and adaptive management. Despite the many SOPs and protocols in HSWRI's plans, circumstances may arise during operations that challenge the decision-making process and undermine the plans. These circumstances may include unexpected occurrences such as the discovery of a new pathogen affecting White Seabass, or a lack of staff training in following SOPs. While plans are necessary for the operation of the hatchery, they must be coupled with adaptive management strategies to allow for quick decision-making, and with adequate staff training and expert consultation, to minimize mistakes.

Currenty, HSWRI's senior culturists generally train technical staff in a hands-on manner, following HSWRI's SOPs. HSWRI also employs a cross-training approach, so that many staff members can complete and understand other staff members' jobs (M. Drawbridge pers. comm.) Senior culturists are allowed to attend external trainings (such as workshops on fish health or Recirculation Aquaculture Systems (RAS)) to expand their knowledge, as funding is available. While staff training appears to be thorough, there have been a few instances of

human error over the course of the program that have resulted in fish mortality; such occurences are not unusual among pubic and private hatchery operations. For instance, in 2011, 33 newly caught White Seabass broodfish died when personnel failed to maintain adequate levels of oxygen in the fish's quarantine tanks (Annual Report 10-11), and in 2006, approximately 28,000 juvenile fish (99% mortality in that raceway) died when an employee working alone on the night shift adjusted a seawater supply valve incorrectly and restricted water flow to the fish (M. Drawbridge email to T. Larinto, 24 August 2006). In response to the latter incident, HSWRI conducted an internal investigation, issued a written warning and probationary assignment for the employee responsible, and reviewed its training protocols in order to prevent the occurrence of a similar error in the future. No other examples of these types of errors were found during this review.

The process to modify or update standard protocols is on an as-needed basis. Major changes to any of the plans or protocols are discussed in Annual Reports and, according to the hatchery leadership team, protocols are updated as follows: "Generally, protocols are reviewed on an annual basis. Hatchery management staff update protocols as needed, or when significant changes are implemented. The date of protocol modification is stamped on the protocol and changes must be approved by senior hatchery management personnel" (M. Drawbridge email to T. S. Talley, 8 February 2016).

A common problem in these complex physical/biological systems involves drifting protocols, wherein small changes made to procedures over time eventually result in the creation of a major problem appearing, such as a decreased survival. Tracing the source of such a problem is difficult when protocols are not well defined and accurately followed. Fully computerized and hyperlinked protocols and SOPs are required to minimize drifting protocols. Creating a database of hyperlinked SOPs can also allow for the creation of daily job task lists and checklists with easy access to SOP details, which can also help with cross-training and integration of different work groups.

Expert reviews of protocols. Over the course of the OREHP, HSWRI has occasionally hired marine fish culture experts (including Dr. John Tucker and Dr. Allen Davis, and most recently, Dr. Jennifer Cobcroft, an expert on malformations) as consultants to review its protocols. While bringing experts to HSWRI to review protocols can be expensive, it is worthwhile and should be done when resources allow. Some things that could use focused review by external experts include power outage procedures (see below), sampling methods for fish batch health and growth checks (see Section 1.6.1), dry feed storage protocols (namely, whether dry feed for juveniles should be retained for up to 6 months; Pellet Feed Storage and Handling SOP 2016), and the organization of HSWRI's SOPs (for example, whether water quality and husbandry plans could be hyperlinked).

5.2.1.2. Emergency decisions.

HSWRI staff make emergency decisions according to the type and scope of the emergency at hand, guided by the many SOPs and hatchery plans in place (e.g., Power Outage SOP 2015, Infectious Disease Emergency SOP 2016, Water Quality Contingency Plan – Carlsbad SOP 2016,

HSWRI Fish Health Management Plan 2016). Emergency prevention procedures for risks to human safety are managed by HSWRI's full time Safety Officer, who, with some help from HSWRI's Dive Safety Officer, is responsible for coordinating a comprehensive safety training program for land- and sea-based operations. Emergencies relating to fish health and welfare are handled on a case-by-case basis by HSWRI's Fish Health Management Team, comprised of three senior hatchery personnel and three veterinarians (two of whom are HSWRI staff members). Trained hatchery personnel can usually address problems, such as mechanical failure, that jeopardize fish health; in the case of disease concerns, HSWRI veterinarians and other trained staff work with CDFW pathologists to address the issue (see Section 1.6.1.6 for greater detail). Infrastructure emergencies, such as equipment failures, are handled on a case-by-case basis by senior hatchery management staff, who call on external contractors and experts in pump mechanics, electrical infrastructure, and Heating, Ventilation and Air Conditioning (HVAC) technology, among other things, as well as trained staff from SeaWorld for support.

While HSWRI's emergency protocols generally do a good job of outlining the actions to take in emergency situations, there are some gaps in HSWRI's emergency preparedness. Alarms and SCADA (Supervisory Control and Data Acquisition) are in place, but have not been updated for years, and there are no protocols about the system.

Furthermore, some protocols reveal equipment design flaws that may increase the likelihood of accidents due to human error and/or insufficient training. For example, tasks such as raising pipes, closing drains and turning off power to pumps, are required to reduce water loss during power surges that result when power is restored after an outage (Power Outage SOP 2015). Water loss during power surges, however, is due to design flaws (e.g., breaker and panel designs can be used for time delays to ease startup surges). Similarly, air blowers, which provide air for the hatchery building during power outage, currently need to be manually set up (Power Outage SOP 2015); a more failsafe design would be an automatic start with blowers plumbed into the system using check valves. Post-power outage tasks, such as priming the main seawater system (Power Outage SOP 2015) also reveal design flaws, including faulty check valves or a suction side air leak. These air leaks are adding to the supersaturation issue at the hatchery (see Section 1.8.1.2). A leak tight system with a working check valve will not lose time in the incidence of a power outage. All of this being said, as far as we can tell HSWRI has never lost fish to a power outage at the hatchery (though one batch of about 3,000 fish was lost to a power outage at the King Harbor growout facility (Annual Report 14-15)), so these changes could be made to improve efficiency, and to eliminate the supersaturation issue from this particular source.

5.2.1.3. Research decisions.

One complication that may impact the decision-making process of a hatchery management staff like HSWRI, which is charged with both fish production and research functions, is that these two objectives can have different operating and decision-making priorities. Achieving the quality of research needed for scientific publication requires enough samples and repetitions to achieve statistical confidence (e.g., 95% confidence levels). From a functional hatchery

perspective this can take a lot of time and money, and as such can limit the number of hypotheses that can be tested. It is important, therefore, to choose and prioritize research topics carefully, including matching research choices with the biggest challenges being faced and determining which questions should remain as in-house trials to improve production as compared with those worthy of formal, statistically rigorous study. Research needs require a regular re-assessment of the costs compared with the outcomes of scientific research, with the willingness to end a line of research if no longer relatively productive.

Thus far, HSWRI's approach to the challenge of being tasked with these two goals – production on the one hand and research on the other – with limited funding, has been to address them separately, funding research through means other than its OREHP budget. One potential drawback to this approach is that in order to get funded, projects must answer calls for research in specific areas. Thus, projects falling within some areas of interest and research need, such as nutrition, have been funded relatively successfully, while others, such as disease, have not, resulting in research gaps.

In order to address the need for research, HSWRI also facilitates graduate student projects at the hatchery and its second laboratory at Mission Bay, San Diego. Additionally, taking a more exploratory approach, HSWRI staff will sometimes manipulate a variable in the culture process to see how fish respond in order to gain insights into improvements that could be made to culture operations (e.g., experiments on egg stocking density (standard 150 ml egg density vs. lower 100 ml egg density); Annual Report 15-16). However, because HSWRI is also required to produce and release fish, these explorations are usually executed with little or no replication.

HSWRI's Aquaculture Program Director is responsible for coordinating the process by which OREHP research decisions are made. HSWRI adheres to a 5-year science plan, which includes a section on Aquaculture, and within it, a section on Replenishment, which encompasses the OREHP. This plan helps guide HSWRI's OREHP research efforts, as do agency Requests for Proposals (RFPs), which are reviewed by HSWRI's Aquaculture Research Team and pursued based on research need and anticipated fundability. For exploratory research conducted at the hatchery, HSWRI senior culture staff (including the clinical veterinarian) discuss potential SOP modifications that might improve fish quality and survival at a specific life stage, and collectively decide which modifications to implement, and the methods to use. Results are reported back to Team members and documented in the Crop Summary (a HSWRI internal document). For certain issues that are enduring or severe, HSWRI will form a more permanent team to plan exploratory modifications, gather data, and report results so that further modifications can be tested. HSWRI has recently taken this step with three recurring problems: (1) bacterial management in larval systems, (2) bony malformations among juveniles, and (3) eye lesions associated with gas bubble disease. Other areas where HSWRI might focus its research efforts include the etiology of hatchery-reared White Seabass malformations (See Section 1.9), including the effects to larvae of exposure to ozonated makeup seawater and disinfected seawater (formalin treatment); the practicality and effect of HSWRI's broodfish replacement scheme as outlined by Gruenthal et al. (Broodstock Management Plan 2011), which has not been followed, up to this point; see Section 3.2.1; and follow-up studies on the

impacts of feeding young larvae enriched rotifers rather than *Artemia* (see Section 1.5.1 for information on trials done at the hatchery).

5.2.2. Data and Information Gaps.

1. No plan or protocol for updating BMPs (other than "as needed"). Major changes to standard operating procedures are reported in the Annual Reports; more common, minor changes were not recorded and cited as not practical to compile over the 20 year history. Turnover of lower level staff reduces institutional memory and makes a strong case for comprehensive computerized record keeping and having all detailed protocols in accessible database. It is uncertain as to the depth and number of changes and whether those warrant edits to the various plans and procedures.
2. Lack of knowledge about the effectiveness of the SOP documentation approach to codifying hatchery procedures. This is a very standard and also a very useful management decision-making tool. However, to be useful in these complex systems, SOPs must be very detailed, as most of the HSWRI procedures are, but also evolving documents that are corresponding to specific tasks or functions. Their usefulness depends upon the hatchery management's ability to have staff follow the standards, achievable through an effective staff training program focusing on this topic.
3. HSWRI's Fish Health Management Plan is stated as being an evolving document and appears to have the associated SOPs as hyperlinks to the separate documents (note that the version of the Plan supplied for this review was, and had to be, a standalone document). This method of linking procedural documents to the main plan is what makes it truly an evolving document. This does not appear to be true of the other plans and associated SOPs.
4. Information about the effectiveness of another approach, the HACCP (Hazard Analysis and Critical Control Points) approach where all steps in all processes and procedures are evaluated. This approach has been discussed in the hatchery and some initial steps have been made, but there is no reference to progress along these lines in the database for this analysis. This type of approach is good at defining and controlling the "things that can fall through the cracks" in a top down type organizational structure.

5.2.3. Recommendations.

1. Strengthen the adaptive management strategy, including developing, updating and reviewing SOPs with staff prior to the start of each fish production season to accommodate adaptive management practices and to serve as a quality control and quality assurance hatchery management tool.
2. Develop a more computerized and transparent system for handling task lists, check lists and SOPs, and include hyperlinks between relevant documents (e.g., SOPs and Plans). Using computer databases and linking systems allows easier access by all staff.

3. Increase the use of available human capital and creativity of the above average staff in order to improve operations. For example, allow staff to create and use spreadsheets and databases in place of daily email messages within each section of the hatchery. This will ultimate save time by entering data/information into cells instead of typing daily email messages, will increase adaptive management potential and efficiency, and will empower and boost morale of staff. This may require a stronger system of accountability and that the decision-making processes to be less centralized.
4. Develop a full HACCP analysis.
5. Improve recognition of the production function of the hatchery (improving survival) as opposed to the research function and make decisions accordingly. That will mean more short-term, possibly non-statistically significant, results to guide changes in SOPs, but not for publication. Every fish batch going through the system should be a learning experience to further pin down some variable of interest. Again, an effective computerized database of information will be needed to view each batch or sub-batch as an experiment.
6. Consider revising guidelines that cannot be feasibly implemented and address these vs. not being compliant (e.g., broodstock replacement rates being too slow).
7. Develop a research priority plan to determine which research has been adequately answered and/or which topics are disproportionately costly as compared to the potential lessons learned, and that guides decisions about other urgent research questions and needs.

5.3. Decision-maker structure: The relationship among CDFW, the Ocean Resources Enhancement Advisory Panel (OREAP), HSWRI, and Regulatory Agencies, including the roles that each group plays in decision-making.

5.3.1. Key Findings.

5.3.1.1. CDFW.

The CDFW Marine Region administers the OREHP, manages all of the OREHP contracts, and has authority over OREHP funds (currently comprised of Ocean Enhancement Stamp funds and SFRA funds), and discretion over the proportions allocated to the various OREHP contracts (including HSWRI's contract, gill net sampling contracts, and pathology contracts) each year. The CDFW Marine Region also manages the White Seabass fishery and associated obligations. CDFW Fisheries Branch makes OREHP pathology decisions, yet, interestingly, despite the existence of the CDFW Aquaculture Program, no aquaculture staff are assigned to the program.

5.3.1.2. OREAP.

The OREAP is a ten-member advisory panel comprised of representatives of stakeholders in the OREHP, including sport fishermen, commercial fishermen, aquaculturists, academics, and CDFW staff (FGC § 6594). The OREAP was founded with the intent of aiding the CDFW director "in establishing policy and direction for the research and enhancement programs to be supported

from the Fish and Game Preservation Fund" (FGC § 6594). In theory, this group, along with the CDFW director, has the power to approve the program budget (FGC § 6595), but the budgets presented to the OREAP tend to be very low resolution, and there is not enough detail or understanding of detail to provide any effective control. The OREAP approves the budget as a recommendation to the Director. Occasionally there are significant discussions about shifting around some of the budget for specific purposes or significant capital investments at OREAP meetings (e.g., OREAP Meeting Minutes, 8 March 2011, OREAP Meeting Minutes, 4 March 2008, OREAP Meeting Minutes, 12 April 2004).

Over the course of the program, there have been some issues with the OREAP. At times, there has been concern over OREAP members' low attendance at meetings (OREAP Meeting Minutes, 18 January 2005). Additionally, some of the organizations that are required to be represented on the OREAP (as dictated in FGC § 6594) no longer exist, including the United Anglers of Southern California and the Gillnet Association (OREAP Meeting Minutes, 30 March 2015). Despite the disbanding of these organizations, it seems that the individuals who previously represented them continued as members on the OREAP, purportedly representing sport and commercial fishermen.

5.3.1.3. HSWRI.

HSWRI is contracted by CDFW to rear and monitor White Seabass, and perform associated environmental monitoring, for the OREHP. HSWRI owns and operates the Leon Raymond Hubbard, Jr. Marine Fish Hatchery in Carlsbad, California, where most of the OREHP's activities take place. The Hatchery was built on land leased from San Diego Gas & Electric (SDGE), with mitigation funds awarded to CDFW from the San Onofre Nuclear Generating Station (SONGS), funds donated to HSWRI, and regular OREHP funds. Equipment used within the hatchery has been purchased with OREHP funds and funds from donations and grants awarded to HSWRI, thus some equipment items are owned by the State (though all have extended beyond their depreciable life), and others are owned by HSWRI (D. Kent email to R. Starr, 8 January 2017).

HSWRI is responsible for the culture of White Seabass, and has developed the protocols by which White Seabass are raised and released. As part of its contract, HSWRI collects and cares for White Seabass broodstock, raises hatchery-spawned fish, tags fish and conducts QA/QC measures, coordinates growout operations, oversees the release of fish, collection of fishery dependent tag recovery data, and conducts research relevant to the program. Some of HSWRI's decision-making processes are outlined in Section 5.2.1.

HSWRI runs the White Seabass Program with OREHP funds administered by CDFW. HSWRI also obtains funds for research from agencies outside of CDFW to fund specific research projects (as described in Section 5.2.1). For example, studies on White Seabass genetics (HSWRI OREAP Meeting Presentation, 29 September 2009), larval nutrition (see Section 1.6.1), and benthic monitoring (HSWRI OREHP Overview Presentation, 20 May 2015) have been funded by sources independent from and outside the control of CDFW (e.g., NOAA, Western Research Aquaculture Center) and do not show up on the CDFW budget numbers.

5.3.1.4. Regulatory Agencies.

Although the OREAP, CDFW, and HSWRI are the primary entities with management responsibility over the OREHP, it is also subject to regulatory oversight by local, state and federal resource agencies because it includes development activities and operations on coastal lands and in public waterways (See Section 5.5).

City and County governments oversee growout operations sited within the harbors and marinas located in their jurisdictions while regional and statewide agencies such as the California Coastal Commission (CCC), Regional Water Quality Control Board, U.S. Army Corps of Engineers, and National Marine Fisheries Service are also involved in authorizing aspects of the program through the issuance or review of discretionary permits. In many cases, these permits establish limitations, safeguards, or monitoring measures for the program to implement in order to ensure its consistency with applicable regulations. This structure of multi-party program management and regulatory oversight likely adds both complexity and costs to the overall project and may limit overall fish production if limited program resources are divided between basic operations and satisfying regulatory monitoring and compliance requirements.

Furthermore, the large number of regulatory agencies involved in overseeing the OREHP's activities at times makes it difficult to ensure that monitoring requirements and limitations imposed by different agencies fit together in the most efficient and effective way and are not at odds. It can also make it difficult to adapt permits based on monitoring results and "lessons-learned" over time. For example, the environmental monitoring program for growout sites in the Los Angeles area was recently discontinued after ten years while the data it amassed indicated that an open discussion of the initial results with the regulatory agencies likely could have resulted in efforts to streamline, scale back or eliminate it much earlier (see Section 2.1.1.1). Further, adherence to regulations surrounding the Broodstock Management Plan, despite revelations that protocols were based on false assumptions about required male to female ratios, limited the application of new scientific findings about the disproporationate genetic contributions of few broodstock indivdiuals (OREAP Meeting Minutes, 3 March 2009; see Chapter 3 for more information on broodstock genetics).

At the same time, however, the involvement of multiple oversight and management bodies can help the program to be carried out in an environmentally safe and responsible manner that is consistent with its various protocols, guidelines, and permit requirements if all parties maintain open communication, coordination and responsiveness.

5.3.2. Data and Information Gaps.

The extent that regulations can be streamlined or eliminated because of consistent compliance of operations and/or outdated scientific information (See Section 5.5.2).

5.3.3. Recommendations.

Review all regulations and determine whether there are some that have consistently been in compliance or the measured impacts are insignificant and could be potentially removed, in particular those with reporting that may take a disproportionate amount of time or resources to complete. Submit requests for any removals to the appropriate agencies (See Section 5.5.3)

5.4. Technology and information: Methods used to ensure that OREHP is using the most current information and technology.

5.4.1. Key Findings.

HSWRI staff have done an excellent job over the years developing, evaluating, and improving OREHP operations. During times of program funding shortages, the incorporation of new technologies and the translation of research and development to operations have tended to be delayed.

5.4.2. Data and Information Gaps.

Experimental research results that could potentially improve hatchery operations have been slow to be incorporated into hatchery operations at times. For example, the lack of resolution of the issues associated with gas supersaturation despite awareness of the cause.

5.4.3. Recommendations.

1. Cycle between developing technology or procedure and implementation should be shortened.
2. Implement refinements and/or improvements to hatchery operations on a small-scale basis, and expand as positive results are achieved and necessary funding becomes available.

5.5. Regulatory compliance: Are operations carried out in compliance with applicable state, local, and federal authorizations?

5.5.1. Key Findings.

Permit requirements for development and operations in California's Coastal Zone requires involvement of the following agencies/regulatory entities in the permitting process (See also Section 5.3.1.4).

Federal:
U.S. Army Corp of Engineers- U.S. Army Corps 404 (intake and moorings)
EPA/NPDES (hatchery discharge; LA County net pens)
U.S.FWS (Endangered Species Permit; Marine Mammal Protection Act)
U.S. Coast Guard (net pens)

State:
California Coastal Commission- Coastal Development permit

Regional/Local:
Regional Water Quality Control Boards

- City of Carlsbad Conditional Use Permit (CUP)
- Wastewater discharge permit (Carlsbad Municipal Water District, and Regional Boards in charge in growout facility locations)

The California Coastal Commission (CCC) has ultimate jurisdiction over permits needed for the OREHP to operate, including discharges from the hatchery and growout operations, thus the OREAP, CDFW, and HSWRI works with the CCC in advance of decisions to facilitate permit processes. Based on regulatory compliance data from over the years (e.g., Objective 2) for the OREHP, including the hatchery and growout facilities, all work conducted under the OREHP appears to represent more than just a "spirit" of regulatory compliance. Although this review did not focus on verifying or documenting the program's status and history of regulatory compliance, HSWRI staff and volunteers appear to have done an exemplary job in being responsible environmental stewards by complying with permits and avoiding or minimizing the potential adverse impacts of its operations. As an example, CDFW and HSWRI were highly involved over the last 10 years in a California Environmental Quality Act (CEQA) review of the program that required significant resources (e.g., HSWRI paid $50,000 for an initial environmental assessment), and coordination among local, state, and federal oversight agencies to facilitate the review and permitting processes. The CEQA review resulted in an Initial Study and, recently, a Negative Declaration (No Impact) (OREHP Final Negative Declaration 2012). This is despite regulatory compliance in California being challenging with agencies often having overlapping authority and jurisdiction with different, and sometimes inconsistent, rules and regulations that are occasionally based on outdated science.

5.5.2. Data and Information Gaps.

More detailed accounting information on the regulatory compliance cost would provide guidance about how this cost could be streamlined. In the case of permits, the CDFW and HSWRI were highly involved with significant resources devoted to producing an Initial Study and Negative Declaration (No Impact) to meet the program's requirements under the California Environmental Quality Act (CEQA), which was submitted to local, state, and federal oversight

agencies to facilitate their review and permitting processes. These costs must be included in the accounting as a line item in the budget.

5.5.3. Recommendations.

Prioritize permit elimination requests by both the amount of effort and resources needed to fulfill permit requirements, and the existence of long term evidence of no or negligible impacts. An example of a high priority elimination would be for procedures that have a proven track record of being in compliance, and for which the permits require much effort. An alternative to reporting could be an inspection program (See Section 5.3.3).

Chapter 6

Objective 6. Develop quantitative measures of success.

6.1. OREHP evaluation frequency: How often has the OREHP been evaluated and what is an appropriate frequency in the future?

6.1.1. Key Findings.

The fifth objective of the OREHP was to "continue to develop, evaluate, and refine hatchery operations to maximize the potential for achieving the goal of the program"; a portion of this objective is assessed in this section. Overall, the OREHP has not been evaluated, as a single entity, since its inception, though the program has most definitely developed and been refined throughout its history. Additionally, components of the OREHP, such as hatchery production and growout operations have been internally assessed periodically or on an as-needed basis. Nevertheless, the current review represents the first program-wide comprehensive assessment of the OREHP and it could be argued that a regular schedule of similar reviews should be instituted to better meet the intent of Objective 5.

The reason for the lack of a comprehensive, program-wide review to this point is not clear. One could speculate that it may have stemmed from a lack of funding or resources dedicated to conducting such a review, a reduced priority given to reviewing the program, or possibly from a conscious decision not to undertake a review until a set of reviewing criteria and responsible parties were established. Regardless of the reason, it is clear that had the OREHP planned to initiate a review, there were no tangible goals or metrics that the reviewers could have used to evaluate the program success. While the OREHP enabling legislation does require periodic review of the program, it neither specifies the metrics that should be used in a review nor establishes a desired frequency of such reviews.

6.1.2. Data and Information Gaps.

1. A clear evaluation plan with established time frames and responsible parties is not available.
2. No clear metrics by which to evaluate program success (See Section 6.2).

6.1.3. Recommendations.

The OREHP and its primary stakeholders need to work together to develop a specific evaluation plan to solicit regular external reviews of the program's progress towards meeting the original OREHP objectives, or, if decided by consensus, an amended version of those objectives. Such an evaluation plan should include the following components:

1. A set of evaluation metrics that can be used to accurately judge the progress the OREHP has made towards meeting the stated program objectives. The nature of these metrics should be decided upon by the OREHP and the stakeholder groups, and should be reviewed (perhaps every 2 OREHP review cycles) so that they can be updated to reflect the ongoing progress made by the OREHP and possible changes in the ecological, social, or regulatory environment in which it works.
2. The OREHP should undergo a full evaluation every 7 to 10 years. This is a long enough interval for the effects of changes of OREHP project actions to manifest themselves.
3. The 7 – 10 year interval is relatively long, it should not be taken to suggest that individual components of the OREHP should not conduct independent evaluations of their progress on a more frequent basis (e.g., as part of the annual reporting process) nor should it be construed to mean that major changes to program components should wait for the end of the next review session if there are clear and justifiable reasons to make changes sooner.

6.2. Success measures: The quantitative measures currently used for assessing ecological, fishery, and socio-economic success.

6.2.1. Key Findings.

Defining the success of the OREHP in terms of its ecological, fishery, and socio-economic contributions is made difficult by the lack of specific goals or metrics that the program, or an external review body, could use to quantify their progress. Although the OREHP enabling legislation specifies the need for such reviews, as mentioned above the legislation does not specify the actual metrics, which would likely need to be both qualitative and quantitative in nature.

An acceptable contribution of the OREHP to the fishery will require clarity and consensus with regards to the selected benchmarks. Criteria for success of a fisheries enhancement or rebuilding program involving use of hatchery fish are necessarily complex and should include inter alia effects on total stock abundance and catches, effects on the abundance of the truly wild and/or naturally recruited population components, effects on the fitness of the naturally recruited population component, and effects of enhancement on fishing effort/mortality (Walters and Martell 2004, Lorenzen 2005, Camp et al. 2017). Moreover, economic and social costs and benefits should be assessed.

Fisheries enhancement or rebuilding programs often involve trade-offs between production or socio-economic and conservation objectives (Lorenzen 2005, Camp et al. 2016 (in press)). Such trade-offs are often poorly understood by stakeholders and managers but must be recognized and made explicit (Lorenzen 2014, Garlock and Lorenzen 2017). Programs oriented towards enhancement of fisheries production or recreational fishing opportunities may require very different hatchery, release and fishery management measures than programs oriented towards

stock conservation or rebuilding objectives (Paquet et al. 2011, Lorenzen et al. 2012). It is therefore crucial that overarching program objectives are made explicit because they affect program design – it is equally important that as the program objectives evolve (e.g., from the "can we rear these fish in captivity" to "how do we rear large numbers of these fish in captivity" phases) that the evaluation programs evolve in parallel.

Given that the CDFW has oversight over the OREHP, it follows that the Department should bear some of the responsibility for establishing review criteria. However, because of the collaborative nature of the program, it is important that other stakeholders, including the OREAP, HSWRI, state and regional regulatory bodies, and private citizens or their representatives also participate in the development of review criteria. Further, guidance from an independent science and technical advisory committee with expertise in hatchery science and associated issues (e.g., fish pathology and health, fisheries and population biology) would help to unbiasedly set and later help to evaluate review criteria.

A review panel can, to an extent, assess whether the program is meeting the legislative objectives, in lieu of an existing set of quantitative measures, but an expanded set of measures (beyond those listed in 6.1) would be needed. However, for the review findings to be of greatest utility to the OREHP, they need to be based on both qualitative and quantitative measures that establish clear goals or milestones for subsequent reviews.

6.2.2. Data and Information Gaps.

1. Little to no socio-economic data are available for use in an evaluation.
2. Few social, economic, production or fish survival goals are established.

6.2.3. Recommendations.

1. OREHP researchers, advisors and stakeholders, and CDFW need to develop a set of adaptive quantitative benchmarks; the schedule to evaluate the benchmarks could be set by the suggested schedule in 6.1.
2. Socio-economic studies of the program are needed. These analyses should include direct, indirect and induced effects (see Section 6.4).
3. Develop specific evaluation metrics that provide quantitative and qualitative assessments of the OREHP goal and objectives. A list of general metrics intended to be used as the basis of more specific criteria to assess the OREHP in the future is provided. The specific quantitative and qualitative criteria used for each of these metrics would be decided jointly by the CDFW, the OREAP and/or the program's stakeholders depending upon the State's priorities for the program. CDFW should work with experts in the fields of policy, sociology, economics, aquaculture, fisheries, and ecology to define these useful and tractable quantitative and qualitative metrics.

These metrics could be assessed for both long term (i.e., since the inception of the OREHP) and short-term (since the last program review) evaluations. The OREHP legislative intent, primary goal and the OREHP objectives form the framework for which metrics and corresponding quantitative and qualitative measures of each metric are chosen.

1. **Decide on the priority outcomes of the OREHP**
    a. Should the OREHP meet the modified, current legislative intent of the program (FGC § 6590, 6592) to conduct a program of basic and applied research into the artificial propagation, rearing and stocking of important marine fish species occurring in ocean waters off southern California in order to determine if hatchery-released fish can enhance stocks of wild species through increased hatchery production of fish, and the monitoring of fisheries to assess hatchery contributions?
    b. Should the OREHP meet the ultimate goal, which is to enhance populations of marine finfish species important to California for their sport and commercial fishing value (White Seabass Enhancement Plan 2010)?
    c. Should the OREHP meet the primary goal of the program, as stated in the Comprehensive Hatchery Plan (2007) and the White Seabass Enhancement Plan (2010), to evaluate the economic and ecological feasibility of releasing hatchery-reared fish to restore depleted, endemic, marine fish populations to a higher, sustainable level?

2. **Suggested metrics based on OREHP objectives**
    a. Develop and implement hatchery operation and growout methods that provide a supply of healthy and vigorous fish.
        i. Demonstrated proficiency in the development and implementation of spawning, rearing and growout protocols for producing fishes from depressed, economically-valuable species.
        ii. Feasibility of those same protocols to be scaled up to produce the numbers of fish that would be needed for a production-driven stock enhancement program.
        iii. Percent of hatchery fish produced that have a particular percent similarity to wild fish in terms of morphology, and/or that does not stray from a list of acceptable malformation types and severity levels in both hatchery and grow out facilities.
        iv. Percent of fish produced that are lost to the diseases most commonly seen in the hatchery and grow out.
    b. Conduct the replenishment program in a manner that will avoid any significant environmental impacts resulting from operation of either the hatchery or pen rearing facilities
        i. Level of change or threshold values of key indicator water quality and sediment variables.
        ii. Continued compliance with environmental regulations (as they change with time, and with hatchery production levels).

c. Maintain and assess a broodstock management plan that results in progeny being released that have genotypic diversity very similar to that of the wild population.
    i. Level of genetic variability in hatchery fish, as calculated with consideration of released adult survival and reproductive success, that is similar to wild fish genotypes and variability
    ii. Level of evenness of famly contributions at each stage of the rearing process.
    iii. Levels of genetic diversity and effective population size that remain unaffected by supplementation
    iv. Measures to reduce domestication selection, e.g. natural spawning and rearing, early release
d. Quantify contributions to the standing stock in definitive terms by tagging fish prior to release and assessing their survival in the field.
    i. Percent contribution of hatchery fish to the wild population, including fishery-independent and/or commercial and recreational fishery dependent contributions based on recapture data and a stock enhancement population model.
    ii. Cost or restitution value per recaptured hatchery fish (or cost per released hatchery fish).
e. Continue to develop, evaluate, and refine hatchery operations to maximize the potential for achieving the goal of the program. Again, the metrics chosen may depend upon the whether the priority is on the current legal intent and interpretation and/or the stated goal of the OREHP.
    i. A measure of scientific contributions (e.g, number of publications, white papers, external research grants awarded *OR* meeting participation per year).
    ii. The extent to which the basic and applied research conducted by the OREHP and connected entities has helped increase our knowledge of the artificial propagation, rearing, stocking and distribution of adversely affected marine species, such as White Seabass?
    iii. A measure of public education contributions (e.g., number of volunteers and students engaged, outreach events hosted, and/or people receiving newsletters).
    iv. Breadth and depth of stakeholder engagement, e.g., ensuring that a particular diversity stakeholder groups is engaged, e.g, recreational fishers, commercial fisheries, educators, seafood system reps, etc).
    v. Clear identification of questions related to effective propagation, rearing, and stocking of target species that remain to be addressed.
    vi. Level of external funds brought in relative to OREHP funds.
    vii. Levels of direct indirect and/or induced economic contributions of the OREHP.
    viii. A measure or indicator of a true adaptive management process being achieved.
    ix. Assessment of whether the culture technologies and techniques have matured to a point where the OREHP can transfer those efforts from HSWRI to another entity (e.g., CDFW hatchery) (or been unsuccessful to a point that

the effort should be abandoned) so that the OREHP can focus on developing techniques for another target species.

6.3. Scientific accomplishments stemming from the OREHP.

6.3.1. Key Findings.

Despite HSWRI researchers being challenged by their dual role as researchers and producers, they have been productive in advancing our knowledge of the biology and culture of White Seabass as evidenced by the number of publications that have been produced throughout the program. Research related to the OREHP has resulted in 56 publications in peer reviewed journals and books, 28 theses and dissertations, and at least 3 more peer reviewed papers that acknowledge specimens or data stemming from the OREHP (Appendix 1). Each peer reviewed paper was cited an average (±1 SE) of 17±2.4 times (range of 0 to 67 citations) by authors of subsequent research papers as calculated for the 50 peer reviewed publications for which citation rates were available (Appendix 1).

Published topics involving White Seabass have included life history, ecology, physiology, basic genetics, diet in culture, common diseases and health conditions, tagging and tracking methods, and influences of enhancement on wild stocks (Appendix 1). Research has also addressed the culture of other species, including California Halibut (Appendix 1: Drawbridge 1990, Stransky 1998, Louie 2005, Vizcaíno-Ochoa et al. 2010), Yellowtail (Appendix 1: Trushenski et al. 2014, Smith 2015, Wrobleski (in progress)) and Sheephead (Appendix 1: Jirsa et al. 2007). The research areas most often applied to other cultured and sport species, include enhancement effects (Hervas et al. 2010), catch methods (Appendix 1: Aalbers et al. 2004), fisheries status (Appendix 1: Allen et al. 2007, Pondella and Allen 2008), feed (Appendix 1: Lopez et al. 2009) and disease in culture (Appendix 1: Chen et al. 1995, Arkush et al. 2005) as indicated by the high citation rate including studies on other species (e.g., Toranzo et al. 2005, Noga 2011, Brownscombe et al. in press).

6.3.2. Data and Information Gaps.

Despite the scientific knowledge gained through the OREHP, there are research gaps that remain and could be addressed. These research areas include:

1. Empirical information on the effectiveness of release strategies, including (more) information on release microhabitat and timing, size at release, and stocking magnitude that will optimize survival and ecological and economic efficiency of the program (see Sections 1.2, 1.3, 1.7, 4.1, 4.4).

2. Data on the effects of stocking on wild White Seabass population size and genetics, including information on interactions with wild populations, and the reproductive success of hatchery fish in the wild (see Sections 1.2, 3.3, 4.4).
    a. Genetic research on hatchery fish, including data on the effective size of broodstock (through individual reproductive success and estimates from parentage assignment of released juveniles), selective mortality throughout the rearing process (by genotyping dead and surviving individuals, estimating family specific mortality, estimating batch specific mortality), and genetic diversity of released juveniles and recaptured adults (see Chapter 3).
    b. Genetic research on wild fish, including data on population structure, the effective size of the wild population (especially variance in reproductive success within age classes), and Ne/N ratio in the wild population (see Chapter 3).
    c. Information on the rate at which tagged fish disperse and leave the Southern California Bight survey area; information on the contributions of domestication effects, diseases and deformities, and release strategies to natural mortality of released White Seabass (see Section 4.4).
    d. Assessments of how hatchery contributions compare with other White Seabass recovery tactics, such as fisheries management, in terms of changes in White Seabass population size, economic costs and benefits, social costs and benefits, and genetic risks (e.g., see Sections 1.1, 3.3.1).
3. Research on the specific causes of deformity in hatchery fish (e.g., genetic, water quality, feed quality), the incidence of different types and severity of deformity in wild fish, and the effects of deformity on growth, reproduction, and survival throughout the life of the fish (see Section 1.9).
4. Empirical data on the triggers and vectors of disease that may impact hatchery-raised White Seabass, including research on effects of time in growout, environmental pathogens, physical and chemical conditions, genetics, feed quality, fish density, and the incidence and rates of disease in wild fish and the potential risks if fish are released following infection with certain pathogens (see Sections 1.2, 1.3, 1.6, 1.7, 1.8, 1.9).
5. Information on wild population dynamics; this may include collaborative research with Mexico to determine population size, structure, catch data, and the age/length of fish at first maturity and the size at which 50% of White Seabass are mature (see Section 4.5).

6.3.3. Recommendations.

Consider developing a decision-making process among CDFW, OREAP and HSWRI, and input from an independent expert advisory team to prioritize research areas using the data gaps listed above (and see Sections 5.2 and 5.3) as a guide.

6.4. The real and perceived social benefits of the OREHP.

6.4.1. Key Findings.

While the monetary costs of hatchery fish production exceed the value of recaptured fish (See Section 4.6.1.2), there are likely many social benefits of the OREHP. To date, however, there have been no formal social analyses conducted to assess the economic and non-economic social aspects of the program. Some social benefit may be derived from the information transfer from the OREHP to other aspects of the research community and industry. Given the pioneering work on the culture of species that are of sport and commercial interests that has resulted from the OREHP, most of the social benefit likely lies in the real and perceived benefits to the recreational and commercial fishing communities.

6.4.1.1. Real benefits.

Most of the real social benefits of the OREHP are likely educational and personal growth experiences that may translate into greater public environmental literacy and stewardship. HSWRI, partly under the auspices of the OREHP, engages in public outreach through its bimonthly newsletter, its Seabass in the Classroom Program, and its frequent participation in public environmentally oriented fairs and events. These outreach and education efforts likely increase public awareness of the OREHP and enhancement programs in general, White Seabass and other marine species, and marine conservation. HSWRI, more directly under the OREHP, provides experiential opportunities through the Save Your White Seabass Head tagging retrieval program, and the volunteer run growout facilities. These experiential opportunities, especially the growout facilities, provide aquaculture and project management training, a sense of identity (a sense of involvement, belonging and responsibility), and ultimately environmental stewardship.

The recreational fishing community most regularly participates in the operation and maintenance of growout pen facilities, so likely receives most of the direct educational and personal growth benefits associated with that part of the program. A recent survey of 14 growout volunteers from eight growout facilities revealed that each volunteer spends (or has spent) an average of 345 hours per year volunteering, and that each of these volunteers has served for an average of 14 yrs (range: 2-25 yrs; Marshall and Shedd 2016). The respondents acted as managers, co-managers, active past managers and/or long-term volunteers whose contributions included recruiting, training and managing volunteers; maintaining, operating, fundraising for and publicizing their facility; transporting, monitoring and caring for the fish; and interacting with HSWRI (Marshall and Shedd 2016). The informational and experiential values of the program also extend to the greater public, such as through the involvement of other programs (e.g., Sea Scouts, Boy Scouts, citizen science programs) and events at the facilities (e.g., tours, field trips, lectures, open house events) (Marshall and Shedd 2016). An average of 761 people per facility were reached through the education efforts of these eight growout facilities between June 2015-June 2016 (Marshall and Shedd 2016). Most (80% or more) volunteers helped with hand-feeding and monitoring the fish, and maintaining the pens, with other duties including recruitment, training and management of other volunteers,

donating money and/or fundraising, donation of vessel or vehicle time, and supporting HSWRI staff.

6.4.1.2. Perceived benefits.

Perceived benefits may be substantial. Anecdotally, recreational anglers feel that the White Seabass Enhancement Program is enhancing wild White Seabass stocks, and that the funds from the Ocean Enhancement Stamp program which go to the OREHP are being directly used to improve their fishing experiences (e.g., Marshall and Shedd 2016). The high-visibility operations, programs and events may have more positive social impact through engendering goodwill and ownership of the program than actual biological impact (contributions of hatchery fish to the wild stocks (see Section 4.6)). The social impacts of the OREHP should be formally and comprehensively assessed, and while a cursory look reveals positive social impacts that should continue to be touted, a priority should be placed on actual benefits and costs.

6.4.2. Data and Information Gaps.

1. A socio-economic analysis of the program is needed, including direct, indirect and induced effects for all elements of the program (RFP for such a study drafted as part of this evaluation; Appendix 2).
2. A survey of the angler views with respect to the White Seabass fishery and the OREHP, and willingness to pay for the program, would be very valuable.
3. Surveys of the costs/benefits of the program to commercial anglers and others who benefit socially and/or economically from the program are needed.

6.4.3. Recommendations.

1. Perform an assessment of the social and economic benefits of the OREHP, including direct, indirect, and induced effects of all elements of the program. Consider using the draft RFP developed by California Sea Grant as the starting point (See Appendix 2). The economic assessment might include an economic impact model (e.g., IMPLAN (Impact Analysis for Planning)) or other quantitative approach.
2. CDFW should engage a broad range of stakeholders (along with the recreational fishermen and growout volunteers that are already targeted), including commercial fishermen, Ports, public/private sector, to gain insight into perceptions of the program and values of different groups, as related to stock enhancement.

6.5. Other species that could be successfully reared using existing facilities.

6.5.1. Key Findings.

CDFW compiled and provided the SAC with a list of 16 species (12 finfishes, 4 invertebrates, all abalone species) that could potentially be targeted for enhancement under the OREHP (CDFW

OREHP Potential Species Table 2016). The list includes fishery, population and distribution information for each species. In 2014, HSWRI established the Dick Laub Fisheries Replenishment Program to investigate the potential of rearing other species for stock enhancement (MacNamara et al. 2016a). As part of the Dick Laub Program, HSWRI worked with the Coastal Conservation Association California to survey recreational fishermen and determine their preferences for alternative enhancement species (MacNamara et al. 2016a). Surveys were distributed at the following events in Southern California: Marina Del Rey Anglers meeting, Fred Hall Tackle Show in Long Beach, Pacific Coast Sportfish Show in Newport Beach, Oceanside Senior Anglers meeting, Fred Hall Tackle Show in Del Mar, San Diego Anglers Open Bay Bass Seminar, San Diego Anglers Open Bay Bass Tournament, San Diego Anglers meeting, and Coastal Conservation Association California meeting; a total of 494 surveys were usable (MacNamara et al. 2016a). Fishermen were provided with a list of 13 marine finfish species or finfish groupings (Table 6.1), and asked to rank the top three species they were most interested in seeing replenished; fishermen were also able to write in a species that was not on the list (MacNamara et al. 2016a).

Table 6.1. List of 17 species that are potential candidates for enhancement under the OREHP. Noted are the 16 species identified by CDFW as appropropriate candidates (CDFW OREHP Potential Species Table 2016), and the 13 finfish species or groupings that HSWRI presented to recreational fishermen in a survey to determine their preferences for alternative enhancement species (MacNamara et al. 2016a).

List containing the potential enhancement species	HSWRI's sportfisher survey ranking	Common name	Scientific name
CDFW & HSWRI	1*	(California) Halibut*	*Paralichthys californicus*
HSWRI	2	Yellowtail	*Seriola lalandi*
CDFW & HSWRI	3	Kelp (Calico) Bass	*Paralabrix clathratus*
CDFW & HSWRI	4	Giant (Black) Sea Bass	*Stereolepis gigas*
CDFW & HSWRI	5	Spotted Sand Bass	*Paralabrax maculatofasciatus*
CDFW & HSWRI	6	Corbina	*Menticirrhus undulatus*
CDFW & HSWRI	7*	California Sheephead*	*Semicossyphus pulcher*
CDFW & HSWRI	8	Barred Sand Bass	*Paralabrax nebulifer*
CDFW & HSWRI	9	Cabezon	*Scorpaenichthys marmoratus*
HSWRI	10	Other Rockfish	*Sebastes* spp.
CDFW & HSWRI	11	Scorpionfish (Sculpin)	*Scorpaena guttata*
HSWRI	12	Spotfin Croaker	*Roncador stearnsii*
CDFW & HSWRI	13	Brown, Gopher or Grass Rockfish	*Sebastes auriculatus, S. carnatus, S. rastrelliger*
CDFW	n.a.	Red, Pink, Green or White Abalone	*Haliotis rufescens, H. corrugata, H. fulgens, H. sorenseni*

* California Halibut and California Sheephead were ranked first and second, respectively in a species selection assessment performed by MacNamara et al. (2016a) using their sportfisher survey results and CDFW's list. Note that only finfish were assessed by HSWRI, not invertebrates.

The top four preferred species were California Halibut (≈25% of all responses), California Yellowtail (≈19% of responses), Kelp Bass (≈14% of responses), and Giant Sea Bass (≈13% of responses), with California Halibut being the top ranked species at every survey location

(MacNamara et al. 2016a). California Yellowtail was the only highly ranked species that CDFW had not identified as an alternative species for stock enhancement.

Using the results from the HSWRI survey and the list of potential stock enhancement species from CDFW, HSWRI ranked species for suitability using criteria it developed in 2016, "A species selection framework for marine finfish stock enhancement in Southern California" (MacNamara et al. 2016b). These criteria included: (1) biological knowledge (number of research articles, population genetics, life history data for model development); (2) status and management (IUCN Red List classification, local population status, recognized as an enhancement candidate by CDFW, ease of protection until recruit to the fishery); (3) user group (high demand among stakeholders, recreational catch index, market value); (4) life history (time to recruit to fishery, movement/dispersal, mortality to growth ratio); (5) culture (extent of rearing success, broodstock availability); and (6) enhancement (previous stock enhancement, genetic resource management, and habitat utilization) (MacNamara et al. 2016b). The results showed that California Halibut was the highest ranked species in 97% of the resampling iterations, and California Sheephead was second on 62% of occasions (MacNamara et al. 2016b).

There are a number fish species that have an economic importance to the state, and these fishes have potential to be cultured for purposes of stock enhancement. In addition to White Seabass, HSWRI has a parallel California Halibut initiative examining the potential of this species as an enhancement candidate. Other potential species include California Yellowtail, California Sheephead, Rockfishes, Giant Sea Bass, Cabezon, Bocaccio, and Striped Bass. All are native species with the exception of Striped Bass, which was historically released for enhancement in central California. HSWRI, in collaboration with various groups, has developed intensive hatchery technologies for most of these species. HSWRI has demonstrated production capabilities by maintaining egg, larvae, and juvenile for most of these species. The existing hatchery infrastructure required to produce these fishes is already supported so there would be no adverse impact to current operational efficiencies.

The OREHP challenges will be to obtain approval to culture additional species, and to manage the hatchery husbandry aspects of these species that have different spawning regimes, specific larval rearing conditions, and floor space at the hatchery. For example, California Halibut or Yellowtail will require coordinated, complex feeding systems, including enriched, live rotifer production capacity. Other issues found with the California Halibut trials include malpigmentation, where half or so of fish have a light or white coloration instead of the normal dynamic pigmentation, and that California Halibut is fairly slow growing (slower growing than White Seabass) so would require extended rearing cycles. Populations, however, are known to be depressed. Approval to conduct research on a new species must the obtained from the CDFW, and the OREAP. Each species should be evaluated individually to determine its capacity for large-scale hatchery production. Additional hatchery operating funds will be needed to conduct the research and to evaluate the production effort over several years.

6.5.2. Data and Information Gaps.

1. Preliminary assessments by CDFW and HSWRI evaluating the potential of culturing new species have been conducted and reported. Additional research is needed to determine the social and economic benefits and costs, and the efficacy of developing large-scale hatchery operations for each new species being assessed.
2. The HSWRI survey discussed in section 6.6.1 (MacNamara et al. 2016a) focused on recreational fishermen to determine their preferences for the proposal of culturing new (alternative) species. As such, there is a potential of survey bias as it would be valuable to survey a broader range of stakeholders, including commercial fishermen, to estimate demand for different species.

6.5.3. Recommendations.

As part of the OREHP legislative mandate, investigations of other species' potential are required and have, to some extent, been done, including the assessment of finfish of sportfishing interest conducted by MacNamara et al. (2016b) and the OREHP potential species information compiled by CDFW (CDFW OREHP Potential Species Table 2016). However, the species assessment should be part of a more comprehensive, *a priori*, publicly participatory assessment conducted with the guidance of an independent advisory committee that compares the economic, social and ecological trade-offs of candidate species in scenarios with enhancement and with no enhancement, only fishery management actions (See Sections 7.4.1-7.4.3). The focal species to be assessed should include those identified already to be of interest (Table 6.1). If species enhancement is deemed to be ecologically and economically more beneficial than management action alone (Section 7.4.2), then the following criteria should be used to prioritize species by enhancement potential:

1. Status of stock (e.g., consistently low enough to offset genetic risks associated with enhancement).
2. Ease of culture (e.g., relatively high growth rates, no overly specialized habitat or dietary requirements).
3. Ease of assessing post-stocking survival, health, and growth (e.g., availability of effective tagging and recapture methods, not highly dispersive, geographic range that can be feasibly sampled), and both contributions to and influences on wild stocks (e.g., genetics effects).
4. Life history that is amenable to enhancement (e.g., relatively fast growth rates, not highly dispersive)
5. Availabililty of ecological and biological information relative to culture and post-stocking assessments.
6. Comprehensive economic and, if feasible, social values of species.
7. Preferences (needs, impacts) of broad stakeholder groups, including both recreational and commercial fishing industries, as well as others who may be affected by enhancement efforts.

8. A feasible transition scheme for production of a new species, if a new species is recommended, into the hatchery's operating plan and infrastructure.

6.6. Commercial opportunities that could be pursued with the existing facilities.

6.6.1. Key Findings.

The question arises of whether the OREHP can venture into commercial opportunities, and if so how will this affect Federal Sport Fish Restoration Act (SFRA) funding. According to the Wildlife and Sport Fish Restoration Program guidelines, the State Fish and Wildlife agency determines what commercial activities and related facilities are allowed on Federal Assistance. For Federal Assistance programs on private lands, it is the responsibility of the State and the private landowner to agree on allowable commercial activities and related facilities. Venturing into commercial opportunities would be considered under the SFRA Financial Reporting "Program Income" and the OREHP would need to work with the SFRA Regional Office to make sure they have everything documented properly[8]. Commercial options within the OREHP also require the approval of the CDFW and the OREAP. Options outside of the OREHP will also require the governing entities to approve the sale of excess White Seabass produced at the Carlsbad hatchery to licensed aquaculture businesses. Once approval is obtained to sell excess fish, the potential of selling small, slightly 'deformed' fish (nose bump) that are not certified for stock enhancement release purposes but could be reared for commercial food consumption becomes a possibility to support the OREHP operations (e.g., Section 1.2.2.1 Recommendation 3c).

Information outlining the Wildlife and Sport Fish Restoration Program guidelines pertinent to allowing commercial activities under the OREHP can be found at the following websites:

U.S. Fish and Wildlife Service Manual Chapters Pertaining to WSFR Grants

- https://fawiki.fws.gov/display/WTK/Service+Manual+Chapters+Pertaining+to+WSFR+Grants

Part 516 FWS Financial Assistance - Award Administration

- *Chapter 1 Financial Reporting for Grant and Cooperative Agreement Awards:* http://www.fws.gov/policy/516fw1.html
- *Chapter 22 Allowable Commercial Activities and Related Facilities on FA Lands:* http://www.fws.gov/policy/522fw22.html

6.6.2. Data and Information Gaps.

An economic, biological and/or social analysis of the costs and benefits of potential alternative commercial opportunities is needed.

[8] SFRA Regional Office. U.S. Fish and Wildlife Service Pacific Southwest Region Wildlife and Sportfish Restoration Program, 2800 Cottage Way, W-1729, Sacramento, CA. 95825; Marie Strassburger Division Chief (916) 414-6727.

6.6.3. Recommendations.

The OREHP hatchery in Carlsbad is recognized as a model for marine hatchery operations. An expanded capability to include the sale of excess fish from the hatchery would make this facility a model for potential commercial development as well for fisheries replenishment. With the existing framework for commercial sale in place, the OREHP should cautiously pursue/investigate the sale of surplus fish. Some caveats/cautions include:

a. Research/enhancement production is still the primary focus.
b. Development of a market is necessary (marketing).
c. The cost/benefit of commercial production should be determined.

Chapter 7. Program-wide conclusions and recommendations.

7.1. Fulfillment of the ultimate goal of the OREHP: Enhancement of marine fish populations.

It is clear from the SAC review that the OREHP has met the original intent of the California State Legislature to conduct basic and applied research on the propagation, rearing, stocking, and distribution of an important marine fish, White Seabass (FGC § 6592). In 1983, little was known about the techniques needed to successfully spawn, rear, and release saltwater fishes. Since then, the OREHP has significantly contributed to the world's knowledge about marine enhancement science and techniques. Similarly, the OREHP has been consistent with the modified legislative intent of *determining* if hatchery released fish can enhance wild stocks (FGC § 6590); however it has shown that, at least for White Seabass, enhancement has not been effective to date, thereby falling short of the ultimate legislative goal of enhancing wild marine fish stocks.

It should also be noted that, while the White Seabass stock was considered depleted when OREHP was initiated and White Seabass chosen as its focal species, the stock has since increased likely due to a combination of high recruitment related to favorable environmental conditions and fisheries management measures (e.g., closure of the coastal gill net fishery).

7.2. Budget conclusions.

The operating budget needed to achieve all aspects of the OREHP objectives exceeds the base funding level of approximately $1.6 million per year that has been available for the program. With inadequate funding, the OREHP objectives suffer. Restricted funding has reduced or limited several OREHP capabilities, including the ability to exchange broodstock at appropriate rates (Objectives 1 and 3), provide stricter oversight of growout facilities (Objective 1), address reoccurring gas supersaturation issues (Objective 1), consistently and extensively perform and address challenges related to recapture surveys (Objective 4), and perform fisheries enhancement modeling (Objective 4). Limited resources have also likely prevented initiation of a genetic monitoring program (Objective 3) and (socio-) economic assessments (Objective 5 and 6). It is important to note that HSWRI has contributed in excess of $400,000 annually to meet operational expenses that are at least in part related to the OREHP and has sought grants and contributions from a mix of private and government sources to make infrastructure repairs and improvements to the hatchery facility; HSWRI has also brought in external funds to cover research and outreach efforts that are related to, but not part of, the OREHP.

7.3. Program-level observations and recommendations.

Although the SAC did not conduct a comprehensive review of OREHP management processes, it believes that the organizational structure of those groups overseeing the OREHP is potentially

sufficient to achieve OREHP goals and objectives. The SAC noted several program-level weaknesses, however, and made recommendations for strengthening the OREHP.

7.3.1. Need to strengthen and update organizational structure.

The ultimate authority for many programmatic decisions within the OREHP was unclear. It is necessary to clarify, for example, who has the authority to make decisions relating to research priorities and issues that influence or put hatchery operations and scientific research into conflict with one another. Part of this uncertainty is caused by the OREHP's dual focus on production and research, two activities which can be very different and for which there are limited resources. Additional uncertainty may be due to the change in the internal interpretation and communication of OREHP intent, goals and objectives through time in the absence of periodic program reviews.
The SAC noted that the program's advisory panel (OREAP) has not been as effective or valuable as it could be, and that CDFW should reconsider how to best use an advisory panel. The current OREAP does not have the representation by the groups detailed in the original legislation, as some of these groups no longer exist or have changed focus. CDFW should restructure and reform the OREAP, and form an independent science and technical advisory group with expertise in hatchery science (and associated issues), population dynamics, release and recapture strategy optimization, and genetics to develop and, later, evaluate quantitative criteria, benchmarks, and timelines to be used in the future evaluation of the program.

7.3.2. Need for external, independent guidance.

Fish health guidance. The SAC was greatly concerned with the differences in opinions between CDFW and HSWRI pathologists regarding the definition of malformed, or deformed fish, and the implications of the range of morphological variability found in hatchery fish on vigor. Currently, this difference in opinion causes a large public relations problem and inhibits smooth operations at the Carlsbad hatchery, thereby resulting in reduced juvenile production due to diversion of resources and delays in decisions about diagnoses and appropriate responses. Further, differences in opinion and therefore the outcome of diagnoses and actions taken may ultimately affect release numbers and post-release survival. Although the risk of introduction of disease or unwanted genetic characteristics to the wild fish population may be low due to the low likelihood that these factors are linked with malformations, it is critical to have consistent decision-making criteria and set appropriate policy for dealing with malformed fish. The SAC strongly recommends that CDFW and HSWRI engage an independent panel of experts that would be charged with the following:

1. Compare the morphological diversity of wild fish with that of hatchery fish.
2. Determine which unique hatchery morphologies pose a genetic or other biological threat to wild populations.
3. Determine which morphologies cause loss in post-release fitness.

4. Develop a set of criteria and protocols for identifying and responding to fish that have unacceptable types and/or levels of deformity that both CDFW and HSWRI staff agree upon.
5. Develop approaches that minimize frequencies and levels of deformities.

Science and technical advice. The SAC developed assessment topics within each OREHP objective to help in determining the extent that each objective has successfully been met. Having a more clearly defined set of assessment metrics in place, such as those suggested in Chapter 6 of this evaluation report, would allow for more efficient, and maybe more frequent, assessments of the program and would provide clearer guidance to OREHP staff and researchers. Although the assessment topics in the evaluation report can currently be used to guide future assessments, more focused, clear, feasible, and occasionally updated metrics agreed upon by CDFW and OREHP contractors are still needed to identify future successes related to stock enhancement. Again, the SAC strongly recommends that CDFW periodically enlist an independent external group of science and technical experts to work with CDFW and stakeholders to develop (and later help to evaluate) a set of quantitative criteria, benchmarks, and timelines for each of the established OREHP objectives (for more discussion on the potential roles of an advisory committee, see Sections 1.9.1; 5.1.3, Recommendation 3; 6.1.3, Recommendation 3; 6.3.3).

7.3.3. Need to strengthen public communication and transparency.

HSWRI has led public outreach, stakeholder engagement and public relations for the OREHP, without provision of communications and development assistance or adequate resources to support this task. This responsibility taxes HSWRI's already limited resources for the OREHP and adds the risk of public scrutiny. The SAC occasionally had to dig deeply to find information needed to assess the status of various aspects of the OREHP and noticed the presence of potentially confusing statistics about various aspects of the program in reports and non-peer reviewed publications (e.g., newsletters). It is recommended that HSWRI and CDFW make greater efforts to keep information about the OREHP openly available to each other and to the public, and to improve consistency and transparency of outcomes and incidences, particularly for issues of public interest (e.g., contribution of the program to wild stocks, recapture rates of tagged fish in gill nets, incidences of disease and deformity, occasional accidents or die-offs, costs and benefits of the program, etc.). Improved transparency may include the development of a process that allows communication with a broad range of stakeholders, including those not already associated with the program, to collect input regarding priorities and development of the program. Further, it is recommended that CDFW assist more with this duty, or find and support knowledgeable public communications professionals to help.

7.4. The future of the OREHP: Review and reform.

This evaluation of the OREHP objectives, goals, intent and budget revealed that it is timely for the relevant authorities and stakeholders to review the overall focus and strategy for the

OREHP in terms of focal species and stocks, and the potential role of enhancement as an additional tool used in the management of those fisheries. The program evaluation has shown that, while the research and technology development objectives of the OREHP have largely been met, the program is not currently in a position to substantially enhance the White Seabass fishery due to a variety of factors.

Post-release survival and therefore contributions to the wild population are low. Further, the California White Seabass stock, which was depleted when the OREHP was established and White Seabass was chosen as its focal species, has since reached a higher level of abundance. These factors, together with changes in the status and management of other California stocks, and increased understanding of the potentials and limitations of stock enhancement to contribute to fisheries management outcomes, suggest that it is timely to reassess the program's utility, and to review and reform the OREHP's priorities and the approaches used to fulfill each of the OREHP objectives.

The following steps are recommended for assessing the future of the OREHP (Fig. 7.1), noting that these recommendations were made without consideration of cost and thus would need to be evaluated with respect to program priorities and levels of available funding.

7.4.1. A science-based and participatory public process.

The future of the OREHP should be determined through a process that is both science-based and participatory with respect to the program's stakeholders (Fig. 7.1). Overall guidance for such a process can be found in the Updated Responsible Approach to Marine Fisheries Enhancement (Lorenzen et al. 2010) and in the Hatchery Reform processes implemented for several salmon hatchery programs in the Pacific Northwest (NWFSC 2017). Scientific methods, such as fisheries models used to assess the potential effectiveness of stock enhancement and other fisheries management measures in achieving desired fisheries management outcomes, enable a systematic approach to the planning of enhancement programs. Stakeholders, principally recreational and commercial fishermen, have played a major role in the operation and funding (through license fees) of the OREHP. It is therefore imperative to involve stakeholders systematically and constructively, and to use current scientific information in making the following decisions about the program's future direction.

7.4.2. Assess the potential role of enhancement in California fisheries management.

The list of candidate species identified by CDFW and HSWRI, including White Seabass, should be honed using analysis of the biological, economic and social costs and benefits of the OREHP as compared to relying solely on (non-OREHP) fishery management strategies (e.g., updating catch quotas and/or size limits) for each species (Fig. 7.1A). If the analysis indicates that conventional fishery management strategies alone may be sufficient for the conservation and management of all or most of the candidate species, then discontinuation of the OREHP should be considered as one option, if legislatively feasible. If some species' stocks are deemed to be extremely low (i.e., severely depleted), and/or if responses to conventional fishery management actions alone

are predicted to be ineffective, then further development or modification of the enhancement program should be considered, and funding adjusted to enable the OREHP to meet its objectives. The candidate species lists put forward by CDFW and HSWRI were generally supported by the SAC, with California Halibut of particular interest for inclusion in this initial assessment given the available information on its biology, ecology, and culture practices, its depressed populations, and the high recreational and commercial fishing demand.

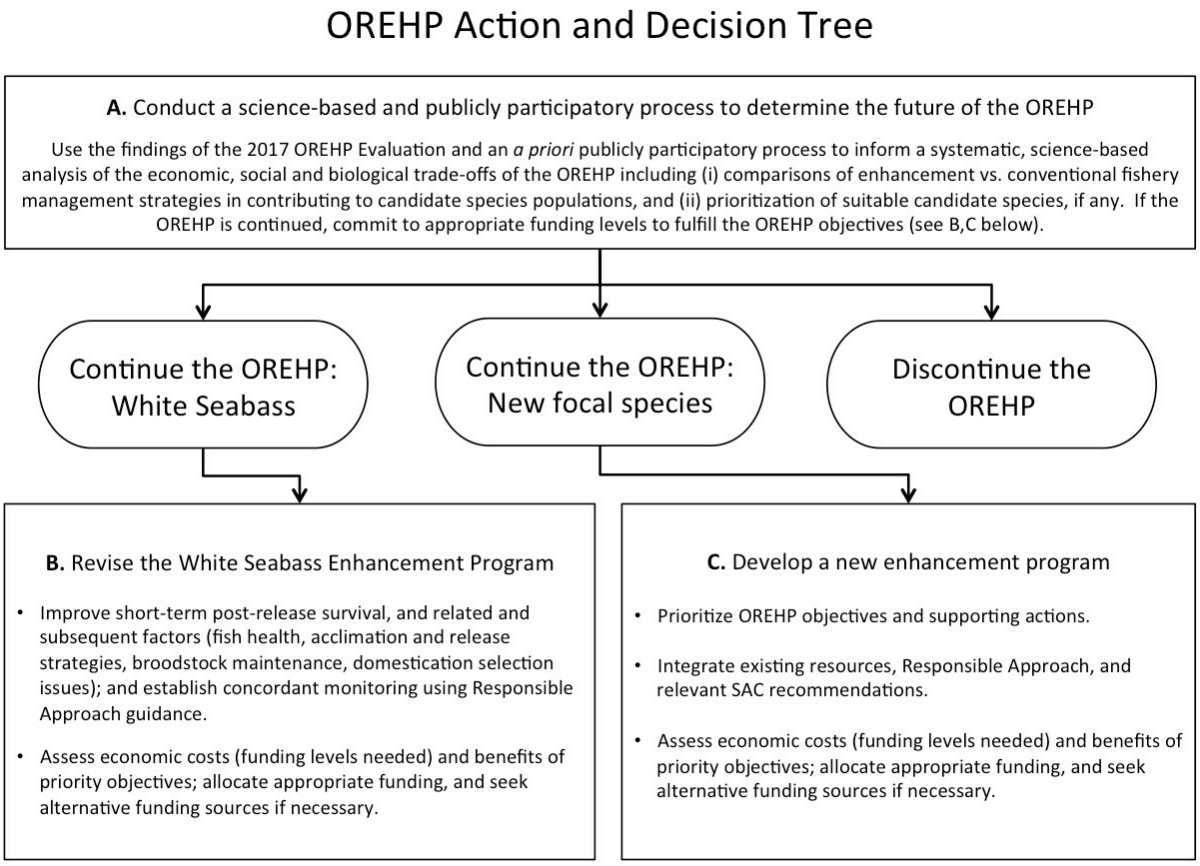

Fig. 7.1. Flow chart of decisions and actions recommended by the Ocean Resources Enhancement and Hatchery Program (OREHP) Evaluation Science Advisory Committee (SAC) to California Dept. of Fish and Wildlife (CDFW) to aid in decisions about the future of the OREHP.

7.4.3. Prioritize candidate focal species by enhancement potential.

If the initial assessment of the value of enhancement in relation to other fishery management strategies indicates that the OREHP could likely contribute to some of the candidate species, then the SAC recommended that those species remaining on the candidate list be prioritized. Specifically, the SAC recommended an *a priori* systematic and quantitative assessment of each candidate species (Fig. 7.1A) similar to the assessment developed by HSWRI (MacNamara et al. 2016b), but with input from a broader range of stakeholders, inclusion of economic and social costs and benefits, more consideration of fit with fisheries management strategies, and conducted in cooperation with an independent advisory committee. Criteria should include, but

are not limited to, depressed stock numbers (e.g., consistently low enough to offset genetic risks associated with enhancement), ease of culture, life history that is amendable to rearing, tracking and enhancement (e.g., relatively high growth rates, not highly dispersive), geographic range that can be feasibly sampled (e.g., most common in U.S. waters), availability of existing biological information, and high demand and value within commercial and recreational fishing industries and throughout the food supply chain (see Section 6.5.3 for full list). Clearly, the findings of the economic, social and ecological (e.g., environmental, genetic and population-level) trade-offs analyses used to narrow the candidate species list may be used to inform this process.

The challenges associated with each candidate species should be assessed and applicable recommendations from the OREHP evaluation report should be used. For example, a fish with a range that extends into Mexico will require collaborative efforts for population/fishery assessments, and relatively slow growth rates will still require decisions surrounding size at release trade-offs. New challenges should also be assessed; for example, the demersal California Halibut would require different tank designs than those established for the pelagic White Seabass, and as such would require a significant capital contribution to reconfigure hatchery systems.

If a change of focal species is decided, White Seabass should be phased out by ceasing breeding efforts while completing the rearing and release of existing early life stages. The rate of phasing, however, may depend upon space, resources (including availability of new species broodstock), and other logistical considerations. An independent advisory panel should be used for guidance on planning of the phasing and/or the development and initiation of a new enhancement program (Fig. 7.1C).

7.4.4. White Seabass enhancement: Focus on reducing post-release mortality.

The results of this evaluation stress the importance of minimizing post-release mortality of hatchery White Seabass to increase the potential of the enhancement program. The same would likely be true of new focal species that might be chosen for enhancement. Greater emphasis should therefore be placed within the OREHP on research of factors that affect post-release mortality, and on husbandry and release strategies that minimize this mortality (Fig. 7.1B). This focus may require increased funding to OREHP in order to fulfill a commitment to reducing pre-release and short-term (e.g., 6-month) post-release mortality rates. Increasing production to compensate for high mortality rates is not recommended because of the increased expenses, infrastructure and resource needs (e.g., staff time, supplies), and increased risk of fish health issues that are associated with higher production rates.

In particular, to improve survival and stock contribution rates, greater attention should be given to:

1. Domestication issues (Objectives 3 and 4).

2. Resolution of fish health challenges (e.g., resolving gas suppersaturation and its health effects, understanding effects of deformity types and severity on fitness, consistency in diagnosis and response to health findings; Objective 1).
3. Improved placement and oversight of growout facilities (Objective 1).
4. More research focused on optimizing release strategies such as timing, size, location and magnitude of releases (Objectives 1 and 4).
5. More effort on post-release monitoring needed to optimize release strategies and estimate recapture rates (Objective 4).
6. Greater integration with fishery management to understand relationships between enhancement efforts and wild populations/fisheries (Objective 4).

If White Seabass production is increased or if there is a change in focal species, however, potential environmental impacts associated with these changes should be reassessed (Objective 2), and monitoring efforts should be modified appropriately to account for higher production levels and/or different impacts depending upon system changes (all Objectives). If survival rates increase, improved genetic practices and monitoring should also be implemented in order to address the potential genetic effects associated with enhancement, which to date have not been an issue because of the extremely small possibility that a hatchery fish will survive to spawn with wild fish. If higher survival rates become the focus, then the broodstock management plan should be reassessed and reworked to include more frequent rotation of wild-caught broodstock, more emphasis should be placed on reducing domestication selection and increasing the proportion of spawns that go on to be reared, and monitoring of family contributions throughout the rearing process should take place to maintain the desired levels of genetic diversity and limit domestication selection (Objectives 1 and 3).

Further, a framework for conducting, evaluating and refining the enhancement program (Objectives 5 and 6) should be developed and used, regardless of the focal species selected. For example, the Updated Responsible Approach to Marine Stock Enhancement provides guidance on goal setting and evaluation, research and technology development, and adaptive management strategies (Objectives 5 and 6). In particular, the SAC recommended that an economic analysis be performed for whichever program approaches are selected in order to ensure that the financial benefits of the program outweigh potential costs, and to inform future assessments (Objectives 5 and 6). More attention should also be placed on adaptive management. The OREHP has many hatchery and growout protocols and plans in place, but data collection, record keeping, and reporting are not currently structured to allow formal assessment of whether protocols are being followed, and how findings and changes are contributing to protocol updates. For example, release strategies need to be optimized, and more formal data collections, record keeping and reporting of results (i.e., adaptive management experiments) can inform the evaluation of model assumptions about survival and the effects of fish size at release, release (micro)habitat, season, acclimation and acclimatization, and release magnitude. Adaptive management would also be useful for addressing many of the other challenges identified.

7.4.5. Address the economics of the program.

Assess economic benefit of the OREHP. Given that funds for the OREHP are largely public and much of the benefit of the program may be social, an economic (and social) analysis would make program expenditures more defensible, help to indicate social and economic strengths and weaknesses of the program, and may provide insights into stakeholder priorities. Improved economic awareness and efficiency is important because the accomplishment of priority Objectives, and the breadth and depth of actions needed to fulfill those Objectives, will be dependent upon available funds (Fig. 7.1B,C). The extent that recommendations made by the SAC through this review can be implemented will also be dependent upon funding levels. For example, if OREHP funding remains static, it may be necessary to narrow the focus of the program to solving the challenges to enhancement that were identified as highest priority by the SAC (e.g., reducing post-release mortality), but if funding is increased then there may be opportunity to also test and address the challenges of a program that contributes more significantly to wild populations (e.g., developing and initiating genetic monitoring). However, resolution of all identified challenges seems beyond a relatively small increase in funding and may require alternative funding sources, such as private organizations.

Need to expand public-private partnerships. There is a need to expand public-private partnerships such as those established already within the OREHP. HSWRI, the primary contractor for the OREHP, has forged partnerships with private groups, such as recreational fishing groups and private foundations, which have provided a substantial supplement of non-OREHP funds and in-kind resources (e.g., volunteer time, boat time, supplies) to operate the hatchery and growout facilities. Because of the infusion of supplemental funding from HSWRI, the SAC considered the potential for conflict of interest, and concluded that the State has benefited from the private funding, and that all information has been publically shared so that there is no conflict of interest among partners associated with the OREHP. If the OREHP continues, CDFW should consider expanding the public-private partnership concept to bring in additional partners (and funds), such as other foundations and commercial fishing communities, to expand capabilities of the OREHP including implementing recommendations made by the SAC for fulfilling each OREHP objective.

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M. Clifford email to V. Taylor, 1 May 2017

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V. Taylor email to T. S. Talley, 27 March 2017

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OREHP-Related Datasets

CDFW OREHP Budgets, Budgets Summaries, and Expenses Summaries

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California Department of Fish and Wildlife (CDFW). 2016. OREHP FY 2016-17 Budget. V. Taylor (ed.).

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OREHP Expenses Summary 2009-2015

California Department of Fish and Wildlife (CDFW). 2015. OREHP Expense Detail FY10 to FY16. Excel file.

Datasets

CDFW OREHP Potential Species Table 2016

California Department of Fish and Wildlife (CDFW). 2016. OREHP Potential Species Table. Prepared by CDFW for the Science Advisory Committee (SAC). Microsoft Word file.

HSWRI Releases Dataset 2016

Hubbs-SeaWorld Research Institute (HSWRI). 2016. Releases SAC 2016b. Prepared by HSWRI for the Science Advisory Committee (SAC). Microsoft Excel file.

Appendix 1. Table of publications stemming from the OREHP, and the number of citations associated with these publications, when available.

Table of publications stemming from the OREHP, including publications in (A) peer reviewed journal articles and book chapters, (B) student theses and dissertations, and (C) publications that acknowledge specimens and/or data stemming from the OREHP. Included are the number of citations of each publication, when available, as of 22 August 2016 from Google Scholar or Research Gate.

A.	Publications in peer reviewed journals and books	No. Citatio
1	Margulies D. 1989. Size-specific vulnerability to predation and sensory system development of White Seabass, *Atractoscion nobilis*, larvae. Fish Bull 87(3):537-52.	43
2	Bartley DM and Kent DB. 1990. Genetic structure of White Seabass populations from the southern California bight region: Applications to hatchery enhancement. CalCOFI Rep 31:97-105.	19
3	Dutton P. 1992. Effects of experience on feeding success by larval White Seabass, *Atractoscion nobilis*. J Fish Biol 41(5):765-73.	19
4	Bartley DM, Kent DB, Drawbridge MA. 1995. Conservation of genetic diversity in a White Seabass hatchery enhancement program in southern California. Schramm HJ and Piper R, editors. Uses and effects of cultured fishes in aquatic ecosystems. American Fisheries Society, Bethesda, MD (USA).	na
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7	Chen M, Henry-Ford D, Groff JM. 1995. Isolation and characterization of *Flexibacter maritimus* from marine fishes of California. J Aquat Anim Health 7(4):318-26.	55
8	Shane, M. A., W. Watson, and H. G. Moser. 1996. Polyprionidae: Giant seabasses and wreckfishes. In: H. G. Moser (ed.), The early stages of fishes in the California current region, p. 873-875. California Cooperative Oceanic Fisheries Investigations Atlas No. 33.	na
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11	Chen MF, Yun S, Marty GD, McDowell TS, House ML, Appersen JA, Guenther TA, Arkush KD, Hedrick RP. 2000. A *Piscirickettsia salmonis*-like bacterium associated with mortality of White Seabass *Atractoscion nobilis*. Dis Aquat Org 43(2):117-26.	27
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14	Shane, M. A. 2001. First records of Mexican barracuda (Sphyraena ensis) and additional records of scalloped hammerhead (*Sphyrna lewini*) in southern California. Bull. So. Calif. Acad. Sci. 100(3):160-166.	1
15	Drawbridge, M.A. 2002. Chapter 11: The Role of Aquaculture in the Restoration of Coastal Fisheries. In: Ecological Aquaculture, the Evolution of the Blue Revolution. Barry Costa-Pierce (ed). Blackwell Science. Osney Mead, Oxford. p. 314-336.	na
16	Aalbers, SA, Stutzer GM, Drawbridge MA. 2004. The effects of catch-and-release angling on the growth and survival of juvenile White Seabass captured on offset circle and J- type hooks. N Am J Fish Manage 24(3):793-800.	67
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19	Arkush KD, Edes HL, McBride AM, Adkison MA, Hedrick RP. 2006. Persistence of *Piscirickettsia salmonis* and detection of serum antibodies to the bacterium in White Seabass *Atractoscion nobilis* following experimental exposure. Dis Aquat Org 73(2):131-9.	3
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21	Chen MF, Apperson JA, Marty GD, Cheng YW. 2006. Copper sulfate treatment decreases hatchery mortality of larval White Seabass *Atractoscion nobilis*. Aquaculture 254(1-4):102-14.	17
22	House ML, Hedrick RP, Winton JR, Fryer JL. 2006. An isolate of *Piscirickettsia salmonis* from White Seabass is fully virulent for coho salmon. J Aquat Anim Health 18(4):252-6.	3
23	Hayward CJ, Bott NJ, Itoh N, Iwashita M, Okihiro M, Nowak BF. 2007. Three species of parasites emerging on the gills of mulloway, argyrosomus japonicus (temminck and schlegel, 1843), cultured in Australia. Aquaculture 265(1-4):27-40.	28
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26	Smiley JE and Drawbridge MA. 2008. A simple apparatus for maintaining gas-supersaturated seawater in the laboratory for experimental purposes. N Am J Aquacult 70(1):61-7.	4
27	Aalbers SA and Drawbridge MA. 2008. White seabass spawning behavior and sound production. Trans Am Fish Soc 137(2):542-50.	25
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	California bight. Mar Biol 154(2):307-13.	
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31	Oakes, Christopher T; Pondella, Daniel J. 2009. The Value of a Net-Cage as a Fish Aggregating Device in Southern California. Journal of the World Aquaculture Society. 40(1): 1-21.	5
32	Lopez LM, Durazo E, Viana MT, Drawbridge M, Bureau DP. 2009. Effect of dietary lipid levels on performance, body composition and fatty acid profile of juvenile White Seabass, *Atractoscion nobilis*. Aquaculture 289(1-2):101-5.	48
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34	Hervas S, Lorenzen K, Shane MA, Drawbridge MA. 2010. Quantitative assessment of a White Seabass *(Atractoscion nobilis*) stock enhancement program in California: Post-release dispersal, growth and survival. Fish Res 105(3):237-43.	23
35	Durazo E, Cruz AC, Lopez LM, Lazo JP, Drawbridge M, Viana MT. 2010. Effects of digestible protein levels in isonitrogenous diets on growth performance and tissue composition of juvenile *Atractoscion nobilis*. Aquacult Nutr 16(1):54-60.	6
36	Vizcaíno-Ochoa, V., J.P. Lazo, B. Barón-Sevilla, and M. Drawbridge. 2010. The effect of dietary docosahexaenoic acid (DHA) on growth, survival and pigmentation of California Halibut *Paralichthys californicus* larvae (Ayres, 1810). Aquaculture. Vol. 302, no. 3-4, pp. 228-234.	18
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38	Jirsa D, Davis DA, Drawbridge M. 2010. Development of a practical soy-based diet for White Seabass. Prog Fish-Cult 72(4):332-7.	12
39	Galaviz MA, Garcia-Gasca A, Drawbridge M, Alvarez-Gonzalez C, Lopez LM. 2011. Ontogeny of the digestive tract and enzymatic activity in White Seabass, *Atractoscion nobilis*, larvae. Aquaculture 318(1-2):162-8.	24
40	Smiley JE, Drawbridge MA, Okihiro MS, Kaufmann RS. 2011. Acute effects of gas supersaturation on juvenile cultured White Seabass. Trans Am Fish Soc 140(5):1269-76.	4
41	Gruenthal, K.M., and M.A. Drawbridge. 2012. Toward responsible stock enhancement: broadcast spawning dynamics and adaptive genetic management in White Seabass aquaculture. Evolutionary Applications 5:405-417.	12
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46	Stuart, K., and M. Drawbridge. 2012. The effect of photoperiod on larval culture performance of two marine finfish species. Aquaculture 360-361:54-57.	6
47	Jirsa D, Deng D, Davis DA, Wang W, Hung S, Drawbridge M. 2013. The effects of dietary lipid levels on performance and heat-shock protein response of juvenile White Seabass, *Atractoscion nobilis*. Aquacult Nutr 19(2):227-32.	12
48	Trushenski J, Mulligan B, Jirsa D, Drawbridge M. 2013. Sparing fish oil with soybean oil in feeds for White Seabass: Effects of inclusion rate and soybean oil composition. N Am J Aquacult 75(2):305-15.	17
49	Jirsa D, Salze GP, Barrows FT, Davis DA, Drawbridge M. 2013. First-limiting amino acids in soybean-based diets for White Seabass *Atractoscion nobilis*. Aquaculture 414:167-72.	2
50	Stuart, K., M. Losordo, P. Olin, and M. Drawbridge. 2013. Effects of stocking density and water conditioners on yolk-sac larvae of two marine finfish during simulated air transport. Aquaculture Research doi: 10.1111/are.12368	0
51	Gruenthal, K.M., B.J. Gauger, and M.A. Drawbridge. 2014. Maternal reproductive exhaustion in a broadcast spawning marine finfish cultured for conservation. Aquaculture 422-423:129-135.	0
52	Jirsa D, Davis DA, Barrows FT, Roy LA, Drawbridge M. 2014. Response of White Seabass to practical diets with varying levels of protein. N Am J Aquacult 76(1):24-7.	2
53	Jirsa D, Davis DA, Salze GP, Rhodes M, Drawbridge M. 2014. Taurine requirement for juvenile White Seabass (*Atractoscion nobilis*) fed soy-based diets. Aquaculture 422:36-41.	9
54	Trushenski JT, Rombenso AN, Page M, Jirsa D, Drawbridge M. 2014. Traditional and fermented soybean meals as ingredients in feeds for White Seabass and Yellowtail jack. N Am J Aquacult 76(4):312-22.	5
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56	Shen SG, Chen F, Schoppik DE, Checkley DM. 2016. Otolith size and the vestibulo-ocular reflex of larvae of White Seabass *Atractoscion nobilis* at high pCO_2. Mar Ecol Prog Series 553:173-183.	0
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2	Mahboudi, F. G. 1987. In vivo and in vitro immunization of White Seabass (*Atractoscion nobilis*) with Vibrio parahaemolyticus O-antigen. Masters Thesis, San Diego State University. 90 pp.	na
3	Orhun, M. R. 1989. Early life history of White Seabass (*Atractoscion nobilis*). Masters Thesis, San Diego State University. 162 pp.	na
4	Dutton, P. H. 1989. The feeding ecology and growth of White Seabass larvae (*Atractoscion nobilis*). Masters Thesis, San Diego State University. 146 pp.	na
5	Donohoe, C. J. 1990. The distribution, abundance, food habits, age and growth of late larval and early juvenile White Seabass *(Atractoscion nobilis*) off San Diego County, California. Masters Thesis, San Diego State University. 95 pp.	na

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7	True, C. D. 1994. Influence of different salinities and temperatures on the metabolism and osmoregulation in juvenile White Seabass, *Atractoscion nobilis*. Masters Thesis, Autonomous University of Baja California, Ensenada, Mexico (in Spanish). 62 pp.	na
8	Rudolph, J.D. 1995. Feeding and predator avoidance strategies of cultured White Seabass, *Atractoscion nobilis*. Masters Thesis, San Diego State University. 106 pp.	na
9	Shane, M. (1996). A study of the fish community in the La Jolla kelp forest off San Diego, California through video transects and gill net sampling. Masters Thesis, San Diego State University. 61 pp.	na
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11	Franklin, M. P. 1997. An investigation into the population structure of White Seabass (*Atractoscion nobilis*) in California and Mexican waters using microsatellite DNA analysis. Ph.D. dissertation, University of California Santa Barbara, Santa Barbara, CA.	na
12	Stransky, B.C. 1998. Assessment of sediment quality effects in Mission Bay and San Diego Bay on the growth, behavior and survival of juvenile White Seabass *(Atractoscion nobilis*) and juvenile California Halibut (*Paralichthys californicus*). Masters Thesis, San Diego State University. 287 pp.	na
13	Viveros, D.O. (1999). Ionic and osmotic regulation in juveniles of Totoaba macdonaldi following changes in salinity. Master's Thesis, Autonomous University of Baja California, Mexico. 67 pp.	na
14	Zimmerman, A. (1999). Skeletal muscle growth in the White Seabass (*Atractoscion nobilis*): in vitro myosatellite cell growth and in vivo muscle fiber development. Master's Thesis, University of San Diego. 238 pp.	na
15	Buhr, G. (2002). The effects of exercise conditioning on the growth and development of juvenile White Seabass (*Atractoscion nobilis*). Master's Thesis, University of San Diego.	na
16	Swaney, J. (2002). Effects of temperature and ration levels on growth of White Seabass, *Atractoscion nobilis*. Master's Thesis, University of San Diego. 97 pp.	na
17	Smiley, J. E. (2004). Effects of gas saturation levels on larval and juvenile White Seabass, *Atractoscion nobilis*. Master's Thesis, University of San Diego.	na
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19	Aalbers, S. (2005). Spawning Activity and Associated Sound Production in the White Seabass, *Atractoscion nobilis*. Master's Thesis, California State University, Fullerton.	na
20	Louie, L. (2005). Behavioral Comparisons Between wild and cultured juvenile California Halibut (*Paralichthys californicus*). Master's Thesis, University of San Diego.	na
21	Cepuritas, A. (2005). Exercise effects on growth rate, IGF activity, and cortisol in juvenile White Seabass. Master's Thesis, University of San Diego.	na
22	Coykendall, D. K. 2005. Population structure and dynamics of White Seabass (*Atractoscion nobilis*) and the genetic effect of hatchery supplementation on the wild population. Ph.D. dissertation,	na

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23	Oakes, C.T. 2007. The value of a mariculture net-cage as a fish aggregating device (FAD) in southern California. Masters Thesis, Occidental College, Los Angeles, CA. 70 pp.	na
24	Peters, C. (2009). Enhancement of growth rates and swimming performance in juvenile marine finfish in aquaculture. Master's Thesis, University of San Diego.	na
25	Gauger, B. (2010). Variations of egg and larval viability of captive White Seabass (*Atractoscion nobilis*) through a complete spawning season. Master's Thesis, University of San Diego.	na
26	Tardy, K. (2011). Functional morphology and swimming behavior in larval and juvenile White Seabass (*Atractoscion nobilis*). Master's Thesis, University of San Diego.	na
27	Velazquez, J.J. (2012). Effect of replacing fish oil with plant-based oils on the performance of juvenile White Seabass (*Atractoscion nobilis*). University Autonomous of Baja California, Ensenada, MX	na
28	Smith, E. (2015). Evaluation of reproductive strategies in captive California Yellowtail (*Seriola lalandi*) using genetic parentage analyses. Master's Thesis, University of San Diego.	na
29	Wrobleski, D. (in progress). Effect of dietary inclusion of Spirulina (*Arthrospira platensis*) on the growth performance, body composition, and hematology of juvenile White Seabass (*Atractoscion nobilis*) and California Yellowtail (Seriola lalandi). Master's Thesis, University of San Diego.	na
C.	**Publications with acknowledgements for specimens or data contributed as a result of the OREHP**	
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2	Schultz, E. T. 1993. The effect of birth date on fitness of female dwarf perch, *Micrometrus minimus* (Perciformes: Embiotocidae). Evolution 47:520-539	na
3	Williams, G.D., K.S. Andrews, S.L. Katz, M.L. Moser, N. Tolimieri, D.A. Farrer, and P.S. Levin. 2012. Scale and pattern of broadnose sevengill shark *Notorynchus cepedianus* movement in estuarine embayments. J Fish Biol. 80(5):1380-1400.	na

Appendix 2. Draft RFP to solicit proposals for a social and economic assessment of the OREHP.

Request for Proposals
Socioeconomic Analysis of the Ocean Resources Enhancement and Hatchery Program (OREHP)

Problem Statement/Background

General Introduction Information

World per capita fish consumption has more than doubled over the past 50 years, from 9.9 kg in the 1960s to 19.2 kg in 2012 (FAO 2014, p. 3). Driven in part by population growth, rising incomes, and urbanization, the increased demand for seafood has resulted in the depletion of some wild caught fish stocks around the world (FAO 2014, p. 7). Over the past five decades, efforts in aquaculture established to meet the rising demand for seafood have resulted in a steady 3.2% per year increase in the supply of farmed fish (FAO 2014, p. 3). Still, there is much work to be done toward meeting food demands and achieving stable populations of popular fishery species, and stock enhancement, "the release of cultured juveniles into wild population(s) to augment the natural supply of juveniles and optimize harvests by overcoming recruitment limitation" (Bell et al. 2008), is one strategy that can be used to close the gap (FAO 2014, p. 219).

Stock enhancement provides biological benefits by contributing to wild stock recovery, helping to protect endangered species, and enhancing scientific knowledge about the life history and environmental requirements of aquatic organisms (Leber et al. 2012). It also offers socioeconomic benefits by preserving fishing heritage and cultures, and providing opportunities for commercial and recreational fishing, helping to maintain the multi-billion dollar recreational fishing industry (Leber et al. 2012). Nevertheless, enhancement programs face important challenges. These include engaging stakeholders in the planning and execution of programs, determining habitat availability for enhanced species, ensuring that wild fish are not displaced by hatchery fish, monitoring survival rates of released fish, and conducting cost-benefit analyses (Lorenzen et al. 2010, SCORE 2014). Evaluating the social and economic benefits and costs of enhancement efforts is crucial for assessing the efficiency of current efforts, and planning for future programs (Lorenzen et al. 2010).

Introduction to the OREHP

The Ocean Resources Enhancement and Hatchery Program (OREHP) is an experimental hatchery program that investigates the economic and ecological feasibility of using cultured marine finfish to successfully enhance wild fish populations off the coast of California, south of a line extending due west from Point Arguello (CDFW 2010). The OREHP was started in 1983 by California State Legislature (FGC § 6590 et seq.) to address declines in numbers of desirable fish that were adversely affecting recreational and commercial fishing, and related industries. A special fund was established to support the research pertaining to propagation, rearing, and stocking. The fund is supplied by user fees placed on State recreational and commercial fishermen, who stand to directly benefit from the resurgence of depressed fisheries, and Federal matching funds including Sportfish Restoration Act account funds. While the OREHP

initially focused on California halibut (*Paralichthys californicus*) and White Seabass (*Atractoscion nobilis*), the focus shifted to primarily White Seabass in 1990, due to its depressed stock conditions and its higher value to recreational and commercial fishermen (CDFW 2010). An independent evaluation of the success of the OREHP in meeting its original objectives was conducted in 2015-2017, and a need for a current socio-economic analysis of the program was identified.

White Seabass Enhancement Program

White Seabass broodstock are collected between Point Conception, California and central Baja California, Mexico under supervision of Hubbs-SeaWorld Research Institute (HSWRI) staff (CDFW 2010). Surplus broodstock are kept in HSWRI's net pen at Santa Catalina Harbor, or at growout facilities, until they replace broodstock at the Leon Raymond Hubbard, Jr. Hatchery in Carlsbad that is owned and operated by HSWRI (CDFW 2010). At the hatchery, a total of 200 broodstock are maintained in four separate, temperature and photoperiod-controlled pools (CDFW 2010). Spawning and larval-rearing take place at the hatchery (CDFW 2010). When juveniles reach a size of 20 – 40 g (about 80 days post hatch or 10 cm length), they are tagged and transferred to concrete, flow-through raceways until they are 91-150 days post hatch. The raceways are flushed with water that comes from, and returns to, Agua Hedionda Lagoon (Drawbridge and Okihiro 2007, CDFW 2010). The fish may then be brought to one of thirteen volunteer-operated growout facilities or, if more than 20 cm long and/or if the growout facilities are full, the fish may be released directly from the hatchery (Drawbridge et al. 2007). Gill net surveys (since 1988), acoustic tracking (since 2002), and collection of adult seabass heads from recreational and commercial fishermen (since 2001) are three methods used to assess the success of restocking efforts (Drawbridge and Okihiro 2007).

Timeline of White Seabass Enhancement Program

October 1986	The first experimental release of more than 2,000 juvenile White Seabass took place in Mission Bay (San Diego, California). Fish were propagated and raised at HSWRI's Mission Bay laboratory.
March 1992	The first legal-sized, oxytetracycline-marked, hatchery-raised White Seabass was recaptured.
October 1995	The marine fish hatchery became operational, built on land donated by San Diego Gas & Electric Company (SDGE) on Agua Hedionda Lagoon in Carlsbad, CA; and funded as an environmental mitigation measure for the San Onofre Nuclear Generating Station (SONGS), owners of which include Southern California Edison, SDGE, and the cities of Anaheim and Riverside. Contributions for the construction of the hatchery also came from private, corporate, and foundation donors.
June 1999	The first legal-sized, coded-wire tagged, hatchery-raised White Seabass was recaptured.
2001	The first year more than 100,000 White Seabass were released in southern California waters.
October 2004	The one-millionth White Seabass was released.

June 2007	Oldest adult fish recovery (13.3 yr); The fish was released off Santa Barbara, CA in 1994 and recovered near Ventura, CA.
June 2008	One-hundredth legal-sized hatchery-raised White Seabass recaptured.
September 2010	A tagged fish was recovered from Monterey, CA that had been released at Dana Pt. in August 2000.
August 2013	A total of 2 million fish had been released since the beginning of the OREHP.
December 2016	To date, 199 adult and 1,772 juvenile White Seabass have been recaptured.

Over the course of the last three decades, the OREHP has resulted in best management practices for White Seabass spawning, rearing, and growout, and has advanced scientific knowledge of White Seabass biology and aquaculture. While much progress has been made toward achieving the OREHP's goal of assessing the ecological feasibility of releasing hatchery-reared White Seabass to restore wild populations, the socioeconomic impacts of the program remain less well known. In 2010, Hervas et al. developed models to assess the dispersal, growth, and survival rates of released White Seabass, quantitative measurements that can be applied to socioeconomic evaluations of the OREHP (Hervas et al. 2010). However, an economic model of the OREHP has not been published since 1989, when Botsford et al. conducted a bioeconomic evaluation of the cost per individual fish entering the fishery, and the value of each fish caught in the fishery (Botsford et al. 1989). While insightful, Botsford et al.'s evaluation is outdated, having been completed before the OREHP hatchery was constructed in 1994, and using data from other species to fill in the models. It is necessary to evaluate the current social and economic costs and benefits of this enhancement effort, so as to determine its current efficiency and efficacy, and to plan for the future.

Scope of Work (Different phases of the project, and their goals and deliverables)
The goal of the proposed project should be to assess the socioeconomic costs and benefits of the OREHP White Seabass Enhancement program, and to identify opportunities for improvement of socioeconomic benefit. The socioeconomic analysis process should be transparent and designed in a way that allows other enhancement programs to learn from and even adapt and duplicate it, for example through the development of a conceptual model or worksheet templates and instructions. The socioeconomic analysis may be completed in two parts, with social costs and benefits discrete from economic costs and benefits, or may be completed in an integrated manner, taking both social and economic costs and benefits into account within one model or framework. The final analysis may be purely quantitative, or may include elements of qualitative assessment. This work should be completed in three phases.

Phase I. Comparative case studies. Compile and synthesize examples of socioeconomic evaluations of enhancement programs based in different regions of the world with similar or relevant environmental and/or economic conditions to Southern California, including native species and potential stakeholders. The **goal** of this phase is to investigate the different models or frameworks used to evaluate the socioeconomic benefits and costs of hatchery programs, and to assess which methods, models, or portions of models, are most effective. Each case study should include:

1. *A description of the enhancement program itself*
    a. Its history and goals, reasons for its growth, and past and current obstacles to growth
    b. Its current social and economic impacts
    c. A discussion of similarities and contrasts to Southern California, including but not limited to native species present, transportation, workforce, energy costs, environmental conditions, existing industry, infrastructure, business climate, regulations, government support programs, marketing, research and development, along with relevance and applicability of the program's experience to Southern California
2. *An assessment of the methods used to evaluate the socioeconomic impacts of the program*
    a. Identification of who conducted the socioeconomic analysis, the year in which it was conducted, and what data was available at the time
    b. A description of the specific variables assessed, the methods or models used to evaluate these variables, and the type and effectiveness of the output information.
    c. Assessment of the model's or framework's effectiveness and applicability to the OREHP

Phase I Deliverable: Due 00 Month YEAR. The first deliverable will be a white paper (or publishable manuscript) with bibliography that presents a review and synthesis of existing, relevant enhancement socioeconomic analyses, and explicit recommendations for the types of models, frameworks, and variables needed for conducting a socioeconomic analysis of the White Seabass Enhancement Program. Case studies may include analyses of enhancement programs in South Carolina, Texas, Florida, or other Gulf States (Red Drum), Arkansas, Tennessee, or Colorado (Rainbow Trout), Alaska (salmon, crab), Washington State (lobster, geoduck, salmon), New Zealand, Canada, and/or Japan. The final recommended type of socioeconomic model and the method by which the White Seabass Enhancement Program will be assessed (including whether social and economic impacts will be kept separate or be integrated) will be decided in consultation between California DFW, HSWRI, and the contractor prior to commencing Phase II.

Phase II. Socioeconomic model/framework for the OREHP White Seabass enhancement. The **goal** of Phase II is to use the information garnered in Phase I to develop a socioeconomic model or framework for measuring the social and economic impacts of the OREHP White Seabass Enhancement Plan.

Regardless of whether an integrative or separate, a quantitative or combined quantitative-qualitative approach is taken, the analysis conducted should capture **direct**, **indirect**, and **induced** impacts of the OREHP. Inputs to a model (for e.g., Cost Benefit, Regional Input-Output, FEAM) may include, among other things: hatchery and growout related costs (operations, transportation, food, tanks, broodstock collection, fish survival rates), jobs and income generated by the hatchery program, value to recreational fishing (angler days, per day angler

expenditures, satisfaction per trip (both catch-rate related and non-catch-rate related)), value to commercial fishing (harvest volume, product mix, yield for product forms, ex-vessel value, first wholesale prices, labor cost, angler utility), environmental impacts (enhancement of wild populations, cost of possible environmental damages), educational value (volunteer growout facilities, classroom programs, graduate student work, knowledge of species biology and life history, contributions to BMPs for hatcheries and enhancement efforts), federal and state funding, and private funding (e.g., Hilborn 1998, USFWS 2005, WSC 2009, FOC 2010, Sheeran and Hesselgrave 2013, Camp et al. 2014). Outputs from the model may include, among other things: regional economic impact, net benefit or cost per individual fish entering the fishery, total angler retail expenditures, net economic value or consumer surplus, ex-vessel value, costs of hatchery production, income and employment generated by the OREHP, and total social utility (e.g. Radtke, et al. 2009, Sheeran and Hesselgrave 2013, Camp et al. 2014).

The contractor will be encouraged to hold workshops and conduct surveys to engage a broad range of stakeholders, and to determine stakeholders' perceptions of the OREHP White Seabass Enhancement Plan and any value they may associate with the program.

Phase II Deliverables: DUE 00 MONTH YEAR. The deliverables for this Phase will be a functional socioeconomic model or framework with instruction manual, and a report of the assumptions, sensitivities and results of the model run or the analysis completed using OREHP White Seabass enhancement data.

Phase III. Socioeconomic assessment. The **goal** of Phase III is to compile information from the first two phases of the project and both assess current practices and make recommendations to improve the White Seabass Enhancement Program and the OREHP in general. This information may also inform the establishment of new enhancement efforts or the expansion of already established enhancement efforts by helping stakeholders and agencies understand the social and economic costs, benefits, and impacts of one approach to stock enhancement.

Phase III Deliverables: DUE 00 MONTH YEAR. The deliverable for this Phase will be a white paper that summarizes the OREHP's socioeconomic costs and benefits, identifies gaps in information, and presents recommendations for increasing efficiency.

Timeline of work. Please provide a brief timeline for completion of major tasks or accomplishments within each Phase. Deadlines for each Phase are provided here.

a. Phase I by
b. Phase II by
c. Phase III by

Funding availability

Funding is available from… to be determined by CADFW and the OREAP.

Proposal Elements: What to Submit
FORMAT OF PROPOSAL, PAGE LIMIT, FONT, MARGINS
Previous experience with such analyses, resumes, team members/team manager, schedule, fee structure (estimated costs per phase, according to the proposed timeline)

Selection/Evaluation Process, Judging Criteria
25% Demonstration of knowledge and understanding of the project
25% Past experience on relevant and/or similar projects
25% Demonstration of capacity to complete all of the deliverables within the required timeline
25% Costs are reasonable and fall within available/projected funding levels

Terms and Conditions
6 Right to reject any proposal deemed unfit

Due date of proposal, how to submit it (address)
7 Date and time:
8 Submission instructions: Contact name, email address, phone

Contact information for questions
Contact name, email, phone, affiliation

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