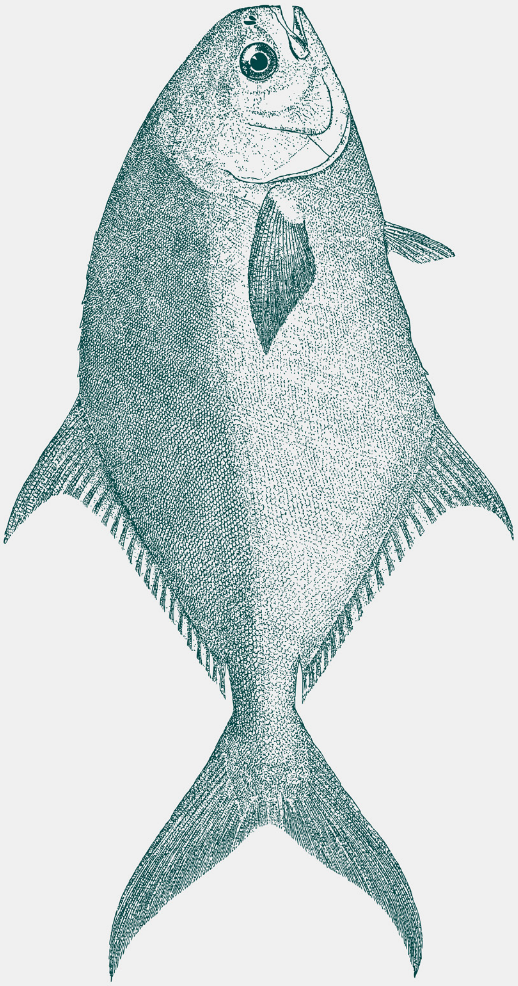
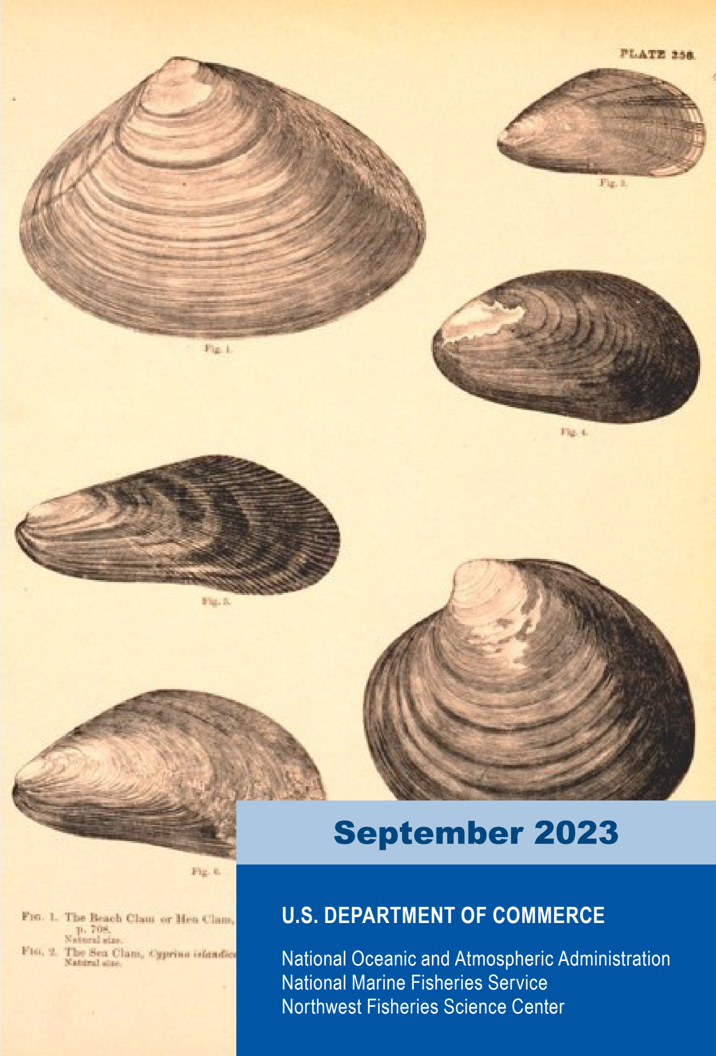
NOAA Technical Memorandum NMFS-NWFSC-186

https://doi.org/10.25923/55c9-ts52

Scientific Support for Health Management and Biosecurity for Marine Aquaculture in the United States





September 2023

U.S. DEPARTMENT OF COMMERCE

National Oceanic and Atmospheric Administration
National Marine Fisheries Service
Northwest Fisheries Science Center

NOAA Technical Memorandum Series NMFS-NWFSC

The Northwest Fisheries Science Center of NOAA's National Marine Fisheries Service uses the NOAA Technical Memorandum NMFS-NWFSC series to issue scientific and technical publications that have received thorough internal scientific review and editing. Reviews are transparent collegial reviews, not anonymous peer reviews. Documents within this series represent sound professional work and may be referenced in the formal scientific and technical literature.

The Northwest Fisheries Science Center's NOAA Technical Memorandum series continues the NMFS-F/NWC series established in 1970 by the Northwest and Alaska Fisheries Science Center, which subsequently was divided into the Northwest Fisheries Science Center and the Alaska Fisheries Science Center. The latter uses the NOAA Technical Memorandum NMFS-AFSC series.

NOAA Technical Memorandums NMFS-NWFSC are available from the NOAA Institutional Repository, https://repository.library.noaa.gov.

Cover images: (top) Drawing of sugar kelp. (bottom left) Drawing of Florida pompano.) (bottom right) Historical drawing of mussels and clams. See Image Credits page for details.

Reference this document as follows:

Rhodes, L. D., K. L. Parrish, and M. W. Rub. 2023. Scientific Support for Health Management and Biosecurity for Marine Aquaculture in the United States. U.S. Department of Commerce, NOAA Technical Memorandum NMFS-NWFSC-186.

https://doi.org/10.25923/55c9-ts52

Scientific Support for Health Management and Biosecurity for Marine Aquaculture in the United States

Linda D. Rhodes,[1*] Kellen L. Parrish,[1] and Michelle W. Rub[2]

https://doi.org/10.25923/55c9-ts52

September 2023

[1]Environmental and Fisheries Sciences Division
Northwest Fisheries Science Center
2725 Montlake Boulevard East
Seattle, Washington 98112

[2]Conservation Biology Division
Northwest Fisheries Science Center
2725 Montlake Boulevard East
Seattle, Washington 98112

[*]Retired

U.S. DEPARTMENT OF COMMERCE

National Oceanic and Atmospheric Administration
National Marine Fisheries Service
Northwest Fisheries Science Center

Contents

Figures

Tables

Image Credits

The cover drawings and decorative images that appear at the beginnings of major sections were sourced online and are either licensed for use or belong to the public domain.

Cover:

(top) © Liliya Butenko, Dreamstime ID 180703284. https://www.dreamstime.com/brown-algae-saccharina-latissima-sugar-kelp-realistic-drawing-illustration-encyclopedia-seas-oceans-isolated-image180703284

(bottom left) © Markusmayer1, Dreamstime ID 172521685. https://www.dreamstime.com/florida-pompano-fish-atlantic-ocean-side-view-trachinotus-carolinus-image172521685

(bottom right) Drawing by NOAA Fisheries. https://photolib.noaa.gov/Collections/Historic-Fisheries/Natural-History-of-Useful-Aquatic-Animals/Shellfish-and-Miscellaneous-Sea-Creatures/emodule/516/eitem/4711

Pathogen Transfer and Transmission:

(left) Male and female sea lice. Photograph © Sea Lice Research Centre, University of Bergen, Norway. http://slrc.w.uib.no/files/2016/03/BMC-_news_150206-1.jpg

(middle) Dissected fish containing many worms (*Lingula intestinalis*). Photograph © Craig Banner, Fish Pathogens Network. https://fishpathogens.net/sites/fishpathogens.net/files/images/cestode_Lingula_intestinalis_CB01.jpg

(right) Histological section of tissue showing embedded Ichthyophonus (fungus). Photograph © Bob Olsen, Fish Pathogens Network. https://fishpathogens.net/sites/fishpathogens.net/files/images/BO_Ichthyophonus_x20_38.jpg

Endemic Species of Concern in Each Region:

(top left) Black seabass. Drawing by NOAA Fisheries. https://media.fisheries.noaa.gov/styles/original/s3/2022-08/640x427-Black-Sea-Bass-NOAAFisheries.png

(top right) Red snapper. Drawing by NOAA Fisheries. https://www.fisheries.noaa.gov/s3/styles/original/s3/2022-09/640x427-Snapper-Red-NOAAFisheries.png

(bottom) California market squid. Drawing by NOAA Fisheries. https://www.fisheries.noaa.gov/s3/styles/original/s3/2022-08/640x427-Squid-California-Market-NOAAFisheries.png

Finfish Aquaculture Disease Factors:

(left) Shoal of greater amberjack in the Adriatic Sea. Photograph © Goran Šafarek, Dreamstime ID 201138355. https://www.dreamstime.com/shoal-greater-amberjack-adriatic-sea-croatia-image201138355

(middle) Juvenile sablefish. Photograph by NOAA Fisheries. https://media.fisheries.noaa.gov/styles/original/s3/2021-07/Young_sablefish_2-3.jpg?itok=fjI9UNxx

(right) Shoal of striped bass (*Morone saxitalis*). Photograph © Slowmotiongli, Dreamstime ID 195794369. https://www.dreamstime.com/striped-bass-morone-saxatilis-shoal-fishes-image195794369

Invertebrate Aquaculture Disease Factors:

(left) Tanks for incubating young shellfish (FLUPSY system). Photograph © California SeaGrant. https://caseagrant.ucsd.edu/sites/default/files/xFLUPSY_HumboldtBay.png.pagespeed.ic.Fy0tfPe22B.webp

(middle) Bay scallops on eelgrass. Photograph by NOAA, National Ocean Service. https://aambpublicoceanservice.blob.core.windows.net/oceanserviceprod/podcast/oct15/czma.jpg

(right) Oyster farmer holding tray full of fresh oysters. Photograph © Auremar, Dreamstime ID 102051134. https://www.dreamstime.com/oyster-farmer-holding-basket-full-fresh-oysters-oyster-farmer-holding-basket-full-fresh-oysters-image102051134

Seaweed/Macroalgae Aquaculture Disease Factors:

(left) Several types of seaweed grown in culture tanks. Photograph © SOLSEA Ltd. https://techpartnerships.noaa.gov/wp-content/uploads/2022/12/SOLSEA_02a.jpg

(middle) Salad containing kelp, red ginger, and sesame seed. Photograph © Liliia Kondratenko, Dreamstime ID 166986240. https://www.dreamstime.com/laminaria-salad-kelp-laminaria-salad-kelp-red-ginger-sesame-seeds-seafood-rich-iodine-minerals-healthy-eating-image166986240

(right) Underwater bull kelp sporophytes. Photograph by L. Rhodes, NOAA Fisheries.

Climate Change and Aquaculture:

(left) Floating net pens with service boat. Photograph © Vladislav Gajic, Dreamstime ID 21897529. https://www.dreamstime.com/royalty-free-stock-image-fish-farm-image20656136

(middle) Global ocean map displaying change in ocean heat content, 1993–2021. Image by NOAA Climate Program. http://www.climate.gov/media/14846

(right) Diagram showing changes in carbonate chemistry associated with changes in seawater pH. Image by NOAA, National Ocean Service, Pacific Marine Environmental Laboratory. https://www.pmel.noaa.gov/co2/files/noaa-oa-researchplan2020-2029.pdf

Federal Agencies with Responsibilities for Aquatic Animal Health, Medicine, and Water Quality:

(left) A variety of petri dish cultures in a microbiological laboratory. Photograph © Evgeniy Salov, Dreamstime ID 131289824. https://www.dreamstime.com/petri-dish-microbiological-laboratory-mold-fungal-cultures-bacterial-research-petri-dish-microbiological-laboratory-image131289824

(middle) Images of DNA sequences. Photograph © Anna Illarionova, Dreamstime ID 19070393. https://www.dreamstime.com/stock-photos-dna-sequence-electrophoresis-photo-image19070393

(right) A glass slide being placed on a microscope stage. Photograph © Anamaria Mejia, Dreamstime ID 103163885. https://www.dreamstime.com/young-male-scientist-looking-slides-under-microscope-male-scientist-looking-slides-under-microscope-image103163885

Disease Modeling:

(left) Diagram of hydrodynamic features used in modeling. Image by NOAA. https://nauticalcharts.noaa.gov/learn/images/hydrodynamic-model-development/modeling-landscape.jpg

(middle) Diagram of infectious disease states. Image from Gomez et al. (2021).[1] https://journals.plos.org/plosone/article/figure/image?size=large&id=10.1371/journal.pone.0245787.g001

(right) Diagram of ocean chemistry interactions. Image from Fennel et al. (2011).[2] https://bg.copernicus.org/articles/8/1881/2011/bg-8-1881-2011.pdf

Candidate Organisms for Marine Aquaculture:

(left) Three Pismo clams sitting on the beach. Photograph by California Department of Fish & Wildlife. https://centralca.cdn-anvilcms.net/media/images/2021/02/17/images/Cal_Outdoors_pismo_clams_pix.max-2400x1350.jpg

(middle) Drawing of a red drum. Drawing by D. R. Peebles, Florida Fish & Wildlife Conservation Commission. https://myfwc.com/media/13859/reddrum.jpg?format=webp&quality=65

(right) Giant kelp underwater. Photograph by C. Fackler, Channel Islands National Marine Sanctuary (NOAA). https://photolib.noaa.gov/Portals/0//GravityImages/35729/ProportionalFixedWidth/sanc006419074x800x800.jpg

---

[1] Gomez, J., J. Prieto, E. Leon, and A. Rodriguez. 2021. INFEKTA—An agent-based model for transmission of infectious diseases: The COVID-19 case in Bogotá, Colombia. PLOS ONE 16(2):e0245787.

[2] Fennel, K., R. Hetland, Y. Feng, and S. DiMarco. 2011. A coupled physical–biological model of the Northern Gulf of Mexico shelf: Model description, validation and analysis of phytoplankton variability. Biogeosciences 8:1881–1899.

Plain Language Summary

Although the United States produced 5.4% of the global marine capture fisheries, it also led all other nations in seafood imports in 2020, creating a gap between domestic consumption and production. Aquaculture, which is the farming of water-based organisms such as shellfish, finfish, and seaweed, can help to close that gap. Marine aquaculture production in the United States is expected to increase in the near future, especially into federally managed waters (generally >3 miles from shore). Diseases of aquatic organisms are recognized as a major limitation for aquaculture, requiring effective health management and biosecurity. Biosecurity includes planning and actions to protect against disease, primarily by preventing the introduction and spread of disease agents.

This document contains an overview of the available science for health management and biosecurity throughout the marine aquaculture industry for finfish, invertebrates (e.g., bivalves), and seaweed/algae (e.g., kelp). It describes factors for aquaculture diseases, discusses anticipated effects of climate change on aquaculture diseases, and identifies federal agencies with responsibilities for aquatic organism health. At this time, NOAA Fisheries is conducting assessments to identify areas suitable for commercial marine aquaculture in the Gulf of Mexico and the Southern California Bight, known as Aquaculture Opportunity Areas (AOAs). Information relevant to these regions is included throughout this document, such as important wild organisms that may occur in the AOAs, and region-specific climate change issues.

The intent of this document is to provide current scientific information on health management and biosecurity for marine aquaculture planning. However, it is not comprehensive, because knowledge about the subject is constantly evolving. It also does not contain detailed information about specific biosecurity procedures, which is presented in a complementary technical memorandum. Both documents are robust starting points for resources and planning, as well as timely sources of information as NOAA Fisheries considers the AOA options in the Gulf of Mexico and the Southern California Bight.

Links used in this section:

- Seafood imports in 2020: https://www.fao.org/documents/card/en/c/cc0461en
- Major limitation for aquaculture: https://onlinelibrary.wiley.com/doi/10.1111/jwas.12966
- Gulf of Mexico: https://coastalscience.noaa.gov/data_reports/an-aquaculture-opportunity-area-atlas-for-the-u-s-gulf-of-mexico/
- Southern California Bight: https://coastalscience.noaa.gov/data_reports/an-aquaculture-opportunity-area-atlas-for-the-southern-california-bight/
- Complementary technical memorandum: https://repository.library.noaa.gov/view/noaa/49079

Executive Summary

Although the United States produced 5.4% of the global marine capture fisheries (~4 million tonnes), it also led all other nations in seafood imports in 2020 (*1*). Among marine aquaculture sectors (finfish, shellfish, seaweed/macroalgae), the United States is only a significant producer of mollusks, where it ranks seventh at ~180,000 tonnes annually (*1*). Increasing marine aquaculture production in the United States is an approach to reducing import demand. Concurrent with increased production is a need for scientific support on health management and biosecurity to ensure production sustainability, to inform environmental consultations and appropriate site selection for aquaculture operations, and to protect both aquatic stocks and surrounding marine resources. Fortunately, there is an existing wealth of knowledge about aquaculture-based health management and biosecurity in peer-reviewed scientific literature and in current production practices and regulations. This technical memorandum contains an overview of the available science regarding health management and biosecurity throughout the marine aquaculture industry for finfish, invertebrates, and seaweed/macroalgae value chains. This report intends to provide understandable and citable information for non-specialists in marine aquaculture diseases and biosecurity, within the context of NOAA Fisheries' mission of responsible stewardship of U.S. ocean resources and habitats, and using sound science and an ecosystem-based management approach.[1]

At points throughout this document, there is information specific to two regions: the Gulf of Mexico and the Southern California Bight. Within each of these regions, NOAA Fisheries previously characterized areas of interest and study areas that included potential Aquaculture Opportunity Area (AOA) options. This information was published in two marine spatial planning atlases, collectively referred to as Aquaculture Atlases (*2*, *3*). The information included here that is specific to those two regions seeks to inform NOAA's ongoing assessment of the study areas.

This document introduces a basic conceptual model of factors affecting disease likelihood, namely host susceptibility, pathogen abundance or virulence, and environmental conditions that favor disease. Lists of pathogens and diseases of known concern for marine aquaculture, and brief descriptions of diseases with common prevention and management actions, are included in the appendices. To help understand how diseases are introduced and disseminated, pathogen transfer mechanisms are briefly described, including:

- Waterborne transmission.
- Physical contact between infected and susceptible individuals.
- Association with organisms (including feeds) that can carry pathogens, either as reservoirs or as intermediate hosts.
- Association with substrates and structures.
- Active movement by pathogens from infected to other susceptible individuals.

---

[1] https://www.fisheries.noaa.gov/about-us

The concepts of pathogen transfer from cultured to wild individuals and from wild to cultured individuals are also presented. Because proximity of endemic aquatic species to an aquaculture facility can increase the hazard of pathogen transfer, results of an analysis of geographic overlap of endemic species of concern (federally listed as endangered or threatened, commercially important) with the AOA study areas are presented, with data tables included in the appendices.

Although pathogen transmission routes are common for each aquaculture sector (i.e., finfish, shellfish, seaweed/macroalgae), requirements and practices for each sector are different. Health management and biosecurity topics specific to each sector are presented in separate sections. Because factors affect disease throughout the production cycle, these are presented sequentially (e.g., hatchery or nursery, transport, grow-out) for each sector.

Modern commercial finfish aquaculture formally began in the United States in 1853 with brook trout (*Salvelinus fontinalis*) in Ohio, and rearing salmonids for release to supplement commercial and recreational harvests became established in the 1870s (*4*). Information on marine finfish aquaculture in the United States is dominated by the culture of salmonids (*5*), and the wealth of information on biosecurity from that sector of the industry is often applied to other marine finfish species. However, there are significant differences between salmonid and non-salmonid aquaculture practices, such as differences in early-stage feeding strategies and differences in pathogen susceptibilities. Influent water security and reliability are essential during hatchery and nursery phases, and recirculating aquaculture systems are increasingly used for these life stages due to the feasibility and economics of managing the reduced volume of influent water. The hope of disease-resistant fish stocks remains mostly unrealized at this time. Transport and grow-out practices rely increasingly on better equipment (e.g., self-contained transport boats), operational planning for both routine and emergency events, and staff biosecurity training. Because marine finfish facilities attract and aggregate wildlife, which increases the potential for pathogen or disease transmission, a discussion about the causes, effects, and mitigations is included. Potential water chemistry changes from a finfish facility, such as dissolved nutrients and benthic deposition, are briefly described. An examination of the average depths for the AOA options in the Gulf of Mexico and Southern California indicate that most are deep enough to sustain low benthic impact from a farm.

The long history of molluskan aquaculture in the United States and current production scale implies that it may have more advanced procedures in place for disease management and biosecurity relative to finfish and seaweed/macroalgae. However, the open-environment nature of molluskan aquaculture makes it vulnerable to pathogen exposure, and using healthy seed is one of the best biosecurity measures. The United States does have some advanced planning, such as the Regional Shellfish Seed Biosecurity Program (RSSBP), developed to streamline animal movement through evaluations and audits of shellfish nurseries (*6*). Nonetheless, pathogens such as OsHV-1 (virus) and *Perkinsus* spp. (protozoan parasites) pose persistent threats to the industry. Hatchery and nursery operations require a secure incoming water supply, similar to the needs in finfish culture. Potential exposure to pathogens can be reduced through filtration, UV treatment (alone or following filtration), and by timing plantings and grow-out transfers for favorable temperature regimes or a lower likelihood of pathogen presence in seawater. Because invertebrates lack the adaptive immunity found in vertebrates, traditional vaccination is not possible. Currently, much of invertebrate health management relies on stress reduction through environmental optimization, including good seawater quality, density management, temperature management, and minimizing handling. Breeding for disease resistance is possible in shellfish, and genomic sequencing is likely to identify gene targets for selection.

Although commercial-scale seaweed/macroalgae aquaculture has been established in Asia since the 1950s, it is a relatively young industry in the United States and Europe, where the cultivated species are different from those reared in Asia. Knowledge of diseases for seaweed/macroalgae cultured in the United States and Europe is not as well developed as for finfish and invertebrates. Seaweed/macroalgae naturally carry other organisms, including other algal species, on their surfaces, and preventing introduction of non-native species is a biosecurity concern for broodstock and seedling selection and before outplanting. Unfavorable environmental conditions, such as temperature shocks, appear to be primary initiating factors for disease development. A genetic approach to identifying specific pathogens of seaweed/macroalgae is increasingly feasible with the expansion of molecular tools to characterize and identify beneficial and harmful microorganisms.

The effects of future climate change on aquaculture may be anticipated from observations of changes in natural settings and from controlled experiments. For the AOA areas of interest of the Gulf of Mexico and Southern California, International Panel on Climate Change Coupled Model Intercomparison Project 6 (IPCC CMIP6) models project that average sea surface temperatures will rise between 0.6°C and 0.8°C, that surface pH will decrease 0.1 units, and that dissolved oxygen and salinities will decline (*7*). Cultured species unable to move from unfavorable environmental conditions will experience higher physiological stress and changes in their associated beneficial microbial communities (their "microbiomes"), potentially increasing susceptibility to infection and disease. Temperature fluxes, such as marine heat waves, are expected to be a major driver to vulnerability to disease, because adjustment and adaptations can be more metabolically demanding. Changes in temperature and pH can independently and synergistically alter ocean chemistry, such as dissolved oxygen, dissolved nutrients, and bioavailable carbonate for calcification. Lower pH can reduce immune responsiveness in shellfish and finfish, and early life stages are especially vulnerable to negative effects. Climate changes are already altering the distribution of fisheries species, changing the endemic species composition and the potential for disease transmission near aquaculture facilities. There will also be impacts on indigenous pathogens, which can change their geographic distribution or ability to cause disease. For example, certain ectoparasites can tolerate a wider range of seawater pH than the hosts, possibly resulting in more severe infestations. In the two AOAs of interest, increased hypoxia (Gulf of Mexico) and ocean acidification (Southern California Bight) are already occurring and are expected to persist.

Animal health in aquaculture is managed by several federal agencies and, if located in state waters, by the state where a facility is located. The U.S. Department of Agriculture's Animal and Plant Health Inspection Service (USDA-APHIS) is the lead federal agency for animal and plant health, and it collects data for reportable diseases nationwide for submission to the World Organisation for Animal Health (WOAH). Other federal agencies involved in aquatic animal health are the U.S. Food and Drug Administration (FDA) Center for Veterinary Medicine and the U.S. Fish and Wildlife Service. The U.S. states in which facilities are located or to which organisms are transported also have health authority, defined by each state's legislative codes, regulations, and policies. Depending on the state, the authority can include required health inspection and reporting to issuing transfer permits. Drugs and biologics used in foodfish aquaculture are federally reviewed and approved for use with defined species under defined conditions, and sometimes for specific diseases. Concern around antimicrobial resistance is stimulating exploration of probiotics and prebiotics for invertebrates and finfish, although no approved commercial product is available at this time.

Improved environmental intelligence and increased computational capability make modeling a powerful tool for characterizing past disease events and for anticipating future disease scenarios. Currently, many aquatic disease models rely on hydrodynamic information to model dispersion through particle tracking, but the emergence of coupled physical–biological models permits the addition of situation-specific information, such as stocking density or pathogen extinction rates. As more models are developed and validated, their utility in data-poor scenarios becomes important for both prediction and for near real-time response.

This technical memorandum was generated in anticipation of expanded marine aquaculture in the United States, and it covers a range of topics related to marine aquaculture health management and biosecurity to provide a basis for understanding and planning. However, this document is not comprehensive, and our knowledge of the subject is constantly expanding and evolving. For clarity and conciseness, more detailed information about practical biosecurity procedures is not included in this document, but is contained in a complementary technical memorandum (*8*). In addition to the references cited here, there is a wealth of information and knowledge available from organizations such as the United Nations Food and Agriculture Organization and the World Organisation for Animal Health. The intent is that this document serves as a robust starting point for resources and planning, as well as a timely source of information as NOAA Fisheries considers AOA options in the Gulf of Mexico and the Southern California Bight.

Acknowledgments

Subject matter expert reviewers made a major contribution to the quality and accuracy of the information contained in this document. Like many experts, they have many demands on their intellect and time, so the authors deeply appreciate their frank and informed reviews. These reviewers include (in alphabetical order):

Marilyn "Guppy" Blair (U.S. Fish & Wildlife Service)
Colleen Burge (California Department of Fish & Wildlife)
Ken Cain (NOAA Fisheries)
William Fairgrieve (NOAA Fisheries)
Lori Gustafson (USDA Animal & Plant Health Inspection Service)
Kathleen Hartman (USDA Animal & Plant Health Inspection Service)
Melissa Karp (NOAA Office of Science & Technology)
Cicilia Kambey (Sea Six Energy)
Theodore Meyers (Alaska Department of Fish & Game)
Meghana Parikh (NOAA Fisheries)
Shane Ramee (U.S. Fish & Wildlife Service)
Andrew Richard (NOAA Office of Aquaculture)
Christopher Schillaci (NOAA Office of Aquaculture)
Pam Sponholtz (U.S. Fish & Wildlife Service)
Janet Whaley (NOAA Fisheries)
Gary Wikfors (NOAA Fisheries)
Diane Windham (NOAA Office of Aquaculture)
Marcus Zobrist (U.S. Environmental Protection Agency)

This document also benefited from input from unnamed experts, particularly the sections for federal agencies (e.g., the FDA's Center for Veterinary Medicine).

Finally, and importantly, we are indebted to the Northwest Fisheries Science Center's superb technical editor for scientific communications, Dr. Al Brown. He reads every word of these publications with a critical eye, ensures conformance to NOAA standards, and provides excellent logical and aesthetic guidance. His role is invaluable.

Glossary

Causative agent: A factor, such as a chemical or an organism, that is responsible for disease development.

Climate change: As defined by the United Nations, long-term shifts in temperatures and weather patterns.

Conspecific: Belonging to the same species, usually used in this context to describe cultured and wild animals that are taxonomically identical but will not necessarily be phenotypically identical.

Crustaceans: Organisms belonging to the subphylum Crustacea, which are commonly known as shrimp, crab, lobster, krill, copepods, and barnacles.

Decontamination: Reduction of a contaminant to a level assumed to be reasonably free of transmission risk. **Disinfection** and **sterilization** are forms of decontamination.

Disease: Clinical or non-clinical infection with one or more pathogenic agents (from WOAH Aquatic Code), characterized by specific signs or symptoms in which normal functions are disturbed or altered at a cellular, tissue, organ, or whole-organism level.

Disinfection: Elimination of most, but not necessarily all, infectious agents.

Drug: Any article intended for use in the diagnosis, cure, mitigation, treatment, or prevention of disease in man or other animals. Any article (other than food) intended to affect the structure or any function of the body of man or other animals (U.S. Code, Title 21, Chapter 9, Subchapter II, §321(g)(1).

Epibiont: An organism that lives on the surface of another organism.

Endemic species or population: Taxon or population that naturally occurs in a specified geographic area.

Etiologic agent: Substance known to or reasonably expected to contain a pathogen.

Facultative pathogen: An organism capable of causing disease but which can replicate independently from the host.

Finfish: Aquatic vertebrates breathing by gills throughout life and having limbs, if any, in the form of fins.

Fomite: Inanimate objects that can carry infectious agents and spread disease. Fomites can also be called passive vectors.

Global warming: As defined by the U.S. National Aeronautics and Space Administration, the long-term heating of Earth's surface observed since the pre-industrial period (1850–1900).

Holobiont: The collection of species that live in close and intimate contact with an organism.

Invertebrates: In the context of this document, refers to crustaceans and mollusks.

Landraces: Subspecies of an organism that are genetically identical to conspecifics in other areas, but have adapted to local conditions.

Mollusks: Organisms belonging to the phylum Mollusca, commonly known as clam, oyster, mussel, scallop, geoduck, chiton, abalone, snail, octopus, squid, and cuttlefish.

Obligate pathogen: A pathogen that requires a host to replicate and to complete its life cycle.

Ocean acidification: Refers to the long-term decrease in ocean pH, caused primarily by the ocean's uptake of carbon dioxide from the atmosphere.

Opportunistic pathogen: An organism that can be present normally without causing disease and can replicate independently from a host, but that causes disease following a change in the host's resistance due to stress and/or environmental change.

Parasite: An organism that lives on or within a second organism by obtaining nutrients from the second organism.

Pathobiome: Host-associated organisms (prokaryotes, eukaryotes, and viruses) causing reduced (or potentially reduced) health of the host from interactions between member organisms and the host.

Pathogen: An organism that can cause disease. Most pathogens are infectious.

Pathogenicity: The ability to cause disease.

Phenology: The study of cyclic and seasonal natural phenomena, especially in relation to climate and plant and animal life.

Seaweed/macroalgae: Visible, multicellular marine algae, classified into three major groups—red algae (Rhodophyta), brown algae (Phaeophyta), and green algae (Chlorophyta).

States: Entities that include states, tribes, and territories of the United States of America.

Sterilization: Removal of all microorganisms.

Shellfish: Invertebrate organisms including crustaceans (shrimp, prawn, crab, lobster), bivalve mollusks (oyster, clam, mussel, scallop), gastropod mollusks (abalone, snail, whelk, conch), and echinoderms (sea urchin, sea cucumber, octopus, squid). Cephalopods (e.g., squid, octopus) are taxonomically in the same phylum as these organisms, but not colloquially called shellfish.

Virulence: Severity or degree of harm or injury that a disease or pathogen can cause.

Wild: Not domesticated or cultivated.

Abbreviations

Abbreviations in this document are described with first use. Those used more than once are represented in this list.

AADAP	Aquatic Animal Drug Approval Partnership
AFS FHS	American Fisheries Society Fish Health Section
AMR	antimicrobial resistance
AOA	Aquaculture Opportunity Area
BOD	biological oxygen demand
CWA	Clean Water Act
DO	dissolved oxygen
DPS	distinct population segment
DSS	decision support system
EFH	essential fish habitat
EPA	U.S. Environmental Protection Agency
ESA	Endangered Species Act
FDA	U.S. Food and Drug Administration
FMP	Fishery Management Plan
FVCOM	Finite-Volume Community Ocean Model
HGT	horizontal gene transfer
HUA	high use area
HYCOM	Hybrid Coordinate Ocean Model
IMTA	integrated multi-trophic aquaculture
ISAV	infectious salmon anemia virus
ISSC	Interstate Shellfish Sanitation Conference
MAB	maximum allowable biomass
MARPOL	International Convention for the Prevention of Pollution from Ships
MSX	multinucleated sphere unknown
MUMS	Minor Use and Minor Species
NADA	new animal drug application
NAHPS	National Aquaculture Health Plan and Standards

NDZ	no-discharge zone
NPDES	National Pollutant Discharge Elimination System
OA	ocean acidification
OIE	Office International des Epizooties (now called World Organisation for Animal Health)
PAR	photosynthetically active radiation
PEIS	programmatic environmental impact statement
QPX	quahog parasite unknown
RAS	recirculating aquaculture system
RSSBP	Regional Shellfish Seed Biosecurity Program
SIR	susceptible–infected–recovered
SPF	specific pathogen-free
TMDL	total maximum daily load
USDA-APHIS	U.S. Department of Agriculture Animal and Plant Health Inspection Service
USFWS	U.S. Fish and Wildlife Service
WOAH	World Organisation for Animal Health
WS-RLO	withering syndrome by Rickettsiales-like organism

Introduction

The United States is a leader in wild capture fisheries, but it is also a leading importer of seafood, creating a gap between domestic consumption and production (*9*). Marine aquaculture production in the United States is anticipated to increase rapidly in the near future, which may help reduce the gap between demand and production. An expansion of marine aquaculture, especially into federally managed waters, supports a need for documented and updated scientific information on health management and biosecurity to inform safe, productive, and sustainable decisions and operations. Aquaculture diseases have long been recognized by the Food and Agriculture Organization of the United Nations (FAO) as a major limitation for global aquaculture (*10*), and the FAO is developing a framework and toolkit for a Progressive Management Pathway for Improving Aquaculture Biosecurity (*11*).

The goal of the current document is to identify health management and biosecurity considerations for marine aquaculture. This document addresses three types of organisms anticipated to be cultivated: finfish, invertebrates (mollusks and crustaceans), and seaweed/macroalgae. It synthesizes peer-reviewed literature; industry, government, and non-governmental organization reports; and subject matter expert opinions to provide relevant species, disease, and location information. This document is written to be readable by non-specialists in organism health and disease management who are engaged in planning for marine aquaculture. For clarity and conciseness, more detailed information about practical biosecurity procedures is not included in this document, but is instead presented in a complementary technical memorandum (*8*).

In addition to topics on marine aquaculture health management and biosecurity, specific information for the Gulf of Mexico and the Southern California Bight is included in this document. These two regions encompass locations where the National Oceanic and Atmospheric Administration (NOAA) is working to identify Aquaculture Opportunity Areas (AOAs) for marine aquaculture. Marine aquaculture planning atlases (hereafter called "Aquaculture Atlases") specific to each region were produced by NOAA's National Centers for Coastal Ocean Science (NCCOS; *2*, *3*). Within each region, areas of interest were identified using water depth and distance from shore requirements based on input from the marine industry and prior permit applications. For the Gulf of Mexico, stakeholders recommended a focus on water depths between 50 and 150 m, while distance from shore was not a constraint (*2*). For the Southern California Bight, stakeholders recommended a focus on water depths between 10 and 150 m, with a maximum distance from shore of 46 km (*3*). Using a rigorous marine spatial data inventory with more than 200 data layers, spatial suitability modeling, and extensive stakeholder knowledge, the Aquaculture Atlases identified 19 discrete locations, or AOA options, most suitable for aquaculture development in federal waters. Nine AOA options were identified in the Gulf of Mexico (depth range: 49.5–93.7 m), and ten AOA options were identified in the Southern California Bight (depth range: 23.1–154.6 m; *2*, *3*). The AOA options identified in the the Aquaculture Atlases are the geographic focus of this document.

Planning and implementing any aquaculture activity is a complex, multidisciplinary process. Protecting and managing the health of the cultured organisms is central to a successful operation, and can be challenging in systems that are open to ocean waters. This document presents topics of health management and biosecurity for marine aquaculture, specifically for diseases caused by infectious agents. There are a variety of health threats to cultured organisms from noninfectious sources that can require policies and actions for prevention and mitigation. These threats include: chemical pollution, such as oil spill toxics; harmful algal blooms, such as ichthyotoxic *Pseudochattonella* spp.; and increasing weather extremes related to climate change. Chemical pollution and harmful algal blooms are occasionally referenced in this document but are not a formal discussion topic. However, the likely impacts of climate change on infectious diseases are included in a separate section.

Infectious Disease Conceptual Model

Disease is a normal and integral part of life, and infectious diseases are a subset of ecological interactions involving two or more organisms. Infectious diseases require three elements: a susceptible host; a pathogen capable of infection; and an environment favorable to disease development (Figure 1). If any one of these elements is absent, disease will not occur. If one or more of the elements (circles) moves along the red arrow toward the center, the likelihood of disease increases. For example, when a host has a debilitating condition such as poor nutrition, susceptibility to infection rises, which increases disease likelihood. This conceptual model is beneficial for understanding the disease likelihood and for identifying preventative and mitigating actions. Disease and biosecurity management planning and actions are typically directed toward one of these elements to minimize or reduce disease risk.

This conceptual model is also useful for certain non-infectious diseases, where "pathogen" can be replaced by a causative agent, such as toxic chemicals.

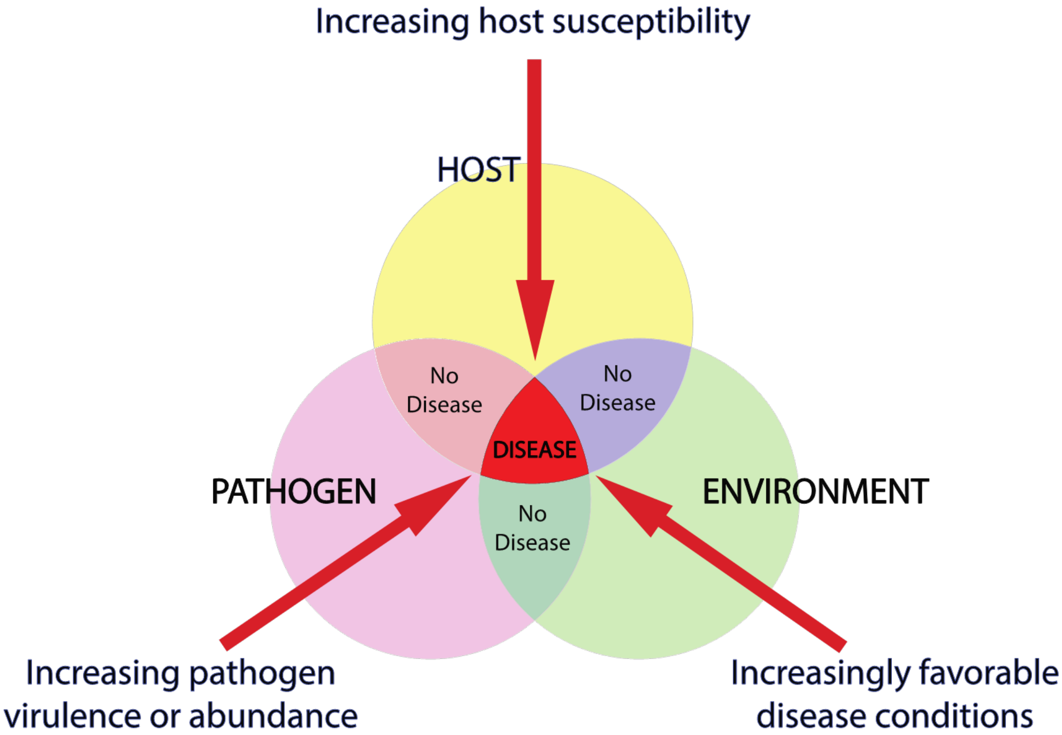


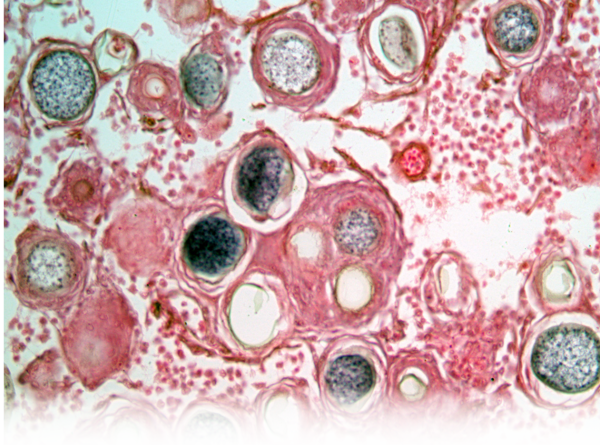
Figure 1. Conceptual model of infectious disease development. The Venn diagram displays the three elements required for disease occurrence as circles. The red arrows represent vectors that drive the elements toward greater likelihood of disease occurrence. Adapted from Snieszko 1974 (*12*).

Diseases and Pathogens of Concern for Marine Aquaculture

Pathogens are classified into three categories: viruses, bacteria, and parasites (e.g., protozoa, fungi, worms). Viruses are obligatory intracellular pathogens and are wholly dependent upon the host cell for persistence and replication. Viral genomes are typically devoted to sequences encoding only components of the virion itself, while the machinery required to replicate the viral particles is provided by the host cell. In contrast, bacteria can often survive and replicate independently of a host, and the overwhelming majority of bacteria that have been identified have no recognized pathogenic role. Furthermore, a significant number of pathogenic bacteria are considered opportunistic; that is, they do not depend upon a host for survival or replication, but are able to colonize a host that cannot limit or eliminate the bacteria, usually due to injury, stress, other illnesses, or a compromised immune system. Opportunistic bacterial infections are of particular concern in marine aquaculture because the etiological agents are often present in the natural environment, and exposure is difficult or impossible to limit or avoid. For example, *Aeromonas* bacterial species are commonly present in water columns and/or associated with invertebrates in the water column (*13*). Cultured sablefish develop severe skin ulcers and septicemia due to atypical *A. salmonicida* during periods of thermal stress, due in part to that strain's ability to grow at higher temperatures (*14*). Parasites can range in size and complexity from unicellular microorganisms (e.g., *Microsporidia*) to metazoans (e.g., sea lice). Although viruses and pathogenic bacteria are semantically also parasites (i.e., they derive existence at the expense of the host), this document considers parasites as pathogens other than viruses and bacteria.

Experiences from marine aquaculture globally have identified pathogens and diseases of concern, including those considered reportable by the World Organisation for Animal Health (WOAH, formerly the Office International des Epizooties or OIE). Appendix A contains diseases and associated pathogens in three tables by type of pathogen (virus, bacteria, parasite), the host (shellfish, finfish, seaweed/macroalgae), and whether the pathogen is reportable by WOAH (as of 2022).

While detailed descriptions of known diseases of concern are beyond the scope of this report, it is useful to provide information about the nature of these diseases to readers who are not disease specialists. Appendix B contains short, general descriptions of diseases and current management tactics for them. These narratives are intended to provide general information, and are not comprehensive or meant to be thorough discussions of the listed diseases.



Pathogen Transfer and Transmission

Awareness of the modes and mechanisms of pathogen transfer and transmission is fundamental to any discussion of the hazards of infection and likelihood of disease. Although some disease-causing agents have a high likelihood to infect a susceptible host if present, many others routinely occur on the host or in the environment without causing disease. This latter group, typically referred to as opportunistic pathogens, becomes pathogenic when host susceptibility increases or environmental conditions favor infection or pathogenicity. The following sections on transfer and transmission are applicable to infectious agents that are *not* opportunistic, and therefore, highly likely to infect or cause disease when present with a host.

Mechanisms for Pathogen Transfer and Transmission

Pathogen transfer and transmission can occur between susceptible hosts (known as horizontal transmission), or from broodstock to offspring through gametes (known as vertical transmission). Horizontal transmission includes direct and indirect transfer between susceptible hosts, and it is the principal pathway of pathogen spread. Common modes of horizontal transmission or transfer include:

- Waterborne transmission by release from an infected host into water and uptake directly from water by a susceptible host.
- Direct or physical contact between infected and susceptible individuals, including predation, ingestion of shed material such as feces or mucus, and feeds.
- Association with organisms that can carry pathogens, either as reservoirs or as intermediate hosts.
- Association with contaminated inanimate materials (fomites) such as substrates and structures (e.g., equipment, sediments) contaminated with a pathogen.
- Active pathogen movement from infected host to susceptible host (e.g., motile stages of parasitic copepods).

Vertical transmission is important during broodstock and seed procurement and during spawning. Pathogens can enter ova or eggs, infecting the developing zygote. Alternatively, pathogens can attach to the exterior of gametes or be associated with reproductive tissues, such as ovarian fluid in fish, to infect the emerging larva. Control of vertical transmission relies on quarantine and screening of broodstock and seed stock for specific pathogens, and surface decontamination (e.g., iodophor treatment of eggs). In the case of certain diseases, adults may be injected with therapeutic drugs, eggs from infected adults may be culled, or juvenile fish may be fed therapeutants at early feeding. Methods to control horizontal transmission of pathogens, including culling or segregation of diseased fish and therapeutant treatment, often are more flexible and diverse than methods for vertical transmission.

Waterborne transmission

Aquatic organisms can be infected by pathogens suspended in seawater through attachment and invasion of surfaces (e.g., gill, gastrointestinal tract, skin). This is the most common mode of natural aquatic virus transmission (e.g., *15*), and immersion is a common mode of exposure in laboratory studies. Immersion trials can determine the infectious dose of a virus, i.e., the concentration of pathogen for a specific time duration required to result in a specified percentage of infected organisms. Laboratory-determined infectious doses can be applied to natural or aquaculture settings, although real-world conditions will differ from the laboratory exposure and may affect potential for infection. Variables such as temperature, water chemistry, and strain-dependent variations in pathogen virulence can alter infection severity and likelihood of disease (Figure 2). Furthermore, the health status of the host organisms will alter the degree of susceptibility to a given dose. As a result, an understanding of hydrodynamic transport of the pathogen, in combination with pathogen survival or decay, can be used to estimate the probabilities of disease transfer between farms. This is becoming an important tool for planning farm locations (*16*–*18*).

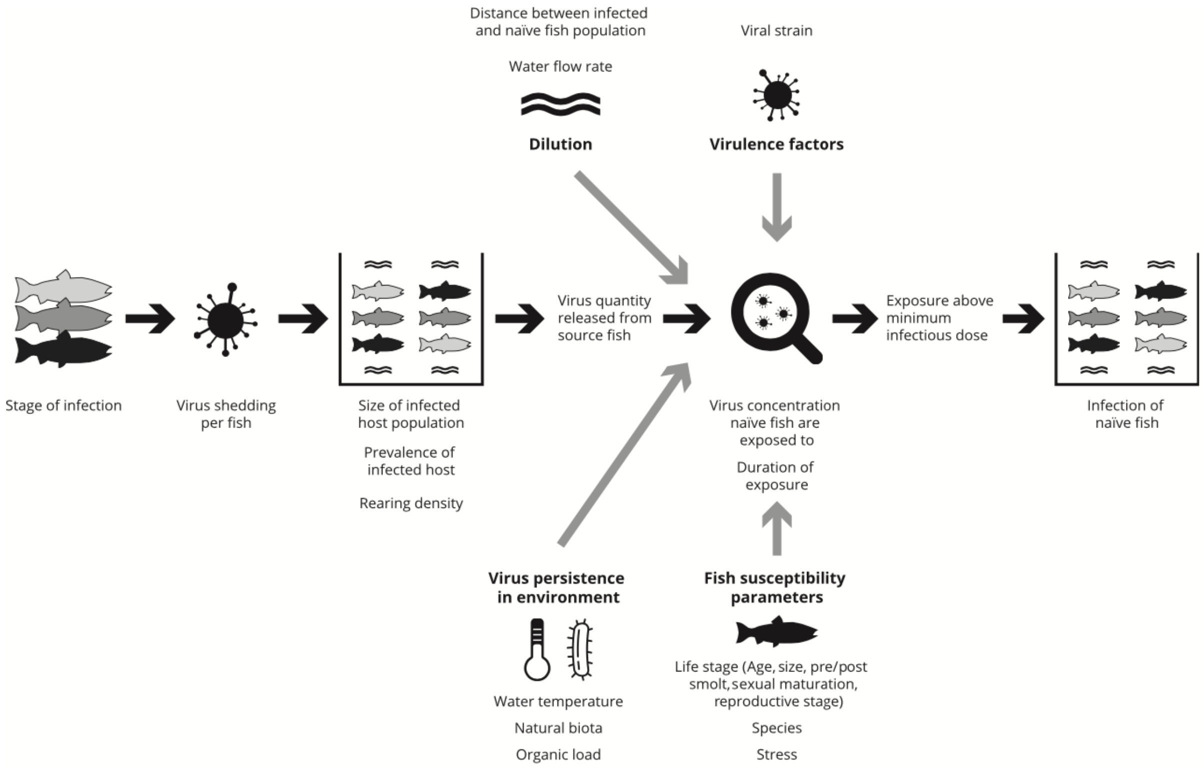


Figure 2. Summary of factors affecting water-borne transmission of fish viruses (from Oidtmann et al. 2018 [*15*], with permission).

In addition to hydrodynamic transport, waterborne transmission can be mediated by human transfer of seawater, most typically by ballast water or tank water (*19*, *20*).

Direct or physical contact between infected and susceptible individuals

Host species engage in a wide range of behaviors, including predation, that result in physical contact and provide opportunities for the direct transfer of pathogens. Trophic transfer of pathogens through predation is also an efficient unidirectional mode of transmission for some pathogens (e.g., *Ichthyophonus*, *21*), and it is common among parasites with a complex life cycle that require more than one host (e.g., *Toxoplasma gondii*, *22*). Organisms also shed biological materials such as gametes, mucus, and feces, which can contain pathogens; the potential for fecal–oral transmission is well documented in hatchery salmonids that have been observed ingesting fecal threads (*23*). Behaviors such as aggression or environmental factors (e.g., abrasives) that cause breaks in integument or surface epithelia create a direct entry route for pathogens (*24*). Loss of mucus, which contains antimicrobial peptides and enzymes, reduces protection against pathogens (*25*), and is the basis of concerns about finfish handling during hatchery manipulations and the impacts of catch-and-release programs.

Although all infected individuals are reservoirs for pathogens, not all may exhibit clinical signs of disease, support pathogen replication, or advance pathogen development. Asymptomatic reservoirs for aquatic pathogens can produce complex epidemiological patterns and confound disease management plans. Asymptomatic reservoir discovery can be accidental—from surveys conducted for other purposes—or via active surveillance for a specific pathogen across a broad spectrum of prospective hosts. Transmission vectors such as prey or predators also function as reservoirs. For example, planktivorous fishes such as herring or shad infected with *Ichthyophonus* can transfer the parasite to salmon that feed on them (*21*). Piscivorous birds such as herons, cormorants, and egrets carry viable pathogenic bacteria such as *Edwardsiella ictaluri* in their gastrointestinal tracts (*26*), and gulls can transmit viable viral particles after feeding on infected shrimp (*27*). Reservoirs that are naturally occurring around aquaculture operations could have a role in pathogen transmission if the aggregating features of the site, such as lights or elevated nutrient levels, attract vectors into proximity.

Association with substrates and structures

Substrates that can harbor pathogens, including aquaculture equipment (e.g., netting) and sediments, are considered fomites or inanimate reservoirs because they can be a source of pathogens. Unless treated or constructed to prevent biological attachments, these substrates will invariably contain living communities, such as biofilms and algal growth, which provide a suitable environment for many pathogens. Bacterial pathogens that are incorporated into biofilms may obtain nutrients from the extracellular matrices and protection against chemicals or antibiotics (*28*). Pathogens can have longer survival in organic-rich sediments, which receive biological fallout such as phytoplankton bloom deposits or feces (*28*). Biofouling of aquaculture facilities is a well known and expensive problem, beginning with biofilms and microorganisms and progressing to macrofouling organisms (*29*). Biofouling also occurs on shell exteriors in shellfish culture (*29*), and epibionts rapidly colonize the surfaces of cultured seaweeds/macroalgae (*30*).

Active pathogen movement from infected to susceptible host

Certain pathogens, particularly ectoparasites, actively move between hosts, resulting in direct transmission. Timing of release from one host for transfer to another is typically coupled to a life-history stage, such as appearance of a sexual maturation organ in *Gyrodactylus gasterostei* (*31*). Some species display active swimming behavior (*32*), while others appear to take advantage of water turbulence for passive transport (*31*). The prolific salmon ectoparasitic copepod, *Lepeophtheirus salmonis*, utilizes a range of sensory inputs for activating and targeting transfer, including visual, chemical, and mechanoreceptive information (e.g., *33*, *34*). In addition to causing disease directly, ectoparasites can act as vectors for bacteria and viruses, providing a secondary route of transmission for these infectious agents (*35*).

Pathogen Transfer Between Wild and Aquaculture Organisms

There are multiple potential disease transfer scenarios between wild and aquaculture organisms depending on species, system design, culture setting, husbandry operations, and other factors. Most offshore aquaculture systems will have free seawater interchange with few barriers to pathogen flow between the culture system and the open ocean.

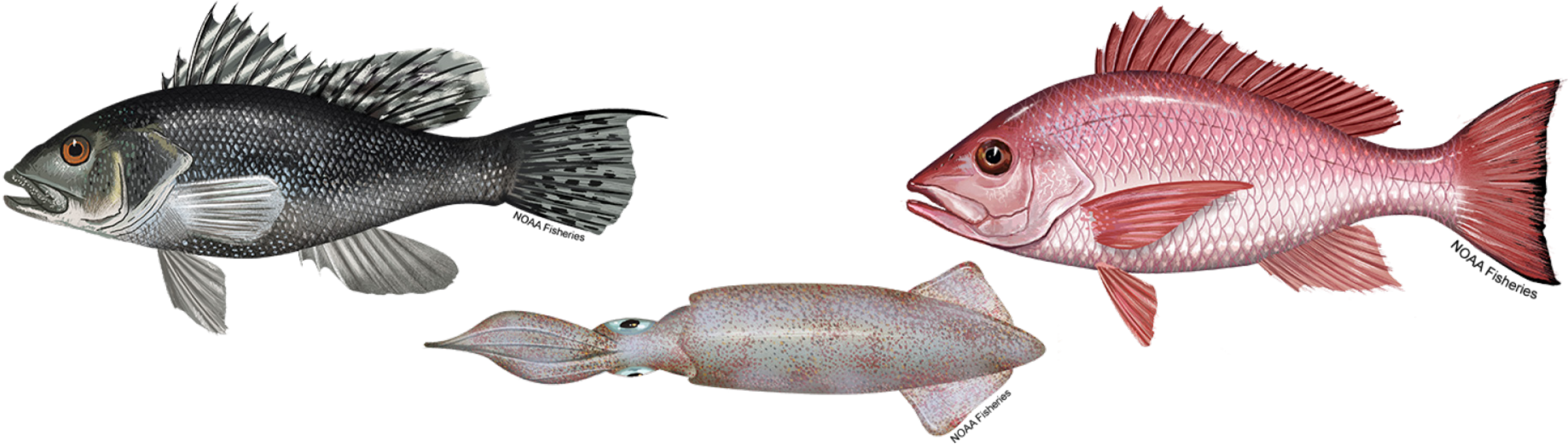
Tracing the direction of pathogen transmission is difficult, and many claims of disease transfer between wild and farmed populations are based on correlation or coincidence (*36*). A major barrier to assessing risk of transfer is the lack of data, especially for wild populations (*37*). Higher mutation rates in viruses present an opportunity to use strain evolution as a tool, allowing documentation of virus transfer from wild to domestic stocks and from domestic to wild stocks (*38*). Bacteria and parasites contain genetic markers such as plasmids, pathogenicity islands, and microsatellites, but these have had limited utility for tracing pathogen transmission.

Pathogens with broad host specificity (i.e., ability to infect hosts of different species) can increase the opportunity for pathogen transmission (*38*). The potential for direct contact or water-borne transmission may be enhanced when farms attract significant numbers of other fish, marine mammals, and/or seabirds that may act as vectors or reservoirs (*39–41*). Understanding transmission pathways requires knowledge of pathogen burden, effectiveness of farms in attracting fish and other aquatic animals, infectious particle transport, pathogen or parasite behavior, and oceanography specific to the location.

Expansion of aquaculture into new regions presents the opportunity for pathogen transmission between aquaculture and endemic species. Bouwmeester et al. (*42*) summarized five pathways for pathogen transfer between aquaculture populations and wild populations, which are summarized in Table 1.

Table 1. Brief descriptions of pathways for pathogen transfers between aquaculture and wild populations. “Intraspecific” and “interspecific” refer to host organisms. (Adapted from Bouwmeester et al. [2020; *42*].)

Pathway or Interaction	**Description**	**Example(s)**
Intraspecific or interspecific pathogen transfer from introduced to endemic populations (spillover).	Pathogen present in introduced species is transferred to the same host species (intraspecific) or to different host species (interspecific) in endemic populations.	Monogenean trematode (*Gyrodactylus salaris*) in Norway (*43–45*); parasitic copepod (*Mytilicola orientalis*; *46*).
Intraspecific or interspecific pathogen transfer from endemic to introduced populations (spillback).	Pathogen present in endemic species is transferred to the same host species (intraspecific) or to different host species (interspecific) in introduced populations. If the introduced population is cultivated at densities higher than endemic densities, infection levels can increase.	Copepod ectoparasite (*Caligus rogercresseyi*) and nematode (*Hysterothylacium aduncum*) in Chile (*47*).
Pathogen transmission interference or pathogen dilution.	Presence of cultured species or nearby wild species affects the transmission of wild pathogens, such as a sink for pathogens.	Pathogen removal by filter feeders (*48*); parasitic worm (*Acanthocephalus galaxii*) in New Zealand (*49*).



Endemic Species of Concern in Each Region

The transmission of disease or pathogens between wild and cultured organisms is an important concern for aquaculture in the Gulf of Mexico and the Southern California Bight. Because most of the mechanisms discussed in Pathogen Transfer and Transmission require spatial proximity of host organisms, spatial overlap between cultured species in AOA study areas and endemic species provides that opportunity. To begin an assessment of that overlap, endemic species of concern for each region were identified and are listed in the tables below. These tables include federally designated endangered or threatened species[1,2] and species that are commercially important.[3,4] Table 2 lists species for the Gulf of Mexico region, and Table 3 lists species for the Southern California Bight region. Although some species listed in these tables are more susceptible to pathogen transfer from aquaculture than others, we do not make that distinction here, as that would be part of the scoping process for individual aquaculture sites.

Spatial Overlap of Endemic and Farmed Species Within AOA Study Areas

The proximity of endemic species to aquaculture facilities presents a potential opportunity for pathogen transmission and transfer, and this type of information would be foundational for any risk assessment for specific proposed projects within the AOA study areas. The determination of essential fish habitat (EFH), High Use Areas (HUA), and Endangered Species Act (ESA) listed species habitat data by NOAA Fisheries permits spatial comparison with the AOA study areas to estimate the potential for proximity with endemic species. To document potential spatial overlap, the geospatial coordinates for the AOA study areas were compared to NOAA Fisheries' spatial distribution information for endemic species. While this analysis is limited to the AOA study areas within the Gulf of Mexico and the Southern California Bight, this approach could be applied to other locations where marine aquaculture is anticipated or planned.

---

[1] https://www.fisheries.noaa.gov/species-directory/threatened-endangered?oq=&field_species_categories_vocab=All&field_species_details_status=All&field_region_vocab=1000001126&items_per_page=100

[2] https://www.fisheries.noaa.gov/southeast/consultations/gulf-mexico

[3] https://www.fisheries.noaa.gov/region/west-coast#fisheries

[4] https://www.fisheries.noaa.gov/southeast/sustainable-fisheries-gulf-mexico

Table 2. Marine species in the Gulf of Mexico region that are federally designated as endangered or threatened, or that were found to represent important fisheries. Endangered or threatened species are marked with an asterisk, and commercially important species are in bold.

Mammals	Dwarf sperm whale* (*Kogia sima*)	False killer whale* (*Pseudorca crassidens*)	Fin whale* (*Balaenoptera physalus*)	Fraser's dolphin* (*Lagenodelphis hosei*)	Harbor porpoise* (*Phocoena phocoena*)
	Harbor seal* (*Phoca vitulina*)	Killer whale* (*Orcinus orca*)	Long-finned pilot whale* (*Globicephala melas*)	Melon-headed whale* (*Peponocephala electra*)	Minke whale* (*Balaenoptera acutorostrata*)
	North Atlantic right whale* (*Eubalaena glacialis*)	Pygmy killer whale* (*Feresa attenuata*)	Rice's whale* (*Balaenoptera ricei*)	Risso's dolphin* (*Grampus griseus*)	Rough-toothed dolphin* (*Steno bredanensis*)
	Sei whale* (*Balaenoptera borealis*)	Short-beaked common dolphin* (*Delphinus delphis*)	Short-finned pilot whale (*Globicephala macrorhynchus*)	Sperm whale* (*Physeter macrocephalus*)	Spinner dolphin* (*Stenella longirostris*)
	Striped dolphin* (*Stenella coeruleoalba*)	True's beaked whale* (*Mesoplodon mirus*)			
Fishes	**Almaco jack** (*Seriola rivoliana*)	**Atlantic bigeye tuna** (*Thunnus obesus*)	Atlantic common thresher shark (*Alopias vulpinus*)	Atlantic goliath grouper* (*Epinephelus itajara*)	**Atlantic mackerel** (*Scomber scombrus*)
	Atlantic menhaden (*Brevoortia tyrannus*)	Atlantic sharpnose shark (*Rhizoprionodon terraenovae*)	Atlantic shortfin mako shark (*Isurus oxyrinchus*)	**Atlantic skipjack tuna** (*Katsuwonus pelamis*)	Atlantic spiny dogfish (*Squalus acanthias*)
	Atlantic striped bass (*Morone saxatilis*)	**Atlantic wahoo** (*Acanthocybium solandri*)	**Atlantic yellowfin tuna** (*Thunnus albacares*)	Banded rudderfish (*Seriola zonata*)	**Black grouper** (*Mycteroperca bonaci*)
	Black sea bass (*Centropristis striata*)	Blackfin snapper (*Lutjanus buccanella*)	Blue catfish (*Ictalurus furcatus*)	**Blueline tilefish** (*Caulolatilus microps*)	**Butterfish** (*Peprilus triacanthus*)
	Cobia (*Rachycentron canadum*)	**Cubera snapper** (*Lutjanus cyanopterus*)	Dwarf seahorse* (*Hippocampus zosterae*)	Gag grouper (*Mycteroperca microlepis*)	Giant manta ray* (*Mobula birostris*)
	Golden tilefish (*Lopholatilus chamaeleonticeps*)	**Goldface tilefish** (*Caulolatilus chrysops*)	**Gray snapper** (*Lutjanus griseus*)	Gray triggerfish (*Balistes capriscus*)	**Greater amberjack** (*Seriola dumerili*)
	Gulf sturgeon* (*Acipenser oxyrhynchus desotoi*)	Hogfish (*Lachnolaimus maximus*)	**King mackerel** (*Scomberomorus cavalla*)	Lane snapper (*Lutjanus synagris*)	Lesser amberjack (*Seriola fasciata*)
	Monkfish (*Lophius americanus*)	**Mutton snapper** (*Lutjanus analis*)	Nassau grouper* (*Epinephelus striatu*)	**North Atlantic albacore tuna** (*Thunnus alalunga*)	**North Atlantic swordfish** (*Xiphias gladius*)
	Oceanic whitetip shark* (*Carcharhinus longimanus*)	**Opah** (*Lampris guttatus*)	**Queen snapper** (*Etelis oculatus*)	**Red grouper** (*Epinephelus morio*)	**Red snapper** (*Lutjanus campechanus*)
	Scalloped hammerhead shark* (*Sphyrna lewini*)	Scamp (*Mycteroperca phenax*)	**Scup** (*Stenotomus chrysops*)	Shortfin mako shark* (*Isurus oxyrinchus*)	Silk snapper (*Lutjanus vivanus*)
	Smalltooth sawfish* (*Pristis pectinata*)	Snowy grouper (*Hyporthodus niveatus*)	**Spanish mackerel** (*Scomberomorus maculatus*)	Speckled hind (*Epinephelus drummondhayi*)	**Summer flounder** (*Paralichthys dentatus*)
	Vermillion snapper (*Rhomboplites aurorubens*)	Warsaw grouper (*Hyporthodus nigritus*)	Wenchman (*Pristipomoides aquilonaris*)	**Western Atlantic bluefin tuna** (*Thunnus thynnus*)	**Winter flounder** (*Pseudopleuronectes americanus*)
	Winter skate (*Leucoraja ocellata*)	Wreckfish (*Polyprion americanus*)	Yellowedge grouper (*Hyporthodus flavolimbatus*)	Yellowmouth grouper (*Mycteroperca interstitialis*)	Yellowtail snapper (*Ocyurus chrysurus*)

Table 2 (continued). Marine species in the Gulf of Mexico region that are federally designated as endangered or threatened, or that were found to represent important fisheries. Endangered or threatened species are marked with an asterisk, and commercially important species are in bold.

Invertebrates	Atlantic surfclam[a] (*Spisula solidissima similis*)	**Blue crab** (*Callinectes sapidus*)	**Brown rock shrimp** (*Sicyonia brevirostris*)	Caribbean spiny lobster (*Panulirus argus*)	Hard clam/northern quahog (*Mercenaria mercenaria*)
	Ocean quahog (*Arctica islandica*)	**Pink shrimp** (*Pandalus borealis*)	**Stone crab** (*Menippe mercenaria*)	**White shrimp** (*Litopenaeus setiferus*)	Boulder star coral* (*Orbicella franksi*)
	Lobed star coral* (*Orbicella annularis*)	**Longfin squid** (*Doryteuthis pealeii*)	Mountainous star coral* (*Orbicella faveolata*)	Pillar coral* (*Dendrogyra cylindrus*)	Queen conch* (*Strombus gigas*)
	Rough cactus coral* (*Mycetophyllia ferax*)	Staghorn coral* (*Acropora cervicornis*)	Elkhorn coral* (*Acropora palmate*)		
Reptiles	Green sea turtle* (*Chelonia mydas*)	Hawksbill sea turtle* (*Eretmochelys imbricate*)	Kemp’s Ridley sea turtle* (*Lepidochelys kempii*)	Leatherback sea turtle* (*Dermochelys coriacea*)	Loggerhead sea turtle* (*Caretta caretta*)
	Olive Ridley turtle* (*Lepidochelys olivacea*)				

[a] Subspecies.

Table 3. Marine species in the Southern California Bight region that are federally designated as endangered or threatened, or that were found to represent important fisheries. Endangered or threatened species are marked with an asterisk, and commercially important species are in bold.

Mammals	Baird's beaked whale* (*Berardius bairdii*)	Blainville's beaked whale* (*Mesoplodon densirostris*)	Blue whale* (*Balaenoptera musculus*)	Bryde's whale* (*Balaenoptera brydei*)	California sea lion* (*Zalophus californianus*)
	Common bottlenose dolphin* (*Tursiops truncatus*)	Cuvier's beaked whale* (*Ziphius cavirostris*)	Dall's porpoise* (*Phocoenoides dalli*)	Dwarf sperm whale* (*Kogia sima*)	False killer whale* (*Pseudorca crassidens*)
	Fin whale* (*Balaenoptera physalus*)	Gray whale* (*Eschrichtius robustus*)	Harbor porpoise* (*Phocoena phocoena*)	Harbor seal* (*Phoca vitulina*)	Humpback whale* (*Megaptera novaeangliae*)
	Killer whale* (*Orcinus orca*)	Long-beaked common dolphin* (*Delphinus capensis*)	Melon-headed whale* (*Peponocephala electra*)	Minke whale* (*Balaenoptera acutorostrata*)	North Pacific right whale* (*Eubalaena japonica*)
	Northern elephant seal* (*Mirounga angustirostris*)	Northern right whale dolphin* (*Lissodelphis borealis*)	Sei whale* (*Balaenoptera borealis*)	Sperm whale* (*Physeter macrocephalus*)	
Fishes	Gulf grouper* (*Mycteroperca jordani*)	**Bocaccio** (*Sebastes paucispinis*)	Oceanic whitetip shark* (*Carcharhinus longimanus*)	Scalloped hammerhead shark* (*Sphyrna lewini*)	Steelhead trout* (*Oncorhynchus mykiss*)[a]
	Arrowtooth flounder (*Atheresthes stomias*)	**Canary rockfish** (*Sebastes pinniger*)	**Dover sole** (*Solea solea*)	**English sole** (*Parophrys vetulus*)	**Lingcod** (*Ophiodon elongatus*)
	North Pacific Swordfish (*Xiphias gladius*)	**Northern anchovy** (*Engraulis mordax*)	**Opah** (*Lampris guttatus*)		
Invertebrates	Black abalone (*Haliotis cracherodii*)	White abalone* (*Haliotis sorenseni*)	**Blue mussel** (*Mytilus edulis*)	**California market squid** (*Loligo opalescens*)	**Geoduck** (*Panopea generosa*)
Reptiles	Leatherback turtle* (*Dermochelys coriacea*)	Loggerhead turtle* (*Caretta caretta*)	Olive Ridley turtle* (*Lepidochelys olivacea*)		

[a] Southern California DPS.

Review overlap analysis

Building on the work in the Aquaculture Atlases that documented ESA-designated species habitat, High Use Areas (HUA), and essential fish habitat (EFH) in the AOA study areas (*2*, *3*), we conducted additional analyses of individual species overlap. The first analysis consisted of using the NOAA EFH mapper tool, fishery management plans, and EFH reports to determine whether an individual species occurred in the AOA study areas. The second analysis used spatial coordinates and weight-catch-per-unit-effort (wtCPUE, in kg/ha) data for individual species caught in fishery-independent bottom trawl surveys conducted in the Gulf of Mexico and on the West Coast (for the Southern California Bight). These data were obtained from the NOAA Fisheries Distribution Mapping and Analysis Portal (DisMAP). DisMAP data were analyzed to check for species overlap with the AOA high–high cluster areas. High–high clusters are GIS shape files that consist of multiple AOA option sites. Survey catch data were analyzed by plotting data points on a GIS map with the AOA study area and high-high cluster polygons to observe overlap with wild species (*50*). This catch distribution analysis is part of a larger spatial analysis manuscript in progress.

Methods for the overlap analyses are described in the appendices. Final EFH, HUA, or ESA overlap determinations are shown in Appendix C1 and Appendix C2. Additionally, Appendix D1 and Appendix D2 show final determinations of potential overlap based on the fishery-independent survey data (for clarity, data from the DisMAP tool are based on catches from trawl surveys and do not include data from commercial or recreational fisheries). Some species appear in both the C and D appendices. Some species in the appendices were mentioned in the Aquaculture Atlases but were found to have no likely overlap with study areas. Depending on the species, EFH and survey data may include one or multiple life stages. Further analysis is needed to determine the specific life stage in which a species is most likely to overlap with the AOA. Seaweed and macroalgae species are excluded from this analysis.

Gulf of Mexico

We identified EFH, HUA, and/or ESA protected areas for 25 species that overlapped with the AOA study area in the Gulf of Mexico. These data include finfish, sharks, and crustaceans. A full list of species evaluated via EFH, HUA, and ESA data is provided in Appendix C1. Catch-per-unit-effort data from the SEAMAP groundfish survey[5] for 35 species in the Gulf of Mexico were evaluated for overlap with AOA high–high cluster areas. Twenty species were shown to have points of overlap since 2008. Of these 20 species found to occur in high–high cluster areas, 19 were observed in clusters that contained AOA options. These data include finfish, shellfish, and crustaceans. A full list of the species evaluated via survey catch distribution data is provided in Appendix D1.

---

[5] Raw survey data can be downloaded at https://seamap.gsmfc.org/. However, the processed data used for this analysis were obtained from the NOAA Fisheries Distribution Mapping Analysis Portal (DisMAP), v. 20220516.

The Aquaculture Atlas for the Gulf of Mexico mentions that the study areas have potential overlap with green sea turtles, multiple shark species, rays, shrimp, and multiple pelagic and reef fish species (*2*, *51*).

Southern California Bight

We identified EFH, HUA, and/or ESA protected areas for 20 species that overlapped with the AOA study areas in the Southern California Bight, including three species for which potential overlap was unsure. These species include finfish, sharks, crustaceans, and mollusks. A full list of species evaluated via EFH, HUA, and ESA data is provided in Appendix C2. Catch-per-unit-effort data from the annual U.S. West Coast Groundfish Bottom Trawl Survey[6] for 17 species in the Southern California Bight were evaluated for overlap with AOA high–high cluster areas. Eleven species showed points of overlap since 2003. Of these 11 species found to occur in high–high cluster areas, all were observed in clusters that contained AOA options. These data include finfish, shellfish, and crustaceans. A full list of the species evaluated via catch distribution data is provided in Appendix D2.

The Aquaculture Atlas for the Southern California Bight mentions that the study areas have potential overlap with multiple sea turtle species, manta rays, multiple shark species, some highly migratory pelagic species, and multiple cetaceans. However, many of these species are seasonally and sporadically present (*3*, *52*, *53*).

Review of spatial analysis

Of the species reviewed, many appear to have some overlap with the AOA study areas and the more spatially limited high–high clusters. The extent to which these overlapping species present a risk for disease transfer to or from farmed organisms is beyond the scope of this document. Of the candidate species for aquaculture (Table 2), survey CPUE data were available for five in DisMAP:[7] sablefish, California halibut, southern flounder, greater amberjack, and black sea bass. Sablefish (*Anoplopoma fimbria*) were documented nearby and may require additional study given their status as a species with aquaculture potential.[8] California halibut (*Paralichthys californicus*) were not observed to be in the high–high clusters, with a population that appears to be distributed north of the Southern California Bight study area. Southern flounder (*Paralichthys lethostigma*) was not observed as overlapping with the high–high clusters, but qualitative observations did place some points near the 10-km radius established to check nearby occurrences. Greater amberjack (*Seriola dumerili*) was not observed in the high–high clusters in the Gulf of Mexico. Finally, black sea bass (*Centropristis striata*) was not documented in the high–high clusters, with observations that appeared to concentrate nearer to shore along the western coast of Florida.

---

[6] Raw survey data can be downloaded at https://www.webapps.nwfsc.noaa.gov/data/map. However, the processed data used for this analysis were obtained from the NOAA Fisheries Distribution Mapping Analysis Portal (DisMAP), v. 20220516.

[7] https://apps-st.fisheries.noaa.gov/dismap/DisMAP.html

[8] https://www.fisheries.noaa.gov/west-coast/aquaculture/sablefish-aquaculture-pacific-northwest

In the most recent (2023) update to the DisMAP tool, multiple species were removed because their occurrences in the trawl surveys were deemed too infrequent to provide robust estimations of abundance.[9] Although California halibut, greater amberjack, and black sea bass were not included in the latest DisMAP version, they were retained in this overlap analysis using data from a 2022 DisMAP version (20220516) because they do occur in the AOA study areas.

While this analysis can provide a baseline for potential species overlap, there are limits to what the current data can tell us about the distribution of wild species near AOAs, high–high clusters, and AOA options. The absence of a species in an area does not necessarily mean it does not, or could not, occur in that area. The DisMAP data were collected via bottom trawling, potentially excluding pelagic species in favor of demersal species. Additional commercial and recreational fishery coordinate data acquired via different methods would significantly aid further analysis and refine our understanding of the distribution of relevant commercial species. Finally, additional statistical processing of these data can help estimate species abundance beyond the data points used for this analysis; as stated, this research is part of another ongoing manuscript in which the authors expect to build on the data presented in this document.

This spatial analysis of endemic species in the AOA study areas helps to determine potential interactions between wild and cultured animals by identifying species that are more likely to occur near marine aquaculture facilities. Because interactions between wild and cultured species present an opportunity for pathogen transfer, this kind of information can inform future disease risk assessments for specific aquaculture projects that are proposed for marine waters. It can also serve as a basis for monitoring wild species for impact assessment or as potential pathogen sources to aquaculture activities. Because a spatial analysis can focus assessment efforts on relevant endemic species, it can be considered a fundamental activity for future specific projects.

Application to other geographic regions

Because NOAA collects fisheries data from the entire U.S. coastline, including Alaska and the Pacific Islands, this analysis can be performed regionally and at different scales. Any region under consideration for marine aquaculture can develop a catalog of endemic organisms that may cohabitate with proposed aquaculture projects. Existing distribution data can serve as a baseline for subsequent analysis of a marine aquaculture facility's potential impact on behavior and distribution of wild populations as one of the variables (e.g., climate change, food distribution, fishing pressure) that impact species distribution. Aquaculture facilities, and the tendency of wild species to aggregate around them, may also affect local species distributions. NOAA Fisheries' development of multispecies, regional tools such as DisMAP provides a replicable, data-based framework for analysis across different geographies.

---

[9] Species were kept for display in the portal if they were caught in at least 5% of tows in a given year for at least 75% of the survey years in a given region. Therefore, many species were removed in the update. Data from the earlier trawl surveys that include these organisms are available upon request from dismap.contact@noaa.gov.

Finfish Aquaculture Disease Factors

Although culture of marine finfish might be considered a recent enterprise, fishponds in Hawai'i have existed for at least 800 years (*54*, *55*), and there are contemporary efforts to re-establish them there.[10,11] Fishponds have elements of integrated multitrophic aquaculture, including cohabiting predatory fish that can remove weak, diseased, or invasive fish (*56*). Because modern finfish aquaculture is principally monospecific, active health and disease management must be exerted by the culturists. The following discussion on vertebrate aquaculture centers on marine finfish species reared in net pens. Health and biosecurity concerns occur at many points between spawning and harvest. Most species reared in net pens undergo a hatchery phase at a land-based facility before stocking on marine-sited farms. Principal pathogen introduction pathways include: introduction and health status of broodstock, influent water source, cross-contamination within a hatchery or nursery, biosecurity failure during transfer of animals to and from net pens (e.g., fish escapes, discharge of contaminated materials), and the free exchange of water through net pens (see *8*, their Figure 2). The following sections address concerns throughout the marine finfish production cycle.

Factors Associated with Hatchery or Nursery Activities

Many aquaculture organisms are likely to begin life in a hatchery or nursery operation on land or in the nearshore zone. Land-based hatcheries help manage the demands of early life-history stages where transitions in feeding strategies (often coupled with prey size) and nutrient demands (for organismal development) require significant human interactions. For example, several larval marine fish species require live feeds such as rotifers and *Artemia* spp. that require daily attention (*57*–*60*). This phase of the culturing cycle may be a point of entry for pathogens into the reared population.

To produce animals for the nursery phase, wild broodstock may, depending on the species, be captured for breeding or sourced from an established breeding program (*60*–*63*). Gametes and eggs (fertilized or unfertilized) may introduce pathogens into the culture facility if brought in from the wild or from external sources (e.g., out-of-area producers). Quarantine with pathogen screening, or obtaining from verified pathogen-free sources (e.g., specific pathogen free (SPF) stocks), are effective measures for minimizing this risk (*64*). However, SPF finfish have been confirmed only for laboratory fish such as zebrafish (*65*), not for commercial

---

[10] https://paepaeoheeia.org/
[11] https://www.seagardens.net/

finfish. Screening broodstock for the presence of specific pathogens—especially for those that are vertically transmitted, such as infectious hematopoietic necrosis virus—is an important biosecurity measure employed by Washington state and tribal salmon hatcheries for egg selection (*66*). The National Aquaculture Health Plan and Standards (*67*) states that rearing facilities should be stocked only with organisms of known health status, and requires hatcheries to participate in health inspection standards. The U.S. Fish and Wildlife Service has clear and explicit procedures for inspecting, testing, and managing incoming broodstock, including a substantial section on isolation and quarantine in their *Handbook of Aquatic Animal Health Procedures and Protocols* (*68*). The American Fisheries Society's Fish Health Section (AFS FHS) publishes a fish health manual known as the "Blue Book" outlining health inspection and diagnostic methods for fish health.[12] Because Blue Book protocols are curated by the largest organization of fish health professionals in the United States, they are widely adopted by government and industry entities for fish health management.

Hatchery or nursery water is another potential route of pathogen introduction into the culture cycle. Hatcheries for anadromous species such as salmonids are often located on rivers and streams in proximity to spawning grounds, where infected adult fish can contaminate incoming water. Use of well water for rearing eggs and larvae has been a favored alternative, although fish are often transferred to surface water at the parr stage, due to water volume requirements. For flow-through hatcheries and nurseries, managing water biosecurity is a major task. Untreated or inadequately treated intake water poses the obvious risk of introducing pathogens already present in the water source, especially if that source contains wild or free-ranging populations of the cultured or other species. Treating water (e.g., filtration, ultraviolet irradiation) can be expensive, and if the flow-through volume is large (e.g., >3,000 gpm), maintenance associated with filter clogging or adequate ultraviolet exposure (typically 30–40 mJ/cm^2) may not be feasible. For recirculating aquaculture system (RAS) hatcheries, influent water volume is 5–10% of a flow-through system (*69*), with a reduced pathogen burden from replacement water and reduced disease issues for RAS hatcheries (*59*). This level of water quality control is making RAS production an emerging best practice for marine aquaculture hatcheries.

A genetic approach to reducing pathogen impact is the development and use of disease-resistant strains of animals (*70*). The obvious advantage is that protection can be conferred not only at the early stages, but throughout the lifespan of the organism. Selective breeding is well established in agriculture and aquaculture, and it is feasible for species with shorter reproductive cycles (*64*). However, resistance for one pathogen may not necessarily provide resistance to another pathogen (*64*).

Factors Associated with Transport

Marine aquaculture involves the transport of organisms, personnel, equipment, and supplies between land and the marine sites, as well as between sites. This anthropogenic activity poses the potential for release of pathogens along the route and transmission of pathogens between locations. The likelihood of pathogen transfer is higher when moving fish or harvested product, rather than other types of aquaculture activities (*20*).

---

[12] https://units.fisheries.org/fhs/fish-health-section-blue-book-2020/

One of the greatest concerns around animal transport is the use of well boats and boats with significant storage to transport fish and equipment. These boats are often too costly to dedicate to each farm site, and they may visit multiple farms in a day (*71*). During transfer between farms, from nursery to farm, or from farm to processing plant, there are several points at which disease-inducing agents might transfer between animals or into the environment. Older versions of the boats are difficult to disinfect between trips to farms due to vessel design, but newer versions are easier and less cumbersome to disinfect as a regular part of operations; newer boats have designs specifically for marine aquaculture purposes, with closed holding areas and equipment for disinfection (*71*). Ballast water in watercraft servicing aquaculture facilities is also a concern, and Norwegian salmon operations recommend that ballast water not be taken in or discharged by watercraft within 5 km of any marine aquaculture facility (*71*).

In response to epidemic events of infectious salmon anemia (ISA) among Atlantic salmon production in the United States, the U.S. Department of Agriculture's Animal and Plant Health Inspection Service (USDA-APHIS) developed the Maine Infectious Salmon Anemia Virus (ISAV) Control Program Standards (*72*) for minimizing and mitigating spread of the virus, including detailed procedures for transport biosecurity. Farms are classified by infection status (ranging from negative to diseased; *72*, Part IV.A and their Table 2), and the level of cleaning and disinfection of personnel gear, equipment, vessels, and wharves is determined by infection status classification and travel destination (see *72*, their Appendix A). These procedures, in combination with other elements of the Maine ISAV Control Program Standards such as surveillance, biosecurity audits, quarantine, and depopulation, successfully suppressed ISA outbreaks by February 2006, and no further disease events have occurred since.

Factors Associated with Finfish Grow-Out

Finfish grow-out practices to maintain healthy stocks are well developed, such as maintaining fish densities that minimize stress, single year-class stocking, regular health inspections, and synchronized fallowing (*8*, *72*). Finfish farms do deposit nutrients into the surrounding environment in the form of uneaten feed and fish excretions. However, the quantity and impact are subject to geographic location, seasonal changes, stocking density, husbandry practices, currents, and other oceanographic factors (*73*, *74*). Primary production, stimulated by animal waste and uneaten feed, can contribute to aggregation of wild species near aquaculture facilities (*75*), with trophic progression to planktivorous species and larger predators including finfish, marine mammals, and sea birds. However, research thus far has not made strong connections between fish farms and major shifts in primary production that cause substantial impacts to other trophic levels (*73*). Facilities are also wild animal aggregation sites due to their structural presence, and may even be preferred given their combination of protective structure and both direct and indirect food availability (*75*).

The size of the aquaculture facility usually reflects the biomass of the cultured animal population. Research and observation from the Norwegian salmon industry indicates that surveillance of cultured populations may be more difficult in larger operations and may increase time to detection of a disease situation (*71*). Daily health checks are best for rapidly detecting disease events, although it may increase operational costs, and a cost–benefit analysis for biosecurity preparedness can help establish effective surveillance (*11*).

Secure management of fish carcasses due to mortalities is critical for limiting release of any possible pathogen and for reducing attraction of predators such as marine mammals and piscivorous fish. Carcasses resulting from grow-out mortality are important for diagnosing cause of death and for disease surveillance (*76*), and should be handled as presumptive infected material. Biosecurity measures include non-leaking carcass containers for transport to shore, disinfection of gear in contact with carcasses, and avoiding transport of carcasses between farm sites (*77*). Disposal of carcasses and offal directly into seawater is not permissible under National Pollutant Discharge Elimination System (NPDES) discharge permits (e.g., *78*–*80*).

Aggregation of wild organisms at farms and disease transfer

Causes of aggregation attraction

Aggregations of predatory marine mammals, seabirds, and finfish are observed near marine aquaculture farms worldwide (*39*, *81*), presumably representing only a subset of interactions between cultured and wild species. Long-term observations (99 months) show that there can be many populations of wild species near or interacting with the aquaculture species (*39*). This proximity between cultured and wild organisms poses a hazard of pathogen transfer; facilities that attract and aggregate wild species can increase that hazard. Aggregating cues include predation opportunities, physical protection by floating structures, and conditioned responses to sounds, lights, and farm operations. Fish farms offer predators alternative food sources that could become a more attractive option than hunting wild prey over wide spatial ranges.

Marine mammal and bird aggregation

Observational research suggests that marine mammals and birds exhibit changes in behavior in the presence of marine fish farms (*39*, *41*). Over multiple years, bottlenose dolphins were often observed foraging directly next to farms (*41*). Any impacts on bottlenose dolphin health as related to fish farms have yet to be substantially researched and documented.

Seabirds near aquaculture farms can be a biosecurity hazard. Meta-analysis of bird abundances shows elevated bird species richness at aquaculture facilities (*75*). Observational studies suggest that wholly piscivorous marine birds, notably cormorants or shags, are prolific in preying on both cultured species and aggregated species near farms (*39*). Similar to marine mammals, seabirds likely habituate toward regular visits to ocean farms (*75*). Some birds, such as gulls, can interfere with fish feeding operations and cause sufficiently stressed fish to stop feeding (*39*).

Birds also carry bacterial pathogens in their gut and on their feet (*26*), and are intermediate or definitive hosts to numerous cestodes, nematodes, trematodes, and other parasites (*82*). For example, greater amberjack (*Seriola dumerili*) is a species with demonstrated susceptibility to infection by trematodes and cestodes (*83*). In addition to serving as vectors and hosts, the presence of and active hunting by avian and mammalian predators can worsen a disease outbreak in fish species by increasing the stress levels of cultured animals (*84*).

Finfish aggregation

There is strong evidence from Europe, South America, North America, and Asia that wild fish attraction to finfish farms is a global issue, regardless of species cultivated (*81*). However, wild fish aggregate at various depths near farms and exhibit variable behaviors, indicating that their purposes for aggregation may differ among species. Fish farms also appear to influence the abundance of species at greater distances from the farms than previously thought. Research from fish farms in the Aegean Sea suggests that increases in wild fish abundance related to farms may be evident up to 3 miles from farm sites (*81*).

Excess or uneaten food is a primary driver for the large aggregations of wild fish at net pen farms (*39*). During day-to-day observations, aggregations most frequently coincided with feeding times for cultured animals, and research on Norwegian salmon farms concluded that the main attractant for wild species was uneaten feed from the farms (*75*). Furthermore, there is a statistical relationship between wild fish abundance and feed supply (*81*). Biofouling communities that form on farm infrastructure, such as algae, barnacles, anemones, and mussels, can act as an additional source of food for aggregating wild fish species (*81*).

There may be secondary effects via trophic subsidy from uneaten feed and fish feces that augment aggregation near farms (*39*). In one study, wild fish collected near aquaculture sites averaged 1.2 times larger and 1.7 times heavier than the same species in reference locations. Analysis revealed dietary shifts in wild fish species near farms as well; feed pellets from farms were found in fish stomachs in addition to elevated fat content in their tissue, suggesting a source of lipids via pelleted feed (*75*).

The physical features of an aquaculture facility can provide protection from predation and structural complexity preferred by many wild fish species, stimulating an “artificial reef” effect (*81*, *85*). Biofouling can enhance that effect by providing food subsidies for lower trophic levels and invertebrates, attracting successive trophic levels to the facility (*81*, *85*).

Aggregations and disease transfer

Proximity between cultured and wild organisms is hard to control due to the open nature of offshore marine aquaculture. When a facility serves as an aggregating site, the potential for pathogen transfer is exacerbated (*39*, *41*). Much of the research on pathogen transfer between wild and cultured finfish comes from salmonid production systems, which constitute ~60% of marine finfish aquaculture (*86*). The direction of transfer (wild-to-wild, farmed-to-wild, or wild-to-farmed) is usually difficult to determine or confirm (*42*). Molecular assessments of viral genetics in wild Atlantic salmon indicated transmission between stocks derived from Europe and North America at shared feeding grounds in the northwestern Atlantic Ocean (*87*). A rare demonstration of directional transmission of infectious hematopoietic necrosis virus from wild to hatchery salmon required extensive molecular genotyping over multiple years in both wild and cultured populations (*88*). This level of monitoring and pathogen-specific knowledge is unlikely to be initially available for marine aquaculture due to cost and complexity of detection methods. Ectoparasites, particularly sea lice (*Lepeophtheirus salmonis* and *Caligus* spp.), continue to be a leading concern in salmonid aquaculture. Substantial correlative data between infection burdens in net pen salmon and proximal wild fish infestations have supported the likelihood that spillover occurs (*89–93*), although it is not clear that aquaculture farms are the sole contributor to infestations in wild fish (*94*). Nonetheless, sea lice management is stringent in salmonid aquaculture, with a wide range of preventative and mitigating tactics (summarized in *95*), and incorporation of relevant environmental data (e.g., water temperature, salinity) into coupled hydrodynamic–biological dispersion models can offer better risk management and understanding of the ectoparasite interactions (*37*).

In addition to pathogen transfer, persistent presence of predators can elevate stress levels in cultured animals. Stress can contribute to immunosuppression and greater susceptibility to disease (*39*, *96*) and increase the dynamics of pathogen transfer (*42*). Therefore, chronic stress-inducing events, such as a routine presence of predators, are health management considerations for producers. For producers, a conservative approach to reduce disease potential can be achieved through rearing robust, healthy stocks, maintaining appropriate rearing densities, and minimizing handling or physiological stress.

Reducing and mitigating aggregation

Actions and technologies to reduce and mitigate the attractiveness of farms to wild species are under continual development. Predator exclusion plans and devices consist primarily of barriers or excluders, deterrence, and removal (*97*). Barriers are typically mesh or netting, either above or below water. Depending on the barrier characteristics, they have a costly requirement of frequent monitoring for release of entangled wild animals. Deterrence can include visuals (e.g., flashing lights, threatening shapes or silhouettes), sounds, physical actions (e.g., water sprays), and movement. However, aggregating animals will habituate to deterrence actions (*81*) unless they are irregular or random in occurrence. Furthermore, certain deterrence options, such as underwater explosions and predator removal, have unfavorable collateral effects (*81*), and are prohibited in some areas such as Scotland.[13]

Uneaten feed is a major attraction of fish farms, and the development and use of properly designed feeds for the cultured species (e.g., floating, sinking, slower-dissolving) can help reduce feed waste. Adjustment of feeding schedules is improving with the application of artificial intelligence to control feed schedules and delivery (*98*, *99*), as well as the development of appropriately designed feeds (e.g., sinking or floating feeds) that cater to species feeding habits (*100*). These systems can use photogrammetry for estimating fish growth for more accurate feed calculations, and monitor fish behavior during feeding for terminating delivery.[14] Furthermore, improvements in fish feed formulations have improved feed-conversion ratios, resulting in more efficient assimilation of nutrients and reduced waste (*5*). These types of advances carry economic benefits for the producer while reducing aggregation attractiveness.

---

[13] https://scottishseafarms.com/sustainability/co-existing-with-marine-life/

[14] https://www.innovasea.com/open-ocean-aquaculture/open-ocean-aquaculture-feeding-systems/

Water quality, farm discharges, and disease

Offshore waters might be considered less impacted by anthropogenic factors, but they are still subjected to river outflows, terrestrial runoff, and discharges from shipping. While offshore waters could be a higher-quality environment for rearing finfish, they may be more vulnerable to farm discharges, depending upon farm site characteristics (*101*). Pollution from ships may have a direct impact on offshore water quality, including oil, harmful or noxious substances, sewage, and garbage. The International Maritime Organization (IMO) is a specialized agency of the United Nations which is responsible for measures to improve the safety and security of international shipping and to prevent pollution from ships.[15] The IMO develops treaties for adoption into law by participating nations, allowing harmonization of national and international policies. The International Convention on Prevention of Pollution from Ships, or MARPOL, is the main international convention covering prevention of pollution of the marine environment by ships from operational or accidental causes.[16] The MARPOL convention was adopted at IMO. In the United States, the convention is implemented through the Act to Prevent Pollution from Ships (APPS).[17] It applies to all vessels, whether seagoing or not, regardless of flag, operating in U.S. navigable waters and the Exclusive Economic Zone (EEZ). It is administered by the Coast Guard. The regulatory mechanism established in APPS to implement MARPOL is separate and distinct from the Clean Water Act (CWA)[18] and other federal environmental laws (*102*). Generally, CWA jurisdiction for discharges from vessels extends only to 3 nautical miles from shore (*103*). In the United States, several federal agencies have some jurisdiction over ships in U.S. waters, but no single agency is responsible for or coordinates all of the relevant government functions. The U.S. Coast Guard and the Environmental Protection Agency (EPA) have principal regulatory and standard-setting responsibilities (*102*). In addition to point sources of pollution such as shipping, the two AOAs of interest have different features relevant to finfish aquaculture.

Region-specific water quality features

The Gulf of Mexico receives runoff from approximately two-thirds of the continental United States, and the northern coastal areas have poor status for nutrients, dissolved oxygen, and sediment contaminants (see *104*, their Figure 2.10). Development of hypoxia and “dead zones,” often due to eutrophication, is routinely monitored and reported by NOAA,[19,20] and these zones can extend to the northwestern and central continental shelf (see *104*, their Figure 2.77). Additionally, petroleum contaminants resulting from natural seeps, extraction (e.g., oil drilling), product transportation, and consumption (e.g., combustion) are significant water-quality topics for offshore areas of the Gulf of Mexico (*104*, *105*).

Southern California Bight waters tend to become strongly stratified in summer and fall, which is weakened or disrupted by upwelling events in winter and spring, and there is a nearshore poleward (south-to-north) undercurrent and countercurrent flow most of the year (*106*, *107*).

---

[15] https://www.imo.org/en/About/Pages/FAQs.aspx

[16] https://www.imo.org/en/KnowledgeCentre/ConferencesMeetings/pages/Marpol.aspx

[17] APPS (Act to Prevent Pollution from Ships). 1980. U.S. Code, title 33, sections 1905–1915.

[18] CWA (Clean Water Act). 1972. Federal Water Pollution Control Act of 1972. Clean Water Act of 1977. U.S. Code, title 33, sections 1251–1387.

[19] https://www.ncei.noaa.gov/maps/hypoxia/

[20] https://gulfhypoxia.net/

The under- and countercurrents can transport water from heavily urbanized and industrialized waters of Los Angeles and Orange Counties into the AOA study areas, although this tends to be a greater hazard to humans than finfish (e.g., *108*). The Southern California Bight Regional Monitoring Program is focused on benthic, rather than pelagic, conditions (*109*),[21] while shelf and offshore monitoring is conducted primarily by large wastewater treatment plants as a component of National Pollutant Discharge Elimination System (NPDES) compliance (e.g., *110*). Unlike in the Gulf of Mexico, prominent hypoxic events do not occur, but significant and complex eddies in both Santa Monica Basin and Santa Barbara Channel can entrain and trap waters carrying contaminants, such as oil from natural seeps or anthropogenic sources (*111*, *112*).

Particulate discharges

Marine finfish aquaculture farms release particulates and nutrients to both the pelagic and benthic environments. Dispersion of particles and propagules in marine environments is estimated to be approximately twice that of terrestrial environments (*113*, *114*). Nutrient enrichment may influence primary production (*74*, *115*) and species aggregation. These processes are factors in pathogen spread through their ability to increase the overall biomass near farms, and thus elevate the probability of contact and pathogen transfer (*26*, *39*, *41*, *42*, *81*, *84*). Dissolved organic nutrients from feed pellets and feces from farms can increase heterotrophic microbial activity, although mechanisms of impact on the food web are not well described (*40*, *73*). One study near fish farms in the Mediterranean Sea suggested a rapid transfer of fixed carbon from farm byproducts to higher trophic levels (*115*).

Pathogen persistence in the marine environment is significantly affected by temperature, salinity, and exposure to ultraviolet radiation (i.e., sunlight); conditions vary for each organism (reviewed in *116*, *117*). Organic material in the seawater can serve as a supportive environment for pathogens associated with particulate and organic matter, allowing longer persistence (*23*, *118*, *119*) and increased opportunity to infect a new host. Microorganisms attached to organic matter and particulates are better shielded from the damaging effects of sunlight, resulting in longer survival. Organic matter may also serve as microscopic niches providing a nutrient source in oligotrophic seawater (reviewed in *116*, *117*), and, in conjunction with dissolved nutrients, can promote biofilm formation on equipment surfaces that increases survival of associated microorganisms (reviewed in *28*). Adsorption to organic particulate material also alters transport dynamics for pathogens, potentially resulting in pathogen deposition in the benthos closer to the farm due to the larger size of the particle–bacteria complex.

The pelagic and benthic impacts of nutrient release will be site-specific and, although pelagic effects in well flushed locations are not detected beyond 100 m, the far-afield effects are poorly understood and difficult to characterize (*74*, *120*). However, research suggests negligible impacts from farm discharge and nutrient enrichment in far-afield locations due to dilution by current and depth (*121*). Farm sites at greater depths can alleviate the impacts on benthic sediments because of greater potential for horizontal dispersal through the water column that is dependent on the capacity of local currents to carry particles (*120*).

---

[21] https://www.sccwrp.org/about/research-areas/regional-monitoring/southern-california-bight-regional-monitoring-program/

Overall, the current practices in siting, feed efficiency, and extensive monitoring of sites in Europe, Canada, and the United States are considered sufficient to limit nitrogen increases to modest levels without significantly impacting the chemical and biological processes of surrounding marine environments (*74*). A 2022 NOAA biological opinion on the subject states that existing marine aquaculture sites in Washington State, and their nutrient discharge, are not likely to jeopardize the welfare of wild species and habitats under the Endangered Species Act (ESA; *122*).[22,23]

Nitrogen and phosphorous

Nitrogen is a primary nutrient discharged from aquaculture facilities, usually as feces or uneaten feed. Exact discharge estimates vary depending on the cultured population, type of feed, and other factors (*74*). Research on the effects of nitrogen discharge on surrounding seawater quality and primary production provide mixed results. A study in Taiwan suggested water-quality impairment when a finfish farm was located in a semi-enclosed lagoon farm site (*74*). It is worth noting that the AOA options from the Aquaculture Atlases are not characterized as semi-enclosed sites (*2*, *3*) and presumably are not subject to water flow restrictions.

In the marine environment, phosphorus is not usually a limiting nutrient compared to nitrogen, but phosphorous can be responsible for changes in biological and chemical processes. Research suggests limited negative effects on seawater quality from phosphorous discharge by fish farms in Newfoundland and Puerto Rico, although local phosphorous loading may be more significant where seawater exchange is restricted (*74*).

Dissolved oxygen

Adequate dissolved oxygen (DO) in aquaculture is a critical component for fish husbandry and welfare to maintain feed conversion efficiency while preventing respiratory stress and illness. Depletion of DO can be due to the biological oxygen demand (BOD) from dissolving waste particles, fish respiration, elevated water temperatures, or a combination of these factors (*123*). In marine aquaculture, exogenous sources of BOD are larger in impact than discharge from farms. Existing research has yet to discover any farm-induced DO depletion in seawater bodies that warranted significant or prolonged concern (*74*).

Benthic deposition and recovery

Benthic recovery periods from aquaculture depositions can vary among farm situations. An intensive salmon net pen culture in shallower depths (31–40 m) near an exposed rocky coastal island cluster in Norway produced significant benthic deposition during grow-out, and the benthic fauna and biogeochemical processes beneath the farm returned to the pre-stocking state after a seven-month post-harvest fallowing (*120*). A study in Scotland found that benthic communities up to 49 m in depth remain impacted 15 months after the cessation of

---

[22] ESA (Endangered Species Act of 1973). 1973. U.S. Code, title 16, sections 1531–1544.

[23] https://repository.library.noaa.gov/view/noaa/41678

farm operation, which was located in a fjordic sea loch (*124*). These and similar assessments highlight that benthic recovery can vary between locations and depends greatly on depth, currents, benthic substrate composition, and geomorphology (e.g., fjord vs. open bay). The Norwegian study recommended 2 km of spacing between farm sites to mitigate organic loading impacts. However, local hydrodynamics need consideration when determining standards for the depth of sites and the distance between farms. Additionally, routine monitoring of benthic communities during fallowing is recommended to analyze trends between farming cycles (*120*).

The minimum, maximum, and average depths reported in the Aquaculture Atlases for the AOA options (*2*, *3*) show that minimum depths exceed 49 m except for the N2 options in Southern California (Tables 4 and 5). Among the five N2 options, three have maximum depths exceeding 49 m and average depths exceeding 40 m (Table 5). None of the AOA options are located in fjords or enclosed embayments, and all have exposure to larger marine circulation patterns (*2*, *3*). These features suggest that potential farm sites in the AOA options are not likely to present negative impacts on benthic sediment quality. However, specific assessments incorporating farm discharge rates and hydrodynamic features need to be conducted for specific proposals for accurate evaluation.

Table 4. Minimum, maximum, and average depths, in meters, for the AOA options in the Gulf of Mexico. Extracted from the NOAA Aquaculture Atlas for the Gulf of Mexico (*2*).

AOA Option	Min Depth	Max Depth	Avg Depth
W-1	84.4	93.7	90.8
W-4	80.6	88.4	84.6
W-8	78.9	82.3	80.6
C-3	59.8	61.4	60.5
C-11	76.4	87.8	82.3
C-13	56.4	69.2	62.2
E-4	49.5	52.9	51.1
E-3	49.6	51.9	51.0
E-1	50.1	51.0	50.6

Table 5. Minimum, maximum, and average depths, in meters, for the AOA options in the Southern California Bight. Extracted from the NOAA Aquaculture Atlas for the Southern California Bight (*3*).

AOA Option	Min Depth	Max Depth	Avg Depth
N1-A	88.60	108.5	95.0
N1-B	78.00	92.6	84.7
N1-C	77.30	101.3	90.0
N2-A	41.90	60.5	51.2
N2-B	38.90	56.0	47.3
N2-C	34.80	63.7	48.1
N2-D	25.50	39.0	31.5
N2-E	23.10	29.8	25.4
CN1-A	58.99	154.6	98.9
CN1-B	55.40	101.3	66.6

Invertebrate Aquaculture Disease Factors

Invertebrate aquaculture can include a wide variety of organisms, ranging from echinoderms (e.g., sea urchin, sea cucumber) and crustaceans (e.g., shrimp, lobster) to bivalves (e.g., oysters, mussels) and cephalopods (e.g., squid, octopus). In the Pacific Northwest, indigenous cultivation of bivalve mollusks through enhancements colloquially called "clam gardens" dates to over 3,000 years ago (*125*), and the clam garden approach is enjoying a contemporary revival.[24] Commercial production of certain bivalves and shrimp in the United States has a long history, with well developed captive broodstock programs. Shrimp are the only commercially produced crustacean for which SPF broodstocks have been developed. Disease concerns commonly arise from culture exposure to hazards in the natural environment, such as culture density, lapses at critical biosecurity points (e.g., water filtration failure), and, in the case of oysters, seasonal elevation of oyster herpesvirus in warmer months (*126*, *127*). Early-life-stage feeding often includes algae and protozoa specifically cultured as forage. Many aquacultured invertebrates (with the exclusion of shrimp) do not receive formulated exogenous feed during grow-out phases, relying instead on extracting nutrients from the ambient environment. The production cycle usually includes a nearshore or estuarine phase. Systems range in design from estuarine ponds to bottom culture to diverse methods of suspending animals during grow-out.

Factors Associated with Hatchery or Nursery Activities

Because of reliance on wild sources for broodstock for some species, there is a likelihood of pathogen introduction into a hatchery or nursery if the broodstock are infected. To minimize this risk, there are efforts toward generating pathogen-free broodstock to reduce that hazard. Development of SPF broodstock has improved biosecurity significantly for some sectors of U.S. shrimp production by reducing reliance on wild collections that caused repeated introduction of the white spot syndrome virus into the industry over many years (*128*). However, requirements for moving broodstock vary by state (e.g., see the Pacific Coast Shellfish Growers Association document on state requirements[25]), which can cause confusion for growers. The creation of a Regional Shellfish Seed Biosecurity Program (RSSBP) offers a voluntary opportunity to streamline intra- and interregional shipments (including East Coast-wide transfers) by following specific health evaluations and audits for the pathogen status of a given shellfish nursery (*6*).[26]

---

[24] https://nativenewsonline.net/sovereignty/indigenous-knowledge-revives-ancient-clam-garden-practices
[25] https://pcsga.org/wprs/wp-content/uploads/2013/04/State-Shellfish-Health-Info-and-Contacts.pdf
[26] https://rssbp.org/hatchery-certification-program/

Larval bivalves are susceptible to viruses (e.g, for oysters, OsHV-1 and oyster velar virus) and bacteria (vibriosis) that have variable effects on adult bivalves (*129*, *130*). Viruses with a broad host range that are able to produce asymptomatic infected adults, such as OsHV-1, are a constant threat to larval rearing operations exposed to untreated seawater and to other age classes of bivalves (*130*). Viral hazards can be reduced by avoiding locations with existing bivalve operations or wild stocks that may act as potential reservoirs of pathogens (*130*). Pretreatment of incoming seawater can also protect against pathogen introduction, if feasible and cost effective. Filtration to 5 µm (or to 1 µm, as recommended by the RSSBP) is effective for removing parasites and pathogens attached to larger particles. Aging seawater for 48 hours before use can reduce viral exposure through sedimentation of particles or natural decay (*131*), while pretreatment by ozonation or ultraviolet radiation can also reduce pathogen loads, especially viral ones (*130*, *132*).

Factors Associated with Invertebrate Grow-out

Disease factors and pathogens of concern during invertebrate grow-out vary depending on culture method and life-history biology. Water biosecurity cannot be controlled in open culture structures such as cages, and this free exchange of seawater provides the main disease transmission route. Transfer into open systems and aligning husbandry and harvest practices require planning for periods of time when there is low pathogen prevalence at virus-infected locations, which can lower disease-caused mortality for juveniles and adults in nearshore environments (*8*). Other successful methods for reducing infection include reducing open system immersion time or delayed exposure to untreated seawater, and movement between low and high salinity environments (*131*, *133*). The need for developing and applying avoidance actions will depend on the concentrations of pathogens at offshore sites.

Culling and frequent removal of mortalities is very effective in finfish aquaculture for reducing disease spread, but may be less useful for molluskan aquaculture, as it depends on disease signs (including death) and accessibility of organisms for inspection (*134*). If a disease is density-dependent and the transmission threshold is known, culling to a density below that threshold could control or even eliminate the disease (*134*).

Reducing the number of susceptible hosts could be extended to adjacent wild sympatric species (e.g., *Mercenaria* spp.) that are reservoirs for pathogens (*135*). Selective culling based on pathogen detection is a proposed strategy for conserving valuable red abalone populations threatened by withering disease (*136*). A similar approach could be used to reduce intermediate hosts for invertebrate parasites, such as whelks in scallop apicomplexan mortality (*137*). Obviously, this type of approach would require extensive interaction and permitting with natural resource authorities, and is likely to generate objections to culling non-aquaculture species.

Biofouling can contribute to animal stress and disease occurrence by reducing water flow, growth, and survival while increasing crowding and potentially harboring pathogens (*138*, *139*). Other factors such as sedimentation, loss from predation, wave action, and hypoxic conditions can either contribute to disease issues or act as primary causes of mortality.

Waterborne pathogen transmission can depend on other factors that may enhance the spread of disease, including seawater temperature and salinity extremes, culture density, and stress due to handling, movement of animals (*129*), or inclement weather conditions. Water temperatures and salinities have predictable effects for some diseases (e.g., Dermo disease in oysters; *140*). Mitigation for unfavorable temperatures would require shifts in production cycle (*127*), choosing temperature-resilient species, and/or selective breeding for temperature hardiness (*141*). Other common practices to reduce disease spread include maintaining low-stress culture densities (which need to be determined empirically for the species), avoiding mingling different age classes or groups from different sources, and minimizing handling (*142–145*).

Although bivalves have simple immune systems compared to vertebrates, immune responses can be measured—such as hemocyte phagocytic activity, abundance of humoral lectins, and humoral antimicrobial peptides (*129*). Although evidence for ontogenetic immune maturation is found in the differential anti-viral responses of larval and adult oysters (*146*), there is no evidence of an acquired or adaptive immunity comparable to that of vertebrates. This makes conventional vaccination approaches useless for invertebrates. The innate ability of mollusks to recognize and react to foreign antigens, including an interferon-like system (*129*, *147*, *148*), has been exploited in the application of immunogens to stimulate an immune response (e.g., *149*). Deeper understanding of cellular pathways exploited by pathogens, such as parasite inhibition of a protective programmed cell death by immune cells (*150*), is providing new opportunities to develop interventions.

Although disease-resistant mollusks can develop naturally through strong selective pressure (*151*), breeding for disease resistance is a popular concept with a long history in aquaculture, with most efforts on bivalves (*152–154*). These programs have focused on major oyster pathogens with severe consequences (OsHV-1, *Bonamia ostreae*, *B. roughleyi*, *Marteilia syndeyi*, *Roseovarius crassostreae*, *Haplosporidium nelsoni*, *Perkinsus marinus*), and were able to show positive advantages after only several generations (*153*, *154*). In contrast, triploidy (for accelerated growth) has exhibited neither an advantage nor a disadvantage for disease resistance (*153*). Genome sequencing revealed that mollusks have good genetic potential for disease resistance selection: there are high single-nucleotide and indel polymorphism rates and significant expansion of genes related to immunity and stress responses, providing an abundance of markers for linkage and quantitative trait locus mapping (e.g., see *154*, their Table 3). A caveat about selecting for disease resistance: the protection conferred against a particular pathogen does not necessarily extend to other pathogens (e.g., *155*), and resistance against one pathogen could increase susceptibility to a different pathogen, although this latter possibility has not been formally studied.

Although seafood safety is not a focus of this document, water quality issues for invertebrate pathogens and public health pathogens are related. Most cultured invertebrates are suspension or detritus feeders, capable of accumulating pathogens or contaminants that are public health hazards (e.g., norovirus, *Vibrio* spp. bacteria, and biotoxins). For shellfish, the National Shellfish Sanitation Program (NSSP)[27] manages seafood safety of shellfish through partnerships among the shellfish industry, state

---

[27] https://www.fda.gov/food/federalstate-food-programs/national-shellfish-sanitation-program-nssp

agencies, and federal agencies, such as the Interstate Shellfish Sanitation Conference (ISSC).[28] The NSSP provides guidance on classification of growing areas based on product and water quality testing through regular updates of the Guide for the Control of Molluscan Shellfish (*156*). The latest version contains provisions for aquaculture harvest in federal waters (3–200 miles from shore), particularly for marine biotoxins. Section 312 of the Clean Water Act[29] prohibits vessel discharge of pollutants, including sewage, in areas designated as "no discharge zones" (NDZ),[30] although a state may impose stricter vessel rules for waters within its jurisdiction. These policies and regulations are jointly enforced by the U.S. Environmental Protection Agency and the U.S. Coast Guard. If an offshore aquaculture facility is located more than 3 miles from shore and is not positioned in an NDZ, it could be exposed to pollutant discharges from vessels.

---

[28] https://www.issc.org/

[29] https://www.epa.gov/vessels-marinas-and-ports/vessel-sewage-discharges-statutes-regulations-and-related-laws-and

[30] https://www.epa.gov/vessels-marinas-and-ports/vessel-sewage-no-discharge-zones

Seaweed/Macroalgae Aquaculture Disease Factors

Seaweeds/macroalgae have been harvested for human use in Asia for approximately 1,500 years, and commercial-scale cultivation became established in the 1950s in Korea, Japan, and China (*157*). Temperate-zone seaweed/macroalgae aquaculture emerged after 2000, primarily in European waters, with large brown kelp as the major cultivated species (*158*). Worldwide seaweed/macroalgae production tripled between 2000 and 2018 (*159*), making up approximately half of the global marine and estuarine aquaculture production by volume (*160*).

Seaweed/macroalgal culture is an increasing priority for marine aquaculture in the United States (*161*). Farmed algae are susceptible to bacteria, protists, viruses, and other algae, as well as grazing pests such as limpets, copepods, and herbivorous fishes (*162*)—often concurrently. Additionally, some macroalgae species native to the United States share susceptibility to the same pathogens and diseases as their cultured counterparts in Asia, often being from a similar genus. As an example, *Pyropia* spp. are commonly cultured in Asia, but several members of the genus are also native to the U.S. West Coast (*162–164*). Several simple biosecurity measures have proven effective in Asian aquaculture, including use of uninfected propagules, cleaning biofouling from organisms and farm ropes, and early identification of infected stock (*160*). However, disease transmission between cultured and wild macroalgae is poorly understood and requires significant additional research on the potential impacts of marine algae culture on wild populations.

Factors Associated with Introducing Non-Native Species

Introduction of non-native species, either intentionally or unintentionally, is a concern for expanding aquaculture, particularly for seaweed/macroalgae, where over half of the invasive introductions have been due to aquaculture (*165*). Some of these introductions have had dire consequences, including overwhelming valuable coral reef habitat (*165*). The best prevention strategy is to avoid cultivating species outside of their native range, and there is a potential adaptive advantage in using locally sourced cultivars (*166*). The most commonly cultured seaweeds/macroalgae have varying degrees of domestication through vegetative propagation or selection, and even if cultivation is within a native range, there is potential for gene flow with indigenous strains or landraces (*167*) .

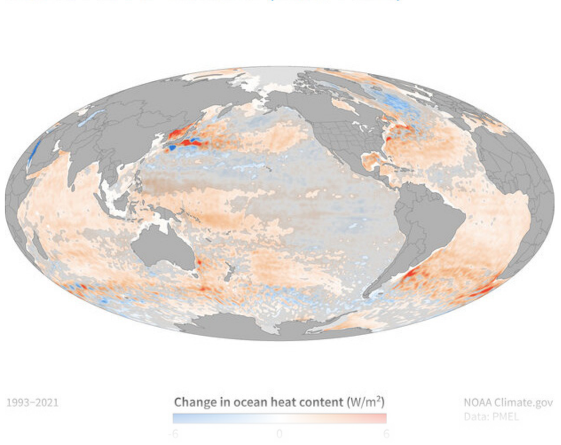
Importing organisms from non-local areas can also introduce non-native species associated with the seaweed/macroalgae—known as its holobiont. While the best prevention is to use broodstock, propagules, or seedlings from sources that do not harbor undesired or invasive epiphytes, endo-epiphytes, or plankton, this knowledge may not be available. A biosecurity program that includes quarantine prior to out-planting is considered a standard practice to allow for observation and control when mitigating the introduction of non-native species (*168*). Multiple jurisdictions have versions and templates of biosecurity plans for marine aquaculture, although there are no standardized versions yet for operations in the United States (*8*, *145*, *169*).

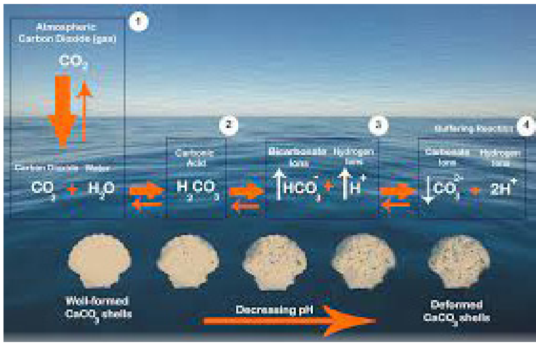
Factors Associated with Seaweed/Macroalgae Grow-Out

Wild macroalgae growing near farms, whether conspecific with cultured varieties or not, represent an important component of wild ecosystems in the areas under evaluation for marine aquaculture. Kelp forests and other macroalgae-dense environments provide habitats for feeding, breeding, and safety for other organisms (*170*, *171*).

Because seaweed/macroalgae depend upon photosynthetically active radiation (PAR), their cultivation is typically located in near-surface waters over a broad area to permit light exposure. Temperatures or light intensities that are too high or too low for the species can induce heat shock or oxidative stress, respectively (*172*). Suspended cultures also rely on dissolved inorganic nutrients that require replenishment through water flow as nutrients are drawn from the water column during growth. Nutrient depletion under high density stocking can occur (*158*), and depletion can be exacerbated if the suspended culture slows the water flow (*173*). Site selection controls most of these environmental parameters, and good knowledge of farm site hydrodynamics, including seasonal and tidal variations, is important for siting a seaweed/macroalgae farm.

Environmental conditions are the dominant factors for cultured seaweed/macroalgae health, including sufficient nutrients, adequate light for photosynthesis, appropriate temperatures, and good salinities—emphasizing the importance of routine water quality monitoring. If one or more of these conditions is not optimal, disease can occur. An example is ice-ice disease in eucheumoid seaweeds, which has been studied for many years. An etiology has not yet been conclusively established (*162*), although the environmental factors known to contribute to the disease are well characterized (*174–176*). Studies suggest there are interactions of multiple microorganisms contributing to ice-ice disease involving the seaweed/macroalgae epibiont community. Because pathobiomes are the suite of organisms associated with a diseased host and the interactions among the associated organisms and with the host (*177*), a pathobiome approach may be better for understanding, preventing, and mitigating diseases of these organisms (*162*, *177*).





Climate Change and Aquaculture

Host, Pathogen, and Environment

For an organism to succumb to a particular disease, a combination of host, pathogen, and environmental conditions favorable to disease is required (*12*, *178*, *179*). Planning for the disease impacts of climate change on aquatic organisms requires consideration of effects on the host–pathogen–environment combination that is altered by climate change (*180*, *181*). An assessment of 375 infectious diseases of humans revealed that 58% of them were aggravated by climate conditions driven by greenhouse gas emissions (*182*). Changes in water flow due to drought, snowpack accumulation and release, and wildfire have already affected inland aquaculture and survival of hatchery salmon (e.g., (*178*). As the U.S. marine aquaculture sector develops, research is needed to understand the disease dynamics of climate change for individual species selected for culture. This will require information on environmental, economic, and biological suitability of selected culture species; disease and biosecurity hazards for those species; and their interactions with climate change effects.

Climate Change Projections for the Gulf of Mexico and the Southern California Bight

Relative to the other drivers, long-term climate trends are largely driven by greenhouse gas emissions (*7*, *183*). Greenhouse gasses include water vapor, carbon dioxide, methane, nitrous oxide, and ozone; these gasses trap heat within the earth's atmosphere, causing global warming. Additionally, carbon dioxide absorbed by the ocean in increasing amounts lowers the pH of seawater over time, causing ocean acidification (OA). The combined effects of global warming and ocean acidification drive changes in other physical characteristics, such as sea level, salinity, dissolved oxygen, wind speed and direction, precipitation, nutrient and sediment loads, and ocean currents. These physical changes, in turn, can have biological impacts such as changes in species distribution and abundance; organism development and growth (e.g., shell formation in certain invertebrates); disease prevalence; and occurrence of harmful algal blooms. Table 6 presents an example of a framework for understanding the climate change-induced hazards for wild species that are also relevant to the aquaculture sector.

According to the United Nations Intergovernmental Panel on Climate Change's (IPCC) CMIP6 climate models, it is "very likely" that the Earth's atmosphere will warm an additional 1.5°C between 2021 and 2040 (*7*). The 1.5°C increase is consistent across emission projections, with the exception of one scenario that predicts a 1.6°C increase. The current projections are for a 0.6°C increase in mean sea surface temperature in the Southern California Bight, and an increase of 0.7°C to 0.8°C in the Gulf of Mexico, over the next two decades (Table 6). Sea level is estimated to rise by 0.1 m and 0.2 m for the Southern California Bight and Gulf of Mexico respectively; ocean surface pH is expected to decrease by 0.1 units in both sites. While coastal precipitation is predicted to increase between 2.4% and 3.1% for the Gulf of Mexico, the data present a more uncertain picture for the Southern California Bight, where estimates range from a 2% decrease to a 0.8% increase in precipitation (*184*, *185*). Weather events, such as extreme heat, heavy precipitation, drought, and tropical cyclones are also predicted to become more frequent and intense for the two study areas. Dissolved oxygen and salinity are predicted to decrease in many areas/depths within the two geographic sites.

A warming atmosphere's effect on physical environmental conditions at an aquaculture site can influence the vulnerability of both cultured and wild species to aquatic pathogens (*5*, *186*–*189*). Changes in environmental conditions can influence pathogen transfer indirectly by altering the distribution and behavior of animal vectors and alternate or definitive hosts, including invertebrates, fishes, birds, and marine mammals (*190*).

Table 6. IPCC CMIP6 Climate Projections for AOA study areas. IPCC CMIP6 projected change in climate driven attributes reported as change in annual mean between the periods 1995–2014 and 2021–40, by geographic region. *Emissions Scenario* is the projected outcome based on the Shared Socioeconomic Pathway (SSP), which is a description of socioeconomic policies and actions that affect greenhouse gas emissions. Lower SSP values are expected to greatly reduce emissions, while higher SSP values are expected to have less emission reduction (*184*, *185*). *Low* has an SSP value of 1.26, and *Intermediate* has an SSP range of 2 to 4.5.

Attributes	**Emissions scenarios**	**Projected changes in annual means between 1995–2014 and 2021–40, by region**	
		Southern California Bight	**Gulf of Mexico**
Sea surface temperature (°C)	Low	+0.6 (0.3–0.9)	+0.7 (0.2–1.2)
	Intermediate	+0.6 (0.3–0.9)	+0.8 (0.3–1.3)
Surface pH	Low	−0.1 (−0.1 to −0.0)	−0.1 (−0.1 to −0.1)
	Intermediate	−0.1 (−0.1 to −0.1)	−0.1 (−0.1 to −0.1)
Sea level rise (m)	Low	+0.1 (0.1–0.2)	+0.2 (0.1–0.3)
	Intermediate	+0.1 (0.1–0.2)	+0.2 (0.1–0.3)
Coastal precipitation (%)	Low	+0.8 (−6.2 to −6.6)	+3.1 (0.6–5.2)
	Intermediate	−2.0 (−5.5 to −4.8)	+2.4 (1.0–5.8)
Cooling days (mean daily temperature above 65°F) (n)	Low	+111.2 (73.9–199.6)	+114.4 (47.3–168.6)
	Intermediate	+121.1 (82.8–215.1)	+116.9 (51.6–182.2)

Aquaculture and Disease Under Climate Change

Cultured and sessile species do not have the ability to seek better conditions as their environment changes, while wild, mobile species can move to more favorable environments or remain in a particular location for specific resources (e.g., prey, structural protection). Poor environmental conditions can cause chronic stresses that contribute to lowered immune function and disease development (*178*, *188*, *191*). Under these conditions, the host's physical barriers to infection can be compromised. For finfish, this could manifest as a disruption in the skin mucosal layer or in a biological disruption of protective microflora associated with this layer (*192*, *193*). For marine shellfish (particularly marine bivalves), decreasing pH can interfere with an organism's ability to produce and maintain a protective shell (*194*–*196*).

Disease in a warmer environment

Temperature is a prime modulator of metabolism in ectotherms, and climate-driven temperature change is expected to alter future disease distribution and prevalence. Aerobic performance is maximized in and around temperature optima, and as the temperature shifts out of optimal range, metabolism also shifts toward anaerobic metabolism, which is more energy-expensive (e.g., *197*). Increases in sea surface temperature are generally associated with increased disease prevalence (*198*–*200*). Disease outbreaks induced by temperature stress are already a common seasonal occurrence for many species (*201*–*203*). Compounding this issue are data suggesting that high-temperature events, such as the marine heatwave that originated in the Gulf of Alaska in 2013, will become more frequent and intense as warming continues (*204*, *205*). Outcomes from warming, such as changes in salinity, pH, and nutrient cycling, can significantly affect immune response, and can render both wild and farmed animals more susceptible to disease (*44*, *188*, *201*, *204*, *206*–*214*). Low dissolved oxygen (hypoxia) combined with temperature stress are cited as major contributors to stress-induced disease outbreaks (*206*, *209*). Conversely, temperature-driven changes in phenology could reduce disease severity; shifts to earlier molting by lobsters can physically segregate the host from infections (*197*). Consistent and reliable environmental monitoring is important to inform both husbandry decisions by producers and regulatory decisions to protect wild and farmed species from temperature and acidification-induced health events.

As poikilotherms, finfish and invertebrates are vulnerable to environmental temperature shifts, and seaweed/macroalgae have no known thermoadaptive mechanisms. Aquaculture strategies to prepare for future temperature changes include using the most robust stocks available, using stocks with demonstrated thermal tolerances, and developing breeding programs for temperature-resilient stocks (*215*). Strategies such as phenotypic selection, quantitative trait loci mapping, and genome-wide association studies demonstrate the feasibility of developing temperature-resilient finfish and shellfish (*216*–*218*).

Disease under changing ocean chemistry

Secondary effects of climate change can also affect the health of cultured and wild species. For example, an increase in nearshore nutrient supply and/or pollution from increased rainfall and coastal erosion can damage the gills of finfish (*203*) and shellfish, making them more vulnerable to secondary infections. An outbreak in Ireland of amoebic gill disease in cultured Atlantic salmon smolts was positively associated with environmental factors including high ammonia, nitrate, and chlorophyll (*219*). Eutrophication and increased turbidity from increased rainfall and runoff along the coasts have also been associated with nearshore increases in parasite, fungi, and disease abundance (*190*).

Warmer ocean temperatures and increased dissolved nutrients are associated with an increase in the frequency and intensity of harmful algal blooms that have direct toxic effects on cultured and wild species (*220*). Increased coastal nutrient supplies may also modify algal species composition, favoring invasive species, depending on the region (*221*).

Some of the best known and characterized climate change impacts are on biological calcification under conditions of ocean acidification, particularly for mollusks (*222*). Negative effects of low pH and low aragonite saturation observed in larval bivalve hatcheries on the Oregon coast (*223*) stimulated research in the United States on the effects of carbonate chemistry changes in both cultured and wild species (e.g., *224*–*226*). OA results in lower bioavailable carbonate (aragonite, calcite) required by marine calcifiers during early developmental stages and as adults (*224*), which can cause developmental delays and morphological abnormalities (*225*, *226*). Molecular analysis indicates that gene expression for biomineralization is not affected by OA conditions, suggesting effects on adult shells are primarily through dissolution (*227*). Beyond impacts on calcification and shell dissolution, acidification directly impacts immune capabilities in mollusks and crustaceans. Lobster exhibit limited ability to modulate hemolymph pH, and when subjected to hypercapnic seawater, immune responses such as hemocyte count and phagocytic capability are reduced by ~50% (*228*). OA suppresses immune response gene expression and induces both oxidative stress and antioxidant activities in the Pacific oyster (*229*). In Eastern oyster and hard clams, hypercapnic seawater enhances the immunosuppressive effects of heavy metal exposure (*230*). In synergy with temperature stress, OA can affect shellfish metabolism, resulting in greater susceptibility to *Vibrio* infection and negative immune alterations associated with greater disease vulnerability (e.g., *231*, *232*).

Under OA conditions, finfish can regulate their blood acid–base balance at the gills and kidney with relatively low metabolic cost (e.g., red drum; *233*, *234*), and this ability helps protect adult fish from acute OA stress. Acidification can even offset deleterious effects of elevated temperatures such as oxidative stress and heavy metal contaminant uptake in adult fish (*235*). Unlike mollusks, finfish exhibit increased mineralization of otoliths and skeletons under acidifying conditions (*236*–*238*), suggesting indirect mechanisms of impact. In contrast, OA has severe negative effects on temperate larval finfish (Atlantic cod, Pacific cod, Atlantic herring, silversides), resulting in reduced survival, stunted growth and development, poor body condition, and/or tissue damage (*239*–*242*). Acidification induces oxidative stress and alters the antioxidant defense response to a heavy metal (cadmium) in flounder larvae (*243*). Elevated CO_2 can elicit behavioral changes by potentially modifying fish neuroreceptors (*244*). Disorientation and altered olfaction have been reported for several coral reef fishes (e.g., *245*, *246*), although these observations cannot be replicated with other reef fish species (*247*), suggesting species-specific effects. OA can change the structure and binding affinity of peptides that mediate behavior in aquatic organisms, providing a mechanism for altered, and potentially harmful, behaviors resulting from acidification (*248*).

In addition to climate change conditions altering the host–pathogen relationship, a pathogen can alter the host–pathogen–environment relationship. For example, oysters naturally resistant to the effects of OA exhibit diminished resistance when challenged with the shellfish pathogen *Vibrio tubiashii* (*249*).

Not all impacts from aquaculture diseases due to climate change-driven alterations are negative. In some regions, increased precipitation may reduce infestations of sea lice and infections by protozoans, such as the oyster pathogen *Haplosporidium nelsoni*, due to reduced parasite survival at lower salinities (*250*, *251*). Levels of acceptable CO_2 in current rearing facilities (e.g., RAS) show that aquaculture has already been operating at concentrations 10-fold or greater than natural acidification (*252*). Agriculture and aquaculture are inherently selective for rearing stock, and producers are continually choosing the most robust and resilient species or characteristics for their purposes. The large difference in effects between early life-history stages and adults (see previous paragraphs) indicates that organisms in grow-out operations may be less affected by OA. Although climate change stressors can induce greater physiological susceptibility to infection, aquaculture organisms are housed in conditions of high nutrient availability and low predation threat, which could offset or partly compensate for those negative environmental effects (*252*).

Climate change impact on species distribution and disease

Long-term temperature changes can change wild species distributions as animals seek favorable conditions for their respective physiologies. In sub-Arctic Icelandic waters, a northwesterly shift in habitat range for 59 of 82 species (72%) occurred over a two-decade period (*253*). This shift coincided with a 1°C increase in sea surface temperature (SST) for this same period, and many of the shifted species lived near the surface or were living near the upper end of their thermal range (*253*). A similar shift in groundfish distribution was noticed from 2014–16 in the Gulf of Alaska that coincided with anomalously warm temperatures (a.k.a. The Blob; *254*, *255*). In this instance, differences in behavior were observed between species and between foraging guilds. There was also a general trend for most species that coincided with increasing sea surface temperature for all life stages to move to deeper and cooler waters. Distribution shifts can alter trophic interactions, adding potential nutritional stress and higher disease susceptibility.

Although physiological requirements and limitations are likely motivators, climate-related distribution changes can be well explained by local spatial and temporal rate changes in temperature (*256*). High-resolution, subregional projections of species distribution shifts under multiple climate models exhibit good agreement for 78–79% of ~550 marine species along the U.S. continental shelf (*257*), suggesting that it is feasible to make predictions about endemic species shifts. Species that shift in spatial distribution may be naive to indigenous pathogens in the new location or may introduce novel pathogens into the new location (*190*, *191*). For the AOA options, species distribution shifts could alter the overlap between wild and cultured populations, creating different sets of disease interaction opportunities over time.

Climate change impact on pathogens

Climate change-induced effects on temperature and ocean chemistry are expected to affect pathogen distribution, abundance, and ability to cause disease (*258*, *259*). Similar to their hosts, each pathogen—whether bacterial, viral, fungal, or parasitic in nature—will have a suite of environmental conditions under which it thrives and persists. Optimal conditions may be life stage-specific, particularly for parasites that alternate between free-swimming infectious, attached and feeding, and sessile reproductive forms (*260*).

Changes in pathogens that can be affected by climate change and its consequences include:

- The ability to infect through changes in pathogen gene expression, altered host microbiomes, and altered host immune responses.
- The degree of virulence or disease-causing abilities through changes in pathogen gene expression and host susceptibility.
- Altering development or the life cycle by activating replication or through faster replication, timing of life-cycle phases, availability of suitable intermediate and definitive hosts, and availability of transmission vectors.
- Passive movement by environmental events such as hurricanes, tsunamis, and airborne transport.

Thermal performance curves are frequently used to understand and predict the theoretical interaction between host and pathogen over a range of temperatures. A simple example for cool-adapted and warm-adapted hosts and pathogens is shown in Figure 3 (see *261* for details about the underlying thermal mismatch hypothesis). Temperatures with differentials between the host and pathogen performance curves (double-headed arrows in Figure 3, panels a and b) are where the pathogen has an advantage over the host (Figure 3, panels c and d). Pathogens with temperature optima higher than their hosts are likely to expand in prevalence or distribution under higher temperature conditions, such as occurred with *Hematodinium* parasites in crab (see *197*, their Figure 2). There are many elements influencing this approach (e.g., curves are often generated under isolated lab conditions rather than real-world conditions), and empirical curves will likely have different shapes than shown in Figure 3. Nonetheless, it provides a practical framework for assessing and predicting specific host–pathogen outcomes with changing temperatures (*261*).

If local temperatures and water-quality parameters push the boundaries of tolerance for host species, pathogens may gain an edge over host resistance. Pathogens, especially viruses and bacteria, have much shorter replication times, allowing them to adapt to new conditions much more quickly than host species. In general, growth and reproduction of pathogenic bacteria (*200, 262*) and parasites are expected to increase within projected temperature changes (*221*), thus increasing the opportunity for encounters with vulnerable

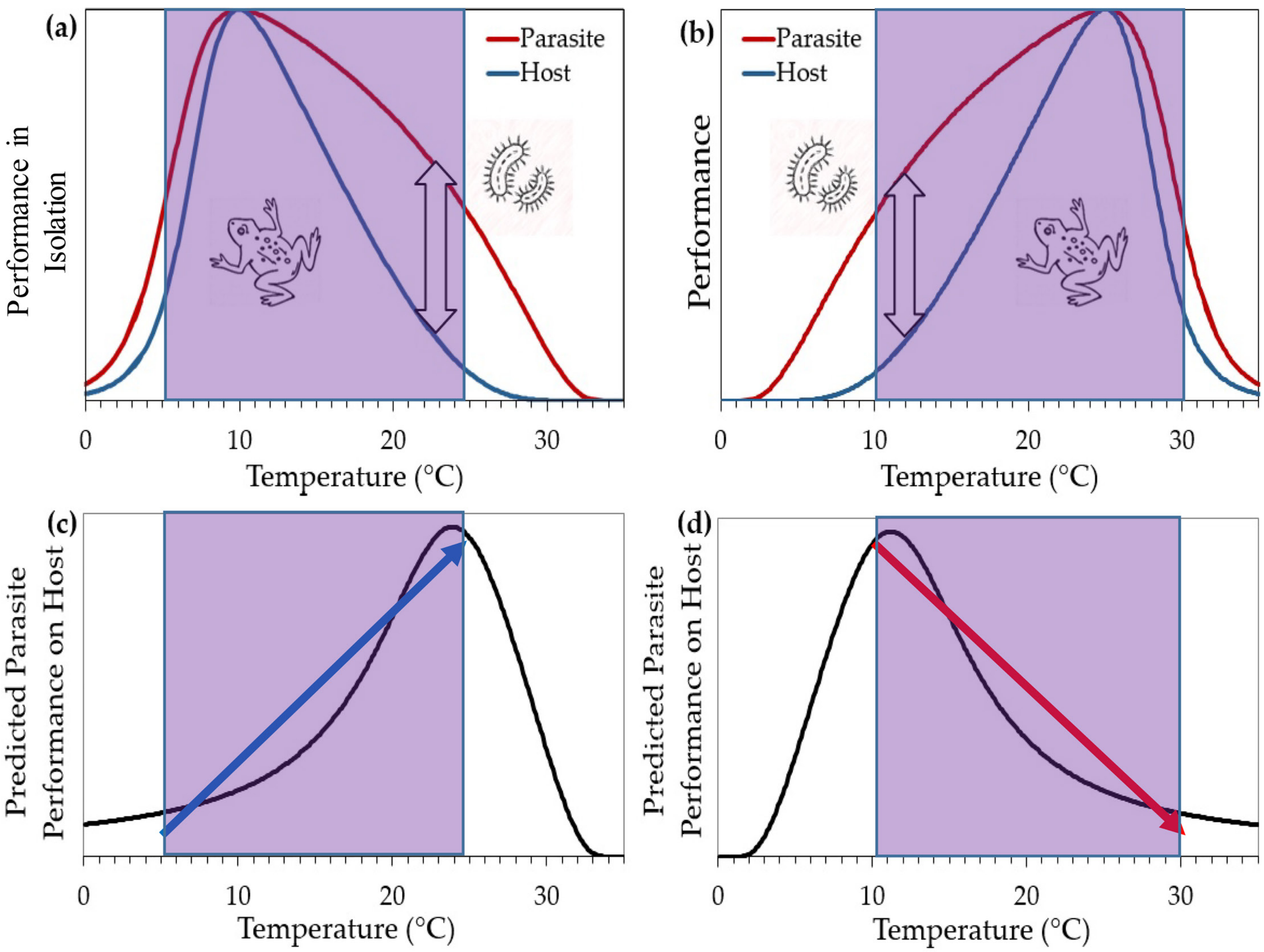


Figure 3. Simple thermal performance curve examples for a host and a pathogen (panels a and b), and resulting predictions of the likelihood of pathogen effect (e.g., prevalence) based on the differential between the curves (panels c and d). From Rohr and Cohen (2020; *261*).

host species. Furthermore, some pathogens may not be as impacted by environmental changes as their hosts. An example would be *Cryptocaryon irritans*, the parasitic cause of marine white spot disease or "marine ich," which can successfully infect, grow, and reproduce in seawater pH ranging from 6 to 9. Therefore, changes in marine white spot disease are more likely due to negative OA effects on the hosts (*263*).

Increased mean temperatures can also alter the window in which conditions are ideal for pathogens to replicate. For parasites, this might mean that they no longer experience a dormant period during winter (*221*). The extended window can result in more generations of organisms, speeding up the process to adapt and overcome the host's natural protections or therapeutics given to farmed species. Changing temperatures may shift the ranges of pathogens, which may include mechanisms of how the pathogen moves through the environment (e.g., changes in currents or stratification), differences in movements of vectors, or changes in host migration (*221*).

Ultimately, disease expression is the combination of pathogen viability and robustness against the host's ability to respond to infection/infestation, which can be modulated by a set of environmental conditions. For example, the responsible agent for Dermo disease, *Perkinsus marinus*, is a warm-water parasite that was historically not present or viable in the northeastern Atlantic (*264*). After the early 1990s, the range of Dermo disease expanded northward of Chesapeake Bay (see *180*, their Figure 3). Although this organism was likely introduced by humans through infected oysters (rather than a natural range expansion of the parasite), long-term increases in winter water temperatures provided the change in physical conditions that likely enabled the expression of disease (*264*). Similarly, Multinucleated Sphere Unknown disease (MSX) in Eastern oyster populations, which is caused by the parasite *Haplosporidium nelsoni,* expanded north of the mid-Atlantic coast with increased winter water temperatures and increased salinity (see *180*, their Figure 3). Disease expression retreated significantly upon the onset of cooler temperatures and wetter conditions that lowered salinity (*251*), providing a natural demonstration of the effect of those environmental conditions on disease distribution.

Region-Specific Climate Change and Disease Concerns

For the Gulf of Mexico and Southern California Bight study areas, multiple changes in environmental conditions are likely to occur, each with potential to affect organism health. Warming temperatures are expected to foment sea level rise, exacerbate erosion and coastal flooding, increase storm frequency and intensity, impact frequency and seasonality of harmful algal blooms, facilitate increased hypoxic conditions, increase precipitation (frequency and/or intensity), and impact the normal phytoplankton biomass (*7*). Additionally, the Southern California Bight is particularly vulnerable to ocean acidification due to seasonal coastal upwelling of deep ocean water that already has a lower pH than surface water (*266*).

Gulf of Mexico

The northern Gulf of Mexico is particularly vulnerable to hypoxia. In addition to warm temperatures during the summer, this area has significant freshwater and nutrient influx from the Midwest via the Mississippi River. Warm surface temperatures and freshwater influx contribute to stratification that inhibits mixing and prevents re-oxygenation of hypoxic bottom waters. Additionally, nutrient-rich organic loading from the Mississippi River can aid the growth of algal blooms that consume oxygen when they die and inhibit sunlight penetration. As a result, part of the Gulf near the river outlet is characterized as having the greatest seasonal hypoxia (dissolved oxygen ≤2 mg/L) in the western hemisphere (*265*).

Predicted increases in the frequency and severity of storms increase the risk of disease spread in the event of finfish escape from damaged net pens, or if shellfish and seaweed/macroalgae structures break loose from their moorings and drift (*259*).

Although ENSO is generated in the Pacific Ocean, the Gulf of Mexico is affected by it. Typical El Niño conditions produce cooler and wetter winter conditions, with stronger storms and more flooding through the Gulf coast.[31] El Niño weather conditions can induce significant changes in temperature and salinity, with potential to affect disease distribution and outbreaks (*180*).

---

[31] https://www.weather.gov/tae/enso

Southern California Bight

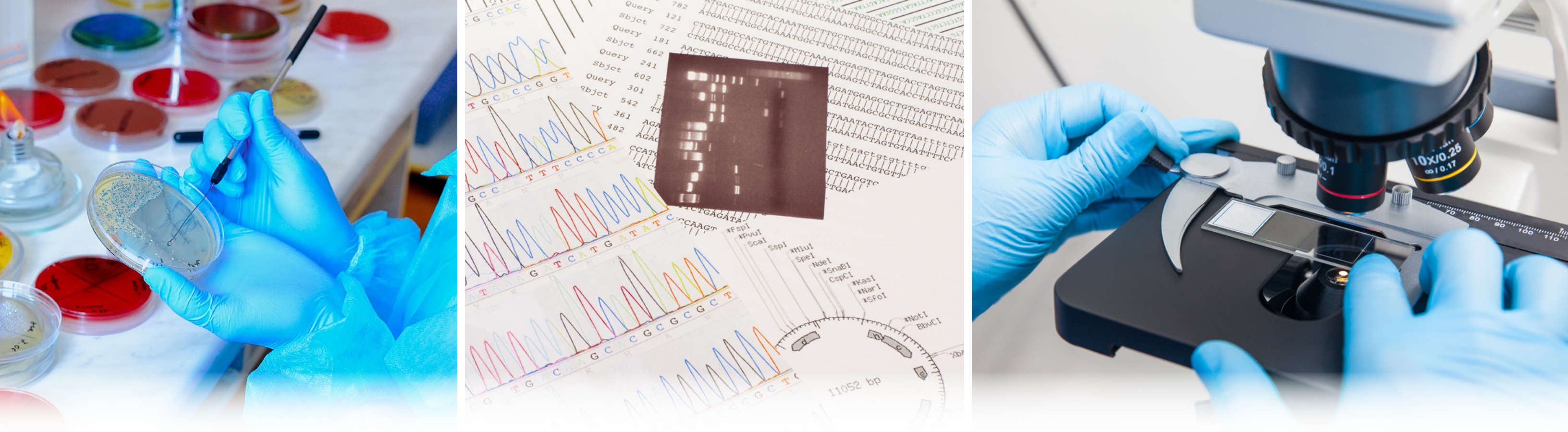
Along the eastern Pacific Ocean, the U.S. West Coast is particularly vulnerable to ocean acidification, with projections of low biologically available carbonate over the next three decades. Seasonal upwelling of acidic waters from the ocean depths is expected to be more frequent and intense, bringing carbonate-depleted water to the surface at nearshore environments (*266*). Although deep water tends to be nutrient-rich, it is also lower in oxygen and higher in CO_2 than surface water, exacerbating acidification. Oyster growers in the Pacific Northwest have been observing the effects of ocean acidification on larval shellfish, resulting in lower growth and higher mortality, since about 2007 (*223*).

Longer-term weather variability is strongly driven by the El Niño/Southern Oscillation (ENSO), a naturally occurring phenomenon of sea surface temperature and atmospheric pressure fluctuations over the southern Pacific Ocean, that has a cycle of approximately 2–7 years.[32] These fluctuations generate general weather trends for the Southern California coast (El Niño = more rainfall; La Niña = less rainfall),[33] and monitoring ENSO provides an important tool for predicting and interpreting weather patterns.

---

[32] https://www.noaa.gov/jetstream/tropical/enso

[33] https://www.weather.gov/lox/elnino



Federal Agencies with Responsibilites for Aquatic Animal Health, Medicine, and Water Quality

In the United States, animal health professionals are asked to report detections of pathogens on the National List of Reportable Animal Diseases (NLRAD)[34,35] to USDA-APHIS Veterinary Services. The NLRAD contains pathogens listed by WOAH as well as pathogens deemed to be of specific concern in the United States. The NLRAD is reviewed and updated annually, and USDA is responsible for reporting detections to WOAH.

Freedom from a Disease

Freedom from a disease is a designation describing confidence that a particular pathogen is absent from a particular geographic area or farm, assuming a given prevalence detection threshold and diagnostic test sensitivity. In the international community, freedom from aquatic diseases is self-declared by WOAH member countries, and WOAH provides guidance for making those declarations.[36] Achievement of pathogen-free status is possible if: a) susceptible species are absent, b) the disease has not historically occurred, or c) structured surveillance shows no disease if disease status is unknown or disease occurred within the past ten years (*267*). An active disease situation requires eradication of the disease, adequate biosecurity measures, and no disease presence based on structured surveillance (*267*). Countries may use zones (based on geographic areas), compartments (based on management and biosecurity practices), or a combination of zones and compartments to declare part of a country free of disease (*134*). To sustain disease-free status, WOAH stipulates three conditions:

1. Detection, or even suspicion, of the disease must be reported to a competent authority.
2. A surveillance program for the disease is in place to provide early detection capability.
3. There are import requirements in place to prevent transfer into the disease-free area.

Establishing and maintaining disease-free status is a complex decision process that often employs a cost–benefit assessment and typically involves behavioral economics (*267*). Because producers are likely to carry most of the burden and benefit of pathogen-free status, their involvement in the decision process is crucial.

---

[34] https://www.aphis.usda.gov/aphis/ourfocus/animalhealth/monitoring-and-surveillance/nlrad/ct_national_list_reportable_animal_diseases

[35] https://www.aphis.usda.gov/animal_health/nahrs/downloads/nlrad-nahrs-disease-list.pdf

[36] https://www.woah.org/en/what-we-offer/self-declared-disease-status/

U.S. Department of Agriculture, Animal and Plant Health Inspection Service (USDA-APHIS)

USDA-APHIS serves as the lead agency for preventing, controlling, and eliminating diseases of commercially farmed aquatic animals, as well as leading the oversight of animal health programs nationwide. USDA-APHIS regulates national import and export of aquatic organisms in relation to organism health (*268*). However, U.S. Fish and Wildlife Service (USFWS) regulates the importation of salmonids under Title 50. The agency regulates the use of veterinary biologics, including commercially marketed vaccines, diagnostic kits, and other products of biological origin (*269*). The Chief Veterinary Medical Officer of USDA is responsible for reporting when WOAH-listed pathogens are detected (*268*).

Individual states have authority over intrastate and interstate movement of aquatic species, typically collaborating to manage regional movements. A state may require state-level reportable pathogens in addition to those on the WOAH list.[37] States are empowered to set their own standards for aquaculture regulations, usually through legislative action, and administration is housed within a competent public authority. For example, as a result of the Florida Aquaculture Policy Act, the Florida Department of Agriculture and Consumer Services is charged with permitting and regulation for the state's aquaculture sector.[38] In contrast, the California legislature makes the California Department of Fish and Wildlife (CDFW) the competent authority for aquaculture regulation in the state, and requires periodic reports to be submitted to the state senate from CDFW.[39] State authority allows for more specific rules that govern aquaculture. California, for example, disallows movement from southern to northern waters to protect the naïve northern California waters against pathogens reported from the state's southern coast,[40] and bivalves may not be transferred from San Diego Bay or Tomales Bay to prevent spread of OsHV-1 (C. Burge, CDFW, personal communication). Florida prohibits the culture of oyster stocks from Atlantic Coast waters in Florida's Gulf Coast waters to minimize introduction of MSX (multinucleated sphere unknown) disease.[41] Although policies differ among states, states can participate with USDA-APHIS monitoring and surveillance through agency programs. For example, under the USDA-APHIS National Animal Health Reporting System (NAHRS), state animal health officials provide monthly reports on reportable diseases and diseases of interest (e.g., emerging diseases) detected in agriculture and aquaculture species.[42]

The 2020 Executive Order 13921, *Promoting American Seafood Competitiveness and Economic Growth*, designated USDA as the lead federal agency for oversight of the health and promotion of farm-raised aquatic livestock (*67*, *161*). USDA's efforts are described in the National

---

[37] https://www.aphis.usda.gov/aphis/ourfocus/animalhealth/nvap/NVAP-Reference-Guide/Animal-Health-Emergency-Management/OIE-and-International-Standards

[38] http://www.leg.state.fl.us/statutes/index.cfm?App_mode=Display_Statute&URL=0500-0599/0597/0597.html

[39] https://nrm.dfg.ca.gov/FileHandler.ashx?DocumentID=203343&inline

[40] https://nrm.dfg.ca.gov/FileHandler.ashx?DocumentID=24619

[41] https://www.fdacs.gov/content/download/64045/file/aquaculture-bmp-manual.pdf

[42] https://www.aphis.usda.gov/aphis/ourfocus/animalhealth/monitoring-and-surveillance/sa_disease_reporting/ct_usda_aphis_animal_health

Aquaculture Health Plan and Standards 2021–23 (NAHPS).[43] The document establishes guidance to enable activities that protect the health of aquatic livestock, including disease reporting; standardization of testing laboratories and methods; biosecurity, training, and education; disease surveillance and response; data management; and health certification programs (*67*). This plan assists in the development of site-based health plans for aquatic animal health, and includes four options for aquaculture health inspection. Once an entity has opted in, the standards become mandatory for compliance. An example of one pathway is the Comprehensive Aquaculture Health Program Standards (CAHPS), based on five management pillars that come together for integral animal health management:

1. An aquatic animal health team (AAHT) consisting of professionals committed to a given site who determine pathogens of concern, risk mitigation strategies, disease surveillance, and reporting strategies.
2. Risk evaluation for the species being cultured, the pathogens of concern, the production method being used, and the end use of the livestock.
3. An early detection system and surveillance plan for pathogens of concern.
4. A disease investigation plan when morbidity and mortality events exceed thresholds.
5. A response, reporting, and recovery plan.

With specific reference to marine aquaculture, NAHPS requires that only animals with a known and approved health status may be stocked in federal marine waters, including negative testing for pathogens of concern relative to the relevant jurisdiction or by the susceptibility of species in the environment around the farm. Pathogen-free feed is also an important component of the standards for marine aquaculture (*67*).

U.S. Department of Agriculture, Center for Veterinary Biologics (USDA-CVB)

Veterinary care often requires biologically derived therapeutants, such as vaccines, bacterins, and disease diagnostic kits. USDA-CVB implements the provisions of the Virus–Serum–Toxin Act (VSTA)[44] to ensure that pure, safe, potent, and effective veterinary biologics are available for diagnosing, preventing, and treating animal diseases.

Vaccines

Vaccines are a highly useful and effective prophylactic measure against diseases in finfish. Because they are typically derived from a biological source, such as bacterial or viral protein, approval for marketing is obtained through USDA-CVB. At this time, there are seven approved vaccines for finfish:[45,46,47]

- Live *Arthrobacter* vaccine against bacterial kidney disease for salmonids.
- Killed *Yersinia* vaccine against enteric red mouth disease for salmonids.

---

[43] https://www.aphis.usda.gov/animal_health/animal_dis_spec/aquaculture/downloads/national-aquacult-health-plan-standards-2021-2023.pdf

[44] VSTA (Virus–Serum–Toxin Act). 1913. U.S. Code, title 21, sections 151–158.

[45] https://www.aphis.usda.gov/aphis/ourfocus/animalhealth/veterinary-biologics/CT_Vb_licensed_products

[46] https://www.aphis.usda.gov/animal_health/vet_biologics/publications/aquaproducts.pdf

[47] https://www.aphis.usda.gov/animal_health/vet_biologics/publications/currentprodcodebook.pdf

- Killed vaccine against multiple bacterial pathogens (*Aeromonas salmonicida*, *Vibrio anguillarum*, *V. ordalii*, and *V. salmonicida*) for salmonids.
- Killed vaccine against viral and bacterial pathogens (infectious salmon anemia virus, *Aeromonas salmonicida*, *Vibrio anguillarum*, *V. ordalii*, and *V. salmonicida*) for salmonids.
- DNA vaccine against infectious salmon anemia virus for salmonids.
- Live *Edwardsiella ictaluri* vaccine for catfish.
- Live *Flavobacterium columnare* vaccine for catfish.

In addition to USDA-approved vaccines, autogenous vaccines may be prepared from inactivated, nontoxic cultured microorganisms, either in a USDA-licensed facility for use by a health professional (licensed use) or under the license of a veterinarian (veterinarian exemption use). Typically, microorganisms isolated from diseased animals are cultured to produce a quantity sufficient for an effective dose, then inactivated to prevent growth after inoculation. Inactivation is often through physical or chemical methods, such as heat or formalin treatment. There are specifications for preparation and administration in the Code of Federal Regulations (9 CFR §113.113),[48] but the full process of review and approval is not required. The use of autogenous vaccines allows customized prevention of diseases, can be very cost-effective, and offers a route to avoid antimicrobial resistance (*270*). The Norwegian salmon industry was able to reduce utilization of antibiotics from >48,000 kg of antibiotics in 1987 to ~1,000 kg in 1996 though the use of autogenous vaccines (*270*). In fact, the three approved vaccines are a result of that success in the reduced use of antibiotics.

U.S. Food and Drug Administration, Center for Veterinary Medicine (FDA-CVM)

In the United States, the Food and Drug Administration's (FDA) Center for Veterinary Medicine (CVM) regulates the manufacture and distribution of food additives and drugs that will be given to aquatic animals. Several offices within the Center for Veterinary Medicine (CVM) play a role with a regard to aquaculture:

- The Office of New Animal Drug Evaluation (ONADE)[49] reviews information submitted by drug sponsors who want approval to manufacture and market drugs for use in the aquaculture industry. As mandated by the Federal Food, Drug, and Cosmetic Act (FD&C Act),[50] a new animal drug may not be sold in interstate commerce unless it is the subject of an approved new animal drug application (NADA), abbreviated NADA (ANADA), or a conditional approval (CNADA). All three types of NADAs are reviewed by ONADE. During the investigational stages of drug development, ONADE may also authorize investigational new animal drug (INAD) exemptions to allow for the use of the drug to generate data to support an approval. Part 21 of the Code of Federal Regulations[51] describes the regulations associated with NADAs and INADs.

---

[48] https://www.govinfo.gov/app/details/CFR-2016-title9-vol1/CFR-2016-title9-vol1-sec113-113
[49] https://www.fda.gov/about-fda/cvm-offices/office-new-animal-drug-evaluation
[50] FFDCA (Federal Food, Drug, and Cosmetic Act). 1938. U.S. Code, title 21, sections 301–399; https://www.fda.gov/regulatory-information/laws-enforced-fda/federal-food-drug-and-cosmetic-act-fdc-act
[51] https://www.accessdata.fda.gov/scripts/cdrh/cfdocs/cfCFR/CFRSearch.cfm?CFRPart=514

- With the passage of the Minor Use and Minor Species Animal Health Act (MUMS Act),[52] the Office of Minor Use and Minor Species Animal Drug Development (OMUMS)[53] was developed to assist in making more drugs legally available to treat minor animal species such as those raised in aquaculture. OMUMS administers the Index of Legally Marketed Unapproved New Animal Drugs for Minor Species ("the Index"). The Index is a list of new animal drugs intended for use in non-food-producing minor species (e.g., ornamental fish) that have had their safety and effectiveness affirmed through an alternate review process involving expert panels external to FDA.[54] OMUMS also manages the MUMS Designation program which provides incentives for sponsors to seek approval of new animal drugs for MUMS indications.
- The Office of Research (OR) conducts aquaculture research and assists in ensuring that fish derived from aquaculture production environments are safe for human consumption.[55] The Office of Surveillance and Compliance (OSC)[56] is responsible for compliance-related actions, post-approval monitoring (e.g., adverse drug event reporting), and animal feed safety. OSC reviews notices that a substance (including an aquaculture feed substance) is generally recognized as safe (GRAS)[57] for a specific use within an animal food, approves food additive petitions (FAP)[58], and regulates medicated animal feeds (i.e., feeds that contain a new animal drug). A medicated feed mill license is required to manufacture some medicated feeds;[59] these licenses are also approved by OSC.

FDA-CVM maintains a list of approved drugs for aquaculture,[60] categorized by the method of administration (immersion, injectable, medicated feed; *271*). The Animal Medicinal Drug Use Clarification Act[61] and FDA's Compliance Guide[62] allow veterinarians to administer approved therapeutic drugs beyond approved uses (known as "extra-label use") under strict conditions. However, administration of antimicrobials and prescription/veterinary feed directive drugs in the United States does require a valid veterinarian–client–patient relationship.[63] Although there have been successful efforts to gain approval of drugs for aquaculture, these have been primarily focused on freshwater aquaculture species, leaving a need for similar efforts for marine aquaculture species.

---

[52] MUMS Act (Minor Use and Minor Species Animal Health Act). 2004. U.S. Code, title 21, section 360.

[53] http://www.fda.gov/AnimalVeterinary/DevelopmentApprovalProcess/MinorUseMinorSpecies/default.htm

[54] https://www.fda.gov/animal-veterinary/minor-useminor-species/index-legally-marketed-unapproved-new-animal-drugs-minor-species

[55] https://www.fda.gov/about-fda/cvm-offices/office-research

[56] https://www.fda.gov/about-fda/cvm-offices/office-surveillance-and-compliance

[57] https://www.fda.gov/animal-veterinary/animal-food-feeds/generally-recognized-safe-gras-notification-program

[58] https://www.accessdata.fda.gov/scripts/cdrh/cfdocs/cfCFR/CFRSearch.cfm?CFRPart=571&showFR=1&subpartNode=21:6.0.1.1.21.1

[59] https://www.fda.gov/animal-veterinary/animal-food-feeds/medicated-feeds

[60] https://www.fda.gov/animal-veterinary/aquaculture/approved-aquaculture-drugs

[61] AMDUCA (Animal Medicinal Drug Use Clarification Act). 1994. U.S. Code, title 21, section 530; https://www.fda.gov/animal-veterinary/guidance-regulations/animal-medicinal-drug-use-clarification-act-1994-amduca

[62] Section 615.115, Extralabel Use of Medicated Feeds for Minor Species; https://www.fda.gov/regulatory-information/search-fda-guidance-documents/cpg-sec-615115-extralabel-use-medicated-feeds-minor-species

[63] https://www.avma.org/resources-tools/pet-owners/petcare/veterinarian-client-patient-relationship-vcpr

U.S. Fish and Wildlife Service, Aquatic Animal Drug Approval Partnership (USFWS-AADAP)

Because the market for drugs for aquatic organisms is much smaller than for terrestrial agriculture, the U.S. Fish and Wildlife Service supports a program to test and obtain approval for aquatic drugs. The Aquatic Animal Drug Approval Partnership (AADAP)[64] is dedicated to developing and coordinating safety and effectiveness studies for approval by FDA, and oversees the Investigational New Animal Drug (INAD) Program. The National INAD Program allows for the legal use of specific investigational drugs by participants. Participants in the INAD Program are required to collect and submit data to AADAP and pay a small fee. AADAP interfaces with drug sponsors to help ensure a drug manufacturer is available, and conducts drug-related research. As marine aquaculture and its health care needs expand and as NOAA increases its focus on developing the information necessary to make drugs available for use in marine species, AADAP can serve as a partner in addressing those needs. NOAA has initiated a marine aquaculture medicine cooperative aimed at partnering with external stakeholders such as AADAP to generate the information necessary to achieve FDA approval of the veterinary drugs needed in marine aquaculture.

As an agency with oversight of aquatic wildlife in the United States, USFWS invests in substantial aquatic animal health services, including regional fish health centers[65] with inspection and diagnostics capabilities, a recently updated Aquatic Animal Health Policy,[66] and an informative handbook detailing procedures and protocols used by USFWS to implement the policy (*68*).[67]

Antibiotics/antiparasitics

The route of administration for antibiotics depends on the rearing system and approved use. Antibiotic baths have shown potential to fight infection and reduce mortality in abalone (*272*), and certain antibiotics on salmon farms are added to feed to treat disease (*273*), but these measures are not approved in the United States. Development of antibiotic resistance in aquatic bacterial pathogens is a concern for aquaculture use because antibiotics in unconsumed feed, or passed via feces, can later be accumulated by microorganisms in the surrounding environment, leading to antibiotic or even antiparasitic resistance (*274*). Resistance occurs when exposure to an antibiotic creates selective pressure that results in microorganisms or parasites that are no longer susceptible to the product (*275*). Research from Chilean salmon farms shows resistance to oxytetracycline, oxolinic acid, and florfenicol in benthic bacteria following antibiotic use (*276*). The study detected elevated levels of resistant bacteria up to 1 km away from the farm site. Additional research suggests that antibiotic resistance in marine sediments near salmon farms positively correlates with a higher use of on-farm antibiotics (*277*).

---

[64] https://www.fws.gov/program/aquatic-animal-health/aquatic-animal-drug-approval-partnership

[65] https://www.fws.gov/program/aquatic-animal-health/fish-health-centers

[66] https://www.fws.gov/policy-library/713fw1

[67] https://www.fws.gov/sites/default/files/policy/files/AquaticAnimalHealthProceduresandProtocols.pdf

Many countries have strong regulations surrounding antibiotic use in animals, and some have banned disease-prevention uses. For example, concerns about human health impacts of resistance development in food animals led to the ban of antibiotics as growth promoters by the European Union in 2006 (*278*, *279*), and the United States has successfully phased out use of medically important antibiotics as growth promoters (*280*). In the United States, all FDA-approved antibiotics for use in animals require a veterinary feed directive (VFD) administered in feed or a veterinary prescription if administered by other routes (e.g., immersion or injection; *281*). Increased regulation of antibiotics in Europe has not resulted in economic losses, and European aquaculture management practices stress the need for producer education and more investment for research on the use of prophylactics (probiotics, prebiotics, and vaccines; *279*).

Prophylactics

In light of concerns surrounding antibiotic resistance in cultured and wild species, supplementation of invertebrate and finfish diets with pro- and prebiotics is receiving increased attention (*282*). While some research has been promising for the use of these products to enhance production and improve disease resistance (*283*), there are still significant research gaps (*282*–*285*).

Probiotics

Probiotics are cultures of live microorganisms that claim to confer a health benefit for the organism consuming them. One potential mechanism supporting the health claims is the ability of beneficial microorganisms in the probiotic culture to successfully compete against pathogenic bacteria (*286*). Research has shown that use of probiotics in aquaculture species can result in improved feed-conversion ratios, in addition to elevated immune response to pathogens. For example, rainbow trout fed a probiotic containing *Clostridium butyricum* showed improved resistance against vibriosis by increasing the phagocytic activity of leucocytes (*284*). Supplementation with probiotics during the larval stage for invertebrates under culture aided in overall growth performance and feed efficiency (*287*).

Research on shellfish probiotics in the United States has established a series of principles for probiotic use (*288*):

- The probiotic should not be harmful, pathogenic, or toxigenic.
- The probiotic should be administered to the host through ingestion for potential colonization and replication within the host digestive system.
- The probiotic should produce the desired effect, whether localized or systemic.
- The probiotic should work in vivo, instead of only in vitro.
- The probiotic should not contain virulence genes or antibiotic resistance genes.

While significant research supports probiotics as useful alternatives to antibiotics in aquaculture, there has been little progress on approval of their use for U.S. aquaculture. The process for approval of viable sources of microorganisms in the United States is regulated by FDA-CVM.

A live microbial product for animals that is intended for use in the cure, mitigation, treatment, or prevention of disease is regulated as a new animal drug. In some instances, the microorganisms in live microbial products may have been selected or genetically modified for production of a novel substance(s) that acts in this manner, and these products are also new animal drugs. Regulatory considerations to approve or accept a viable source of microorganism as an animal food are described in CPG 689.100, *Animal Products Containing Live Microorganisms*. Currently, the best recommendation for seeking approval for a viable source of microorganism product is to contact the FDA at askcvm@fda.hhs.gov to find out what regulatory pathway applies for a particular market formulation (*289*).

While research suggests probiotics can be a net positive for a growing aquaculture sector (*284*, *287*), there are potential risks. Recently, concerns were raised about the accuracy of traceability with the labeling of certain commercial probiotics used in aquaculture. Laboratory analysis of several marketed probiotics revealed that some contained antibiotic-resistant genes, some contained wholly different bacteria than what was labeled, and some contained incorrect amounts of the probiotic advertised, in addition to other bacterial species. Accuracy in labeling of probiotics is crucial given the potential for different species of bacteria to behave differently when ingested by a culture species; mislabeling has the potential to harm operations and endanger wild species via the transfer of pathogens and resistance from cultured animals (*290*).

Prebiotics

In contrast to probiotics, prebiotics are nondigestible food ingredients that modulate the growth or activity of microorganisms in the digestive system of the organisms consuming them (*283*, *285*). Research on marine finfish suggests that there are potential positive benefits, one being the potential to activate innate immunity in fish species (*291*); research also suggests that prebiotics can modify the composition of gastrointestinal microorganisms in fish (*286*). However, most research concludes that the impacts on health, especially disease resistance, are not well elucidated (*286*) and that impacts of prebiotic substances vary considerably among aquaculture species (*291*). Additionally, most research on prebiotics (and probiotics) has not been conducted within the larger scale of commercial aquaculture operations (*282*, *285*), and will required further study to provide justification for an endorsement of commercial application. Multiple species of popular marine finfish used in aquaculture have been subjects of prebiotic research; therefore, there is a growing baseline of information for marine aquaculture species as investigation continues (*285*, *286*).

Of additional interest is the use of synbiotics, or a combination of prebiotics and probiotics that work synergistically to benefit the host (*285*, *292*). Initial research does suggest that there are benefits when the two products are used in tandem, and that synbiotics may offer protection against infectious disease (*292*), but further research—specifically challenge studies—is still needed to bolster these findings (*293*).

U.S. Environmental Protection Agency (EPA)

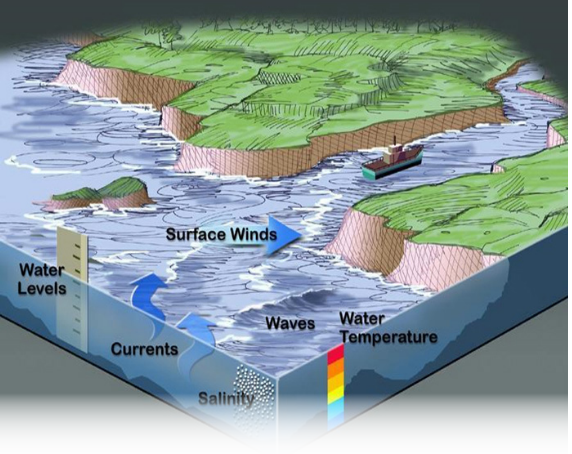
The U.S. Environmental Protection Agency (EPA) has responsibility for protecting and managing the quality of U.S. waters under the authority of the Clean Water Act. Section 301(a) of the CWA prohibits the “discharge of pollutants” except in compliance-prescribed provisions of the CWA, including section 402. Section 402 of the CWA establishes the National Pollutant Discharge Elimination System (NPDES) and authorizes EPA (or states authorized by EPA) to issue permits for point-source discharges of pollutants into U.S. waters, including the territorial seas.[68] For purposes of the CWA, offshore federal waters begin 3 miles from shore for all states. In the contiguous zone and the ocean beyond 3 miles, EPA is the sole permit-issuing authority for NPDES-regulated discharges. The term “pollutant” is defined in CWA section 502(6) and §122.2. The statute defines pollutant very broadly and includes any type of industrial, municipal, or agricultural waste (including heat) discharged into water. Biological materials, including living organisms, can and have been deemed as pollutants in several cases (*294*). EPA has defined Concentrated Aquatic Animal Production Facilities (CAAPFs)[69] and has developed specific regulations for discharges from them. Aquaculture discharges typically regulated under NPDES include unconsumed feed and feces; dissolved nutrients (nitrogen, phosphorus, ammonium); suspended and settleable solids; dissolved oxygen and biological oxygen demand; and unconsumed therapeutic drugs and chemicals.

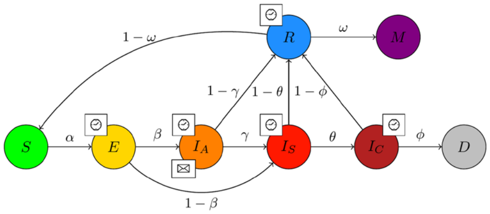
NPDES permits may be individual (specifically tailored to a single facility) or general (tailored to cover multiple operations with similar types of discharges, often within a specified geographic area). EPA publishes notice of the draft permit for public comment, typically for 30–60 days depending on the level of public interest. Following the close of the public comment period, EPA will consider all comments received and, as appropriate, finalize the permit. Depending upon the nature of the proposed discharge and the complexity of the public comments, the permitting process could exceed 180 days from the day the application is received. A clear and complete application package will expedite the issuance process. NPDES permits are issued for a period not to exceed 5 years. Monitoring results must be regularly reported to EPA (the frequency will be identified in the permit), and annual reports may also be required. EPA may also perform compliance inspections at the facility. Permits must be reapplied for every 5 years for as long as the facility continues to discharge.

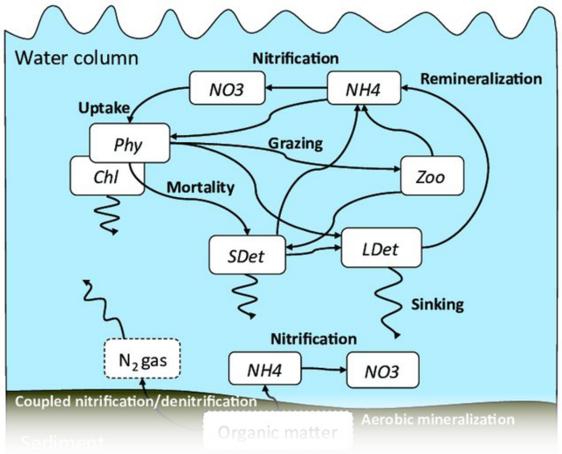
---

[68] https://www.epa.gov/npdes

[69] https://www.ecfr.gov/current/title-40/chapter-I/subchapter-D/part-122#122.24







Disease Modeling

Although empirical observations and experiments provide information supporting or refuting biosecurity decisions or actions, specific data can be inconsistent or scarce. Disease modeling can and has been used to identify important parameters for establishing policy and management actions for processes such as site selection, disease control, disease outbreak factors, or even anticipated effects of climate change (*295*–*297*). Coupled biological–physical modeling can also inform site parameters, such as stocking densities for seaweed/macroalgae (*173*). The power of modeling lies in formulating testable hypotheses and updating the algorithms when new data are provided. The limitations of modeling are its dependence on parameters and data included in the algorithms, which may be incomplete, incorrect, or inappropriate for the intended use. Nonetheless, disease modeling permits inferences across data gaps and insights into complex datasets, often with statistical testing capability that can complement empirical evidence and expert opinion.

Traditional epidemiological modeling of disease spread for terrestrial organisms (including humans) has relied primarily on susceptible–infected–recovered (SIR) models (*298*) or derivatives of SIR (e.g., susceptible–exposed–infected–removed, or SEIR). However, the ability of water to transport pathogens to remote susceptible individuals has added a need to include an environmental feature to account for the absence of direct interaction between susceptible and infected individuals. Although direct transmission models can be applied for cases where shedding rates of pathogens are high or the pathogen is environmentally persistent, environmental transmission models tend to be more accurate for many aquatic pathogens by accounting for time-lagged, indirect routes of pathogen transfer (*299*).

Pathogen dispersion is a common application for modeling the effects or projected effects of aquaculture farms on each other and on the natural environment (*16*). For the Gulf of Mexico, there are well developed models identifying and characterizing transport that influence the distribution of particulate objects, such as larvae and oil droplets from an oil spill (reviewed in *300*), and these have good potential to be applied to pathogens. An obvious caveat is that the transport properties of larvae and oil droplets will be different from most pathogens. Many of the efforts described by Justić et al. (*300*) provide pathogen dispersion models, and rely on the Finite-Volume Community Ocean Model (FVCOM),[70] a scalable, unstructured grid, 3D algorithm that has an open source code. FVCOM is useful for water bodies with complex and seasonally variable movements, such as fjords and inland waters, and it has served as the hydrodynamic basis for biological–physical models exploring

---

[70] https://www.fvcom.org/

pathogen connectivity between farms (*301*). Biological–physical modeling typically has three components: a hydrodynamic model, a particle-tracking model, and a biological model (*301*). Dispersion models can utilize the first two elements (hydrodynamics and particle tracking) to provide information for characterizing pathogen connectivity between farms (*302*). Model specificity can be increased by the addition of biological information about host–pathogen interactions, such as pathogen reproduction rates, contact and density thresholds, and infectious dose (*301*). Both dispersion and biological–physical models have utility for farm siting assessments, establishment of biosecurity zones, and predicting disease spread.

In more open ocean areas such as the southern California coast, use of Lagrangian analysis with oceanic general circulation models—such as the Regional Ocean Modeling System (ROMS)[71] or the Hybrid Coordinate Ocean Model (HYCOM)[72]—can provide both hydrodynamics and particle tracking capabilities (*303*). Lagrangian modeling has been used to trace a wide range of particle sizes, from microbes (*304*) to icebergs (*305*). In southern California, ROMS simulations within 15 km of shore found that particle dispersion from the coast was influenced more by submesoscale currents, rather than tides (*306*). Submesoscales have a strong effect on the distribution and patchiness of plankton both vertically and horizontally (reviewed by *307*), and may require special attention for pathogen transfer.

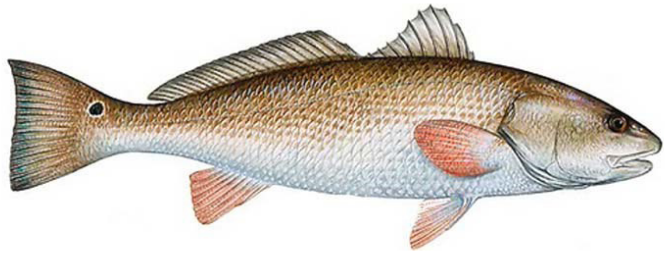
Because pathogen transfer includes active transport by human activities, modeling is used to assess hazards of spread through evaluation of farm networks. This type of effort has found that farms using more than one fish supplier are associated with a wider array of pathogen (or greater pathogen richness) than those using a single supplier (*308*). Network analysis using a susceptible–infected (SI) model for a viral disease in salmon identified human-assisted spread of the pathogen through live fish transport as a much greater factor than local passive spread (*309*). That model accurately reconstructed the pattern and timing of reported infection for approximately one-third of the Irish salmon industry between 2009 and 2017. These types of network models not only provide a method of identifying critical factors in pathogen spread, but also provide opportunities to develop effective interventions, such as targeting biosecurity to farms with the most connections (*309*).

Modeling can also be applied to explore the possiblities of using aquaculture as a tool for environmental improvements. An SI model adapted for parasite transmission in aquatic systems estimated that aquacultured oysters could remove parasites from the water and potentially reduce the infection pressure on sympatric wild oysters (*310*). This shows that modeling efforts have good potential for exploring and testing hypotheses for improving ecosystem services of aquaculture or for restorative aquaculture, prior to real-world testing and implementation.

---

[71] https://www.myroms.org/

[72] https://www.hycom.org/


Candidate Organisms for Marine Aquaculture

Marine organisms have been cultivated for food for centuries (*311*). Clam gardens cultivated by indigenous peoples of the Pacific Northwest have been radiodated to 3,500 years ago (*312*), and production of the seaweed *Pyropia* dates to 1481 in Korea (*313*). Currently, marine aquaculture in the United States generates 90 million pounds of seafood valued at $430 million,[73] and some species already in cultivation are candidates for offshore rearing. Table 7 provides a list of candidate species for cultivation in the Gulf of Mexico and the Southern California Bight in the future. This list is based on multiple factors, including a history of culture, consumption of wild conspecifics in the United States, and/or high potential for economic and biological suitability for culture in U.S. waters. A marine finfish aquaculture feasibility workshop and stakeholder survey held in 2017[74] assessed and reported on eighteen non-salmonid finfish species.[75] A subsequent workshop and symposium in 2019 reported on the aquaculture status and potential of these eighteen finfish species (*314*).

Table 7. Candidate organisms for marine aquaculture and relevant geographic region. Finfish species in bold were assessed for aquaculture status in workshops (*314*). Organisms marked with asterisks are also applicable for conservation or restorative aquaculture. This table is not comprehensive or exclusive for species that may be proposed for aquaculture, and the absence of a species does not necessarily exclude it from cultivation. GOM = Gulf of Mexico, SCB = Southern California Bight.

Group	Organism	Region
Algae	Dead man's fingers (*Codium* spp.)	GOM
	Eucheuma spp.	GOM
	Graciliaria spp.	GOM
	Sargassum spp.	GOM
	Sea lettuce (*Ulva* spp.)	GOM
	Bladderwrack (*Fucus distichus*)	SCB
	Giant kelp (*Macrocystis pyrifera*)	SCB
	Kombu (*Laminaria setchellii*)	SCB
	Nori (*Pyrophia* spp.)	SCB
	Ribbon/winged kelp (*Alaria marginata*)	SCB
	Sea cabbage/sweet kombu (*Saccharina sessilis*)	SCB
	Sea palm (*Postelsia palmaeformis*)	SCB

---

[73] https://www.fisheries.noaa.gov/national/aquaculture/us-aquaculture

[74] https://www.fau.edu/hboi/research/aquaculture-innovation/center-for-marine-and-warm-water-aquaculture/education-outreach/status-of-marine-fish/

[75] https://www.fau.edu/hboi/documents/status-marine-finfish/status-of-marine-finfish.xlsx

Table 7 (continued). Candidate organisms for marine aquaculture and relevant geographic region.

Group	Organism	Region
Algae	Sugar kelp (*Saccharina latissima*)	SCB
	Turkish washcloth (*Mastocarpus papillatus*)	SCB
	Sea spaghetti (*Gracilaria andersonii*)	GOM, SCB
Finfish	**Almaco jack** (*Seriola rivoliana*)	GOM
	Black sea bass (*Centropristis striata*)	GOM
	Cobia (*Rachycentron canadum*)	GOM
	Florida pompano (*Trachinotus carolinus*)	GOM
	Greater amberjack/kampachi (*Seriola* spp.)	GOM
	Red drum (*Sciaenops ocellatus*)	GOM
	Southern flounder (*Paralichthys lethostigma*)	GOM
	Spotted seatrout (*Cynoscion nebulosus*)	GOM
	Tripletail (*Lobotes surinamensis*)	GOM
	California halibut (*Paralichthys californicus*)	SCB
	California yellowtail (*Seriola lalandi*)	SCB
	Olive flounder (*Paralichthys olivaceus*)	SCB
	Sablefish (*Anoplopoma fimbria*)	SCB
	Striped bass (*Morone saxatilis*)	SCB
	White sea bass (*Atractoscion nobilis*)	SCB
Invertebrates	Bay scallop (*Argopecten irradians*)	GOM
	Eastern oyster (*Crassostrea virginica*)	GOM
	Quahog (*Mercenaria mercenaria*)	GOM
	Southern quahog (*Mercenaria campechiensis*)	GOM
	Urchin (*Lytechinus variegatus*)	GOM
	Manila clam (*Venerupis philippinarum*)	GOM, SCB
	*California mussel (*Mytilus californianus*)	SCB
	*Olympia oyster (*Ostrea lurida*)	SCB
	*Pismo clam (*Tivela stultorum*)	SCB
	Abalone (*Haliotis* spp.)	SCB
	Mediterranean mussel (*Mytilus galloprovincialis*)	SCB
	Pacific oyster (*Magallana gigas*)	SCB
	Purple-hinged rock scallop (*Crassadoma gigantea*)	SCB

Final Remarks

This document was developed to provide key information about health management and biosecurity for planning marine aquaculture in U.S. federal waters. Although it presents a wide range of relevant topics and issues that deserve consideration in planning, it does not address seafood safety or public health, nor does it provide management details for specific diseases or pathogens. When aquaculture projects in federally managed waters are proposed, it is anticipated that products such as disease risk assessments for the proposed cultivated species and cost–benefit analyses for biosecurity would be generated as part of the proposal process.

There is considerable international emphasis on aquaculture biosecurity that can be beneficial for U.S. marine aquaculture. FAO’s Progressive Management Pathway identifies four stages of increasing knowledge and actions that can lead to sustainable health management and biosecurity systems: risk definition, initiation of biosecurity systems, enhancement of biosecurity systems, and sustainable biosecurity systems (*10*). Each stage relies on ever-improving knowledge about drivers, factors, and pathways for aquatic diseases. This knowledge requires surveillance or monitoring, good recordkeeping, and dedicated research for advancing our understanding of aquatic diseases (*10*, *160*).

The evolving nature of marine aquaculture requires credible research to identify factors and actions to support a thriving industry while conserving quality marine resources. We have little knowledge of most of the organisms in the oceans, and consequently, are unaware of their ecological roles and contributions to ecosystem functioning. Our desire to provide nutrition for humans should not be to the detriment of the services that oceans provide to Earth’s biosphere—it cannot be a zero-sum situation. By consistently updating knowledge, learning from experiences, and planning thoughtfully, we should be able to achieve the goals of productive aquaculture in a thriving ecosystem.

References

1. FAO (Food and Agriculture Organization of the United Nations). 2022. The State of World Fisheries and Aquaculture 2022: Towards Blue Transformation. Food and Agriculture Organization of the United Nations, Rome, Italy. DOI: 10.4060/cc0461en
2. Riley, K. L., L. C. Wickliffe, J. A. Jossart, J. K. MacKay, A. L. Randall, G. E. Bath, M. B. Balling, B. M. Jensen, and J. A. Morris, Jr. 2021. An Aquaculture Opportunity Area Atlas for the U.S. Gulf of Mexico. National Oceanic and Atmospheric Administration, Silver Spring, Maryland.
3. Morris Jr., J., J. MacKay, J. Jossart, L. Wickliffe, A. Randall, G. Bath, M. Balling, B. Jensen, and K. Riley. 2021. An Aquaculture Opportunity Area Atlas for the Southern California Bight. National Oceanic and Atmospheric Administration, Silver Spring, Maryland.
4. Bowen, J. T. 1970. A history of fish culture as related to the development of fishery programs. Pages 71–93 *in* N. G. Benson, editor. A Century of Fisheries in North America, volume 7. American Fisheries Society, Bethesda, Maryland.
5. Naylor, R. L., R. W. Hardy, A. H. Buschmann, S. R. Bush, L. Cao, D. H. Klinger, D. C. Little, J. Lubchenco, S. E. Shumway, and M. Troell. 2021. A 20-year retrospective review of global aquaculture. Nature 591(7851):551–563. DOI: 10.1038/s41586-021-03308-6
6. Regional Shellfish Seed Biosecurity Program. 2021. Regional Shellfish Seed Biosecurity-Program Hatchery Compliance. Rutgers University, New Brunswick, NJ, USA. Available: rssbp.org/wp-content/uploads/2022/01/RSSBP-Program-Guide_Dec-14-2021.pdf (December 2021).
7. Pörtner, H., D. C. Roberts, H. Adams, C. Adler, P. Aldunce, E. Ali, R. A. Begum, R. Betts, R. B. Kerr, and R. Biesbroek. 2022. Climate change 2022: Impacts, adaptation and vulnerability. IPCC Sixth Assessment Report. Cambridge University Press, Cambridge, U.K.
8. Rhodes, L. D., K. L. Parrish, and M. L. Willis. 2023. Review of Best Practices for Biosecurity and Disease Management for Marine Aquaculture in U.S. Waters. U.S. Department of Commerce, NOAA Technical Memorandum NMFS-NWFSC-180. DOI: 10.25923/b4qp-9e65
9. NMFS (National Marine Fisheries Service). 2022. Fisheries of the United States, 2020. NOAA Current Fishery Statistics No. 2020. National Marine Fisheries Service, Silver Spring, Maryland. Available: media.fisheries.noaa.gov/2022-05/Fisheries-of-the-United-States-2020-Report-FINAL.pdf (July 2023).
10. Subasinghe, R., V. Alday-Sanz, M. G. Bondad-Reantaso, H. Jie, A. P. Shinn, and P. Sorgeloos. 2023. Biosecurity: Reducing the burden of disease. Journal of the World Aquaculture Society 54(2):397-426. DOI: 10.1111/jwas.12966
11. FAO (Food and Agriculture Organization of the United Nations). 2020. Report of the Progressive Mangement Pathway for Improving Aquaculture Biosecurity (PMP/AB): First Technical Working Group Meeting: Rome, 20–22 March 2019. Food and Agriculture Organization of the United Nations, Rome, Italy. DOI: 10.4060/cb0582en
12. Snieszko, S. F. 1974. The effects of environmental stress on outbreaks of infectious diseases of fishes. Journal of Fish Biology 6(2):197–208.
13. Chaix, G., F. Roger, T. Berthe, B. Lamy, E. Jumas-Bilak, R. Lafite, J. Forget-Leray, and F. Petit. 2017. Distinct *Aeromonas* Populations in Water Column and Associated with Copepods from Estuarine Environment (Seine, France). Frontiers in Microbiology 8. DOI: 10.3389/fmicb.2017.01259

14. Vasquez, I., A. Hossain, H. Gnanagobal, K. Valderrama, B. Campbell, M. Ness, S. J. Charette, A. K. Gamperl, R. Cipriano, C. Segovia, and J. Santander. 2022. Comparative Genomics of Typical and Atypical *Aeromonas salmonicida* Complete Genomes Revealed New Insights into Pathogenesis Evolution. Microorganisms 10(1):189. DOI: 10.3390/microorganisms10010189
15. Oidtmann, B., Dixon, P., Way, K., Joiner, C., Bayley, A. 2018. Risk of waterborne virus spread—review of survival of relevant fish crustacean viruses in the aquatic environment and implications for control measures. Reviews in Aquaculture 10:641–669. DOI: 10.1111/raq.12192
16. Salama, N. K. G., and B. Rabe. 2013. Developing models for investigating the environmental transmission of disease-causing agents within open-cage salmon aquaculture. Aquaculture Environment Interactions 4:91–115. DOI: 10.3354/aei00077
17. Salama, N. K. G., and A. G. Murray. 2011. Farm size as a factor in hydrodynamic transmission of pathogens in aquaculture fish production. Aquaculture Environment Interactions 2:61–74. DOI: 10.3354/aei00030
18. Benetti, D. D., D. I. Benetti, J. A. Rivera, B. Sardenberg, and B. O'Hanlon. 2010. Site Selection Criteria for Open Ocean Aquaculture. Marine Technology Society Journal 44(3):22–35. DOI: 10.4031/MTSJ.44.3.11
19. Pagenkopp Lohan, K. M., J. A. Darling, and G. M. Ruiz. 2022. International shipping as a potent vector for spreading marine parasites. Diversity and Distributions 28(9):1922–1933. DOI: 10.1111/ddi.13592
20. Murray, A. G., R. J. Smith, and R. M. Stagg. 2002. Shipping and the spread of infectious salmon anemia in Scottish aquaculture. Emerging Infectious Diseases 8(1):1–5.
21. Kocan, R. M. 2019. Transmission models for the fish pathogen *Ichthyophonus*: synthesis of field observations and empirical studies. Canadian Journal of Fisheries and Aquatic Sciences 76(4):636–642. DOI: 10.1139/cjfas-2018-0166
22. Mazzillo, F. F., K. Shapiro, and M. W. Silver. 2013. A New Pathogen Transmission Mechanism in the Ocean: The Case of Sea Otter Exposure to the Land-Parasite *Toxoplasma gondii*. PLOS ONE 8(12):e82477. DOI: 10.1371/journal.pone.0082477
23. Balfry, S. K., L. J. Albright, and T. P. T. Evelyn. 1996. Horizontal transfer of *Renibacterium salmoninarum* among farmed salmonids via the fecal–oral route. Diseases of Aquatic Organisms 25(1–2):63–69.
24. Sveen, L., C. Karlsen, and E. Ytteborg. 2020. Mechanical induced wounds in fish—a review on models and healing mechanisms. Reviews in Aquaculture 12(4):2446–2465. DOI: 10.1111/raq.12443
25. Rakers, S., L. Niklasson, D. Steinhagen, C. Kruse, J. Schauber, K. Sundell, and R. Paus. 2013. Antimicrobial Peptides (AMPs) from Fish Epidermis: Perspectives for Investigative Dermatology. Journal of Investigative Dermatology 133(5):1140–1149. DOI: 10.1038/jid.2012.503
26. Taylor, P. W. 1992. Fish-eating birds as potential vectors of *Edwardsiella ictaluri*. Journal of Aquatic Animal Health 4(4):240–243.
27. Vanpatten, K. A., L. M. Nunan, and D. V. Lightner. 2004. Seabirds as potential vectors of penaeid shrimp viruses and the development of a surrogate laboratory model utilizing domestic chickens. Aquaculture 241(1):31–46. DOI: 10.1016/j.aquaculture.2004.08.012
28. de Carvalho, C. C. C. R. 2018. Marine Biofilms: A Successful Microbial Strategy With Economic Implications. Frontiers in Marine Science 5. DOI: 10.3389/fmars.2018.00126

29. Fitridge, I., T. Dempster, J. Guenther, and R. de Nys. 2012. The impact and control of biofouling in marine aquaculture: a review. Biofouling 28(7):649–669. DOI: 10.1080/08927014.2012.700478

30. Liu, Y., G. H. Wikfors, P. Clark, S. Pitchford, M. Krisak, M. S. Dixon, and Y. Li. 2022. A deep dive into the epibiotic communities on aquacultured sugar kelp *Saccharina latissima* in Southern New England. Algal Research 63:102654. DOI: 10.1016/j.algal.2022.102654

31. Grano-Maldonado, M. I., J. Moreno-Navas, and M. A. Rodriguez-Santiago. 2018. Transmission Strategies Used by *Gyrodactylus gasterostei* (Monogenea) on Its Host, the Three-Spined Stickleback *Gasterosteus aculeatus*. Fishes 3(2):20.

32. Connors, B. M., M. Krkošek, and L. M. Dill. 2008. Sea lice escape predation on their host. Biology letters 4(5):455–457. DOI: 10.1098/rsbl.2008.0276

33. Fields, D. M., A. B. Skiftesvik, and H. I. Browman. 2018. Behavioural responses of infective-stage copepodids of the salmon louse (*Lepeophtheirus salmonis*, Copepoda:Caligidae) to host-related sensory cues. Journal of Fish Diseases 41(6):875–884. DOI: 10.1111/jfd.12690

34. Mordue, A. J., and M. A. Birkett. 2009. A review of host finding behaviour in the parasitic sea louse, *Lepeophtheirus salmonis* (Caligidae: Copepoda). Journal of Fish Diseases 32(1):3–13. DOI: 10.1111/j.1365-2761.2008.01004.x

35. Cusack, R., and D. K. Cone. 1986. A review of parasites as vectors of viral and bacterial diseases of fish. Journal of Fish Diseases 9(2):169–171. DOI: 10.1111/j.1365-2761.1986.tb01000.x

36. Lafferty, K. D., C. D. Harvell, J. M. Conrad, C. S. Friedman, M. L. Kent, A. M. Kuris, E. N. Powell, D. Rondeau, and S. M. Saksida. 2015. Infectious Diseases Affect Marine Fisheries and Aquaculture Economics. Annual Review of Marine Science 7(1):471–496. DOI: 10.1146/annurev-marine-010814-015646

37. Taranger, G. L., Ø. Karlsen, R. J. Bannister, K. A. Glover, V. Husa, E. Karlsbakk, B. O. Kvamme, K. K. Boxaspen, P. A. Bjørn, B. Finstad, A. S. Madhun, H. C. Morton, and T. Svåsand. 2014. Risk assessment of the environmental impact of Norwegian Atlantic salmon farming. ICES Journal of Marine Science 72(3):997–1021. DOI: 10.1093/icesjms/fsu132

38. Kurath, G., and J. Winton. 2011. Complex dynamics at the interface between wild and domestic viruses of finfish. Current Opinion in Virology 1(1):73–80. DOI: 10.1016/j.coviro.2011.05.010

39. Díaz López, B. 2017. Temporal variability in predator presence around a fin fish farm in the Northwestern Mediterranean Sea. Marine Ecology 38(1):e12378. DOI: 10.1111/maec.12378

40. Holmer, M. 2010. Enviromental issues of fish farming in offshore waters: perspectives, concerns and research needs. Aquaculture Enviroment Interactions 1:57–70. DOI: 10.3354/aei00007

41. Bonizzoni, S., N. B. Furey, E. Pirotta, V. D. Valavanis, B. Würsig, and G. Bearzi. 2014. Fish farming and its appeal to common bottlenose dolphins: modelling habitat use in a Mediterranean embayment. Aquatic Conservation: Marine and Freshwater Ecosystems 24(5):696–711. DOI: 10.1002/aqc.2401

42. Bouwmeester, M. M., M. A. Goedknegt, R. Poulin, D. W. Thieltges, and N. Pettorelli. 2020. Collateral diseases: Aquaculture impacts on wildlife infections. Journal of Applied Ecology 58(3):453–464. DOI: 10.1111/1365-2664.13775

43. Johnsen, B. O., and A. J. Jenser. 1991. The *Gyrodactylus* story in Norway. Aquaculture 98(1):289–302. DOI: 10.1016/0044-8486(91)90393-L

44. Johansen, L. H., I. Jensen, H. Mikkelsen, P. A. Bjørn, P. A. Jansen, and Ø. Bergh. 2011. Disease interaction and pathogens exchange between wild and farmed fish populations with special reference to Norway. Aquaculture 315(3):167–186. DOI: 10.1016/j.aquaculture.2011.02.014

45. Olstad, K. 2013. NOBANIS—Invasive Alien Species Fact Sheet: *Gyrodactylus salaris*. European Network on Invasive Alien Species (NOBANIS), Iceland. Available: www.nobanis.org/globalassets/speciesinfo/g/gyrodactylus-salaris/gyrodactylus_salaris.pdf (May 2023).

46. Goedknegt, M. A., A.-K. Schuster, C. Buschbaum, R. Gergs, A. S. Jung, P. C. Luttikhuizen, J. van der Meer, K. Troost, K. M. Wegner, and D. W. Thieltges. 2017. Spillover but no spillback of two invasive parasitic copepods from invasive Pacific oysters (*Crassostrea gigas*) to native bivalve hosts. Biological Invasions 19(1):365–379. DOI: 10.1007/s10530-016-1285-0

47. Sepúlveda, F., S. L. Marín, and J. Carvajal. 2004. Metazoan parasites in wild fish and farmed salmon from aquaculture sites in southern Chile. Aquaculture 235(1):89–100. DOI: 10.1016/j.aquaculture.2003.09.015

48. Burge, C. A., C. J. Closek, C. S. Friedman, M. L. Groner, C. M. Jenkins, A. Shore-Maggio, and J. E. Welsh. 2016. The Use of Filter-feeders to Manage Disease in a Changing World. Integrative and Comparative Biology 56(4):573–587. DOI: 10.1093/icb/icw048

49. Paterson, R. A., C. R. Townsend, R. Poulin, and D. M. Tompkins. 2011. Introduced brown trout alter native acanthocephalan infections in native fish. Journal of Animal Ecology 80(5):990–998. DOI: 10.1111/j.1365-2656.2011.01834.x

50. NMFS (National Marine Fisheries Service). 2022. DisMAP data records. National Marine Fisheries Service, Silver Spring, Maryland. Available: apps-st.fisheries.noaa.gov/dismap/ (September 2022).

51. Gulf of Mexico Fishery Management Council and National Marine Fisheries Service. 2016. Five-year review of essential fish habitat requirements. Gulf of Mexico Fishery Management Council, Tampa, Florida.

52. PFMC (Pacific Fishery Management Council). 2022. Pacific Coast Groundfish Fishery Management Plan. Available: www.pcouncil.org/groundfish-fishery-management-plan-and-amendments/ (July 2023).

53. PFMC (Pacific Fishery Management Council). 2016. Fishery Management Plan for U.S. West Coast Fisheries for Highly Migratory Species. Pacific Fishery Management Council, Portland, Oregon.

54. Kikuchi, W. K. 1976. Prehistoric Hawaiian fishponds: Indigenous aquaculture influenced the development of social stratification in Hawaii. Science 193(4250):295–299. DOI: 10.1126/science.193.4250.295

55. Costa-Pierce, B. A. 1987. Aquaculture in Ancient Hawaii. BioScience 37:320–331.

56. Akiona, A. K., B. N. Popp, R. J. Toonen, M. C. Siple, K. Kotubetey, H. Kawelo, and E. C. Franklin. 2022. Predatory fish diets shift towards an invasive mullet in a traditional Hawaiian aquaculture system. Aquaculture, Fish and Fisheries 2(5):307–320. DOI: 10.1002/aff2.68

57. Goetz, F. W., B. F. Anulacion, M. R. Arkhoosh, M. A. Cook, W. W. Dickhoff, J. P. Dietrich, W. T. Fairgrieve, E. S. Hayman, M. B. R. Hicks, C. Jensen, R. B. Johnson, J. S. F. Lee, J. A. Luckenbach, K. C. Massee, and T. H. Wade. 2020. Status of Sablefish, *Anoplopoma fimbria*, aquaculture. World Aquaculture Society 52:607–646. DOI: 10.1111/jwas.12769

58. Saillant, E., N. Adams, J. T. Lemus, J. S. Franks, Yonathan Zohar, J. Stubblefield, and C. Manley. 2021. First data on aquaculture of the Tripletail, *Lobotes surinamensis*, a promising candidate species for U.S. marine aquaculture. World Aquaculture Society 52(3):582–594. DOI: 10.1111/jwas.12807

59. Drawbridge, M., M. Shane, and C. Silbernagel. 2021. The status of white seabass, *Atractoscion nobilis* as a commercially ready species for marine US aquaculture. World Aquaculture Society 52(3):647–661. DOI: 10.1111/jwas.12772

60. Weirich, C. R., K. L. Riley, M. Riche, K. L. Main, P. S. Wills, G. Illan, D. S. Cerino, and T. J. Pfeiffer. 2021. The status of Florida pompano, *Trachinotus carolinus*, as a commercially ready species for U.S. marine aquaculture. Journal of the World Aquaculture Society 52(3):731–763. DOI: 10.1111/jwas.12809

61. Stuart, K., C. Silbernagel, and M. Drawbridge. 2021. The status of California halibut, *Paralichthys californicus*, as a technologically feasible species for marine U.S. aquaculture. World Aquaculture Society 52:662–671. DOI: 10.1111/jwas.12768

62. Andersen, L. K., J. Abernathy, D. L. Berlinsky, G. Bolton, M. M. Booker, R. J. Borski, T. Brown, D. Cerino, M. Ciaramella, R. W. Clark, M. O. Frinsko, S. A. Fuller, S. Gabel, B. W. Green, E. Herbst, R. G. Hodson, M. Hopper, L. W. Kenter, F. Lopez, A. S. McGinty, B. Nash, M. Parker, S. Pigg, S. Rawles, K. Riley, M. J. Turano, C. D. Webster, C. R. Weirich, E. Won, L. C. Woods, and B. J. Reading. 2021. The status of striped bass, *Morone saxatilis*, as a commercially ready species for US marine aquaculture. Journal of the World Aquaculture Society 52(3):710–730.

63. Blaylock, R., E. Saillant, A. Apeitos, D. Abrego, P. Cason, and R. Vega. 2021. The status of spotted seatrout (*Cynoscion nebulosus*) as a technologically feasible species for US marine aquaculture. Journal of the World Aquaculture Society 52(3):526–540.

64. Murray, K. N., T. S. Clark, M. J. Kebus, and M. L. Kent. 2021. Specific Pathogen Free—A review of strategies in agriculture, aquaculture, and laboratory mammals and how they inform new recommendations for laboratory zebrafish. Research in Veterinary Science 142:78–93. DOI: 10.1016/j.rvsc.2021.11.005

65. Kent, M. L., C. Buchner, V. G. Watral, J. L. Sanders, J. Ladu, T. S. Peterson, and R. L. Tanguay. 2011. Development and maintenance of a specific pathogen-free (SPF) zebrafish research facility for *Pseudoloma neurophilia*. Diseases of Aquatic Organisms 95(1):73–79. DOI: 10.3354/dao02333

66. Fisheries Co-Manangers of Washington State. 2006. The Salmonid Disease Control Policy of the Fisheries Co-Managers of Washington State. Available: files.nwifc.org/fish-health/FinalDiseasePolicy-July2006_Ver3.pdf (March 2022).

67. APHIS (Animal and Plant Health Inspection Service). 2021. National Aquaculture Health Plan & Standards, 2021–2023. U.S. Department of Agriculture, Riverdale, Maryland. Available: www.aphis.usda.gov/animal_health/animal_dis_spec/aquaculture/downloads/national-aquacult-health-plan-standards-2021-2023.pdf (December 2021).

68. USFWS (U.S. Fish and Wildlife Service). 2022. Handbook of Aquatic Animal Health Procedures and Protocols. U.S. Fish and Wildlife Service, Department of the Interior, Washington, D.C. Available: www.fws.gov/sites/default/files/policy/files/AquaticAnimalHealthProceduresandProtocols.pdf (May 2023).

69. Bregnballe, J. 2015. A guide to recirculation aquaculture. FAO (Food & Agriculture Organization of the United Nations) & EUROFISH International Organisation, Copenhagen, Denmark. Available: www.fao.org/3/i4626e/i4626e.pdf (July 2023).

70. Mugimba, K. K., D. K. Byarugaba, S. Mutoloki, Ø. Evensen, and H. M. Munang'andu. 2021. Challenges and Solutions to Viral Diseases of Finfish in Marine Aquaculture. Pathogens 10(6):673. DOI: 10.3390/pathogens10060673

71. Lillehaug, A., N. Santi, and A. Østvik. 2015. Practical Biosecurity in Atlantic Salmon Production. Journal of Applied Aquaculture 27(3):249–262. DOI: 10.1080/10454438.2015.1066174

72. USDA-APHIS Veterinary Services. 2017. Maine Infectious Salmon Anemia Virus Control Program Standards. U.S. Department of Agriculture, Washington, D.C. Available: www.aphis.usda.gov/animal_health/animal_dis_spec/aquaculture/downloads/isa_standards.pdf (July 2023).

73. Streicher, M. D., H. Reiss, and K. Reiss. 2021. Impact of aquaculture and agriculture nutrient sources on macroalgae in a bioassay study. Marine Pollution Bulletin 173:113025. DOI: 10.1016/j.marpolbul.2021.113025

74. Price, C., K. D. Black, B. T. Hargrave, and J. A. Morris, Jr. 2015. Marine cage culture and the environment: Effects on water quality and primary production. Aquaculture Environment Interactions 6(2):151-174.

75. Barrett, L. T., S. E. Swearer, and T. Dempster. 2019. Impacts of marine and freshwater aquaculture on wildlife: A global meta-analysis. Reviews in Aquaculture 11(4):1022–1044.

76. OIE (Office International des Epizooties). 2021. Aquatic Animal Health Code, Chapter 1.4: Aquatic Animal Disease Surveillance. Office International des Epizooties, Paris. Available: www.woah.org/fileadmin/Home/eng/Health_standards/aahc/current/chapitre_aqua_ani_surveillance.pdf (June 2022).

77. OIE (Office International des Epizooties). 2022. Aquatic Animal Health Code, Chapter 4.8: Handling, Disposal, and Treatment of Aquatic Animal Waste. Office International des Epizooties, Paris. Available: www.woah.org/fileadmin/Home/eng/Health_standards/aahc/current/chapitre_aquatic_animal_waste.pdf (June 2022).

78. Maine, E. 2002. Authorization to Discharge under the National Pollutant Discharge Elimination System. Acadia Aquaculture Inc., Boston, Massachusetts.

79. EPA (U.S. Environmental Protection Agency). 2020. Authorization to Discharge Under the National Pollutant Discharge Elimination System: Permit Number FL0A00001. U.S. Environmental Protection Agency. Available: www.epa.gov/sites/default/files/2020-10/documents/npdes_permit_for_ocean_era_inc._-_velella_epsilon_fl0a00001.pdf (April 2022).

80. WADOE (Washington Department of Ecology). 2021. National Pollutant Discharge Elimination System Waste Discharge Permit No. WA0031542. Available: apps.ecology.wa.gov/paris/DownloadDocument.aspx?Id=349570 (August 2022).

81. Callier, M. D., C. J. Byron, D. A. Bengtson, P. J. Cranford, S. F. Cross, U. Focken, H. M. Jansen, P. Kamermans, A. Kiessling, and T. Landry. 2018. Attraction and repulsion of mobile wild organisms to finfish and shellfish aquaculture: A review. Reviews in Aquaculture 10(4):924–949.

82. Pozio, E. 2003. Foodborne and waterborne parasites. Acta Microbiologica Polonica 52(SUPP):83–96.

83. Alcaide, E., E. Sanjuan, F. de la Gandara, and A. Garcia-Gomez. 2000. Susceptibility of amberjack (*Seriola dumerili*) to bacterial fish pathogens. Bulletin of the European Association of Fish Pathologists 20(4):153–156.

84. Price, I. M., and J. G. Nickum. 1995. Aquaculture and birds: The context for controversy. Colonial Waterbirds 18:33–45.

85. Sanchez-Jerez, P., D. Fernandez-Jover, I. Uglem, P. Arechavala-Lopez, T. Dempster, J. T. Bayle-Sempere, C. Valle Pérez, D. Izquierdo, P.-A. Bjørn, and R. Nilsen. 2011. Coastal Fish Farms as Fish Aggregation Devices (FADs). Pages 187–208 *in* S. A. Bortone, F. P. Brandini, G. Fabi, and S. Otake, editors. Artificial Reefs in Fishery Management. CRC Press, Boca Raton, Florida.

86. Carballeira Braña, C. B., K. Cerbule, P. Senff, and I. K. Stolz. 2021. Towards Environmental Sustainability in Marine Finfish Aquaculture. Frontiers in Marine Science 8:e666662. DOI: 10.3389/fmars.2021.666662

87. Teffer, A. K., J. Carr, A. Tabata, A. Schulze, I. Bradbury, D. Deschamps, C.-A. Gillis, E. B. Brunsdon, G. Mordecai, K. M. Miller, and V. L. Trudeau. 2020. A molecular assessment of infectious agents carried by Atlantic salmon at sea and in three eastern Canadian rivers, including aquaculture escapees and North American and European origin wild stocks. FACETS 5(1):234–263. DOI: 10.1139/facets-2019-0048

88. Anderson, E. D., H. M. Engelking, E. J. Emmenegger, and G. Kurath. 2000. Molecular Epidemiology Reveals Emergence of a Virulent Infectious Hematopoietic Necrosis (IHN) Virus Strain in Wild Salmon and Its Transmission to Hatchery Fish. Journal of Aquatic Animal Health 12(2):85–99. Available: www.tandfonline.com/doi/abs/10.1577/1548-8667%28200006%29012%3C0085%3AMEREOA%3E2.0.CO%3B2 (July 2023).

89. Costello, M. J. 2009. How sea lice from salmon farms may cause wild salmonid declines in Europe and North America and be a threat to fishes elsewhere. Proceeding of the Royal Society B Biological Sciences 276(1672):3385–3394. DOI: 10.1098/rspb.2009.0771

90. Thorstad, E. B., C. D. Todd, I. Uglem, P. A. Bjørn, P. G. Gargan, K. W. Vollset, E. Halttunen, S. Kålås, M. Berg, and B. Finstad. 2015. Effects of salmon lice *Lepeophtheirus salmonis* on wild sea trout *Salmo trutta*—a literature review. Aquaculture Environment Interactions 7(2):91–113. DOI: 10.3354/aei00142

91. Rees, E. E., S. St-Hilaire, S. R. M. Jones, M. Krkošek, S. DeDominicis, M. G. G. Foreman, T. Patanasatienkul, and C. W. Revie. 2015. Spatial patterns of sea lice infection among wild and captive salmon in western Canada. Landscape Ecology 30(6):989–1004. DOI: 10.1007/s10980-015-0188-2

92. Nekouei, O., R. Vanderstichel, K. Thakur, G. Arriagada, T. Patanasatienkul, P. Whittaker, B. Milligan, L. Stewardson, and C. W. Revie. 2018. Association between sea lice (*Lepeophtheirus salmonis*) infestation on Atlantic salmon farms and wild Pacific salmon in Muchalat Inlet, Canada. Scientific Reports 8(1):4023. DOI: 10.1038/s41598-018-22458-8

93. Vollset, K. W., L. Qviller, B. Skar, B. T. Barlaup, and I. Dohoo. 2018. Parasitic sea louse infestations on wild sea trout: separating the roles of fish farms and temperature. Parasites & Vectors 11(1):609. DOI: 10.1186/s13071-018-3189-6

94. DFO (Fisheries and Oceans Canada). 2023. Association Between Sea Lice from Atlantic Salmon Farms and Sea Lice Infestations on Wild Juvenile Pacific Salmon in British Columbia. Fisheries and Oceans Canada, Ottowa, Ontario, Canada. Available: www.dfo-mpo.gc.ca/csas-sccs/Publications/ScR-RS/2022/2022_045-eng.pdf (February 2023).

95. Guragain, P., M. Tkachov, A. S. Båtnes, Y. Olsen, P. Winge, and A. M. Bones. 2021. Principles and Methods of Counteracting Harmful Salmon–Arthropod Interactions in Salmon Farming: Addressing Possibilities, Limitations, and Future Options. Frontiers in Marine Science 8:701793. DOI: 10.3389/fmars.2021.701793

96. Ashley, P. J. 2007. Fish welfare: Current issues in aquaculture. Applied Animal Behaviour Science 104(3–4):199–235.

97. Bevan, D. J., K. P. Chandroo, and R. D. Moccia. 2002. Predator Control in Commercial Aquaculture in Canada. University of Guelph, Guelph, Ontario, Canada. Available: animalbiosciences.uoguelph.ca/aquacentre/files/misc-factsheets/Predator%20Control%20in%20Commercial%20Aquaculture%20in%20Canada.pdf (February 2023).

98. Føre, M., K. Frank, T. Norton, E. Svendsen, J. A. Alfredsen, T. Dempster, H. Eguiraun, W. Watson, A. Stahl, L. M. Sunde, C. Schellewald, K. R. Skøien, M. O. Alver, and D. Berckmans. 2018. Precision fish farming: A new framework to improve production in aquaculture. Biosystems Engineering 173:176–193. DOI: 10.1016/j.biosystemseng.2017.10.014

99. Moe Føre, H., T. Thorvaldsen, T. C. Osmundsen, F. Asche, R. Tveterås, J. T. Fagertun, and H. V. Bjelland. 2022. Technological innovations promoting sustainable salmon (*Salmo salar*) aquaculture in Norway. Aquaculture Reports 24:101115. DOI: 10.1016/j.aqrep.2022.101115

100. Robinson, E. H., S. K. Johnson, and J. T. Davis. 1985. Guidelines for Selecting Feeds and Techniques of Feeding Fish. Texas Agricultural Extension Service, College Station, Texas.

101. Upton, H. F. 2019. U.S. Offshore Aquaculture Regulation and Development. Congressional Research Service, Washington, D.C. Available: www.everycrsreport.com/files/20191010_R45952_5fad4bb891b10e324bea0334d38554371860d6b6.pdf (July 2023).

102. Copeland, C. 2005. Cruise Ship Pollution: Background, Laws and Regulations, and Key Issues. Congressional Research Service, Washington, D.C. Available: www.everycrsreport.com/reports/RL32450.html (July 2023)

103. USEPA (U.S. Environmental Protection Agency). 2013. National Pollutant Discharge Elimination System (NPDES) Vessel General Permit (VGP) for Discharges Incidental to the Normal Operation of Vessels: Fact Sheet. U.S. Environmental Protection Agency, Washington, D.C. Available: www3.epa.gov/npdes/pubs/vgp_fact_sheet2013.pdf (July 2023).

104. Kennicutt, M. C. 2017. Water Quality of the Gulf of Mexico. Pages 55–164 *in* C. H. Ward, editor. Habitats and Biota of the Gulf of Mexico: Before the Deepwater Horizon Oil Spill: Volume 1: Water Quality, Sediments, Sediment Contaminants, Oil and Gas Seeps, Coastal Habitats, Offshore Plankton and Benthos, and Shellfish. Springer, New York.

105. National Research Council (U.S.) Committee on Oil in the Sea: Inputs Fates and Effects. 2003. C. Natural Seepage of Crude Oil into the Marine Environment. National Academy Press, Washington, D.C. Available: www.ncbi.nlm.nih.gov/books/NBK220695/. (August 2022).

106. Hickey, B. M. 1992. Circulation over the Santa Monica–San Pedro Basin and Shelf. Progress in Oceanography 30(1):37–115. DOI: 10.1016/0079-6611(92)90009-O

107. McClatchie, S. 2014. Oceanography of the Southern California Current System Relevant to Fisheries. Pages 13–60 *in* Regional Fisheries Oceanography of the California Current System: The CalCOFI program. Springer Netherlands, Dordrecht.

108. Cao, Y., M. R. Raith, P. D. Smith, J. F. Griffith, S. B. Weisberg, A. Schriewer, A. Sheldon, C. Crompton, G. G. Amenu, J. Gregory, J. Guzman, K. D. Goodwin, L. Othman, M. Manasjan, S. Choi, S. Rapoport, S. Steele, T. Nguyen, and X. Yu. 2017. Regional Assessment of Human Fecal Contamination in Southern California Coastal Drainages. International Journal of Environmental Research and Public Health 14(8):874.

109. Schiff, K., D. Greenstein, N. Dodder, and D. J. Gillett. 2016. Southern California Bight regional monitoring. Regional Studies in Marine Science 4:34–46. DOI: 10.1016/j.rsma.2015.09.003

110. LACSD (Los Angeles County Sanitation Districts). 2022. Joint Water Pollution Control Plant: Biennial Receiving Water Monitoring Report, 2020–2021. Los Angeles County Sanitation Districts, Whittier, California USA. Available: www.lacsd.org/home/showpublisheddocument/9782/638096215929100000 (July 2023).

111. DiGiacomo, P. M., and B. Holt. 2001. Satellite observations of small coastal ocean eddies in the Southern California Bight. Journal of Geophysical Research: Oceans 106(C10):22521–22543. DOI: 10.1029/2000JC000728

112. Dong, C., E. Y. Idica, and J. C. McWilliams. 2009. Circulation and multiple-scale variability in the Southern California Bight. Progress in Oceanography 82(3):168–190. DOI: 10.1016/j.pocean.2009.07.005

113. Kinlan, B. P., and S. D. Gaines. 2003. Propagule Dispersal in Marine and Terrestrial Environments: A Community Perspective. Ecology 84(8):2007–2020. DOI: 10.1890/01-0622

114. McCallum, H., D. Harvell, and A. Dobson. 2003. Rates of spread of marine pathogens. Ecology Letters 6(12):1062–1067. DOI: 10.1046/j.1461-0248.2003.00545.x

115. Pitta, P., M. Tsapakis, E. T. Apostolaki, T. Tsagaraki, M. Holmer, and I. Karakassis. 2009. 'Ghost nutrients' from fish farms are transferred up the food web by phytoplankton grazers. Marine Ecology Progress Series 374:1–6.

116. Carlucci, A. F., and D. Pramer. 1959. Factors Affecting the Survival of Bacteria in Sea Water. Applied Microbiology 7(6):388–392. DOI: 10.1128/am.7.6.388-392.1959

117. Murphy, H. 2017. Persistence of Pathogens in Sewage and Other Water Types. Pages 1–27 *in* J. B. Rose and B. Jiménez-Cisneros, editors. Water and Sanitation for the 21st Century: Health and Microbiolgocial Aspects of Excreta and Wastewater Management (Global Water Pathogen Project). Michigan State University, East Lansing, Michigan. DOI: 10.14321/waterpathogens.51

118. Gerba, C. P., and G. E. Schaiberger. 1975. Effect of particulates on virus survival in seawater. Journal of the Water Pollution Control Federation 47(1):93–103.

119. Kapuscinski, R. B., and R. Mitchell. 1980. Processes controlling virus inactivation in coastal waters. Water Research 14(4):363–371. DOI: 10.1016/0043-1354(80)90084-6

120. Keeley, N., T. Valdemarsen, S. Woodcock, M. Holmer, V. Husa, and R. Bannister. 2019. Resilience of dynamic coastal benthic ecosystems in response to large-scale finfish farming. Aquaculture Environment Interactions 11:161–179.

121. Strain, P. M., and B. T. Hargrave. 2005. Salmon Aquaculture, Nutrient Fluxes and Ecosystem Processes in SouthwesternNew Brunswick. Pages 29–57 *in* B. T. Hargrave, editor. Environmental Effects of Marine Finfish Aquaculture. Springer Berlin Heidelberg, Berlin, Germany.

122. NMFS (National Marine Fisheries Service). 2022. Reinitiation of Endangered Species Act Section 7(a)(2) Biological Opinion, and Magnuson-Stevens Fishery Conservation and Management Act Essential Fish Habitat Response for the Environmental Protection Agency's Approval of Washington State Department of Ecology's Sediment Management Standards (WAC 173-204-412) Regarding Marine Finfish Rearing Facilities. National Marine Fisheries Service, Silver Spring, Maryland. DOI: 10.25923/6p01-b072

123. Ebeling, J. M., and M. B. Timmons. 2012. Recirculating aquaculture. Pages 245–277 *in* J. H. Tidwell, editor. Aquaculture Production Systems. Cayuga Aqua Ventures, Freeville, New York.

124. Pereira, P. M. F., K. D. Black, D. S. McLusky, and T. D. Nickell. 2004. Recovery of sediments after cessation of marine fish farm production. Aquaculture 235(1):315–330. DOI: 10.1016/j.aquaculture.2003.12.023

125. Lepofsky, D., and M. Caldwell. 2013. Indigenous marine resource management on the Northwest Coast of North America. Ecological Processes 2(1):12. DOI: 10.1186/2192-1709-2-12

126. Cotter, E., S. K. Malham, S. O'Keeffe, S. A. Lynch, J. W. Latchford, J. W. King, A. R. Beaumont, and S. C. Culloty. 2010. Summer mortality of the Pacific oyster, *Crassostrea gigas*, in the Irish Sea: The influence of growth, biochemistry and gametogenesis. Aquaculture 303(1–4):8–21.

127. Carrasco, N., I. Gairin, J. Pérez, K. B. Andree, A. Roque, M. Fernández-Tejedor, C. J. Rodgers, C. Aguilera, and M. D. Furones. 2017. A Production Calendar Based on Water Temperature, Spat Size, and Husbandry Practices Reduce OsHV-1 μvar Impact on Cultured Pacific Oyster *Crassostrea gigas* in the Ebro Delta (Catalonia), Mediterranean Coast of Spain. Frontiers in Physiology 8:125. DOI: 10.3389/fphys.2017.00125

128. Lightner, D. V. 2005. Biosecurity in Shrimp Farming: Pathogen Exclusion through Use of SPF Stock and Routine Surveillance. Journal of the World Aquaculture Society 36(3):229–248. DOI: 10.1111/j.1749-7345.2005.tb00328.x

129. Zannella, C., F. Mosca, F. Mariani, G. Franci, V. Folliero, M. Galdiero, P. G. Tiscar, and M. Galdiero. 2017. Microbial Diseases of Bivalve Mollusks: Infections, Immunology and Antimicrobial Defense. Marine Drugs 15(6). DOI: 10.3390/md15060182

130. Rodgers, C., I. Arzul, N. Carrasco, and D. Furones Nozal. 2019. A literature review as an aid to identify strategies for mitigating ostreid herpesvirus 1 in *Crassostrea gigas* hatchery and nursery systems. Reviews in Aquaculture 11(3):565–585. DOI: 10.1111/raq.12246

131. Whittington, R. J., N. K. Dhand, O. Evans, and I. Paul-Pont. 2015. Further observations on the influence of husbandry practices on OsHV-1 μVar mortality in Pacific oysters *Crassostrea gigas*: Age, cultivation structures and growing height. Aquaculture 438:82–97. DOI: 10.1016/j.aquaculture.2014.12.040

132. Summerfelt, S. T. 2003. Ozonation and UV irradiation—An introduction and examples of current applications. Aquacultural Engineering 28(1–2):21–36.

133. Paul-Pont, I., N. K. Dhand, and R. J. Whittington. 2013. Influence of husbandry practices on OsHV-1 associated mortality of Pacific oysters *Crassostrea gigas*. Aquaculture 412–413:202–214. DOI: 10.1016/j.aquaculture.2013.07.038

134. Pernet, F., C. Lupo, C. Bacher, and R. J. Whittington. 2016. Infectious diseases in oyster aquaculture require a new integrated approach. Philosophical Transactions of the Royal Society B: Biological Sciences 371(1689):20150213. DOI: 10.1098/rstb.2015.0213

135. Baker, S., D. Petty, R. Francis-Floyd, R. Yanong, and L. Sturmer. 2021. Introduction to Infectious Diseases in Hard Clams. University of Florida Extension, Gainesville, Florida.

136. Ben-Horin, T., K. D. Lafferty, G. Bidegain, and H. S. Lenihan. 2016. Fishing diseased abalone to promote yield and conservation. Philosophical Transactions of the Royal Society B: Biological Sciences 371(1689):20150211. DOI: 10.1098/rstb.2015.0211

137. Kristmundsson, Á., and M. A. Freeman. 2018. Harmless sea snail parasite causes mass mortalities in numerous commercial scallop populations in the northern hemisphere. Scientific Reports 8(1):1–12.

138. Adams, C. M., S. E. Shumway, R. B. Whitlatch, and T. Getchis. 2011. Biofouling in marine molluscan shellfish aquaculture: A survey assessing the business and economic implications of mitigation. Journal of the World Aquaculture Society 42(2):242–252.

139. Bannister, J., M. Sievers, F. Bush, and N. Bloecher. 2019. Biofouling in marine aquaculture: a review of recent research and developments. Biofouling 35(6):631–648. DOI: 10.1080/08927014.2019.1640214

140. Burreson, E. M., and L. M. R. Calvo. 1996. Epizootiology of *Perkinsus marinus* disease of oysters in Chesapeake Bay, with emphasis on data since 1985. Journal of Shellfish Research 15(1):17–34.

141. Abisha, R., K. K. Krishnani, K. Sukhdhane, A. K. Verma, M. Brahmane, and N. K. Chadha. 2022. Sustainable development of climate-resilient aquaculture and culture-based fisheries through adaptation of abiotic stresses: a review. Journal of Water and Climate Change 13(7):2671–2689. DOI: 10.2166/wcc.2022.045

142. Crosson, L. M., N. Wight, G. R. VanBlaricom, I. Kiryu, J. D. Moore, and C. S. Friedman. 2014. Abalone withering syndrome: Distribution, impacts, current diagnostic methods, and new findings. Diseases of Aquatic Organisms 108(3):261–270.

143. CEFAS (Centre for Environment Fisheries and Aquaculture Science). 2019. Shellfish Biosecurity Measures Plan. Centre for Environment Fisheries and Aquaculture Science, Weymouth, Dorset, UK. Available: assets.publishing.service.gov.uk/government/uploads/system/uploads/attachment_data/file/919656/Shellfish_Biosecurity_Measures_Plan_2020.pdf (June 2022).

144. Arzul, I., D. Furones, D. Cheslett, L. Gennari, E. Delangle, F. Enez, C. Lupo, S. Mortensen, F. Pernet, and E. Peeler. 2021. Manual for bivalve disease management and biosecurity. European Commission, Brussels, Belgium Available: www.vivaldi-project.eu/content/download/158545/file/VIVALDI-Manual%20EN.pdf (March 2022).

145. Spark, E., S. Roberts, M. Deveney, T. Bradley, C. Dang, E. Wronski, M. Walker, and B. Zippel. 2018. National biosecurity plan guidelines for Australian oyster hatcheries. Commonwealth of Australia, Canberra, Australia. Available: www.awe.gov.au/sites/default/files/sitecollectiondocuments/animal-plant/aquatic/oysters.pdf (July 2023).

146. Green, T. J., C. Montagnani, K. Benkendorff, N. Robinson, and P. Speck. 2014. Ontogeny and water temperature influences the antiviral response of the Pacific oyster, *Crassostrea gigas*. Fish and Shellfish Immunology 36(1):151–157. DOI: 10.1016/j.fsi.2013.10.026

147. Qiao, X., L. Wang, and L. Song. 2021. The primitive interferon-like system and its antiviral function in molluscs. Developmental and Comparative Immunology 118:103997. DOI: 10.1016/j.dci.2021.103997

148. de la Ballina, N. R., F. Maresca, A. Cao, and A. Villalba. 2022. Bivalve Haemocyte Subpopulations: A Review. Frontiers in Immunology 13:826255. DOI: 10.3389/fimmu.2022.826255

149. Cawthorn, R. J. 2011. Diseases of American lobsters (*Homarus americanus*): A review. Journal of Invertebrate Pathology 106(1):71–78. DOI: 10.1016/j.jip.2010.09.010

150. Witkop, E. M., G. H. Wikfors, D. A. Proestou, K. M. Lundgren, M. Sullivan, and M. Gomez-Chiarri. 2022. *Perkinsus marinus* suppresses in vitro eastern oyster apoptosis via IAP-dependent and caspase-independent pathways involving TNFR, NF-kB, and oxidative pathway crosstalk. Developmental and Comparative Immunology 129:104339. DOI: 10.1016/j.dci.2022.104339

151. Carnegie, R. B., and E. M. Burreson. 2011. Declining impact of an introduced pathogen: *Haplosporidium nelsoni* in the oyster *Crassostrea virginica* in Chesapeake Bay. Marine Ecology Progress Series 432:1–15.

152. Lynch, S. A., G. Flannery, T. Hugh-Jones, D. Hugh-Jones, and S. C. Culloty. 2014. Thirty-year history of Irish (Rossmore) *Ostrea edulis* selectively bred for disease resistance to *Bonamia ostreae*. Diseases of Aquatic Organisms 110(1-2):113–121. DOI: 10.3354/dao02734

153. Dégremont, L., C. Garcia, and S. K. Allen. 2015. Genetic improvement for disease resistance in oysters: A review. Journal of Invertebrate Pathology 131:226–241. DOI: 10.1016/j.jip.2015.05.010

154. Hollenbeck, C. M., and I. A. Johnston. 2018. Genomic Tools and Selective Breeding in Molluscs. Frontiers in genetics 9:253. DOI: 10.3389/fgene.2018.00253

155. Azéma, P., J. B. Lamy, P. Boudry, T. Renault, M. A. Travers, and L. Dégremont. 2017. Genetic parameters of resistance to *Vibrio aestuarianus*, and OsHV-1 infections in the Pacific oyster, *Crassostrea gigas*, at three different life stages. Genetics Selection Evolution 49(1):23. DOI: 10.1186/s12711-017-0297-2

156. FDA (U.S. Food and Drug Administration) and ISSC (Interstate Shellfish Sanitation Conference). 2020. National Shellfish Sanitation Program (NSSP) Guide for the Control of Molluscan Shellfish: 2019 Revision. U.S. Food and Drug Administration, Washington, D.C. Available: www.fda.gov/media/143238/download (July 2023).

157. Cheng, T.-H. 1969. Production of kelp—a major aspect of China's exploitation of the sea. Economic Botany 23(3):215–236. DOI: 10.1007/BF02860454

158. Campbell, I., A. Macleod, C. Sahlmann, L. Neves, J. Funderud, M. Øverland, A. D. Hughes, and M. Stanley. 2019. The Environmental Risks Associated With the Development of Seaweed Farming in Europe—Prioritizing Key Knowledge Gaps. Frontiers in Marine Science 6(107). DOI: 10.3389/fmars.2019.00107

159. FAO (Food and Agriculture Organization of the United Nations). 2020. The State of World Fisheries and Aquaculture 2020: Sustainability in action. Food and Agriculture Organization of the United Nations, Rome, Italy. DOI: 10.4060/ca9229en

160. Cottier-Cook, E. J., J. P. Cabarubias, J. Brakel, J. Brodie, A. H. Buschmann, I. Campbell, A. T. Critchley, C. L. Hewitt, J. Huang, A. Q. Hurtado, C. S. B. Kambey, P. E. Lim, T. Liu, J. P. Mateo, F. E. Msuya, Z. Qi, L. Shaxson, G. D. Stentiford, and M. G. Bondad-Reantaso. 2022. A new Progressive Management Pathway for improving seaweed biosecurity. Nature Communications 13(1):7401. DOI: 10.1038/s41467-022-34783-8

161. United States, Executive Office of the President [Donald Trump]. 2020. Executive Order 13921: Promoting American Seafood Competitiveness and Economic Growth.

162. Ward, G. M., J. P. Faisan, E. J. Cottier-Cook, C. Gachon, A. Q. Hurtado, P. E. Lim, I. Matoju, F. E. Msuya, D. Bass, and J. Brodie. 2019. A review of reported seaweed diseases and pests in aquaculture in Asia. Journal of the World Aquaculture Society 51(4):815-828. DOI: 10.1111/jwas.12649

163. Kerwin, J. L., L. M. Johnson, H. C. Whisler, and A. R. Tuininga. 1992. Infection and morphogenesis of *Pythium marinum* in species of *Porphyra* and other red algae. Canadian Journal of Botany 70(5):1017–1024.

164. CDFW (California Department of Fish and Wildlife). 2022. Commonly Harvested Kelp and Other Marine Algae. Available: wildlife.ca.gov/Conservation/Marine/Kelp/Commonly-Harvested-Marine-Algae (July 2023).

165. Schaffelke, B., J. E. Smith, and C. L. Hewitt. 2006. Introduced Macroalgae—a Growing Concern. Journal of Applied Phycology 18(3):529–541. DOI: 10.1007/s10811-006-9074-2

166. Assis, J., A. V. Lucas, I. Bárbara, and E. Á. Serrão. 2016. Future climate change is predicted to shift long-term persistence zones in the cold-temperate kelp *Laminaria hyperborea*. Marine Environmental Research 113:174–182. DOI: 10.1016/j.marenvres.2015.11.005

167. Valero, M., M.-L. Guillemin, C. Destombe, B. Jacquemin, C. Gachon, Y. Badis, A. H. Buschmann, C. Camus, and S. Faugeron. 2017. Perspectives on domestication research for sustainable seaweed aquaculture. Perspectives in Phycology 4(1):33–46. DOI: 10.1127/pip/2017/0066

168. Kambey, C. S. B., I. Campbell, C. F. A. Sondak, A. R. M. Nor, P. E. Lim, and E. J. Cottier-Cook. 2020. An analysis of the current status and future of biosecurity frameworks for the Indonesian seaweed industry. Journal of Applied Phycology 32(4):2147–2160. DOI: 10.1007/s10811-019-02020-3

169. SCAAH (Subcommittee on Aquatic Animal Health) 2016. 2017. Aquatic farm biosecurity plan: generic guidelines and template. Canberra, Australia. Available: agriculture.gov.au/animal/aquatic/guidelines-and-resources (July 2023).

170. Steneck, R. S., M. H. Graham, B. J. Bourque, D. Corbett, J. M. Erlandson, J. A. Estes, and M. J. Tegner. 2002. Kelp forest ecosystems: biodiversity, stability, resilience and future. Environmental Conservation 29(4):436–459. DOI: 10.1017/S0376892902000322

171. Graham, M. H. 2004. Effects of Local Deforestation on the Diversity and Structure of Southern California Giant Kelp Forest Food Webs. Ecosystems 7(4):341–357. DOI: 10.1007/s10021-003-0245-6

172. Heinrich, S., K. Valentin, S. Frickenhaus, U. John, and C. Wiencke. 2012. Transcriptomic Analysis of Acclimation to Temperature and Light Stress in *Saccharina latissima* (Phaeophyceae). PLOS ONE 7(8):e44342. DOI: 10.1371/journal.pone.0044342

173. Shi, J., H. Wei, L. Zhao, Y. Yuan, J. Fang, and J. Zhang. 2011. A physical–biological coupled aquaculture model for a suspended aquaculture area of China. Aquaculture 318(3):412–424. DOI: 10.1016/j.aquaculture.2011.05.048

174. Ward, G. M., C. S. B. Kambey, J. P. Faisan, P. L. Tan, C. C. Daumich, I. Matoju, G. D. Stentiford, D. Bass, P. E. Lim, J. Brodie, and S. W. Poong. 2022. Ice-Ice disease: An environmentally and microbiologically driven syndrome in tropical seaweed aquaculture. Reviews in Aquaculture 14(1):414–439. DOI: 10.1111/raq.12606

175. Arasamuthu, A., and J. K. Patterson Edward. 2016. Occurrence of ice-ice disease in seaweed *Kappaphycus alvarezii* at Gulf of Mannar and Palk Bay, Southeastern India. Indian Journal of Geo-Marine Sciences 47(06):1208–1216.

176. Kambey, C. S. B., I. Campbell, E. J. Cottier-Cook, A. R. M. Nor, A. Kassim, A. Sade, and P. E. Lim. 2021. Seaweed aquaculture: a preliminary assessment of biosecurity measures for controlling the ice-ice syndrome and pest outbreaks of a *Kappaphycus* farm. Journal of Applied Phycology 33(5):3179–3197. DOI: 10.1007/s10811-021-02530-z

177. Bass, D., G. D. Stentiford, H.-C. Wang, B. Koskella, and C. R. Tyler. 2019. The Pathobiome in Animal and Plant Diseases. Trends in Ecology & Evolution 34(11):996–1008. DOI: 10.1016/j.tree.2019.07.012

178. Chiaramonte, L., D. Munson, and J. Trushenski. 2016. Climate Change and Considerations for Fish Health and Fish Health Professionals. Fisheries 41(7):396–399. DOI: 10.1080/03632415.2016.1182508

179. Snieszko, S. F. 1978. Bacterial Diseases of Fishes and Their Control. Marine Fisheries Review 40(3):65–68.

180. Burge, C. A., C. Mark Eakin, C. S. Friedman, B. Froelich, P. K. Hershberger, E. E. Hofmann, L. E. Petes, K. C. Prager, E. Weil, B. L. Willis, S. E. Ford, and C. D. Harvell. 2014. Climate Change Influences on Marine Infectious Diseases: Implications for Management and Society. Annual Review of Marine Science 6(1):249–277. DOI: 10.1146/annurev-marine-010213-135029

181. Shields, J. D. 2013. Complex etiologies of emerging diseases in lobsters (*Homarus americanus*) from Long Island Sound. Canadian Journal of Fisheries and Aquatic Sciences 70(11):1576–1587. DOI: 10.1139/cjfas-2013-0050

182. Mora, C., T. McKenzie, I. M. Gaw, J. M. Dean, H. von Hammerstein, T. A. Knudson, R. O. Setter, C. Z. Smith, K. M. Webster, J. A. Patz, and E. C. Franklin. 2022. Over half of known human pathogenic diseases can be aggravated by climate change. Nature Climate Change 12(9):869–875. DOI: 10.1038/s41558-022-01426-1

183. Eggleton, T. 2017. Climate Change: The Physical Picture. Climate Change Impacts on Fisheries and Aquaculture: A Global Analysis 1:1–21.

184. Gutiérrez, J. M., R. G. Jones, G. T. Narisma, L. M. Alves, M. Amjad, I. V. Gorodetskaya, M. Grose, N. A. B. Klutse, S. Krakovska, J. Li, D. Martínez-Castro, L. O. Mearns, S. H. Mernild, T. Ngo-Duc, B. van den Hurk, and J.-H. Yoon. 2021. IPCC WGI Interactive Atlas: Regional information (Advanced). Available: interactive-atlas.ipcc.ch/Accessed (July 2023)

185. Iturbide, M., J. Fernández, J. M. Gutiérrez, J. Bedia, E. Cimadevilla, J. Díez-Sierra, R. Manzanas, A. Casanueva, J. Baño-Medina, J. Milovac, S. Herrera, A. S. Cofiño, D. San Martín, M. García-Díez, M. Hauser, D. Huard, and Ö. Yelekci. 2021. Repository supporting the implementation of FAIR principles in the IPCC-WGI Atlas. CERN (Zenodo), Genève, Switzerland. DOI: 10.5281/zenodo.3691645

186. Brown, A. R., M. Lilley, J. Shutler, C. Lowe, Y. Artioli, R. Torres, E. Berdalet, and C. R. Tyler. 2020. Assessing risks and mitigating impacts of harmful algal blooms on mariculture and marine fisheries. Reviews in Aquaculture 12(3):1663–1688. DOI: 10.1111/raq.12403

187. Maulu, S., Hasimuna, O., Haamblya, L., Monde, C., Musuka,C., Makorwa, T., Munganga, B., Phiri, K., Nsekanabo, J. 2021. Climate Change Effects on Aquaculture Production: Sustainability Implications, Mitigation, and Adaptations. Frontiers in Sustainable Food Systems 5(609097). DOI: 10.3389/fsufs.2021.609097

188. Pörtner, H. O. 2002. Climate variations and the physiological basis of temperature dependent biogeography: systemic to molecular hierarchy of thermal tolerance in animals. Comparative Biochemistry and Physiology Part A: Molecular & Integrative Physiology 132(4):739–761. DOI: 10.1016/S1095-6433(02)00045-4

189. Tigchelaar, M., W. W. L. Cheung, E. Y. Mohammed, M. J. Phillips, H. J. Payne, E. R. Selig, C. C. C. Wabnitz, M. A. Oyinlola, T. L. Frölicher, J. A. Gephart, C. D. Golden, E. H. Allison, A. Bennett, L. Cao, J. Fanzo, B. S. Halpern, V. W. Y. Lam, F. Micheli, R. L. Naylor, U. R. Sumaila, A. Tagliabue, and M. Troell. 2021. Compound climate risks threaten aquatic food system benefits. Nature Food 2(9):673–682. DOI: 10.1038/s43016-021-00368-9

190. Marcogliese, D. 2008. The impact of climate change on the parasites and infectious diseases of aquatic animals. Revue Scientifique et Technique 27(2):467–484.

191. Reid, G. K., H. J. Gurney-Smith, M. Flaherty, A. F. Garber, I. Forster, K. Brewer-Dalton, D. Knowler, D. J. Marcogliese, T. Chopin, R. D. Moccia, C. T. Smith, and S. De Silva. 2019. Climate change and aquaculture: Considering adaptation potential. Aquaculture Environment Interactions 11:603–624.

192. Magarinos, B., F. Pazos, Y. Santos, J. L. Romalde, and A. E. Toranzo. 1995. Response of *Pasteurella piscicida* and *Flexibacter maritimus* to skin mucus of marine fish. Diseases of Aquatic Organisms 21(2):103–108.

193. Sundh, H., and K. S. Sundell. 2015. Environmental impacts on fish mucosa. Pages 171–197 in B. H. Beck and E. Peatman, editors. Mucosal Health in Aquaculture. Academic Press, San Diego, California.

194. Beesley, A., D. Lowe, C. Pascoe, and S. Widdicombe. 2008. Effects of CO_2-induced seawater acidification on the health of *Mytilus edulis*. Climate Research 37(2–3):215–225.

195. Bibby, R., P. Cleall-Harding, S. Rundle, S. Widdicombe, and J. Spicer. 2007. Ocean acidification disrupts induced defences in the intertidal gastropod *Littorina littorea*. Biology Letters 3(6):699–701.

196. Wood, H. L., J. I. Spicer, and S. Widdicombe. 2008. Ocean acidification may increase calcification rates, but at a cost. Proceedings of the Royal Society B: Biological Sciences 275(1644):1767–1773. DOI: 10.1098/rspb.2008.0343

197. Shields, J. D. 2019. Climate change enhances disease processes in crustaceans: case studies in lobsters, crabs, and shrimps. Journal of Crustacean Biology 39(6):673–683. DOI: 10.1093/jcbiol/ruz072

198. Harvell, C. D., D. Montecino-Latorre, J. M. Caldwell, J. M. Burt, K. Bosley, A. Keller, S. F. Heron, A. K. Salomon, L. Lee, O. Pontier, C. Pattengill-Semmens, and J. K. Gaydos. 2019. Disease epidemic and a marine heat wave are associated with the continental-scale collapse of a pivotal predator (*Pycnopodia helianthoides*). Science Advances 5(1):eaau7042. DOI: 10.1126/sciadv.aau7042

199. Maynard, J., R. Van Hooidonk, C. D. Harvell, C. M. Eakin, G. Liu, B. L. Willis, G. J. Williams, M. L. Groner, A. Dobson, and S. F. Heron. 2016. Improving marine disease surveillance through sea temperature monitoring, outlooks and projections. Philosophical Transactions of the Royal Society B: Biological Sciences 371(1689):20150208.

200. Rose, J. B., P. R. Epstein, E. K. Lipp, B. H. Sherman, S. M. Bernard, and J. A. Patz. 2001. Climate variability and change in the United States: potential impacts on water- and foodborne diseases caused by microbiologic agents. Environmental Health Perspectives 109(suppl 2):211–221. DOI: 10.1289/ehp.01109s2211

201. Larsen, A. K., I. H. Nymo, K. K. Sørensen, M. Seppola, R. Rødven, M. P. Jiménez de Bagüés, S. Al Dahouk, and J. Godfroid. 2018. Concomitant temperature stress and immune activation may increase mortality despite efficient clearance of an intracellular bacterial infection in Atlantic cod. Frontiers in Microbiology 9:2963.

202. Groner, M. L., M. E. Eisenlord, R. M. Yoshioka, E. A. Fiorenza, P. D. Dawkins, O. J. Graham, M. Winningham, A. Vompe, N. D. Rivlin, B. Yang, C. A. Burge, B. Rappazzo, C. P. Gomes, and C. D. Harvell. 2021. Warming sea surface temperatures fuel summer epidemics of eelgrass wasting disease. Marine Ecology Progress Series 679:47–58.

203. Harper, C., and J. C. Wolf. 2009. Morphologic Effects of the Stress Response in Fish. ILAR Journal 50(4):387–396. DOI: 10.1093/ilar.50.4.387

204. Le Morvan, C., D. Troutaud, and P. Deschaux. 1998. Differential Effects of Temperature on Specific and Nonspecific Immune Defences in Fish. Journal of Experimental Biology 201(2):165–168. DOI: 10.1242/jeb.201.2.165

205. Walsh, J. E., R. L. Thoman, U. S. Bhatt, P. A. Bieniek, B. Brettschneider, M. Brubaker, S. Danielson, R. Lader, F. Fetterer, and K. Holderied. 2018. The high latitude marine heat wave of 2016 and its impacts on Alaska. Bulletin of the American Meteorological Society 99(1):S39–S43.

206. Pickering, A. 1989. Environmental stress and the survival of brown trout, *Salmo trutta*. Freshwater Biology 21(1):47–55.

207. Pulsford, A. L., S. Lemaire-Gony, M. Tomlinson, N. Collingwood, and P. J. Glynn. 1994. Effects of acute stress on the immune system of the dab, *Limanda limanda*. Comparative Biochemistry and Physiology Part C: Pharmacology, Toxicology and Endocrinology 109(2):129–139. DOI: 10.1016/0742-8413(94)00053-D

208. Espelid, S., G. B. Løkken, K. Steiro, and J. Bøgwald. 1996. Effects of cortisol and stress on the immune system in Atlantic Salmon (*Salmo salar* L.). Fish and Shellfish Immunology 6(2):95–110. DOI: 10.1006/fsim.1996.0011

209. Wendelaar Bonga, S. E. 1997. The stress response in fish. Physiological Reviews 77(3):591–625. DOI: 10.1152/physrev.1997.77.3.591

210. Bly, J. E., and L. W. Clem. 1992. Temperature and teleost immune functions. Fish and Shellfish Immunology 2(3):159–171.

211. Magnadóttir, B. 2006. Innate immunity of fish (overview). Fish and Shellfish Immunology 20(2):137–151. DOI: 10.1016/j.fsi.2004.09.006

212. Abram, Q. H., B. Dixon, and B. A. Katzenback. 2017. Impacts of Low Temperature on the Teleost Immune System. Biology 6(4):39.

213. Ellis, A. E. 2001. Innate host defense mechanisms of fish against viruses and bacteria. Developmental and Comparative Immunology 25(8):827–839. DOI: 10.1016/S0145-305X(01)00038-6

214. Sundh, H., B. O. Kvamme, F. Fridell, R. E. Olsen, T. Ellis, G. L. Taranger, and K. Sundell. 2010. Intestinal barrier function of Atlantic salmon (*Salmo salar* L.) post smolts is reduced by common sea cage environments and suggested as a possible physiological welfare indicator. BMC Physiology 10(1):22. DOI: 10.1186/1472-6793-10-22

215. Sae-Lim, P., A. Kause, H. A. Mulder, and I. Olesen. 2017. Breeding and Genetics Symposium: Climate change and selective breeding in aquaculture. Journal of Animal Science 95(4):1801–1812. DOI: 10.2527/jas.2016.1066

216. Liu, J., W. Peng, F. Yu, Y. Shen, W. Yu, Y. Lu, W. Lin, M. Zhou, Z. Huang, X. Luo, W. You, and C. Ke. 2022. Genomic selection applications can improve the environmental performance of aquatics: A case study on the heat tolerance of abalone. Evolutionary Applcations 15(6):992–1001. DOI: 10.1111/eva.13388

217. Gallardo-Hidalgo, J., A. Barría, G. M. Yoshida, and J. M. Yáñéz. 2021. Genetics of growth and survival under chronic heat stress and trade-offs with growth- and robustness-related traits in rainbow trout. Aquaculture 531:735685. DOI: 10.1016/j.aquaculture.2020.735685

218. Ma, A., Z. Huang, X.-a. Wang, Y. Xu, and X. Guo. 2021. Identification of quantitative trait loci associated with upper temperature tolerance in turbot, *Scophthalmus maximus*. Scientific Reports 11(1):21920. DOI: 10.1038/s41598-021-01062-3

219. Bermingham, M., and M. F. Mulcahy. 2004. Environmental risk factors associated with amoebic gill disease in cultured salmon, *Salmo salar* L., smolts in Ireland. Journal of Fish Diseases 27(10):555–571.

220. Anderson, D. M., E. Fensin, C. J. Gobler, A. E. Hoeglund, K. A. Hubbard, D. M. Kulis, J. H. Landsberg, K. A. Lefebvre, P. Provoost, M. L. Richlen, J. L. Smith, A. R. Solow, and V. L. Trainer. 2021. Marine harmful algal blooms (HABs) in the United States: History, current status and future trends. Harmful Algae 102:101975. DOI: 10.1016/j.hal.2021.101975

221. Callaway, R., A. P. Shinn, S. E. Grenfell, J. E. Bron, G. Burnell, E. J. Cook, M. Crumlish, S. Culloty, K. Davidson, R. P. Ellis, K. J. Flynn, C. Fox, D. M. Green, G. C. Hays, A. D. Hughes, E. Johnston, C. D. Lowe, I. Lupatsch, S. Malham, A. F. Mendzil, T. Nickell, T. Pickerell, A. F. Rowley, M. S. Stanley, D. R. Tocher, J. F. Turnbull, G. Webb, E. Wootton, and R. J. Shields. 2012. Review of climate change impacts on marine aquaculture in the UK and Ireland. Aquatic Conservation: Marine and Freshwater Ecosystems 22(3):389–421. DOI: 10.1002/aqc.2247

222. Gazeau, F., L. M. Parker, S. Comeau, J.-P. Gattuso, W. A. O'Connor, S. Martin, H.-O. Pörtner, and P. M. Ross. 2013. Impacts of ocean acidification on marine shelled molluscs. Marine Biology 160(8):2207–2245. DOI: 10.1007/s00227-013-2219-3

223. Barton, A., B. Hales, G. G. Waldbusser, C. Langdon, and R. A. Feely. 2012. The Pacific oyster, *Crassostrea gigas*, shows negative correlation to naturally elevated carbon dioxide levels: Implications for near-term ocean acidification effects. Limnology and Oceanography 57(3):698–710. DOI: 10.4319/lo.2012.57.3.0698

224. Waldbusser, G. G., B. Hales, C. J. Langdon, B. A. Haley, P. Schrader, E. L. Brunner, M. W. Gray, C. A. Miller, I. Gimenez, and G. Hutchinson. 2015. Ocean Acidification Has Multiple Modes of Action on Bivalve Larvae. PLOS ONE 10(6):e0128376. DOI: 10.1371/journal.pone.0128376

225. Gray, M. W., C. J. Langdon, G. G. Waldbusser, B. Hales, and S. Kramer. 2017. Mechanistic understanding of ocean acidification impacts on larval feeding physiology and energy budgets of the mussel *Mytilus californianus*. Marine Ecology Progress Series 563:81–94.

226. Mekkes, L., W. Renema, N. Bednaršek, S. R. Alin, R. A. Feely, J. Huisman, P. Roessingh, and K. T. C. A. Peijnenburg. 2021. Pteropods make thinner shells in the upwelling region of the California Current Ecosystem. Scientific Reports 11(1):1731. DOI: 10.1038/s41598-021-81131-9

227. Chandra Rajan, K., Y. Meng, Z. Yu, S. B. Roberts, and T. Vengatesen. 2021. Oyster biomineralization under ocean acidification: From genes to shell. Global Change Biology 27(16):3779–3797. DOI: 10.1111/gcb.15675

228. Hernroth, B., H. N. Sköld, K. Wiklander, F. Jutfelt, and S. Baden. 2012. Simulated climate change causes immune suppression and protein damage in the crustacean *Nephrops norvegicus*. Fish and Shellfish Immunology 33(5):1095–1101. DOI: 10.1016/j.fsi.2012.08.011

229. Wang, Q., R. Cao, X. Ning, L. You, C. Mu, C. Wang, L. Wei, M. Cong, H. Wu, and J. Zhao. 2016. Effects of ocean acidification on immune responses of the Pacific oyster *Crassostrea gigas*. Fish and Shellfish Immunology 49:24–33. DOI: 10.1016/j.fsi.2015.12.025

230. Ivanina, A. V., C. Hawkins, and I. M. Sokolova. 2014. Immunomodulation by the interactive effects of cadmium and hypercapnia in marine bivalves *Crassostrea virginica* and *Mercenaria mercenaria*. Fish and Shellfish Immunology 37(2):299–312. DOI: 10.1016/j.fsi.2014.02.016

231. Cao, R., Y. Liu, Q. Wang, D. Yang, H. Liu, W. Ran, Y. Qu, and J. Zhao. 2018. Seawater Acidification Reduced the Resistance of *Crassostrea gigas* to *Vibrio splendidus* Challenge: An Energy Metabolism Perspective. Frontiers in Physiology 9. DOI: 10.3389/fphys.2018.00880

232. Mackenzie, C. L., S. A. Lynch, S. C. Culloty, and S. K. Malham. 2014. Future Oceanic Warming and Acidification Alter Immune Response and Disease Status in a Commercial Shellfish Species, *Mytilus edulis* L. PLOS ONE 9(6):e99712. DOI: 10.1371/journal.pone.0099712

233. Ern, R., and A. J. Esbaugh. 2016. Hyperventilation and blood acid–base balance in hypercapnia exposed red drum (*Sciaenops ocellatus*). Journal of Comparative Physiology B 186(4):447–460. DOI: 10.1007/s00360-016-0971-7

234. Esbaugh, A. J., R. Ern, W. M. Nordi, and A. S. Johnson. 2016. Respiratory plasticity is insufficient to alleviate blood acid–base disturbances after acclimation to ocean acidification in the estuarine red drum, *Sciaenops ocellatus*. Journal of Comparative Physiology B 186(1):97–109. DOI: 10.1007/s00360-015-0940-6

235. Sampaio, E., A. R. Lopes, S. Francisco, J. R. Paula, M. Pimentel, A. L. Maulvault, T. Repolho, T. F. Grilo, P. Pousão-Ferreira, A. Marques, and R. Rosa. 2018. Ocean acidification dampens physiological stress response to warming and contamination in a commercially-important fish (*Argyrosomus regius*). Science of The Total Environment 618:388–398. DOI: 10.1016/j.scitotenv.2017.11.059

236. Checkley, D. M., Jr., A. G. Dickson, M. Takahashi, J. A. Radich, N. Eisenkolb, and R. Asch. 2009. Elevated CO_2 Enhances Otolith Growth in Young Fish. Science 324(5935):1683. DOI: 10.1126/science.1169806

237. Bignami, S., I. C. Enochs, D. P. Manzello, S. Sponaugle, and R. K. Cowen. 2013. Ocean acidification alters the otoliths of a pantropical fish species with implications for sensory function. Proceedings of the National Academy of Sciences 110(18):7366–7370. DOI: 10.1073/pnas.1301365110

238. Di Santo, V. 2019. Ocean acidification and warming affect skeletal mineralization in a marine fish. Proceedings of the Royal Society B: Biological Sciences 286(1894):20182187. DOI: 10.1098/rspb.2018.2187

239. Baumann, H., S. C. Talmage, and C. J. Gobler. 2012. Reduced early life growth and survival in a fish in direct response to increased carbon dioxide. Nature Climate Change 2(1):38–41. DOI: 10.1038/nclimate1291

240. Frommel, A. Y., R. Maneja, D. Lowe, C. K. Pascoe, A. J. Geffen, A. Folkvord, U. Piatkowski, and C. Clemmesen. 2014. Organ damage in Atlantic herring larvae as a result of ocean acidification. Ecological Applications 24(5):1131–1143. DOI: 10.1890/13-0297.1

241. Stiasny, M. H., F. H. Mittermayer, M. Sswat, R. Voss, F. Jutfelt, M. Chierici, V. Puvanendran, A. Mortensen, T. B. H. Reusch, and C. Clemmesen. 2016. Ocean Acidification Effects on Atlantic Cod Larval Survival and Recruitment to the Fished Population. PLOS ONE 11(8):e0155448. DOI: 10.1371/journal.pone.0155448

242. Hurst, T. P., L. A. Copeman, S. A. Haines, S. D. Meredith, K. Daniels, and K. M. Hubbard. 2019. Elevated CO_2 alters behavior, growth, and lipid composition of Pacific cod larvae. Marine Environmental Research 145:52–65. DOI: 10.1016/j.marenvres.2019.02.004

243. Cui, W., L. Cao, J. Liu, Z. Ren, B. Zhao, and S. Dou. 2020. Effects of seawater acidification and cadmium on the antioxidant defense of flounder *Paralichthys olivaceus* larvae. Science of The Total Environment 718:137234. DOI: 10.1016/j.scitotenv.2020.137234

244. Nilsson, G. E., D. L. Dixson, P. Domenici, M. I. McCormick, C. Sørensen, S.-A. Watson, and P. L. Munday. 2012. Near-future carbon dioxide levels alter fish behaviour by interfering with neurotransmitter function. Nature Climate Change 2(3):201–204. DOI: 10.1038/nclimate1352

245. Dixson, D. L., P. L. Munday, and G. P. Jones. 2010. Ocean acidification disrupts the innate ability of fish to detect predator olfactory cues. Ecology Letters 13(1):68–75. DOI: 10.1111/j.1461-0248.2009.01400.x

246. Munday, P. L., D. L. Dixson, J. M. Donelson, G. P. Jones, M. S. Pratchett, G. V. Devitsina, and K. B. Døving. 2009. Ocean acidification impairs olfactory discrimination and homing ability of a marine fish. Proceedings of the National Academy of Sciences 106(6):1848–1852. DOI: 10.1073/pnas.0809996106

247. Clark, T. D., G. D. Raby, D. G. Roche, S. A. Binning, B. Speers-Roesch, F. Jutfelt, and J. Sundin. 2020. Ocean acidification does not impair the behaviour of coral reef fishes. Nature 577(7790):370–375. DOI: 10.1038/s41586-019-1903-y

248. Roggatz, C. C., M. Lorch, J. D. Hardege, and D. M. Benoit. 2016. Ocean acidification affects marine chemical communication by changing structure and function of peptide signalling molecules. Global Change Biology 22(12):3914–3926. DOI: 10.1111/gcb.13354

249. Dang, X., T. H. Lee, and V. Thiyagarajan. 2022. Wild oyster population resistance to ocean acidification adversely affected by bacterial infection. Environmental Pollution 317:120813. DOI: 10.1016/j.envpol.2022.120813

250. Bricknell, I. R., S. J. Dalesman, B. O'Shea, C. P. Campbell, and A. J. Mordue Luntz. 2006. Effect of environmental salinity on sea lice *Lepeophtheirus salmonis* settlement success. Diseases of Aquatic Organisms 71(3):201–212.

251. Hofmann, E., S. Ford, E. Powell, and J. Klinck. 2001. Modeling studies of the effect of climate variability on MSX disease in eastern oyster (*Crassostrea virginica*) populations. Hydrobiologia 460(1):195–212. DOI: 10.1023/A:1013159329598

252. Ellis, R. P., M. A. Urbina, and R. W. Wilson. 2017. Lessons from two high CO_2 worlds—future oceans and intensive aquaculture. Global Change Biology 23(6):2141–2148. DOI: 10.1111/gcb.13515

253. Campana, S. E., R. B. Stefánsdóttir, K. Jakobsdóttir, and J. Sólmundsson. 2020. Shifting fish distributions in warming sub-Arctic oceans. Scientific Reports 10(1):16448. DOI: 10.1038/s41598-020-73444-y

254. Bond, N. A., M. F. Cronin, H. Freeland, and N. Mantua. 2015. Causes and impacts of the 2014 warm anomaly in the NE Pacific. Geophysical Research Letters 42(9):3414–3420. DOI: 10.1002/2015GL063306

255. Yang, Q., E. D. Cokelet, P. J. Stabeno, L. Li, A. B. Hollowed, W. A. Palsson, N. A. Bond, and S. J. Barbeaux. 2019. How "The Blob" affected groundfish distributions in the Gulf of Alaska. Fisheries Oceanography 28(4):434–453.

256. Pinsky, M. L., B. Worm, M. J. Fogarty, J. L. Sarmiento, and S. A. Levin. 2013. Marine Taxa Track Local Climate Velocities. Science 341(6151):1239–1242. DOI: 10.1126/science.1239352

257. Morley, J. W., R. L. Selden, R. J. Latour, T. L. Frölicher, R. J. Seagraves, and M. L. Pinsky. 2018. Projecting shifts in thermal habitat for 686 species on the North American continental shelf. PLOS ONE 13(5):e0196127. DOI: 10.1371/journal.pone.0196127

258. Subcommittee on Aquatic Animal Health. 2017. Aquatic Farm Biosecurity Plan: generic guidelines and template. Commonwealth of Australia, Canberra, Australia. Availablewww.agriculture.gov.au/sites/default/files/sitecollectiondocuments/fisheries/aquaculture/aquaculture-farm-biosecurity-plan.pdf (May 2022).

259. Handisyde, N., L. Ross, M. Badjeck, and E. Allison. 2006. The effects of climate change on world aquaculture: A global perspective. Stirling Institute of Aquaculture, Stirling, Scotland, U.K.

260. Hutson, K. S., A. J. Mooney, I. Ernst, A. K. Brazenor, M. Scheel, and J. Atalah. A decision support tool for parasite management in fish aquaculture. Reviews in Aquaculture 14(3). DOI: 10.1111/raq.12668

261. Rohr, J. R., and J. M. Cohen. 2020. Understanding how temperature shifts could impact infectious disease. PLOS Biology 18(11):e3000938. DOI: 10.1371/journal.pbio.3000938

262. Confalonieri, U., B. Menne, R. Akhtar, K. L. Ebi, M. Hauengue, R. S. Kovats, B. Revich, and A. Woodward. 2007. Human Health. Intergovernmental Panel on Climate Change, Cambridge, U.K.

263. Zhou, L., J. Huang, Y. Jiang, J. Kong, X. Xie, and F. Yin. 2022. pH Regulates the Formation and Hatching of *Cryptocaryon irritans* Tomonts, Which Affects Cryptocaryoniasis Occurrence in *Larimichthys crocea* Aquaculture. Applied and Environmental Microbiology 88(7):e00058-22. DOI: 10.1128/aem.00058-22

264. Cook, T., M. Folli, J. Klinck, S. Ford, and J. Miller. 1998. The Relationship Between Increasing Sea-surface Temperature and the Northward Spread of *Perkinsus marinus* (Dermo) Disease Epizootics in Oysters. Estuarine, Coastal and Shelf Science 46(4):587–597. DOI: 10.1006/ecss.1997.0283

265. Bianchi, T. S., S. F. DiMarco, J. H. Cowan, R. D. Hetland, P. Chapman, J. W. Day, and M. A. Allison. 2010. The science of hypoxia in the Northern Gulf of Mexico: A review. Science of The Total Environment 408(7):1471–1484. DOI: 10.1016/j.scitotenv.2009.11.047

266. Gruber, N., C. Hauri, Z. Lachkar, D. Loher, T. L. Frölicher, and G.-K. Plattner. 2012. Rapid Progression of Ocean Acidification in the California Current System. Science 337(6091):220–223. DOI: 10.1126/science.1216773

267. Peeler, E. J., and M. J. Otte. 2016. Epidemiology and Economics Support Decisions about Freedom from Aquatic Animal Disease. Transboundary and Emerging Diseases 63(3):266–277. DOI: 10.1111/tbed.12278

268. USDA. 2022. Aquaculture. Available: www.usda.gov/topics/farming/aquaculture (July 2023).

269. USDA-APHIS. 2022. Veterinary Biologics. Available: www.aphis.usda.gov/aphis/ourfocus/animalhealth/veterinary-biologics (January 2022).

270. Barnes, A. C., O. Rudenko, M. Landos, H. T. Dong, A. Lusiastuti, L. H. Phuoc, and J. Delamare-Deboutteville. 2022. Autogenous vaccination in aquaculture: A locally enabled solution towards reduction of the global antimicrobial resistance problem. Reviews in Aquaculture 14(2):907–918. DOI: 10.1111/raq.12633

271. FDA (Food and Drug Administration). 2022. Approved Aquaculture Drugs. Available: www.fda.gov/animal-veterinary/aquaculture/approved-aquaculture-drugs (July 2023).

272. Moore, J. D., S. N. Byron, B. C. Marshman, and J. P. Snider. 2019. An oxytetracycline bath protocol to eliminate the agent of withering syndrome, *Candidatus* Xenohaliotis californiensis, in captive abalone populations. Aquaculture 503:267–274. DOI: 10.1016/j.aquaculture.2019.01.014

273. Miranda, C. D., F. A. Godoy, and M. R. Lee. 2018. Current Status of the Use of Antibiotics and the Antimicrobial Resistance in the Chilean Salmon Farms. Frontiers in Microbiology 9. DOI: 10.3389/fmicb.2018.01284

274. Hedberg, N., I. Stenson, M. Nitz Pettersson, D. Warshan, H. Nguyen-Kim, M. Tedengren, and N. Kautsky. 2018. Antibiotic use in Vietnamese fish and lobster sea cage farms; implications for coral reefs and human health. Aquaculture 495:366–375. DOI: 10.1016/j.aquaculture.2018.06.005

275. Holmes, A. H., L. S. Moore, A. Sundsfjord, M. Steinbakk, S. Regmi, A. Karkey, P. J. Guerin, and L. J. Piddock. 2016. Understanding the mechanisms and drivers of antimicrobial resistance. Lancet 387(10014):176–187. DOI: 10.1016/S0140-6736(15)00473-0

276. Buschmann, A. H., A. Tomova, A. López, M. A. Maldonado, L. A. Henríquez, L. Ivanova, F. Moy, H. P. Godfrey, and F. C. Cabello. 2012. Salmon Aquaculture and Antimicrobial Resistance in the Marine Environment. PLOS ONE 7(8):e42725. DOI: 10.1371/journal.pone.0042724

277. Shah, S. Q. A., F. C. Cabello, T. M. L'Abée-Lund, A. Tomova, H. P. Godfrey, A. H. Buschmann, and H. Sorum. 2014. Antimicrobial resistance and antimicrobial resistance genes in marine bacteria from salmon aquaculture and non-aquaculture sites. Environmental Microbiology 16(5):1310–1320. DOI: 10.1111/1462-2920.12421

278. Wegener, H. 2003. Ending the Use of Antimicrobial Growth Promoters Is Making a Difference. ASM News 69(9):443–445.

279. Cabello, F. C., H. P. Godfrey, A. Tomova, L. Ivanova, H. Dolz, A. Millanao, and A. H. Buschmann. 2013. Antimicrobial use in aquaculture re-examined: its relevance to antimicrobial resistance and to animal and human health. Environmental Microbiology 15(7):1917–1942. DOI: 10.1111/1462-2920.12134

280. Sneeringer, S. 2015. Restrictions on Antibiotic Use for Production Purposes in U.S. Livestock Industries Likely To Have Small Effects on Prices and Quantities. Available: www.ers.usda.gov/amber-waves/2015/november/restrictions-on-antibiotic-use-for-production-purposes-in-u-s-livestock-industries-likely-to-have-small-effects-on-prices-and-quantities/ (July 2023).

281. FDA-CVM (Food and Drug Administration-Center for Veterinary Medicine). 2020. Fact Sheet: Veterinary Feed Directive Final Rule and Next Steps. Available: www.fda.gov/animal-veterinary/development-approval-process/fact-sheet-veterinary-feed-directive-final-rule-and-next-steps (February 2022).

282. Merrifield, D. L. 2012. Probiotics, prebiotics in aquatic animals. Available: www.globalseafood.org/advocate/probiotics-prebiotics-in-aquatic-animals/ (July 2023).

283. Ganguly, S., I. Paul, and S. Mukhopadhayay. 2010. Application and Effectiveness of Immunostimulants, Probiotics, and Prebiotics in Aquaculture: A Review. Israeli Journal of Aquaculture - Bamidgeh 62:130–138. DOI: 10.46989/001c.38880

284. Hoseinifar, S. H., Y.-Z. Sun, A. Wang, and Z. Zhou. 2018. Probiotics as Means of Diseases Control in Aquaculture, a Review of Current Knowledge and Future Perspectives. Frontiers in Microbiology 9(2429). DOI: 10.3389/fmicb.2018.02429

285. Ringø, E., R. E. Olsen, T. Ø. Gifstad, R. A. Dalmo, H. Amlund, G. I. Hemre, and A. M. Bakke. 2010. Prebiotics in aquaculture: a review. Aquaculture Nutrition 16(2):117–136. DOI: 10.1111/j.1365-2095.2009.00731.x

286. Guerreiro, I., A. Oliva-Teles, and P. Enes. 2018. Prebiotics as functional ingredients: focus on Mediterranean fish aquaculture. Reviews in Aquaculture 10(4):800–832. DOI: 10.1111/raq.12201

287. Ringø, E. 2020. Probiotics in shellfish aquaculture. Aquaculture and Fisheries 5(1):1–27.

288. Kapareiko, D., H. J. Lim, E. J. Schott, A. Hanif, and G. H. Wikfors. 2011. Isolation and evaluation of new probiotic bacteria for use in shellfish hatcheries: II. Effects of a *Vibrio* sp. probiotic candidate upon survival of oyster larvae (*Crassostrea virginica*) in pilot-scale trials. Journal of Shellfish Research 30(3):617–625.

289. FDA (Food and Drug Administration). 2015. CPG Sec. 689.100 Direct-Fed Microbial Products. Available: www.fda.gov/regulatory-information/search-fda-guidance-documents/cpg-sec-689100-direct-fed-microbial-products (July 2023).

290. Anokyewaa, M. A., K. Amoah, Y. Li, Y. Lu, F. K. Kuebutornye, B. Asiedu, and I. Seidu. 2021. Prevalence of virulence genes and antibiotic susceptibility of *Bacillus* used in commercial aquaculture probiotics in China. Aquaculture Reports 21:100784.

291. Song, S. K., B. R. Beck, D. Kim, J. Park, J. Kim, H. D. Kim, and E. Ringø. 2014. Prebiotics as immunostimulants in aquaculture: A review. Fish and Shellfish Immunology 40(1):40–48. DOI: 10.1016/j.fsi.2014.06.016

292. Yilmaz, S., E. Yilmaz, M. A. O. Dawood, E. Ringø, E. Ahmadifar, and H. M. R. Abdel-Latif. 2022. Probiotics, prebiotics, and synbiotics used to control vibriosis in fish: A review. Aquaculture 547:737514. DOI: 10.1016/j.aquaculture.2021.737514

293. Cerezuela, R. 2011. Current Knowledge in Synbiotic Use for Fish Aquaculture: A Review. Journal of Aquaculture Research and Development S1:1–8. DOI: 10.4172/2155-9546.S1-008

294. Havard Law School Emmett Environmental Law & Policy Clinic, Environmental Law Institute, and The Ocean Foundation. 2012. Offshore Aquaculture Regulation under the Clean Water Act. Harvard College, Boston, Massachusetts. Available: oceanfdn.org/sites/default/files/cwa-aquaculture.pdf (July 2023).

295. Liu, Y., S.-I. Saitoh, I. N. Radiarta, T. Isada, T. Hirawake, H. Mizuta, and H. Yasui. 2013. Improvement of an aquaculture site-selection model for Japanese kelp (*Saccharina japonica*) in southern Hokkaido, Japan: an application for the impacts of climate events. ICES Journal of Marine Science 70(7):1460–1470. DOI: 10.1093/icesjms/fst108

296. Green, D. M. 2010. A strategic model for epidemic control in aquaculture. Preventive Veterinary Medicine 94(1):119–127. DOI: 10.1016/j.prevetmed.2009.12.004

297. Wasserberg, G., E. E. Osnas, R. E. Rolley, and M. D. Samuel. 2009. Host culling as an adaptive management tool for chronic wasting disease in white-tailed deer: a modelling study. Journal of Applied Ecology 46(2):457–466. DOI: 10.1111/j.1365-2664.2008.01576.x

298. Kermack, W. O., A. G. McKendrick, and G. T. Walker. 1927. A contribution to the mathematical theory of epidemics. Proceedings of the Royal Society A: Mathematical, Physical and Engineering Sciences 115(772):700–721. DOI: 10.1098/rspa.1927.0118

299. Benson, L., R. S. Davidson, D. M. Green, A. Hoyle, M. R. Hutchings, and G. Marion. 2021. When and why direct transmission models can be used for environmentally persistent pathogens. PLOS Computational Biology 17(12):e1009652. DOI: 10.1371/journal.pcbi.1009652

300. Justić, D., V. Kourafalou, G. Mariotti, S. He, R. Weisberg, Y. Androulidakis, C. Barker, A. Bracco, B. Dzwonkowski, C. Hu, H. Huang, G. Jacobs, M. Le Hénaff, Y. Liu, S. Morey, J. Nittrouer, E. Overton, C. B. Paris, B. J. Roberts, K. Rose, A. Valle-Levinson, and J. Wiggert. 2021. Transport Processes in the Gulf of Mexico Along the River–Estuary–Shelf–Ocean Continuum: a Review of Research from the Gulf of Mexico Research Initiative. Estuaries and Coasts 45:621–657. DOI: 10.1007/s12237-021-01005-1

301. Cantrell, D. L., M. L. Groner, T. Ben-Horin, J. Grant, and C. W. Revie. 2020. Modeling Pathogen Dispersal in Marine Fish and Shellfish. Trends in Parasitology 36(3):239–249. DOI: 10.1016/j.pt.2019.12.013

302. Foreman, M. G., M. Guo, K. A. Garver, D. Stucchi, P. Chandler, D. Wan, J. Morrison, and D. Tuele. 2015. Modelling Infectious Hematopoietic Necrosis Virus Dispersion from Marine Salmon Farms in the Discovery Islands, British Columbia, Canada. PLOS ONE 10(6):e0130951. DOI: 10.1371/journal.pone.0130951

303. van Sebille, E., S. M. Griffies, R. Abernathey, T. P. Adams, P. Berloff, A. Biastoch, B. Blanke, E. P. Chassignet, Y. Cheng, C. J. Cotter, E. Deleersnijder, K. Döös, H. F. Drake, S. Drijfhout, S. F. Gary, A. W. Heemink, J. Kjellsson, I. M. Koszalka, M. Lange, C. Lique, G. A. MacGilchrist, R. Marsh, C. G. Mayorga Adame, R. McAdam, F. Nencioli, C. B. Paris, M. D. Piggott, J. A. Polton, S. Rühs, S. H. A. M. Shah, M. D. Thomas, J. Wang, P. J. Wolfram, L. Zanna, and J. D. Zika. 2018. Lagrangian ocean analysis: Fundamentals and practices. Ocean Modelling 121:49–75. DOI: 10.1016/j.ocemod.2017.11.008

304. Hellweger, F. L., E. v. Sebille, and N. D. Fredrick. 2014. Biogeographic patterns in ocean microbes emerge in a neutral agent-based model. Science 345(6202):1346–1349. DOI: 10.1126/science.1254421

305. Marsh, R., V. O. Ivchenko, N. Skliris, S. Alderson, G. R. Bigg, G. Madec, A. T. Blaker, Y. Aksenov, B. Sinha, A. C. Coward, J. Le Sommer, N. Merino, and V. B. Zalesny. 2015. NEMO–ICB (v1.0): interactive icebergs in the NEMO ocean model globally configured at eddy-permitting resolution. Geoscientific Model Development 8(5):1547–1562. DOI: 10.5194/gmd-8-1547-2015

306. Romero, L., Y. Uchiyama, J. C. Ohlmann, J. C. McWilliams, and D. A. Siegel. 2013. Simulations of Nearshore Particle-Pair Dispersion in Southern California. Journal of Physical Oceanography 43(9):1862–1879. DOI: 10.1175/jpo-d-13-011.1

307. Lévy, M., P. J. S. Franks, and K. S. Smith. 2018. The role of submesoscale currents in structuring marine ecosystems. Nature Communications 9(1):4758. DOI: 10.1038/s41467-018-07059-3

308. Yatabe, T., S. J. More, F. Geoghegan, C. McManus, A. E. Hill, and B. Martínez-López. 2018. Can biosecurity and local network properties predict pathogen species richness in the salmonid industry? PLOS ONE 13(1):e0191680. DOI: 10.1371/journal.pone.0191680

309. Yatabe, T., B. Martínez-López, J. M. Díaz-Cao, F. Geoghegan, N. M. Ruane, T. Morrissey, C. McManus, A. E. Hill, and S. J. More. 2020. Data-Driven Network Modeling as a Framework to Evaluate the Transmission of Piscine Myocarditis Virus (PMCV) in the Irish Farmed Atlantic Salmon Population and the Impact of Different Mitigation Measures. Frontiers in Veterinary Science 7:385. DOI: 10.3389/fvets.2020.00385

310. Ben-Horin, T., C. A. Burge, D. Bushek, M. L. Groner, D. A. Proestou, L. I. Huey, G. Bidegain, and R. B. Carnegie. 2018. Intensive oyster aquaculture can reduce disease impacts on sympatric wild oysters. Aquaculture Environment Interactions 10:557–567.

311. Nash, C. 2010. The History of Aquaculture. John Wiley and Sons, Hoboken, New Jersey.

312. Smith, N. F., D. Lepofsky, G. Toniello, K. Holmes, L. Wilson, C. M. Neudorf, and C. Roberts. 2019. 3500 years of shellfish mariculture on the Northwest Coast of North America. PLOS ONE 14(2):e0211194. DOI: 10.1371/journal.pone.0211194

313. Hwang, E. K., N. Yotsukura, S. J. Pang, L. Su, and T. F. Shan. 2019. Seaweed breeding programs and progress in eastern Asian countries. Phycologia 58(5):484–495. DOI: 10.1080/00318884.2019.1639436

314. Riche, M. 2021. A special issue of the Journal of the World Aquaculture Society: Status of U.S. marine finfish species for domestic aquaculture expansion. Journal of the World Aquaculture Society 52(3):500. DOI: 10.1111/jwas.12813

315. Kim, C. H., and J.-A. Leong. 1999. Fish Viruses. Pages 558–568 *in* A. Granoff and R. G. Webster. Encyclopedia of Virology, 2nd edition. DOI: 10.1006/rwvi.1999.0100

316. Crane, M., and A. Hyatt. 2011. Viruses of Fish: An Overview of Significant Pathogens. Viruses 3(11):2025–2046. DOI: 10.3390/v3112025

317. Isshiki, T., T. Nagano, K. Kanehira, and S. Suzuki. 2004. Distribution of marine birnavirus in cultured marine fish species from Kagawa Prefecture, Japan. Journal of Fish Diseases 27(2):89–98. DOI: 10.1111/j.1365-2761.2003.00518.x

318. Lightner, D. V., R. M. Redman, C. R. Pantoja, K. F. Tang, B. L. Noble, P. Schofield, L. L. Mohney, L. M. Nunan, and S. A. Navarro. 2012. Historic emergence, impact and current status of shrimp pathogens in the Americas. Journal of Invertebrate Pathology 110(2):174–183. DOI: 10.1016/j.jip.2012.03.006

319. OIE (Office International des Epizooties). 2020. Aquatic Animal Health Code Chapter 9.1: Acute Hepatopancreatic Necrosis Disease. Office International des Epizooties, Paris. Available: www.oie.int/en/what-we-do/standards/codes-and-manuals/aquatic-code-online-access/?id=169&L=1&htmfile=chapitre_ahpnd.htm (December 2021).

320. Wang, M., Y. Chen, Z. Zhao, S. Weng, J. Yang, S. Liu, C. Liu, F. Yuan, B. Ai, H. Zhang, M. Zhang, L. Lu, K. Yuan, Z. Yu, B. Mo, X. Liu, C. Gai, Y. Li, R. Lu, Z. Zhong, L. Zheng, G. Feng, S. C. Li, and J. He. 2021. A convenient polyculture system that controls a shrimp viral disease with a high transmission rate. Communications Biology 4(1):1276. DOI: 10.1038/s42003-021-02800-z

321. Burge, C. A., A. Shore-Maggio, and N. D. Rivlin. 2017. Ecology of Emerging Infectious Diseases of Invertebrates. Pages 587–625 *in* A. E. Hajek, editor. Ecology of Invertebrate Diseases. John Wiley and Sons, Hoboken, New Jersey.

322. Burge, C. A., and C. S. Friedman. 2012. Quantifying Ostreid Herpesvirus (OsHV-1) Genome Copies and Expression during Transmission. Microbial Ecology 63(3):596–604. DOI: 10.1007/s00248-011-9937-1

323. Corbeil, S. 2020. Abalone Viral Ganglioneuritis. Pathogens 9(9). DOI: 10.3390/pathogens9090720

324. Toranzo, A. E., B. Magarinos, and J. L. Romalde. 2005. A review of the main bacterial fish diseases in mariculture systems. Aquaculture 246(1–4):37–61. DOI: 10.1016/j.aquaculture.2005.01.002

325. Paladini, G., M. Longshaw, A. Gustinelli, and A. P. Shinn. 2017. Parasitic Diseases in Aquaculture: Their Biology, Diagnosis and Control. Pages 37–107 *in* B. Austin and A. Newaj-Fyzul, editors. Diagnosis and Control of Diseases of Fish and Shellfish. John Wiley and Sons, Hoboken, New Jersey.

326. Villalba, A., K. S. Reece, M. Camino Ordás, S. M. Casas, and A. Figueras. 2004. Perkinsosis in molluscs: A review. Aquatic Living Resources 17:411–432. DOI: DOI: 10.1051/alr:2004050

327. Carnegie, R., and N. Cochennec-Laureau. 2004. Microcell parasites of oysters: Recent insights and future trends. Aquatic Living Resources 17:519–528. DOI: 10.1051/alr:2004055

328. Frans, I., C. W. Michiels, P. Bossier, K. A. Willems, B. Lievens, and H. Rediers. 2011. *Vibrio anguillarum* as a fish pathogen: virulence factors, diagnosis and prevention. Journal of Fish Diseases 34(9):643–661. DOI: 10.1111/j.1365-2761.2011.01279.x

329. de Souza Valente, C., and A. H. L. Wan. 2021. *Vibrio* and major commercially important vibriosis diseases in decapod crustaceans. Journal of Invertebrate Pathology 181:107527. DOI: 10.1016/j.jip.2020.107527

330. Roux, F. L., and M. Blokesch. 2018. Eco-evolutionary Dynamics Linked to Horizontal Gene Transfer in Vibrios. Annual Review of Microbiology 72(1):89–110. DOI: 10.1146/annurev-micro-090817-062148

331. Gauthier, D. T., and M. W. Rhodes. 2009. Mycobacteriosis in fishes: A review. The Veterinary Journal 180(1):33–47. DOI: 10.1016/j.tvjl.2008.05.012

332. Jacobs, J. M., C. B. Stine, A. M. Baya, and M. L. Kent. 2009. A review of mycobacteriosis in marine fish. Journal of Fish Diseases 32(2):119–130. DOI: 10.1111/j.1365-2761.2008.01016.x

333. Maekawa, S., T. Yoshida, P.-C. Wang, and S.-C. Chen. 2018. Current knowledge of nocardiosis in teleost fish. Journal of Fish Diseases 41(3):413–419. DOI: 10.1111/jfd.12782

334. Gauthier, D. T., R. J. Latour, D. M. Heisey, C. F. Bonzek, J. Gartland, E. J. Burge, and W. K. Vogelbein. 2008. Mycobacteriosis-Associated Mortality in Wild Striped Bass (*Morone saxatilis*) from Chespeake Bay, USA. Ecological Applications 18(7):1718–1727. DOI: 10.1890/07-2083.1

335. Jones, S. 2019. Characterization of *Piscirickettsia salmonis* and salmonid rickettsial septicaemia to inform pathogen transfer risk assessments in British Columbia. Research Document 2019/020. Fisheries and Oceans Canada, Ottawa, Ontario, Canada. Available: waves-vagues.dfo-mpo.gc.ca/library-bibliotheque/40852854.pdf (July 2023).

336. Rhodes, L. D., and C. Mimeault. 2019. Characterization of *Renibacterium salmoninarum* and bacterial kidney disease to inform pathogen transfer risk assessments in British Columbia. Research Document 2019/018. Fisheries and Oceans Canada, Ottawa, Ontario, Canada. Available: waves-vagues.dfo-mpo.gc.ca/library-bibliotheque/40852817.pdf (July 2023).

337. Colquhoun, D. J., and S. Duodu. 2011. *Francisella* infections in farmed and wild aquatic organisms. Veterinary Research 42(1):47. DOI: 10.1186/1297-9716-42-47

338. Munang'andu, H. M. 2018. Intracellular Bacterial Infections: A Challenge for Developing Cellular Mediated Immunity Vaccines for Farmed Fish. Microorganisms 6(2):33. DOI: 10.3390/microorganisms6020033

339. Elliott, D. G., G. D. Wiens, K. L. Hammell, and L. D. Rhodes. 2014. Vaccination against Bacterial Kidney Disease. Pages 255–272 *in* R. Gudding, A. Lillehaug, and Ø. Evensen, editors. Fish Vaccination. John Wiley and Sons, Hoboken, New Jersey.

340. Austin, B., and D. A. Austin. 2016. Bacterial Fish Pathogens: Disease of Farmed and Wild Fish. Springer International Publishing, New York. DOI: 10.1007/978-3-319-32674-0

341. Janda, J. M., and S. L. Abbott. 2010. The Genus *Aeromonas*: Taxonomy, Pathogenicity, and Infection. Clinical Microbiology Reviews 23(1):35–73. DOI: 10.1128/CMR.00039-09

342. Goncalves Pessoa, R. B., W. F. de Oliveira, D. S. C. Marques, M. T. Dos Santos Correia, E. de Carvalho, and L. Coelho. 2019. The genus *Aeromonas*: A general approach. Microbial Pathogenesis 130:81–94. DOI: 10.1016/j.micpath.2019.02.036

343. Cipriano, R. C., and B. Austin. 2011. Furunculosis and other aeromonad diseases. Pages 424–483 *in* P. T. K. Woo and D. B. Bruno, editors. Fish Diseases and Disorders, 2nd edition, volume 3: Viral, Bacterial, and Fungal Infections. Centre for Agriculture and Bioscience International, Wallingford, U.K.

344. Midtlyng, P. J. 2014. Vaccination against Furunculosis. Pages 185–199 *in* R. Gudding, A. Lillehaug, and Ø. Evensen, editors. Fish Vaccination. John Wiley and Sons, Hoboken, New Jersey.

345. Mishra, A., G.-H. Nam, J.-A. Gim, H.-E. Lee, A. Jo, and H.-S. Kim. 2018. Current Challenges of *Streptococcus* Infection and Effective Molecular, Cellular, and Environmental Control Methods in Aquaculture. Molecules and Cells 41(6):495–505. DOI: 10.14348/molcells.2018.2154

346. Ma, J., T. J. Bruce, E. M. Jones, and K. D. Cain. 2019. A Review of Fish Vaccine Development Strategies: Conventional Methods and Modern Biotechnological Approaches. Microorganisms 7(11):569.

347. Park, S. B., T. Aoki, and T. S. Jung. 2012. Pathogenesis of and strategies for preventing *Edwardsiella tarda* infection in fish. Veterinary Research 43(1):67. DOI: 10.1186/1297-9716-43-67

348. Buján, N., A. E. Toranzo, and B. Magariños. 2018. *Edwardsiella piscicida*: A significant bacterial pathogen of cultured fish. Diseases of Aquatic Organisms 131(1):59–71.

349. Leung, K. Y., Q. Wang, Z. Yang, and B. A. Siame. 2019. *Edwardsiella piscicida*: A versatile emerging pathogen of fish. Virulence 10(1):555–567. DOI: 10.1080/21505594.2019.1621648

350. Kumar, G., S. Menanteau-Ledouble, M. Saleh, and M. El-Matbouli. 2015. *Yersinia ruckeri*, the causative agent of enteric redmouth disease in fish. Veterinary Research 46(1):103. DOI: 10.1186/s13567-015-0238-4

351. Tobback, E., A. Decostere, K. Hermans, F. Haesebrouck, and K. Chiers. 2007. *Yersinia ruckeri* infections in salmonid fish. Journal of Fish Diseases 30(5):257–268. DOI: 10.1111/j.1365-2761.2007.00816.x

352. Loch, T. P., and M. Faisal. 2015. Emerging flavobacterial infections in fish: A review. Journal of Advanced Research 6(3):283–300. DOI: 10.1016/j.jare.2014.10.009

353. Avendano-Herrera, R., A. E. Toranzo, and B. Magarinos. 2006. Tenacibaculosis infection in marine fish caused by *Tenacibaculum maritimum*: a review. Diseases of Aquatic Organisms 71(3):255–266. DOI: 10.3354/dao071255

354. Barnes, M. E., and M. L. Brown. 2011. A Review of *Flavobacterium psychrophilum* Biology, Clinical Signs, and Bacterial Cold Water Disease Prevention and Treatment. The Open Fish Science Journal 4:40–48. Available: benthamopen.com/contents/pdf/TOFISHSJ/TOFISHSJ-4-40.pdf (July 2023).

355. Declercq, A. M., F. Haesebrouck, W. Van den Broeck, P. Bossier, and A. Decostere. 2013. Columnaris disease in fish: a review with emphasis on bacterium–host interactions. Veterinary Research 44(1):27. DOI: 10.1186/1297-9716-44-27

356. CAB International. 2021. Pseudomonas infections in fish datasheet. CAB International, Wallingford, U.K. Available: www.cabi.org/isc/datasheet/67006 (December 2021).

357. Balboa, S., H. W. Ferguson, and J. L. Romalde. 2007. Phenotypic, serological and genetic characterization of *Pseudomonas anguilliseptica* strains isolated from cod, *Gadus morhua* L., in northern Europe. Journal of Fish Diseases 30(11):657–664. DOI: 10.1111/j.1365-2761.2007.00849.x

358. Bower, S. M. 2014. *Haplosporidium nelsoni* (MSX) of Oysters. Fisheries and Oceans Canada (DFO), Ottowa, Canada. Available: www.dfo-mpo.gc.ca/science/aah-saa/diseases-maladies/hapneloy-eng.html (May 2023).

359. Crosson, L. M., N. S. Lottsfeldt, M. E. Weavil-Abueg, and C. S. Friedman. 2020. Abalone Withering Syndrome Disease Dynamics: Infectious Dose and Temporal Stability in Seawater. Journal of Aquatic Animal Health 32(2):83–92. DOI: 10.1002/aah.10102

360. Bower, S. M., S. E. McGladdery, and I. M. Price. 1994. Synopsis of infectious diseases and parasites of commercially exploited shellfish. Annual Review of Fish Diseases 4:1–199.

361. Travers, M.-A., K. Boettcher Miller, A. Roque, and C. S. Friedman. 2015. Bacterial diseases in marine bivalves. Journal of Invertebrate Pathology 131:11–31. DOI: 10.1016/j.jip.2015.07.010

362. Davis, C. V., and B. J. Barber. 1999. Growth and survival of selected lines of eastern oysters, *Crassostrea virginica* (Gmelin 1791) affected by juvenile oyster disease. Aquaculture 178(3):253–271. DOI: 10.1016/S0044-8486(99)00135-0

363. Conchas, R. F., J. Santamarina, A. Lama, M. A. Longa, and J. Montes. 2003. Evolution of bonamiosis in Galacia (NW Spain). Bulletin of the European Association of Fish Pathologists 23(6):265–272.

364. Buchmann, K. 2015. Impact and control of protozoan parasites in maricultured fishes. Parasitology 142(1):168–177. DOI: 10.1017/S003118201300005X

365. Urawa, S., N. Ueki, and E. Karlsbakk. 1998. A Review of *Ichthyobodo* Infection in Marine Fishes. Fish Pathology 33(4):311–320. DOI: 10.3147/jsfp.33.311

366. Erkinharju, T., R. A. Dalmo, M. Hansen, and T. Seternes. 2021. Cleaner fish in aquaculture: review on diseases and vaccination. Reviews in Aquaculture 13(1):189–237. DOI: 10.1111/raq.12470

367. Gnanagobal, H., T. Cao, A. Hossain, M. Dang, J. R. Hall, S. Kumar, D. Van Cuong, D. Boyce, and J. Santander. 2021. Lumpfish (*Cyclopterus lumpus*) Is Susceptible to *Renibacterium salmoninarum* Infection and Induces Cell-Mediated Immunity in the Chronic Stage. Frontiers in Immunology 12:733266. DOI: 10.3389/fimmu.2021.733266

368. Klochkova, T. A., S. Jung, and G. H. Kim. 2017. Host range and salinity tolerance of *Pythium porphyrae* may indicate its terrestrial origin. Journal of Applied Phycology 29(1):371–379.

369. Pang, T., J. Liu, Q. Liu, H. Li, and J. Li. 2015. Observations on pests and diseases affecting a eucheumatoid farm in China. Journal of Applied Phycology 27(5):1975–1984. DOI: 10.1007/s10811-014-0507-z

370. Kambey, C. S. B., P.-E. Lim, E. J. Cottier-Cook, I. Campbell, S.-W. Poong, and A. Kassim. 2021. Standard Operating Procedure of Eucheumatoid Cultivation Using Biosecurity-Based Approach. University of Malaya Press, Kuala Lumpur, Malaysia. Available: www.globalseaweed.org/wp-content/uploads/2021/11/Standard-Operating-Procedure_GSS-UM.pdf (July 2023).

371. Hurtado, A. Q., I. C. Neish, and A. T. Critchley. 2019. Phyconomy: the extensive cultivation of seaweeds, their sustainability and economic value, with particular reference to important lessons to be learned and transferred from the practice of eucheumatoid farming. Phycologia 58(5):472–483. DOI: 10.1080/00318884.2019.1625632

372. PFMC (Pacific Fishery Management Council). 2012. Pacific Coast Groundfish 5-Year Review of Essential Fish Habitat. Pacific Fishery Management Council, Portland, Oregon.

373. NMFS (National Marine Fisheries Service). 2022. Essential Fish Habitat Mapper. Available: www.fisheries.noaa.gov/resource/map/essential-fish-habitat-mapper (September 2022).

374. ESRI (Environmental Systems Research Institute). 2022. ArcGIS Pro 3.1.0. Environmental Systems Research Institute, Redlands, California.

Appendix A: Specific Diseases and Pathogens of Concern

The following tables list specific diseases and associated pathogens that are known concerns for marine aquaculture because they are commonly detected in aquaculture organisms and have an economic impact for affected species. They are organized by pathogen category (viruses, bacteria, parasites). It is important to remember that only FDA-approved drugs should be used in aquaculture.[1]

Viruses

Shrimp viruses

Shrimp viruses of concern originated with the practice of using wild shrimp stocks for seed production, which brought pathogens such as white spot syndrome virus (WSSV) and infectious hypodermal and hematopoietic necrosis virus (IHHNV) into the industry (*318*). Over the past three decades, sourcing has shifted away from wild collection to use of specific-pathogen-free (SPF) seed stocks of *Penaeus vannamei*, but these pathogens remain problematic (*318*). Among the 33 WOAH-listed diseases for aquatic organisms, seven of them are penaeid shrimp viral diseases (*319*).

Disease prevention and management: Since the late 1990s, the principal measure to reduce introduction of shrimp viral pathogens into farmed shrimp has been sourcing animals from domesticated SPF broodstock and post-larvae (*128*). Now widely applied in the United States, this approach has not been adopted globally, probably due to the technological demands of producing and maintaining SPF shrimp, which include pathogen diagnostic capabilities, surveillance competence, and biosecure facilities suitable for quarantine and isolation rearing. Furthermore, environmental management of source waters (treated to remove pathogens and vectors), farm fallowing and treatment between cohorts, appropriate stocking densities, and routine sanitary management are necessary to maintain healthy shrimp, especially against agents that are ubiquitous such as *Vibrio parahaemolyticus* (*128*). Although sophisticated technologies such as RNAi or management strategies such as polyculture (e.g., *320*) are currently being explored, the only reliable treatments for shrimp viral diseases are depopulation or harvest.

Mollusk viruses

There are two mollusk viruses of emerging concern at this time. Oyster herpesvirus-1 (OsHV-1) affects bivalves, primarily Pacific oyster (*Crassostrea gigas*), and has a global distribution (*321*). Virus transmission occurs between different bivalve species, and infections can progress to mortality within 15 days with up to 100% mortality (*321*). Although mortalities are associated with the spring and summer seasons, seawater temperature

---

[1] https://www.fda.gov/animal-veterinary/aquaculture/approved-aquaculture-drugs

optima for disease vary widely by geography, and it is likely that the OsHV-1 variants are different in their temperature responses (*322*). Because human transport of live bivalves is the primary mode of spread, limiting movement is a principal control measure (*322*).

Abalone viral ganglioneuritis is caused by the abalone herpesvirus, and has caused severe damage to abalone aquaculture in Taiwan and Australia since the first outbreak reports in 2003 (*323*). To date, no cases have been reported in the United States.

Finfish viruses

Finfish viruses may represent a source of emerging infectious diseases for marine offshore aquaculture, due to the high titer of viral particles normally present in seawater (up to 10^8/mL) and the relatively high mutation rates, especially for RNA viruses. Viral transmission may occur between wild and cultured species (in both directions), and there are many reservoirs for viruses that may be carriers but asymptomatic (*70*, *317*). Human actions can further contribute to transmission through movement of fish and fish products (*20*), transfer of ballast water, and habitat modifications that may increase transmission likelihood, such as closer proximity due to compression of natural feeding areas (*70*).

Table A-1. Viral diseases in aquaculture and their etiologic agents. Diseases or agents in bold are currently listed by WOAH (*315–317*).

Disease	Viral agent	Host(s)
Tetrahedral baculovirus	Baculovirus penaei (BP)	Crustaceans
White spot disease (WSD)	White spot syndrome virus (WSSV)	All decapods
Taura syndrome	Taura syndrome virus (TSV)	Crustaceans
Yellow head disease	Yellow head virus (YHV), gill-associated virus (GAV)	Crustaceans
Infectious hypodermal and hematopoietic necrosis	Infectious hypodermal and hematopoietic necrosis virus (IHHNV)	Crustaceans
Infectious myonecrosis (IMN)	Infectious myonecrosis virus (IMV)	Crustaceans
White tail disease (WTD)	***Macrobrachium rosenbergii* nodavirus (MrNV)**	Crustaceans
Decapod iridescent virus 1 infection	Decapod iridescent virus 1 (DIV1)	All decapods
Infectious pancreatic necrosis (IPN)	Infectious pancreatic necrosis virus (birnavirus)	Finfish
Infectious salmon anemia (ISA)	Infectious salmon anemia virus (orthomyxovirus)	Finfish
Salmon paramyxovirus	Chinook salmon paramyxovirus (paramyxovirus)	Finfish
Salmon reovirus	Chum salmon reovirus (reovirus)	Finfish
Infectious hematopoietic necrosis (IHN)	Infectious hematopoietic necrosis virus (reovirus)	Finfish
Viral hemorrhagic septicemia (VHS)	Viral hemorrhagic septicemia virus (rhabdovirus)	Finfish
Lymphocystis	Lymphocystic virus (iridovirus)	Finfish
Erythrocytic necrosis	Erythrocytic necrosis virus (iridovirus)	Finfish
Adenovirus	Atlantic cod adenovirus (adenovirus), dab adenovirus (adenovirus)	Finfish
Marine aquabirnavirus infection	Marine aquabirnavirus (birnavirus)	Finfish, Shellfish
Viral nervous necrosis (VNN)	Nervous necrosis virus (betanodavirus)	Finfish

Table A-1 (continued). Viral diseases in aquaculture and their etiologic agents.

Disease	Viral agent	Host(s)
Pancreas disease	Salmon alphavirus (alphavirus)	Finfish
Scale drop disease	Scale drop disease virus (megalocytivirus), *Lates calcarifer* herpes virus (herpesvirus), *Lates calcarifer* birnavirus (birnavirus)	Finfish
Megalocytivirus infection	Infectious spleen and kidney necrosis virus (megalocytivirus), red sea bream iridovirus (iridovirus), turbot reddish body iridovirus (iridovirus)	Finfish
Ostreid herpesvirus-1 (OsHV-1)	Ostreid herpesvirus-1	Bivalves
Abalone viral ganglioneuritis	Abalone herpesvirus	Mollusks

Bacteria

There are several bacterial diseases of specific concern for aquaculture. These diseases include aeromoniasis, edwardsiellosis, flavobacteriosis, mycobacteriosis/nocardiosis, pseudomoniasis, streptococcosus, vibriosis, anaerobic infections, and infections with intracellular bacteria. Table A-2 lists the bacteria mostly commonly associated with these diseases.

Table A-2. Common bacterial diseases in aquaculture and their etiologic agents. Diseases or agents in bold are listed by WOAH (*324*).

Disease	Commonly associated bacteria	Host(s)
Aeromoniasis	*Aeromonas caviae*, *A. hydrophila*, *A. jandaei*, *A. salmonicida*, *A. sobria*, *A. veronii*, motile *Aeromonas* species	Finfish
Edwardsiellosis/Yersiniosis	*Edwardsiella anguillarum*, *E. ictaluri*, *E. piscicida*, *E. tarda*, *Yersinia ruckeri*	Finfish
Flavobacteriosis/ Tenacibaculosis	*Chryseobacterium* spp., *Flavobacterium branchiophilum*, *F. columnare*, *F. psychrophilum*, *Tenacibaculum maritimum*	Finfish
Mycobacteriosis/ Nocardiosis	*Mycobacterium fortuitum*, *M. marinum*, Nocardia asteroides, *N. crassostreae*, *N. seriolae*	Finfish
Pseudomoniasis	*Pseudomonas anguillisptica*, *P. fluorescens*, *P. plecoglossicida*	Finfish
ROD (roseovarius oyster disease)	*Roseovarius crassostreae*	Mollusks
Streptococcus/ Lactococcosus/Gaffkemia	*Aerococcus viridans*, *Lactococcus garvieae*, *L. petauri*, *Streptococcus agalactiae*, *S. iniae*, *Streptococcus* spp. (shrimp)	Finfish, Crustaceans
Vibriosis	*Photobacterium damselae*, *Vibrio alginolyticus*, *V. (Listonella) anguillarum*, *V. harveyi*, *V. ordalii*, *V. parahaemolyticus*, *V. (Aliivibrio) salmonicida*, *V. spendidus*, *V. vulnificus*, *V. corallyticus*	Finfish, Crustaceans, Echinoderms
Necrotizing hepatopancreatitis (NHP)	*Hepatobacter penaei*	Crustaceans
Acute hepatopancreas necrosis disease (AHPND)	*Vibrio parahaemolyticus* strain $\mathrm{Vp}_{\mathrm{AHPND}}$	Crustaceans
Anaerobic infections	*Clostridium botulinum*, *Enterobacterium catenabacterium*	Finfish
Intracellular infections	*Chlamydia* spp., *Francisella noatunensis*, *Piscirickettsia salmonis*, *Renibacterium salmoninarum*	Finfish
Withering syndrome	*Xenohaliotis californiensis* (WS-RLO)	Mollusks

Parasites

Parasites can have complex life cycles involving one or more hosts. This can complicate control of infection, especially if intermediate hosts are naturally present in the culture area. However, parasites, such as many protists, that have no need of an intermediate host can be horizontally transmitted (*325*). In general, shellfish tend to have more problematic parasites than finfish, and oysters are particularly affected by a suite of related haplosporidian parasites including *Haplosporidium* spp, *Bonamia* spp, and *Mikrocytos* spp.

Table A-3. Common parasitic diseases in aquaculture and their etiologic agents. Diseases or agents in bold are listed by WOAH (*325–327*).

Disease	Associated parasite	Host
Bonamiosis	*Bonamia ostreae*, *B. exitiosa*	Mollusks
Parasitic white spot disease, marine ich	*Cryptocaryon irritans*	Finfish
Dermo, Perkinsosis	*Perkinsus marinus*, *P. olseni*, *P. chesapeaki*, *P. qugwadi*, *P. andrewsi*, *P. mediterraneus*	Mollusks
Monogenean flatworms	*Neobenedenia* spp.	Finfish
Digenetic trematode	*Galactosomum* spp., *Stephanostomum tenue*, *Paradeontacylix* spp.	Finfish
Ectocommensal ciliates	*Trichodina* spp., *Uronema* spp., *Epistylis* spp.	Finfish
MSX (multinucleated sphere unknown)	*Haplosporidium nelsoni*	Mollusks
Myxozoan infections	*Kudoa* spp., *Myxidium* spp., *Myxobolus* spp.	Finfish
QPX (quahog parasite unknown)	*Labyrinthomorpha* spp., *Thraustochytriales* spp.	Mollusks
Scallop apicomplexan	*Merocystis kathae*	Mollusks
SSO (seaside organism)	*Haplosporidium costale*	Mollusks
Ectoparasites	*Lepeophtheirus salmonis*, *Caligus rogercresseyi*, *Ichthyobodo* spp., *Trichodina* spp.	Finfish

Appendix B: Descriptions of Diseases of Concern for Marine Aquaculture

The following sections provide a brief description of diseases and current management tactics for vertebrates and invertebrates (first section) and seaweed/macroalgae (second section). These narratives are intended to provide general information and are not comprehensive or meant to be a thorough discussion of the listed diseases.

Descriptions of Diseases of Concern for Vertebrates and Invertebrates

Vibriosis

Vibriosis is a leading disease among finfish and shellfish under all temperature regimens; the bacteria causing vibriosis typically have global distribution and widespread environmental presence, and the species causing the economic impact are *Vibrio anguillarum, V. ordalii, V. salmonicida*, and *V. vulnificus* (*324*). Some agents, such as *V. anguillarum*, can infect over 50 freshwater and marine species in temperate and tropical temperature regimens, employing mechanisms to invade and colonize tissues (e.g., proteases, toxins, nutrient sequestration) that are not species-specific (*328*). All crustaceans (shrimp, crab, lobster) are susceptible to vibriosis at multiple life-history stages by several vibrio species, resulting in severe losses (*329*). Acute hepatopancreatic necrosis disease in shrimp is WOAH-listed, and is caused by strains of *V. parahaemolyticus* carrying specific virulence genes (*319*). In addition to causing disease in aquatic animals, certain non-cholera vibrios (*V. parahaemolyticus, V. vulnificus)* are zoonotic; monitoring for these species is well established by public health departments for commercial and recreationally harvested locations. The complex and plastic genomics of vibrios include horizontal gene transfer, which permits occupancy of a wide range of habitats and is possibly responsible for their ability to rapidly adapt from aquatic environments to organisms (*330*).

Disease prevention and management: The most common sign of vibriosis is septicemia. Because the ubiquitous environmental distribution of vibrios consistently poses a threat of infection for farmed organisms, good health management to minimize stresses that increase susceptibility is the best preventative. For finfish vibriosis caused by *V. anguillarum, V. salmonicida,* or *Photobacterium damselae piscicida*, vaccination is widely available, most frequently as bacterin preparations, and additional preventative measures such as probiotics, immunostimulatory molecules, and antimicrobial peptides are increasingly being used (*328*). For shellfish, proper water quality, density management, and avoidance of acute stressors (e.g., temperature shocks) may be augmented with probiotics, prebiotics, synbiotics, and biofloc technology (*329*). Although antibiotics have historically been the therapeutic choice for vibriosis in finfish and shellfish, there is an emphasis on ensuring their use is judicious, and preventative measures such as good husbandry and vaccination, where available, are important to minimize use. However, antibiotic application is sometimes unavoidable and necessary.

Mycobacteriosis/Nocardiosis

Mycobacteriosis/nocardiosis, or fish tuberculosis, has a global distribution and occurs in all temperature regimens (tropical through cold water) and at all salinities (freshwater, brackish, and marine). These diseases tend to be subacute or chronic in nature, characterized by granulomatous lesions (*331–333*). *Mycobacteria* and *Nocardia* species are the etiologic agents, with ten species most frequently isolated and reported as finfish and shellfish pathogens (*M. marinum*, *M. salmoniphilum*, *M. fortuitum*, *M. chelonae*, *M. abscessus*, *M. shottsii*, *M. pseudoshottsii*, *N. asteroides*, *N. seriolae*, *N. salmonicida*, and *N. crassostreae*; *331–333*). Mycobacteriosis in wild striped bass in Chesapeake Bay emerged in 1997 and has persisted since then, but the population-level impacts were only gradually appreciated due to the chronic nature of the disease and cryptic mortality (*334*). An additional concern for mycobacteria is the zoonotic potential, particularly for *M. marinum*, through direct contact with infected animals or water (not foodborne; *332*).

Disease prevention and management: These diseases cannot be resolved with antibiotics, and there are no commercial vaccines available. Competent examination of organisms for signs of disease is essential prior to stocking. Because the diagnostics require specialized media and are more slowly growing than most aquatic pathogens, a skilled laboratory is required.

Intracellular bacterial diseases

Diseases caused by intracellular bacteria not described below include: *Piscirickettsia salmonis* in salmonids; *Renibacterium salmoninarum* in salmonids; *Chlamydia* spp. in salmonids; *Francisella nuatunensis* in cod, salmonids, and cichlids; and necrotizing hepatopancreatitis (NHP) rickettsial-like bacterium in penaeid shrimp. These bacteria have global distributions and range from cold to tropical waters; none have zoonotic potential. *P. salmonis* and *R. salmoninarum* are chronic, severe endemic pathogens in salmon hatcheries, and can cause either acute epidemics or chronic persistent losses. Transmission of *R. salmoniarum* can be via horizontal or vertical mechanisms (*335*, *336*). *F. nuatunensis* has a broad spectrum of hosts, making it a serious threat for finfish aquaculture (*337*).

Disease prevention and management: The intracellular lifestyle of these bacteria poses challenges for delivery of therapeutics to an intracellular location, and for vaccine development for finfish hosts, due to sequestration from detection by humoral immunity such as antibody-based immunity (*338*, *339*). Furthermore, the intracellular location also results in a time-limited ability of antibiotics to reduce pathogen burden. Screening broodstock and eggs for pathogens and culling or segregating infected animals prior to stocking have been the most effective biosecurity measures.

Aeromoniasis

Aeromoniasis occurs primarily in finfish and frequently manifests as external ulcers. Disease can be found in all temperature regimens, affecting fish in fresh, brackish, and marine environments. Aeromonads occur in a variety of aquatic environments, and infections are typically opportunistic (*340*, *341*). Due to genomic heterogeneity, species identification is more reliable with genetic rather than phenotypic methods, focusing on gene loci such as housekeeping genes (e.g., *gyrB*, *rpoD*, *dnaJ*) rather than the 16S rRNA gene (*342*). *Aeromonas salmonicida* and its subspecies, the etiologic agents of furunculosis, are good examples of variation in host tropism, where the typical, pigment-producing subspecies affect salmonids while the atypical, nonpigmented subspecies affect other marine fish such as sablefish and halibut (*343*, *344*). Aeromonads have low zoonotic potential, involving primarily *A. caviae*, *A. veronii*, *A. dhakensis*, and *A. hydrophila* (*342*).

Disease prevention and management: Like vibrios, the constant presence of aeromonads in the aquatic environment compels a strong health management plan. For *A. salmonicida*, vaccines are an important tool (*344*). Polyvalent bacterins can provide better protection, and current formulations commonly combine antigens for both vibrio and *A. salmonicida* (*324*).

Streptococcosis/Lactococcosis

Streptococcosis/lactococcosis manifests with erratic swimming (e.g., spinning or spiraling), exophthalmia, and cutaneous hemorrhages as the most frequent signs. The diseases occur in freshwater and marine finfish. Disease due to the *Streptococcus* species is found at all temperature regimens, while those due to *Lactococcus garvieae* and *Vagococcus salmoninarum* present in temperate and tropical regimens (*324*, *345*). The etiologic agents are ubiquitous and globally present, and their infection is considered opportunistic (*340*, *345*). In addition to causing disease in at least 30 finfish species, the etiologic agents can infect shrimp, amphibians, birds, and mammals, posing a zoonotic risk to humans (*345*). Gaffkemia in lobster and crayfish is a high mortality disease caused by *Aerococcus viridans*, with global distribution due to human transport of infected organisms (*149*). Severe mortality in penaeid shrimp due to *Streptococcus* spp. is an emerging problem in shrimp aquaculture (*318*).

Disease prevention and management: The opportunistic character of streptococcosis indicates that high-quality culture conditions are paramount in reducing disease risks (e.g., appropriate rearing densities, good quality feed and water conditions). Several probiotic formulations have shown protection against lactococcosis (*340*) but, in spite of considerable vaccination efforts (focusing on bacterins), the only reliable and commercially available fish vaccines are for *S. agalactiae* and *S. iniae* (*346*). Antimicrobials have had limited effectiveness against *Streptococcus* and *Lactococcus* species due to the development of resistance (*324*, *340*, *345*). Oxytetracycline has been useful in treating gaffkemia, and is approved by FDA for this use (*149*). Because *A. viridans* can only infect through ruptures in the shell, reducing trauma during handling and holding is important for reducing infection (*149*).

Edwardsiellosis/Yersiniosis

Edwardsiellosis/yersiniosis are diseases caused by *Edwardsiella* and *Yersinia* species, which occur across all temperature regimens with global distribution (*347*). While *Edwardsiella ictaluri* infections are limited to freshwater hosts (*340*), the other *Edwardsiella* species, including *E. piscicida* (recently reclassified from *E. tarda*), do infect freshwater and marine finfish (*347–349*). Because Edwardsiellosis manifests a variety of clinical signs that can be similar to aeromoniasis, vibriosis, and pseudomoniasis, accurate diagnosis requires laboratory identification (*347*). Yersiniosis, or enteric red mouth, is a disease of primarily salmonids caused by *Y. ruckeri*, which has been isolated from reptiles, birds, and mammals that could serve as reservoirs or transmission vectors (*350*, *351*). As enteric pathogens, *Edwardsiella* and *Yersinia* species can be transmitted through direct contact, oral–fecal routes, or even food chains (*349*, *351*). Additionally, *E. piscicida* has zoonotic potential (*347*).

Disease prevention and management: *Edwardsiella* and *Yersinia* species possess a wide array of virulence factors, including immune evasion mechanisms to support their pathogenic lifestyle (*347*, *348*), and screening for infection is important if there is a history of infection at a facility or in the stock. Commercial vaccines are available for *E. ictaluri* and *Y. ruckeri* (*346*), but vaccines against other species that are suitable for aquaculture are still under development (*347*). Probiotics based on *Bacillus* species have shown protective value against *Y. ruckeri* (*350*, *351*). In general, *Edwardsiella* species and *Y. ruckeri* continue to be responsive to antibiotics such as oxytetracycline, although some resistant isolates have been reported (*348*, *350*, *351*).

Flavobacteriosis/Tenacibaculosis

Flavobacteriosis primarily afflicts freshwater fish, including the early life stages of anadromous fish such as salmonids (*352*), while tenacibaculosis occurs in a range of marine fish with a degree of greater susceptibility in younger fish (*353*). The distribution of these diseases is global, occurring in temperate and tropical waters (*352–354*). The entry points for the bacteria that cause these diseases are surfaces such as skin, gills, and mucous membranes, with subsequent systemic distribution during infection progression (*353–355*). None of the agents of flavobacteriosis or tenacibaculosis in fish poses a known zoonotic threat.

Disease prevention and management: Due to the entry route, treatment with surface disinfectants (e.g., formalin) and manipulation of water salinity have been used to control infection (*353*, *355*). There are commercially available vaccines against *Flavobacterium columnare* and *Tenacibaculum maritimum* (*346*), and bacterins have demonstrated protective action (*355*). Both antimicrobials and chemicals such as herbicides (e.g., Diquat), Chloramine-T, copper sulfate, and hydrogen peroxide have been employed as immersion therapeutic agents (*354*, *355*). *T. maritimum* displays susceptibility to a broad array of antimicrobials (*353*). However, rapid development of resistance to oral antimicrobials, as well as natural resistance, limits use for some pathogens such as *F. psychrophilum* (*354*).

Pseudomoniasis

Pseudomoniasis is not as widely prevalent as the previously mentioned diseases, with a principal distribution in Asia and subsequently in Europe. The principal etiologic agents are *Pseudomonas anguilliseptica* and *P. fluorescens,* affecting both freshwater and marine fish (*324*). Pseudomonads are widely present in the environment, and infections are considered opportunistic or secondary (*356*). The species affecting fish have no known zoonotic potential.

Disease prevention and management: As with all opportunistic diseases, stress reduction and good-quality environments reduce the risk of pseudomoniasis. Although there are no commercial vaccines available, the phenotypic and serological homogeneity of the most problematic agent, *P. anguilliseptica*, suggests potential cross-protection among isolates from different host species (*357*).

Multinucleated sphere unknown (MSX)

Multinucleated sphere unknown, or MSX, is a condition in oysters caused by the spore-forming protozoan *Haplosporidium nelsoni*. MSX can cause epizootic mortality among all life-history stages (spat through adults), with seasonal high prevalence in summer and fall; temperature and salinity are potent regulators of infection. Entry appears to be through gill and mantle tissue, with rapid dissemination throughout the animal. The disease can rapidly transmit over large distances, and is not correlated with oyster density, potentially implicating an intermediate host or vector. Geographic distribution is along the eastern coast of the United States and Canada, but it has not yet been confirmed in the Gulf of Mexico (*358*).

Disease prevention and management: Because the complete life cycle of *H. nelsoni* is unknown, preventing or controlling MSX is difficult. Nursery infections can be prevented by filtration (1 µm) and ultraviolet irradiation of influent water. Current prevention strategies include not introducing infected oysters; use of MSX-resistant oysters is a good preventative measure. Low salinity induces expulsion of the parasite, which can be helpful in managing an infected population. Finally, harvesting oysters at 18–24 months can help to avoid the disease risk period (*358*).

Withering syndrome (WS-RLO)

Withering syndrome (WS-RLO), caused by the Rickettsiales-like organism Candidatus *Xenohaliotis californiensis*, is the primary disease of concern for cultured and wild vetigastropods, specifically abalone. It exists along the California coast of the United States and the west coast of Mexico. Higher temperatures have been associated with prominence and lethality of WS-RLO, specifically temperatures at, or in excess of, 17°C. Transmission of this disease is via the fecal–oral route, which is compounded by the gregarious nature of wild abalone and, potentially, by nearby high-density culture settings (*42*, *142*). While red abalone (*Haliotis rufescens*) are the only species in the genus with a commercial aquaculture presence in California, WS-RLO does impact all members of the genus, including all wild species found in California; the endangered black and white abalone is highly susceptible to WS-RLO. Due to declines in the wild population, there are concerns about potential WS-RLO transmission between wild and cultured abalone populations (*42*, *359*).

Disease prevention and management: Some success has been achieved via the use of a 500-ppm oxytetracycline bath for infected abalone with no signs of adverse behavior, survival, or body condition. Some slowing in the growth rate was observed, but overall survival improved substantially. Tissue concentrations of oxytetracycline remained above 100 ppm for 125 days. While this method merits consideration for the introduction of abalone populations to new environments, especially ones in which WS-RLO is not present (*272*), this application of oxytetracycline is not an approved use. Additionally, as the disease exists in wild populations, spacing considerations should be a part of any farm planning near a wild population, to prevent spread.

Dermo disease

The endoparasite *Perkinsus marinus*, and related *Perkinsus* species, are protists that are widely dispersed on the U.S. East Coast, the Gulf of Mexico, and the Caribbean as far south as Brazil. *P. marinus* causes Dermo disease in several oysters (*Crassostrea* spp.). It proliferates in higher temperatures, but can survive lower temperatures in the environment, proliferating further during warmer months. Oysters are susceptible to infection in natural and laboratory settings, with some variance between species. *C. virginica*, the most important commercial species, is quite susceptible to infection, and experiences high mortality during outbreaks. Ectoparasite snails, such as *Boonea impressa*, were also found to transmit the parasite between *C. virginica* specimens in a laboratory setting (*360*). Although hard clams do not develop disease from infection, they can serve as vectors for oysters. Introducing infected clams to areas with susceptible cultured or wild oyster populations, or other susceptible bivalve mollusks, can introduce *Perkinsus* strains and cause significant mortality events (*135*).

Disease prevention and management: Research indicates the presence of resistance in eastern oyster populations to *Perkinsus marinus*, corresponding with a cohort's ancestral exposure to *P. marinus*. Resistant cohorts to the disease can be an effective means of preventing disease. Also, the disease is often transmitted when uninfected oysters ingest the parasite released by the disintegration of dead infected oysters.[1] Therefore, monitoring and expedient removal of dead animals is important for stopping the spread of disease. Furthermore, as with other disease concerns, effective control measures include methods for ensuring that infected animals are not moved into areas with uninfected animals. Dermo prevention and disease management also includes adequate spacing between individuals and groups (especially cohorts) and the selection of species to include in a farm (if multi-species) given that multiple species can serve as hosts for Dermo. Additionally, Dermo is less tolerant of low salinity, and delaying exposure by keeping animals in low/lower salinity (less than 9 ppt[2]) environments, in conjunction with cooler waters, is used to control the disease. Finally, harvesting oysters at 18–24 months can help to avoid the disease risk period.

---

[1] https://www.oyster.umd.edu/dermo-disease-testing

[2] https://www.dfo-mpo.gc.ca/science/aah-saa/diseases-maladies/pmdoy-eng.html

Seaside organism disease (SSO)

Seaside organism disease, or SSO, is due to the protozoan *Haplosporidium costale*, and is distributed in oysters on both the eastern and western coasts of the United States, including Alaska. Mortality tends to occur in late spring and early summer, due to infections that occurred the prior fall. Sporulation causes severe tissue damage, and is the likely proximal cause of mortality. Few or no clinical signs makes it difficult to detect before mortality.[3]

Disease prevention and management: Nursery infections can be prevented by filtration (to 1 µm) and ultraviolet irradiation of influent water. There are no effective control measures for existing infections, although transfer to low salinity can slow disease progression. Harvesting oysters at 18–24 months can help to avoid the disease risk period.[74]

Quahog parasite unknown (QPX)

Quahog parasite unknown, or QPX, is a significant pathogen of hard clams. QPX is a protist associated with mortality events of up to 95%, from eastern Canada to Virginia. QPX has not been detected in seed clams, suggesting that clams become infected in the planting areas. The most significant environmental factor attributed to QPX is high salinity (>30 ppt); however, mortalities are commonly associated with additional stressors, namely low temperatures in northern states and very dense culture conditions (*135*).

Disease prevention and management: Stress on cultured animals, high stocking densities, and poor plot husbandry are considered factors that exacerbate mortalities associated with QPX. Some evidence suggests that extensive mortality can be prevented by prompt removal of infected individuals. Additionally, reduction of stocking density in plots was reported as an effective means of reducing mortalities to negligible levels (*360*).

Roseovarius organism disease (ROD)

Roseovarius organism disease (ROD), formerly known as juvenile oyster disease (JOD), is a mortality syndrome of juvenile eastern oyster (*Crassostrea virginica*) caused by a marine alphaproteobacterium, *Roseovarius crassostreae*. It has been observed only in hatchery juveniles, with high mortalities (40–90%) within a week of the appearance of signs, at water temperatures >20°C (360, 361).

Disease prevention and management: The current management strategy is to shift rearing time to avoid higher water temperatures during periods of vulnerability (15–25-mm shell size). Because infections appear to be linked more strongly to locations than to oyster stocks, maintaining juveniles on filtered (≤25 µm) water is also effective (*360*). For strategic management, selection for ROD-resistant stocks has been demonstrated (*362*).

---

[3] https://www.dfo-mpo.gc.ca/science/aah-saa/diseases-maladies/hcoy-eng.html

Bonamiosis

Bonamiosis is caused by the protozoans *Bonamia ostreae* and *B. exitiosa*, which infects oysters at multiple life-history stages, including larvae. Transmission can occur directly between host oysters, and the parasite occurs on both the eastern (Maine) and western (California and Washington) coasts of the United States.[4] Although disease and mortality are reported only in *Ostrea edulis*, the parasite is detectable in *Crassostrea gigas*, which may be a reservoir. Temperature and salinity have a complex influence on disease, but infection is often fatal.

Disease prevention and management: Use of hatchery seed, instead of naturally settled seed, can reduce introduction into stock (*363*). Selective breeding for resistant stocks can further reduce possibility of infection (*152*). Suspension culture, lower stocking densities, and co-culture with *C. gigas* can also help control mortalities. There are no known chemotherapies available.

Scallop apicomplexan

Merocystis kathae is an apicomplexan parasite that causes scallop apicomplexan disease, characterized by shrunken adductor muscle in scallops due to invasion and rupture of muscle cells. The parasite is readily transmissible between sympatric whelks and scallops, resulting in epizootic mortalities among the latter, and positing a two-molluskan host life cycle (*137*).

Disease prevention and management: Because the parasite requires both whelks and scallops to complete its life cycle, segregation of the hosts is currently the only way to avoid infection. Because whelks are not harmed by the parasite, maintaining scallops on a separate or filtered water system is the only effective prevention tactic.

Ectoparasitism of finfish (sea lice, ichthyobodiasis/costiasis, cryptobiosis)

External parasites are a leading cause of economic losses for salmon farming, and have been a problem since the initiation of salmon aquaculture (*95*, *364*). Because ectoparasites feed directly on tissues, the fish experience physiological stress, osmoregulatory imbalance, and skin lesions that allow secondary infections (*364*, *365*). Due to the severity of impact, many countries with substantial salmonid aquaculture sectors have burden thresholds for some ectoparasites, such as sea lice, that require an action, such as reporting to regulatory authorities and parasite management (see *95*). Some parasites, such as marine *Ichthyobodo* species, affect hosts ranging from benthic (halibut, flounder) and epibenthic (rockfish, cod, sea bass, haddock) to pelagic (salmon), with distributions from open ocean to nearshore waters (*365*), allowing parasite transfers across host species.

---

[4] https://www.gov.scot/binaries/content/documents/govscot/publications/factsheet/2019/11/marine-scotland-topic-sheets-aquaculture/documents/bonamiasis-updated-october-2016/bonamiasis-updated-october-2016/govscot%3Adocument/bonamiasis.pdf

Disease prevention and management: Although breeding resistant fish stocks has promise for preventing infestations, the work is slow and pathogen-specific (*364*). Application of immunostimulants in feed and vaccination has had limited success (*364*), although a commercial vaccine against sea lice is now available. Most ectoparasite management has involved use of chemicals such as orally administered compounds (emamectin benzoate, benzoyl ureas), and bath treatments (organophosphates, pyrethroids, hydrogen peroxide, formalin; *95*, *364*, *365*). Physical and mechanical methods to prevent infestation include use of skirts on cages, snorkels, submerged feedings or cages, and light traps (*95*). Once an infestation occurs, management tactics include parasite removal with rapid thermal exposure, freshwater treatment, and physical removal with water jets (*95*). Several biological control methods, such as cleaner fish and biological pesticides (e.g., louse-specific viruses), are also proposed, but have not yet enjoyed widespread application (*95*), as those approaches have their own disease issues such as pathogens associated with cleaner fish (*366*, *367*).

Descriptions of Diseases of Concern for Seaweed/Macroalgae

Oomycete pathogens and red rot disease

Oomycete pathogens are among the most detrimental and important when discussing the culture of macroalgaes, namely *Pyropia*. *Pythium porphyrae* and *Olpidiopsis porphyrae* are the best-studied and most prominent pathogens, both associated with red rot disease in *Pyropia* and other cultivated edible seaweed species. The presence of the pathogen is usually temperature- and salinity-dependent (*368*). The U.S. West Coast is home to wild *Pyropia* species, as well as additional macroalgae with susceptibility to *Pythium porphyrae* (*163*, *164*). There is still little information on oomycete pathogens to suggest concrete solutions for mitigation and prevention (*162*).

Disease prevention and management: To prevent disease in general, *Pyropia* is washed in acid solutions to control the spread of pathogens such as *Pythium porphyae*. Research from Japan suggests that washing the thalli of algae in a pH 2.0 acid bath for 5 minutes did show suppression of red rot disease (*162*). Research also suggests that siting farms near freshwater sources may contribute to inoculation by *Olpidiopsis*, suggesting a potential terrestrial origin of this pathogen (*368*). Therefore, salinity consideration while locating farms may contribute to a disease prevention framework.

Ice-ice disease

Ice-ice disease impacts eucheumatoid seaweeds (*Kappaphycus* and *Eucheuma* spp.). Disease signs are whitening and decay of the thallus, with eventual death of the organism. The etiologic agents are unknown, with most attention on prokaryotes (bacteria), but the role of eukaryotes and viruses is understudied (*174*). Environmental factors have been mostly closely identified with appearance of the disease, including increases in water temperatures, salinity variations associated with low tides or freshwater runoff, and low water currents (*175*, *369*).

Disease prevention and management: Measures such as using propagules or seedlings from known, low-disease sources; cleaning surface holobionts during initial inoculation and during grow-out; cleaning farm equipment and transport boats; and monitoring and removing whitened and decaying seaweeds are effective in reducing disease incidence (*370*). High reliance on vegetative propagation of non-indigenous strains may contribute to increased susceptibility to disease, so use of local strains and seedlings from sexual reproduction can improve disease resistance (*371*). Because environmental conditions are strongly implicated in initiating disease, farm siting is particularly important for providing stable temperatures between 25–30°C, reducing variations in salinity, and water current flow >0.1 m/s to reduce surface holobiont and biofilm accumulation on thalli (*174*, *175*, *369*, *370*).

Appendix C: Method of Analysis of Overlap with Study Areas and AOA Options

To characterize the potential for co-occurrence of aquaculture and wild species, geographic coordinate data for AOA study areas were compared to catch distribution data, EFH data and reports, high use area (HUA) reporting, and Endangered Species Act (ESA) listed species habitat (*48)*. NOAA resources used for this analysis were the Aquaculture Atlases (*2*, *3*), Gulf of Mexico Fishery Management Council habitat requirements (*49*), essential fish habitat data (*372*, *373*), fishery management plans (*52*, *53*), and the NOAA Fisheries Catch Distribution Mapping tool (*48).* We employed the NOAA-NMFS EFH mapping tool and NOAA DisMap catch distribution mapping tool that allowed us to compare study area coordinates to the various EFH data layers (*373*) and the locations where species were caught in fishery-independent surveys (*50*). When available, we evaluated fishery management plans (FMPs) that included these species and range data or maps to determine whether there was overlap with the study areas (*372*). As a precaution, the authors used conservative estimates when employing this method to estimate overlap. Because an EFH data layer often contained multiple species, we reviewed each species listed in a layer for a more detailed report on species overlap, thereby building on information already in the Aquaculture Atlases.

Essential Fish Habitat Species Analysis

In addition to summarizing the general determinations of overlap with EFH, we individually reviewed the overlap between species within study areas using the NOAA EFH mapping tool in conjunction with the larger study area coordinates for the Aquaculture Atlases. We entered coordinates of each study area and AOA option into the EFH mapping tool and recorded any species' EFH that registered in those coordinates to determine overlap. EFH connected to the selected data points also linked to the resources that contain the EFH data; these resources are used as the ultimate citations for the data. The resources linked in the EFH mapping tool included EFH reports and FMPs. Additionally, some species were mentioned in the Atlases but did not appear in the EFH mapping tool. For analysis of overlap, we searched NOAA Fisheries databases for available shape files and coordinate data of EFH, fisheries, protected resources, and other relevant species distribution data that were compatible with GIS mapping software. Using ArcGIS Pro software, we merged these layers to best view overlap between the larger study areas and data describing wild species distribution. An internet search and visual comparisons of available distribution maps were used to determine potential overlap, although these estimates have less concrete spatial data. A summary of the overlap of the AOA study areas with EFH, HUA, and ESA species is included in Appendices C1 and C2.

Survey Catch-per-Unit-Effort Distribution Mapping Analysis

As a complement to analysis of EFH-, HUA-, and ESA-listed species, the authors used point observation data of where species were caught in fishery-independent surveys conducted in the Gulf of Mexico (operated by SEAMAP) and the U.S. West Coast available in DisMAP to determine specific individual species overlap with the study areas' high–high clusters. High–high clusters are zones within the broader AOA study areas that comprise multiple AOA options. Per discussion with other members of the AOA project teams, it was decided that the high–high clusters were an appropriate level of analysis. Wild species were selected for analysis if: a) they were conspecific with candidate species for marine aquaculture, b) they were in the same genus, c) they were significant for commercial or recreational fishing, or d) they are protected/endangered. Using ArcGIS Pro (*374*), DisMAP species coordinate data were plotted alongside the Aquaculture Atlases' high–high cluster polygon shape files (*2*, *3*) to evaluate overlap.

The survey data were downloaded for relevant species from the NOAA DisMAP tool (*48*) from the "Single species analysis" module. The data contain coordinates (lat/long), the year, and measurements of the biomass of the species documented at that location, calculated as weight-catch-per-unit-effort (wtCPUE, in kg/ha). Data were cleaned to remove any points in which the wtCPUE was zero, indicating the species was not observed at that location. Individual coordinates with a wtCPUE above zero indicate locations where a species was documented. Shape files of AOA study areas were received from the NOAA authors of the Aquaculture Opportunity Area atlases for the Gulf of Mexico and the Southern California Bight. The files were uploaded as polygons into the ArcGIS pro mapping tool. Each area included three different shape files, one for the overall study area (largest), one for high–high cluster areas (second largest), and one for AOA options (smallest).

Once uploaded, the data were then analyzed to count the number of points of overlap that a species had with the high–high cluster areas, as well as to document any points within clusters that contained AOA options. To do this analysis, the authors used the "Summarize Within" feature in ArcGIS. This tool counted the number of points in each cluster, the average biomass across the years, and the latest year in which a point occurred in each cluster. Data from the "Summarize Within" analysis were then used to characterize individual species overlap. Any overlap with high–high clusters that contained AOA options are documented and summarized in Appendices D1 and D2.

Finally, after seeing many points that occurred near, but not inside the high–high clusters, additional analysis was performed to indicate if a species had occurred within 10 km of a cluster, but outside of the "Summarizie Within" analysis (Appendices D1 and D2). These observations were made for all species, and will be the subject of further analysis in an ongoing manuscript to estimate species abundance around AOA options.

Appendix C1: Gulf of Mexico Protected and Commercially Important Species Having Spatial Overlap with AOA Study Areas

Species	Overlap with AOA Study Area?	Classification of Overlap	Source of Data
Smalltooth sawfish (*Pristis pectinata*)	Yes	Endangered Species Act High Use	NOAA Fisheries (https://www.fisheries.noaa.gov/species/smalltooth-sawfish)
Giant manta ray (*Mobula birostris*)	Yes	Endangered Species Act High Use	Aquaculture Opportunity Area Atlas
Gulf sturgeon (*Acipenser oxyrhynchus desotoi*)	No		NOAA Fisheries (https://www.fisheries.noaa.gov/resource/map/gulf-sturgeon-critical-habitat-map-and-gis-data)
Nassau grouper (*Epinephelus striatu*)	Yes	Endangered Species Act listed and occurs in region	Aquaculture Opportunity Area Atlas
Black grouper (*Mycteroperca bonaci*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
Scalloped hammerhead shark (*Sphyrna lewini*)	Yes	Essential Fish Habitat	Atlantic Highly Migratory Species Essential Fish Habitat (https://www.fisheries.noaa.gov/atlantic-highly-migratory-species/atlantic-hms-fishery-management-plans-and-amendments)
Brown rock shrimp (*Sicyonia brevirostris*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
Pink Shrimp (*Pandalus borealis*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
Spiny lobster (*Panulirus spp.*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
White shrimp (*Litopenaeus setiferus*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
Atlantic sharpnose shark (*Rhizoprionodon terraenovae*)	Yes	Essential Fish Habitat	Atlantic Highly Migratory Species Fishery Management Plan: Essential Fish Habitat (https://www.fisheries.noaa.gov/atlantic-highly-migratory-species/atlantic-hms-fishery-management-plans-and-amendments)

Species	Overlap with AOA Study Area?	Classification of Overlap	Source of Data
Atlantic shortfin mako shark (*Isurus oxyrinchus*)	Yes	Essential Fish Habitat	Atlantic Highly Migratory Species Fishery Management Plan: Essential Fish Habitat (https://www.fisheries.noaa.gov/atlantic-highly-migratory-species/atlantic-hms-fishery-management-plans-and-amendments)
Yellowfin tuna (*Thunnus albacares*)	Yes	Essential Fish Habitat	Atlantic Highly Migratory Species Fishery Management Plan: Essential Fish Habitat (https://www.fisheries.noaa.gov/atlantic-highly-migratory-species/atlantic-hms-fishery-management-plans-and-amendments)
Cobia (*Rachycentron canadum*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
Golden tilefish (*Lopholatilus chamaeleonticeps*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
Gray triggerfish (*Balistes capriscus*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
Greater amberjack (*Seriola dumerili*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
King mackerel (*Scomberomorus cavalla*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
Red drum (*Sciaenops ocellatus*)	No		Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
Spanish mackerel (*Scomberomorus maculatus*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
Vermillion snapper (*Rhomboplites aurorubens*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)
Bluefin tuna (*Thunnus thynnus*)	Yes	Essential Fish Habitat	Atlantic Highly Migratory Species Fishery Management Plan: Essential Fish Habitat (https://www.fisheries.noaa.gov/atlantic-highly-migratory-species/atlantic-hms-fishery-management-plans-and-amendments)
Red grouper (*Epinephelus morio*)	Yes	Essential Fish Habitat	Gulf Essential Fish Habitat Report (https://gulfcouncil.org/fishery-management-2/implemented-plans/essential-fish-habitat/)

Appendix C2: Southern California Bight Protected and Commercially Important Species Having Spatial Overlap with AOA Study Areas

Species	Overlap with AOA Study Area?	Classification of Overlap	Source of Data
Green abalone (*Haliotis fulgens*)	Unsure	Other	
Black abalone (*Haliotis cracherodii*)	No	Endangered Species Act listed and occurs in region. Distribution data do not appear to show overlap with study areas.	Endangered Species Act Critical Habitat Map (https://apps.wildlife.ca.gov/bios6/?al=ds2666)
White abalone (*Haliotis sorenseni*)	Unsure	Endangered Species Act listed and occurs in region	Aquaculture Opportunity Area Atlas
Gulf grouper (*Mycteroperca jordani*)	Unsure	Endangered Species Act listed and occurs in region	Aquaculture Opportunity Area Atlas
Oceanic whitetip shark (*Carcharhinus longimanus*)	Unsure	Endangered Species Act listed and occurs in region	Aquaculture Opportunity Area Atlas
Scalloped hammerhead shark (*Sphyrna lewini*)	Yes	Endangered Species Act listed and occurs in region	Aquaculture Opportunity Area Atlas
Steelhead trout[a] (*Oncorhynchus mykiss irideus*)	Unsure	Endangered Species Act listed and occurs in region	Aquaculture Opportunity Area Atlas
California market squid (*Doryteuthis opalescens*)	Yes	Essential Fish Habitat	Coastal Pelagic Species Fishery Management Plan (https://www.pcouncil.org/fishery-management-plan-and-amendments/)
Krill (*Thysanoessa spinifera*)	Yes	Essential Fish Habitat	Coastal Pelagic Species Fishery Management Plan (https://www.pcouncil.org/fishery-management-plan-and-amendments/)
Krill (*Euphausia pacifica*)	Yes	Essential Fish Habitat	Coastal Pelagic Species Fishery Management Plan (https://www.pcouncil.org/fishery-management-plan-and-amendments/)
Arrowtooth flounder (*Atheresthes stomias*)	Yes	Essential Fish Habitat	Pacific Coast Groundfish Fishery Management Plan (https://www.pcouncil.org/managed_fishery/groundfish/)
Bocaccio (*Sebastes paucispinis*)	Yes	Essential Fish Habitat	Pacific Coast Groundfish Fishery Management Plan (https://www.pcouncil.org/managed_fishery/groundfish/)
Canary rockfish (*Sebastes pinniger*)	Yes	Essential Fish Habitat	Pacific Coast Groundfish Fishery Management Plan (https://www.pcouncil.org/managed_fishery/groundfish/)

[a] Southern California DPS.

Species	Overlap with AOA Study Area?	Classification of Overlap	Source of Data
Dover sole (*Microstomus pacificus*)	Yes	Essential Fish Habitat	Pacific Coast Groundfish Fishery Management Plan (https://www.pcouncil.org/managed_fishery/groundfish/)
English sole (*Parophrys vetulus*)	Yes	Essential Fish Habitat	Pacific Coast Groundfish Fishery Management Plan (https://www.pcouncil.org/managed_fishery/groundfish/)
Lingcod (*Ophiodon elongatus*)	Yes	Essential Fish Habitat	Pacific Coast Groundfish Fishery Management Plan (https://www.pcouncil.org/managed_fishery/groundfish/)
North pacific swordfish (*Xiphias gladius*)	Yes	Essential Fish Habitat	Highly Migratory Species Fishery Management Plan (https://www.pcouncil.org/managed_fishery/highly-migratory-species/)
Northern anchovy (*Engraulis mordax*)	Yes	Essential Fish Habitat	Coastal Pelagic Species Fishery Management Plan (https://www.pcouncil.org/fishery-management-plan-and-amendments/)
Albacore tuna (*Thunnus alalunga*)	Yes	Essential Fish Habitat	Coastal Pelagic Species Fishery Management Plan (https://www.pcouncil.org/fishery-management-plan-and-amendments/)
Bigeye tuna (*Thunnus obesus*)	Yes	Essential Fish Habitat	Coastal Pelagic Species Fishery Management Plan (https://www.pcouncil.org/fishery-management-plan-and-amendments/)
Blue shark (*Prionace glauca*)	Yes	Essential Fish Habitat	Coastal Pelagic Species Fishery Management Plan (https://www.pcouncil.org/fishery-management-plan-and-amendments/)
Common thresher shark (*Alopias vulpinus*)	Yes	Essential Fish Habitat	Coastal Pelagic Species Fishery Management Plan (https://www.pcouncil.org/fishery-management-plan-and-amendments/)
Shortfin mako shark (*Isurus oxyrinchus*)	Yes	Essential Fish Habitat	Highly Migratory Species Fishery Management Plan (https://www.pcouncil.org/managed_fishery/highly-migratory-species/)
Yellowfin tuna (*Thunnus albacares*)	Yes	Essential Fish Habitat	Highly Migratory Species Fishery Management Plan (https://www.pcouncil.org/managed_fishery/highly-migratory-species/)
Dorado (*Coryphaena hippurus*)	Yes	Essential Fish Habitat	Highly Migratory Species Fishery Management Plan (https://www.pcouncil.org/managed_fishery/highly-migratory-species/)

Appendix D1: Gulf of Mexico Fishery-Independent Survey Catch Distribution Overlap with AOA Option High–High Cluster Areas

Species[a]	Samples taken in region (since 2008)	Occurrences in region	Occurrences in AOA HH clusters	Occcurrences in HH clusters containing AOA final options	AOA alternative sites in clusters of overlap	Points within 10 km of cluster containing AOA options
Atlantic croaker (*Micropogonias undulatus*)	3,727	1,523	31	1	C-11	14
Banded rudderfish (*Seriola zonata*)*	3,727	38	0	0		0
Black seabass (*Centropristis striata*)*	3,727	63	0	0		0
Blue crab (*Callinectes sapidus*)	3,727	676	1	0		0
Bluefish (*Pomatomus saltatrix*)*	3,727	34	0	0		0
Gag grouper (*Mycteroperca microlepis*)*	3,727	25	2	2	W-4,E-4	0
Gray snapper (*Lutjanus griseus*)	3,727	251	3	3	E-3	0
Gray triggerfish (*Balistes capriscus*)	3,727	438	2	2	W-4,E-4	0
Greater amberjack (*Seriola dumerili*)*	3,727	76	0	0		0
Gulf butterfish (*Peprilus burti*)	3,727	1,543	49	7	W-1, C-13, W-4	8
Gulf menhaden (*Brevoortia patronus*) *	3,727	144	0	0		0
King mackerel (*Scomberomorus cavalla*)*	3,727	71	0	0		0
Spanish mackerel (*Scomberomorus maculatus*)*	3,727	84	0	0		0
Red grouper (*Epinephelus morio*)	3,727	260	0	0		0
Red snapper (*Lutjanus campechanus*)	3,727	1,276	16	8	W-4,C-13, E-3	0
Vermilion snapper (*Rhomboplites aurorubens*)	3,727	546	26	18	C-13, E-1, E-3, E-4	10
Dusky flounder (*Syacium papillosum*)	3,727	1,116	37	32	E-3, E-4	2
Fringed flounder (*Etropus crossotus*)	3,727	563	0	0		0
Gray flounder (*Etropus surinamensis*)*	3,727	129	7	7	E-3, E-4	0
Sash flounder (*Monolene antillarum*)	3,727	493	47	6	W-1, W-4,	9
Shelf flounder (*Etropus cyclosquamus*)*	3,727	77	1	1	E-4	0
Shoal flounder (*Syacium gunteri*)	3,727	10	1	1	W-4	0
Spiny flounder (*Engyophrys senta*)*	3,727	182	5	1	W-4	0
Gulf flounder (*Paralichthys albigutta*)*	3,727	63	0	0		0
Southern flounder (*Paralichthys lethostigma*)*	3,727	195	0	0		0
Atlantic thorny oyster (*Spondylus americanus*)*	3,727	14	1	1	E-3	0

[a] Species with an asterisk represent those whose survey data were removed in the most recent DisMAP update, but are archived and available upon request from DisMAP (https://apps-st.fisheries.noaa.gov/dismap/). Species were kept for display in the portal if they were caught in at least 5% of tows in a given year for at least 75% of the survey years in a given region.

Species[a]	Samples taken in region (since 2008)	Occurrences in region	Occurrences in AOA HH clusters	Occcurrences in HH clusters containing AOA final options	AOA alternative sites in clusters of overlap	Points within 10 km of cluster containing AOA options
Atlantic wing oyster (*Pteria colymbus*)*	3,727	36	0	0		0
Calico scallop (*Argopecten gibbus*)*	3,727	192	1	1	E-3	0
Mossy scallop (*Flexopecten glaber*)*	3,727	48	0	0		0
Paper scallop (*Amusium papyraceum*)	3,727	729	57	8	W-4, C-13, E-3	0
Ravenel scallop (*Euvola raveneli*)*	3,727	116	3	3	E-3, E-4	0
Northern white shrimp (*Litopenaeus setiferus*)	3,727	687	0	0		0
Brown shrimp (*Farfantepenaeus aztecus*)	3,727	2,165	58	7	W-1, W-4, C-13	9
Pink shrimp (*Farfantepenaeus duorarum*)	3,727	791	6	4	E-3, E-4	0
Purple spined sea urchin (*Arbacia punctulata*)*	3,727	177	0	0		0

[a] Species with an asterisk represent those whose survey data were removed in the most recent DisMAP update, but are archived and available upon request from DisMAP (https://apps-st.fisheries.noaa.gov/dismap/). Species were kept for display in the portal if they were caught in at least 5% of tows in a given year for at least 75% of the survey years in a given region.

Appendix D2: Southern California Bight Fishery-Independent Survey Catch Distribution Overlap with AOA Option High–High Cluster Areas

Species[a]	Samples taken in region (since 2008)	Occurrences in region	Occurrences in AOA HH clusters	Occurrences in HH clusters containing AOA final options	AOA alternative sites in clusters of overlap	Points within 10 km of cluster containing AOA options
Arrowtooth flounder (*Atheresthes stomias*)	8,623	793	0	0	—	0
Bocaccio (*Sebastes paucispinis*)	8,623	757	2	2	N1-A, N1-B, N1-C	12
California halibut/flounder (*Paralichthys californicus*)*	8,623	80	1	1	N1-A, N1-B, N1-C	0
Canary rockfish (*Sebastes pinniger*)	8,623	871	0	0	—	0
Dover sole (*Microstomus pacificus*)	8,623	7,239	5	4	N1-A, N1-B, N1-C, CN1-A, CN1-B	33
English sole (*Parophrys vetulus*)	8,623	4,264	31	11	N1-A, N1-B, N1-C, N2-A, N2-B, N2-C, N2-D, N2-E, CN1-A, CN1-B	20
Lingcod (*Ophiodon elongatus*)	8,623	3,440	9	9	N1-A, N1-B, N1-C, N2-A, N2-B, N2-C, N2-D, N2-E, CN1-A, CN1-B	18
Pacific halibut (*Hippoglossus stenolepis*)	8,623	793	0	0	—	0
Pacific pompano (*Peprilus simillimus*)	8,623	564	20	19	N1-A, N1-B, N1-C, N2-A, N2-B, N2-C, N2-D, N2-E, CN1-A, CN1-B	19
Sablefish (*Anoplopoma fimbria*)	8,623	5,053	1	1	CN1-A, CN1-B	7
Starry flounder (*Platichthys stellatus*)	8,623	152	0	0	—	0
California market squid (*Doryteuthis opalescens*)	8,623	2,111	3	3	N1-A, N1-B, N1-C, N2-A, N2-B, N2-C, N2-D, N2-E	10
Vancouver scallop (*Chlamys hastata*)*	8,623	83	0	0	—	0
Weathervane scallop (*Patinopecten caurinus*)*	8,623	67	1	1	CN1-A, CN1-B	3
Northern anchovy (*Engraulis mordax*)	8,623	265	3	3	N1-A, N1-B, N1-C, N2-A, N2-B, N2-C, N2-D, N2-E	9
Yelloweye rockfish (*Sebastes ruberrimus*)	8,623	251	0	0	—	0
Eulachon (*Thaleichthys pacificus*)	8,623	722	1	1	N1-A, N1-B, N1-C	0

[a] Species with an asterisk represent those whose survey data were removed in the most recent DisMAP update, but are archived and available upon request from DisMAP (https://apps-st.fisheries.noaa.gov/dismap/). Species were kept for display in the portal if they were caught in at least 5% of tows in a given year for at least 75% of the survey years in a given region.

Recently published by the Northwest Fisheries Science Center

NOAA Technical Memorandum NMFS-NWFSC-

185 **Kamikawa, D. J., and J. Buchanan. 2023.** Northeast Pacific Invertebrates, 5th Edition. U.S.Department of Commerce, NOAA Technical Memorandum NMFS-NWFSC-185. *In press.*

184 **Somers, K. A., C. E. Whitmire, K. E. Richerson, and V. J. Tuttle. 2023.** Fishing Effort in the 2002–21 U.S. Pacific Coast Groundfish Fisheries. U.S. Department of Commerce, NOAA Technical Memorandum NMFS-NWFSC-184. https://doi.org/10.25923/fa7g-yp86

183 **Keller, A. A., D. L. Draper, A. C. Chappell, K. L. Bosley, J. C. Buchanan, P. H. Frey, J. H. Harms, M. A. Head, and V. H. Simon. 2023.** The 2021 U.S. West Coast Bottom Trawl Survey of Groundfish Resources off Washington, Oregon, and California: Estimates of Distribution, Abundance, and Length Composition. U.S. Department of Commerce, NOAA Technical Memorandum NMFS-NWFSC-183. https://doi.org/10.25923/8hwq-2s75

182 **Somers, K. A., K. E. Richerson, V. J. Tuttle, and J. T. McVeigh. 2023.** Estimated Discard and Catch of Groundfish Species in the 2021 U.S. West Coast Fisheries. U.S. Department of Commerce, NOAA Technical Memorandum NMFS-NWFSC-182. https://doi.org/10.25923/teda-x859

181 **Stefankiv, O., M. Bond, and P. Kiffney. 2023.** Interim Report of Data Supporting Phase 1 Reviews of Essential Fish Habitat for Pacific Coast Salmon. U.S. Department of Commerce, NOAA Technical Memorandum NMFS-NWFSC-181. https://doi.org/10.25923/nahb-s175

180 **Rhodes, L. D., K. L. Parrish, and M. L. Willis. 2023.** Review of Best Practices for Biosecurity and Disease Management for Marine Aquaculture in U.S. Waters. U.S. Department of Commerce, NOAA Technical Memorandum NMFS-NWFSC-180. https://doi.org/10.25923/b4qp-9e65

179 **Gustafson, R., K. E. Richerson, K. A. Somers, V. J. Tuttle, and J. E. Jannot. 2022.** Information in Support of a Five-Year Status Review of Eulachon (*Thaleichthys pacificus*) Listed under the Endangered Species Act: Southern Distinct Population Segment. U.S. Department of Commerce, NOAA Technical Memorandum NMFS-NWFSC-179. https://doi.org/10.25923/p7mr-sk77

178 **Richerson, K. E., J. E. Jannot, J. T. McVeigh, K. A. Somers, V. J. Tuttle, and S. Wang. 2022.** Observed and Estimated Bycatch of Green Sturgeon in 2002–19 U.S. West Coast Groundfish Fisheries. U.S. Department of Commerce, NOAA Technical Memorandum NMFS-NWFSC-178. https://doi.org/10.25923/tkp7-mj29

177 **Morzaria-Luna, H., I. C. Kaplan, C. J. Harvey, R. Girardin, E. A. Fulton, P. MacCready, B. Chasco, P. Horne, and M. Schmidt. 2022.** Design and Parameterization of a Spatially Explicit Atlantis Ecosystem Model for Puget Sound. U.S. Department of Commerce, NOAA Technical Memorandum NMFS-NWFSC-177. https://doi.org/10.25923/tnp6-mf67

NOAA Technical Memorandums NMFS-NWFSC are available from the NOAA Institutional Repository, https://repository.library.noaa.gov.

U.S. Secretary of Commerce
Gina M. Raimondo

Under Secretary of Commerce for
Oceans and Atmosphere
Dr. Richard W. Spinrad

Assistant Administrator for Fisheries
Janet Coit

September 2023

fisheries.noaa.gov

OFFICIAL BUSINESS

National Marine
Fisheries Service
Northwest Fisheries Science Center
2725 Montlake Boulevard East
Seattle, Washington 98112