NORTHEASTERN U.S.

AQUACULTURE MANAGEMENT GUIDE

A manual for the identification and management of aquaculture production hazards

First edition, 2014

Northeastern Regional Aquaculture Center

USDA

United States Department of Agriculture
National Institute of Food and Agriculture

Tessa L. Getchis, *Editor*

NORTHEASTERN U.S.

AQUACULTURE MANAGEMENT GUIDE

A manual for the identification and management of aquaculture production hazards

First edition, 2014

Northeastern Regional Aquaculture Center

United States Department of Agriculture
National Institute of Food and Agriculture

Editor

Tessa L. Getchis, Connecticut Sea Grant and UConn Extension, University of Connecticut

Contributors

Deborah Bouchard, University of Maine
David Bushek, Rutgers University
Joseph Buttner, Salem State University
Ryan Carnegie, Virginia Institute of Marine Science
Michael Chambers, New Hampshire Sea Grant
Anoushka Concepcion, Connecticut Sea Grant and UConn Extension, University of Connecticut
John Ewart, Delaware Sea Grant, University of Delaware
Ann Faulds, Pennsylvania Sea Grant, Pennsylvania State University
Tessa L. Getchis, Connecticut Sea Grant and UConn Extension, University of Connecticut
Gef Flimlin, Rutgers Cooperative Extension, Rutgers University
Doris Hicks, Delaware Sea Grant, University of Delaware
Craig Hollingsworth, University of Massachusetts Extension
Jang Kim, University of Connecticut
Andy Lazur, University of Maryland
Dale Leavitt, Roger Williams University
Scott Lindell, Marine Biological Laboratory
Dennis McIntosh, Delaware State University
Diane Murphy, Woods Hole Sea Grant, Cape Cod Cooperative Extension
Michael Pietrak, University of Maine, Aquaculture Research Institute
Sarah Redmond, University of Maine
Joshua Reitsma, Woods Hole Sea Grant, Cape Cod Cooperative Extension
Michael Rice, URI Extension, University of Rhode Island
Gregg Rivara, Cornell Cooperative Extension
Roxanna Smolowitz, Roger Williams University
Donald Webster, University of Maryland Extension

Graphic Design

Dean Batteson, Office of Communications, College of Agriculture, Health and Natural Resources, University of Connecticut

Editorial Assistance

Sandra Shumway and **Ronald Tardiff**, University of Connecticut

Dedication:

We dedicate this publication to Walt Canzonier, "TSFRHG from Bivalve sur Maurice". Walt, a stalwart supporter of the aquaculture industry in the northeastern U.S., encouraged this effort. He envisions an extension, research, and regulatory community that is more responsive to the needs of the industry, an industry that is better informed about the risks inherent in the business, and one that is better able to address and manage production hazards on the farm.

Acknowledgments:

We gratefully acknowledge the following farmers whose experience proved invaluable to this effort: Charlie Conklin, Paul Dobbins, Betsy Haskin, Rick Karney, George Mathis, George Nardi, Carter Newell, Jules Opton-Himmel, Dale Parsons, Robert Rheault, James Tweed, John Waylon, and Lawrence Williams. These farmers shared an immense amount of knowledge in an effort to educate new entrants to the industry. In addition, several researchers, aquatic animal health professionals, regulators, and extension professionals participated in workshops and focus groups and provided valuable feedback on this publication including: Chris Bartlett, Paul Bowser, Michael Brown, Marta Gomez-Chiarri, David Grunden, Jurij Homziak, Bill Kelleher, John Kraeuter, David McGhie, Jeff Mercer, Dana Morse, David Noyes, Michael Oesterling, Dina Proestau, Nathan Stone, Marc Turano, Colby Wells and Charlie Yarish.

Funding

This project was funded through the USDA National Institute of Food and Agriculture Northeastern Regional Aquaculture Center grant #Z540501 to the Northeast Aquaculture Extension Network.

Preface

Each year, the aquaculture industry experiences significant economic losses as a result of pathogens that cause disease, pests that render product unmarketable, operational mishaps, adverse weather events, and closures of harvest areas due to the presence of organisms with the potential to cause human illness. Collectively, we refer to these as aquaculture production hazards, which present considerable risk to operations. Massive loss of farmed product and human illness caused from ingestion of unknowingly contaminated product both adversely impact profitability, trade, and public perception.

The ability of professionals to respond to problems and assist farmers is often limited by a lack of farm-level monitoring, record keeping, and farmer knowledge of hazards and hazard management strategies. Frequently, the causes of mortality events remain unknown or are identified when it is too late to prevent, control, correct or mitigate. Often, key pieces of information are missing from farmers' requests to identify and correct the hazard, limiting the response from the extension and aquatic health professional community.

To respond to this problem, the Northeast Aquaculture Extension Network (NAEN), a group of extension professionals from universities and industry associations across the northeastern U.S., together with researchers, aquatic animal health professionals, and experienced industry members has developed this comprehensive publication that identifies strategies to address aquaculture production hazards. The manual includes science-based information about major production hazards facing farmers, including: predators, diseases, parasites, organisms that have the potential to cause aquatic animal illness and human illness (e.g. toxic algae), biofouling, spread of invasive species, and other operational and environmental hazards. The manual also includes guidelines for environmental monitoring, evaluation and sampling of stocks, record-keeping procedures, and state-by-state contact information for whom to call when a problem occurs. The manual incorporates best management practices and biosecurity measures developed through research and outreach efforts funded by the USDA Northeastern Regional Aquaculture Center (NRAC) and others.

Improved knowledge of hazards associated with aquaculture production is the first step towards developing or improving risk management strategies. Use of appropriate farm monitoring protocols and record keeping will help aquatic animal health professionals respond better and more efficiently to animal illness or mortality events. If the causes of such events are identified quickly and definitively, future losses may be minimized or prevented, leading to increased production and profitability. The potential for realized economic benefits is significant; operators who plan proactively to minimize production hazards may have a competitive advantage in the marketplace.

Table of Contents

Introduction

Purpose

This manual is intended to help prospective farmers to identify, record, monitor, and manage production hazards, and ultimately minimize production-related risk.

Identify	Identify potential hazards or threats to the operation
Record	Record parameters that might help to indicate a problem
Monitor	Monitor and review records
Manage	Manage for when parameters fall outside of optimum or acceptable levels
Ask	Ask for assistance

Included is information on types of risk, production hazards, and hazard management strategies for major aquaculture crops cultivated in the northeastern U.S. While the region is defined as Maryland to Maine, some variation in hazards and management strategies exist and are noted within the text.

Unique to the Northeast region, the manual includes information on a variety of shellfish, finfish, and seaweed species. It does not describe all species under cultivation, but instead highlights species that are common and for which, until now, complete information on the subject has not been available. The manual does not cover shellfish or finfish processing-related hazards or their management. These issues are addressed in the Fish and Fishery Products Hazards and Control Guidance of the U.S. Food and Drug Administration, 4th Edition, April 2011[1]. Though research is underway, knowledge on processing hazards associated with seaweed culture is limited and will not be covered in this volume.

This publication is a collaborative effort of extension agents and aquatic health professionals in the Northeast region, and is one of many resources available for aquaculturists. A list of reference materials and contact information for these resource providers is contained at the end of the document.

Information presented in this document is up-to-date as of January 2014. Considerable effort has been made to ensure that the text is based on the best available information. Periodic updates will be made available in the online version of this publication.

The document has color-coded tabs for ease of use.

[1] U.S. Food and Drug Administration, 2011. Fish and Fishery Products Hazards and Control Guidance of the U.S. Food and Drug Administration, 4th Edition, April 2011.

How to Use This Manual

The document is divided into several sections; the first reviews basic considerations for starting an aquaculture business; the second introduces the types of risk involved in aquaculture and strategies to minimize risk; and the third covers the basics of record keeping. This information is essential for prospective and new farmers.

Individual chapters focus on shellfish, finfish, and seaweed aquaculture and provide a practical function: to acquaint the reader with production hazards and management strategies. Each chapter begins with an overview of basic morphology and life cycles for each group. Next, there is a pictorial overview of husbandry practices and systems. The bulk of the text includes information on the major types of production hazards, including: environmental conditions, biofouling organisms, predators, diseases, invasive species, and operational conditions. Each section is presented in a similar manner and includes:

Introduction to hazard
Explanation of why the hazard exists
How to measure/monitor for the hazard
Hazard management strategies

While this manual provides hazard management strategies based upon applied research, nothing can substitute for field experience. As the farmer becomes familiar with the cultivation site, it is likely that personal observations and written records will aid significantly in future planning. Where appropriate, national- and geographically- relevant best management practices (BMP) have been incorporated into the text. References and further reading are included at the end of each chapter.

This compilation can serve as a reference or may be used to complete a production-related hazard analysis for the aquaculture operation. Instructions and forms for the hazard analysis process can be found in Appendices 1-3.

CHAPTER 1

Types of Risk in Aquaculture

Overview

Every prospective farmer should develop a comprehensive business plan. That business plan should include a description of the business and aquaculture product, a market analysis for the product, a business implementation strategy, and a multi-year enterprise budget. That plan should include a hazard analysis and a risk management plan. While the focus of this text is on risk related to production, all other types of risk should be considered prior to starting an aquaculture business.

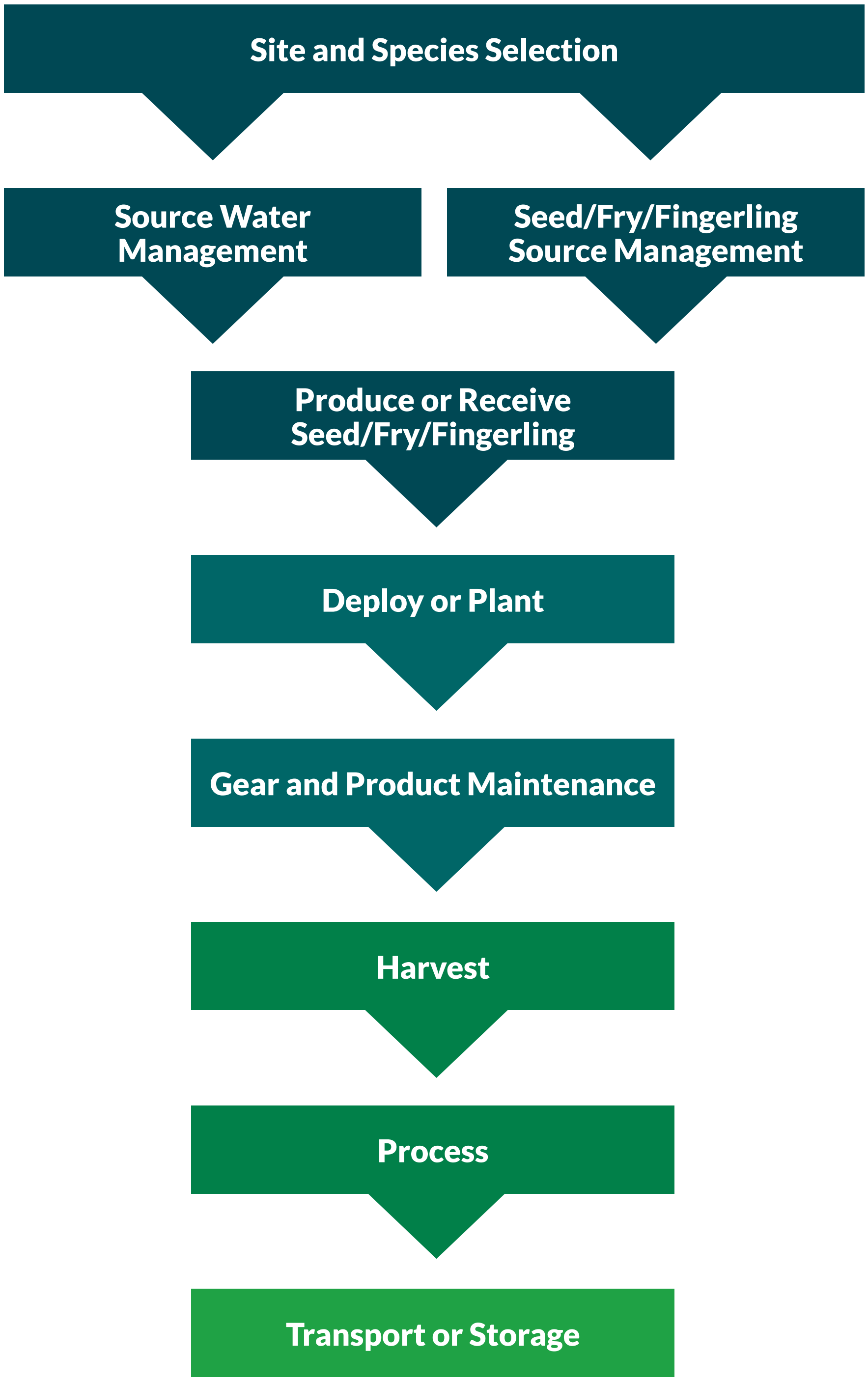
There are five basic types of agricultural risk:

Production
Marketing
Financial
Legal
Human Resource Management

Production Risks

Production risks relate to the possibility that yield will be lower than anticipated. Major sources of production risks arise from adverse environmental conditions and inclement weather events (such as drought, excessive rainfall, extreme temperatures), but may also result from damage due to predators, biofouling organisms, disease, and invasive species. Farmers should review the entire operational flow, and consider the potential hazards at each production step (Figure 1).

Production Flow Chart



Tools and strategies:

- Develop and implement an environmental management plan that includes: a) careful selection of cultivation site(s) that is appropriate for the species to be cultivated; b) routine monitoring and record keeping of environmental parameters important to the cultivation of the target species; c) establish tolerance levels for environmental parameters and have a written plan for what to do when parameters are outside of acceptable tolerance levels.
- Be aware of other threats that may exist (predators, disease, biofouling organisms) and develop and implement a plan that includes: a) the use of prevention and avoidance techniques that have been field-tested and proven economically-viable; b) routine monitoring and record keeping of gear and animals for the presence of these threats and damage resulting from them; c) establishment of tolerance levels for numbers of pests, predators, and biofouling organisms.
- Adopt local and disease resistant strains.
- Inspect and keep on file all import, transport and disposal licenses or certificates. Reject any shipments without proper licenses or certificates.
- Diversify by growing different crops.
- Be aware of, and adhere to, any government policies regarding import/export of seed and the disposal of non-target organisms and material.
- Inspect all shipments of product entering or leaving the facility. Properly dispose of all non-target organisms and material.
- Insurance: Purchase disaster assistance or crop insurance coverage to stabilize income.

Marketing/Price Risks

Marketing risks relate to the possibility that the farmer will lose the market for his/her products or that the price received will be less than expected. Common sources of marketing risk include lower prices due to increased supply or decreased consumer demand; loss of market access due to the relocation or closing of a processor or other buyer; and, lack of marketing power due to the small size of farm sellers relative to others in the market.

Tools and Strategies:

- Develop a marketing plan with realistic sales forecasts and target prices.
- Form or join a marketing cooperative to enhance prices and guarantee a market.
- Increase direct marketing efforts to capture a higher price.
- Market through multiple channels or outlets to reduce reliance on a single market.
- Enter into sales or price contracts with buyers.
- Spread harvest and sales over the season by scheduling planting.
- Conduct basic market research and survey customers.

Financial Risks

Financial risks relate to the possibility of having insufficient cash to meet expected obligations, lower than expected profits, and loss of net worth. Sources of financial risk commonly result from the production and marketing risks described earlier. In addition, financial risks may also be caused by increases in key input costs, increases in interest rates, excessive borrowing, lack of adequate cash or credit reserves, and changes in exchange rates.

Tools and Strategies:

- Develop a comprehensive business plan identifying mission, objectives, and goals.
- Monitor financial ratios and benchmarks related to liquidity, solvency and profitability.
- Conduct a trend analysis to assess what is happening with farm income and net worth over time.
- Purchase whole farm revenue insurance, such as Adjusted Gross Revenue (AGR) or AGR-Lite, to provide a safety net.
- Communicate with suppliers and lenders to review and renegotiate exiting contracts and loan terms.
- Evaluate the possibility of business expansion (getting larger) or contraction (reducing size).
- Use non-farm investments such as IRAs or mutual funds to diversify the business asset portfolio.

Legal and environmental risks

In part, legal risks relate to fulfilling business agreements and contracts. Another major source of legal risk is tort liability, i.e., causing injury to another person or property due to negligence. Legal risk is also related to environmental liability and concerns about water quality, erosion, and pesticide use.

Tools and strategies:

- Review business insurance policies and be certain to carry sufficient liability coverage.
- Evaluate the type of business legal structure; a sole proprietorship is not always the best business organization.
- Understand business contracts and agreements; ask questions if unsure.
- Take time to develop good relationships with neighbors and address their concerns.
- Use best management practices to limit environmental risk.
- Know and follow local, state, and federal regulations related to the farming operation.

Human resource management risks:

Human resource risks pertain to risks associated with individuals and their relationships to each other, their families and the farm business. Sources of human resource risk include the three D's — divorce, death, or disability of a business owner, manager, employee or family member. It also includes risks arising from poor communications and people-management practices.

Tools and strategies:

- Develop and practice good "people skills" for family as well as employees.
- Evaluate alternative sources of labor.
- Provide a safe working environment for all employees and customers who may be on site.
- Provide adequate training for employees.
- Communicate with employees and family members.
- Recognize and reward good performance.
- Review estate and business transfer plans to help insure the farm continues.
- Consider long-term care and life insurance needs.

Managing risk starts with identifying the most crucial risks; understanding the potential impacts and likelihood of undesirable outcomes; and identifying and taking possible steps to mitigate or lessen the impacts. For help with risk management planning, seek assistance from the local Extension office. Contact information appears in Appendix 4.

Adapted and modified, with permission, from the Northeast Vegetable Management Guide, written by Michael Sciabarrasi, Extension Professor, Agricultural Business Management, UNH Cooperative Extension.

CHAPTER 2

Record Keeping

Introduction

Any analysis of the aquaculture business is dependent upon sound information. Accurate, detailed, and complete records can help the farmer to:

- Provide control over the business and improve the management and efficiency of the farm.
- Provide a basis for farm credit and financing.
- Determine the relative profitability of various production techniques or systems.
- Provide information for government programs such as grants, loans, and insurance.
- Provide information for tax purposes.

For the purpose of this manual, the focus is specifically on production-related record keeping. Financial record keeping is not covered in this manual; however, detailed financial records are critical to the aquaculture operation.

Production Records

Two general categories of production records exist. Resource inventory records consist of assessing what materials and products are on hand at the time of the inventory and need only be considered on a periodic basis. Operations records include items monitored on the day-to-day performance of the farm and should be kept on a weekly or daily basis.

1. Resource inventory records include:

- **Lists of machinery and equipment owned:** Having an updated listing of all resources available in one place makes it much easier to determine the farmer's preparedness for upcoming production needs and allows him/her to plan for future purchases, should more equipment be needed.
- **List of property and property utilization schedule:** If a farm has a number of leases, systems, or ponds or multiple species in production, records should be kept of the use of each.
- **List of seed/fingerling/fry source(s) and strain(s):** This is particularly important if there are multiple sources or multiple purchases of varying size classes.
- **Inventory of seed/fingerling/fry purchased:** Document the amount, size, and distribution of each category purchased.
- **Inventory of seed/fingerling/fry planted and product transplanted/harvested:** When continuous stocking and harvesting is practiced, the change (increase or decrease) in the number and value of inventory should be calculated. Therefore, a record of beginning and ending inventory is necessary. Routine inventory of product amounts, size and holding location will provide, over the long term, critical data on survival and growth of specific groupings of product. In addition, generating diagrams or maps of product placement on the farm will reduce confusion as to source and year class of individual groups. Keeping specific lots of animals separate and routinely recording the condition of those stocks will provide important information as to performance of each group that will aid in making future decisions about purchases and placements.
- **Financial Transactions:** The intent of these records is to provide management with an accurate list of products bought and sold. As each lease, pond, or system is seeded or transplanted then harvested, the following items should be recorded – date of seeding/harvest, species seeded/harvested, amount seeded/harvested, price charged/received per unit, and the disposition of the product. Gross revenue of the production should include the cash and credit sales of the products and the imputed values of the quantities consumed on the farm. These records are essential not only for husbandry decisions, but are necessary to be eligible for any type of disaster assistance or crop insurance coverage.

2. Operations records include information such as:

- **Water quality measurements:** Information such as temperature, salinity, alkalinity, hardness, transparency, tidal stage, and a variety of other parameters that may be important to the operation should be recorded on a daily basis, and on occasion, more frequently.
- **Weather conditions:** Weather plays an important role in the performance of most aquaculture stocks. Having records allow the farmer to interpret potential influences of weather on production.
- **Stocking density for hatchery/nursery/growout systems:** What works best in one system or geographic area may not be applicable for all areas. It may take several years of observation to determine what stocking is best for the operation.
- **Daily feed consumption:** Recording daily feed consumption is an essential step in understanding the best feeding strategy for the shellfish. Determining the feed conversion efficiency allows the farmer to adjust feed type and amounts.
- **Predators (presence/absence; level):** Making observations of predator activity may allow the farmer to modify husbandry practices to avoid predation. Some predators may present a seasonal or otherwise predictable threat and can be managed or accounted for.

- **Fouling (presence/absence; level):** Water supply and food flux to the aquaculture organism can be restricted if fouling becomes extensive. Additionally, fouling may affect the appearance of the product and, subsequently, consumer acceptance. Fouling development, however, varies with environmental conditions so documenting the patterns of fouling development can help the farmer plan his/her work schedule to allow for adequate fouling control.
- **Disease (organism observations that would indicate a potential problem):** Disease is a common problem when rearing aquaculture organisms in high-density monocultures. Unexplained mortality or growth suppression may require outside assistance from an animal health professional. Without a detailed accounting of the situation along with details of the growing environment, however, assistance to aid in a diagnosis or recommend a solution to the problem will be difficult.
- **Growth:** The farmer should know the optimum growth rate and time to market for their product. Growth should be measured or estimated on a periodic basis. For all species, marking and measuring individuals allows the farmer to assess how the product is growing. A high variation between individuals could be an indication of food limitation. This information allows the grower to respond by increasing flow, decreasing stocking density, or changing food distribution methods (in fed cultures such as finfish). Though it provides only an estimate, measuring volume (with any graduated container) is easier and faster than individual measurements. This can be done every few weeks or monthly depending on the size/volume of the product.
- **Condition Index (CI) is a measure of the general well being of the animal):** The CI is a tool that can be used to assess the general health, meat quality, or yield of an organism using basic measurements (e.g. length-whole weight, whole weight-shell depth, etc.) that depend upon the organism in culture. CI can also be used to estimate the effect of environmental factors on the cultured organism. Farmers should be familiar with common condition factor measurements for their species.
- **General observations:** The farmer should note any changes in normal behavior (e.g. signs of stress) or if growth slows or stops. They should recognize seasonal (e.g. temperature effects) or predictable changes (e.g. reproductive growth) in animal growth or condition. The farmer should be aware of optimum or acceptable levels for environmental or operational parameters and make note when deviations occur. If there is any indication of a problem, contact the local aquatic health professional (see Appendix 3).

Preparing data collection sheets will help the farmer to be better organized and save time when record keeping. Printing out multiple copies of these documents and placing them in a loose-leaf notebook makes information entry relatively straightforward and self-explanatory, especially if there are multiple individuals recording data on the farm. Examples of record sheets are included at the end of this chapter.

Excerpted, with permission, from Aquaculture Record Keeping written by Robert Pomeroy, and published by Connecticut Sea Grant.

Excerpted, with permission, from Record Keeping for Aquatic Farm Management by Dale Leavitt and Gef Flimlin, and published by the Northeastern Regional Aquaculture Center.

Record Keeping Log Examples

Note that the detail on any given form is dependent on the aquaculture product, the individual farm practices, the complexity of the business, and the production level.

Example 1. Shellfish Inventory Record

<table>
<tr><td colspan="5">Lease/Lot No and Location:</td></tr>
<tr><td colspan="5">Intended Use: seed/live in-shell/shucked or processed</td></tr>
<tr><td colspan="5">Seed Source</td></tr>
<tr><td colspan="2">Seed Purchase Date:</td><td colspan="3">Quantity Purchased:</td></tr>
<tr><td colspan="2">Seed Size:</td><td colspan="3">Seed Certification/License on file: yes/no</td></tr>
<tr><td colspan="2">Financial Transaction on file: yes/no</td><td colspan="3">Original Plant Date:</td></tr>
<tr><td colspan="2">Anticipated Harvest Date:</td><td colspan="3">Anticipated Animal Harvest Size:</td></tr>
<tr><td colspan="2">Anticipated Harvest Value Per Unit:</td><td colspan="3">Anticipated % Cumulative Mortality:</td></tr>
<tr><td>Date</td><td>Description of Activity
(initial stocking, grade, transplant to/ from, gear maintenance, harvest, etc.)</td><td>Quantity planted or removed</td><td>Mortality count</td><td>Observations</td></tr>
<tr><td></td><td></td><td></td><td></td><td></td></tr>
<tr><td></td><td></td><td></td><td></td><td></td></tr>
<tr><td></td><td></td><td></td><td></td><td></td></tr>
<tr><td></td><td></td><td></td><td></td><td></td></tr>
<tr><td></td><td></td><td></td><td></td><td></td></tr>
</table>

Example 2. Environmental Conditions Record

Lease/Lot No.		Month:		
Date	Air Temp	Water Temp	Salinity	Observations (weather, predators, biofouling, disease, etc.)
1				
2				
3				
4				
5				
6				
7				
And so on...				
31				

Example 3. Fish Production System Record

System No:								
Date	Check (☑) the following activities undertaken:							
	Stock	Treat	Sample	Harvest	Drain	Description of Activity	Stocking or System Density (unit)	Observation

CHAPTER 3

Shellfish Aquaculture in the Northeastern U.S.

Overview

Bivalve shellfish production is the largest segment of marine aquaculture in the U.S. The five major species cultivated include:

eastern oyster *Crassostrea virginica*
northern quahog *Mercenaria mercenaria*
blue mussel *Mytilus edulis*
bay scallop *Argopecten irradians*
softshell clam *Mya arenaria*

Two other species, the Atlantic razor clam *Ensis directus* and the European flat oyster *Ostrea edulis*, are cultured on a small scale. While shellfish are grown primarily for human consumption, a portion is produced for fisheries stock enhancement and habitat restoration programs.

Shellfish aquaculture is comprised of three stages: hatchery, nursery, and growout. Farmers rely upon either hatchery seed production or procurement of wild seed. In some geographic locations, large natural beds exist solely for the latter purpose.

Hatcheries are typically land-based facilities that draw seawater from a local water body or a saltwater well. Wild or cultivated adult shellfish are brought into the facility and placed in tanks for broodstock conditioning. Gametes are collected, fertilized, and then maintained in static tanks or recirculating systems and reared through metamorphosis. Some hatcheries are experimenting with flow-through production methods that allow high-density larval culture. Post-set shellfish may remain in the hatchery for a period of time to achieve a larger size or may be moved into the nursery and growout stages.

Nursery culture can continue on land or can involve seeding the shellfish in floating or submerged containers or gear (e.g. upwellers, bags, trays, cages, longlines, etc.) or directly to the bottom. While many farmers choose to rear their product in containers, a large proportion of the industry plants significantly larger seed (presumably less vulnerable to predation) directly on the bottom, or use some combination of both gear and bottom culture. While some privately owned shellfish grounds exist, the majority of the industry cultivates shellfish on public grounds through municipal or state leasing or licensing programs.

This chapter includes an overview of production-related risks for the major species and includes an example of shellfish morphology and the life cycle (Figure 1-2), as well as images of cultivation gear (Figures 3-14). Detailed information on hatchery production and growout practices are documented in a number of references listed at the end of this manual (Appendix 5).

Shellfish Morphology

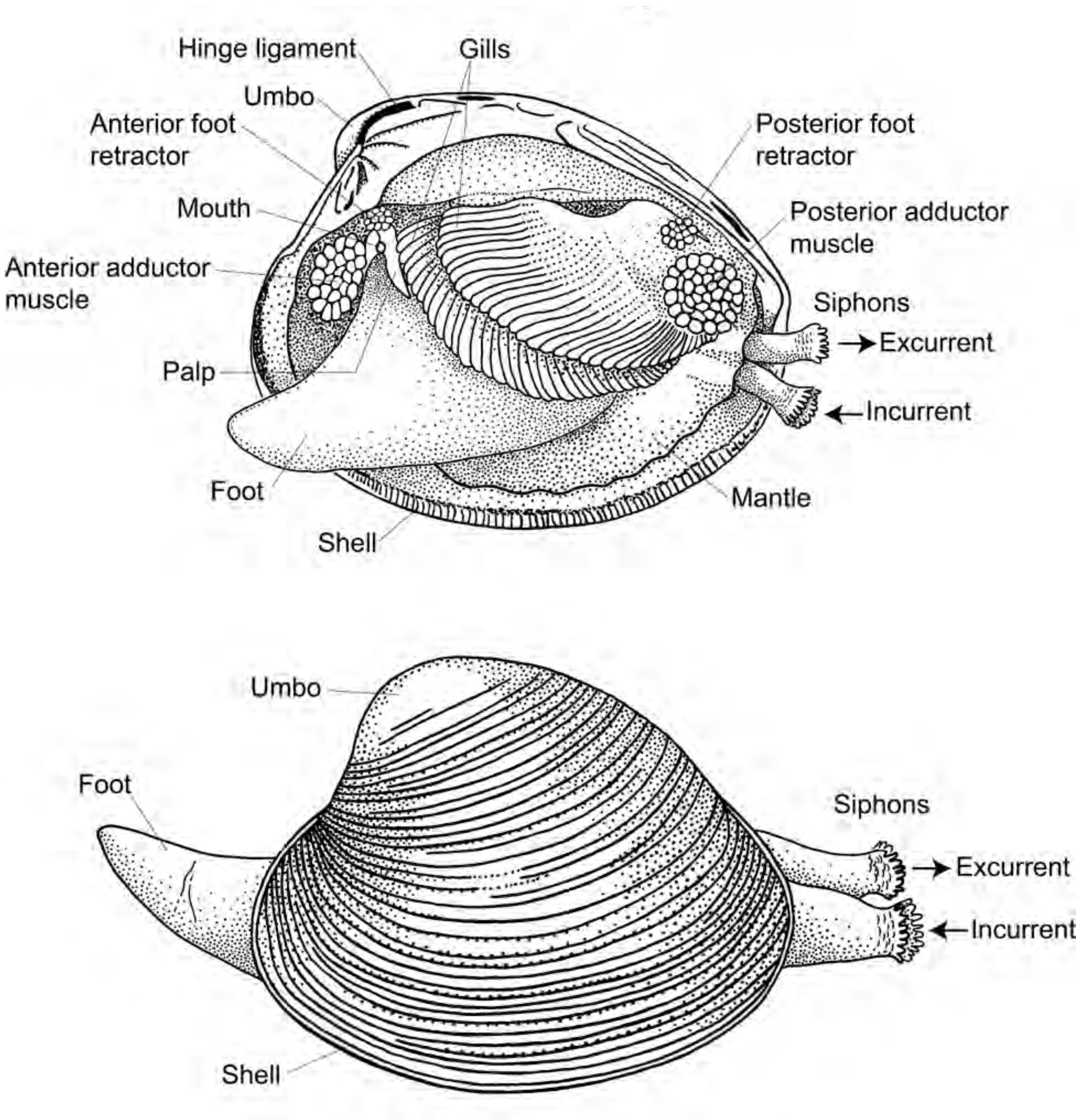


Figure 1. External and internal morphology of the northern quahog, Mercenaria mercenaria*.*
Virge Kask

Shellfish Life Cycle

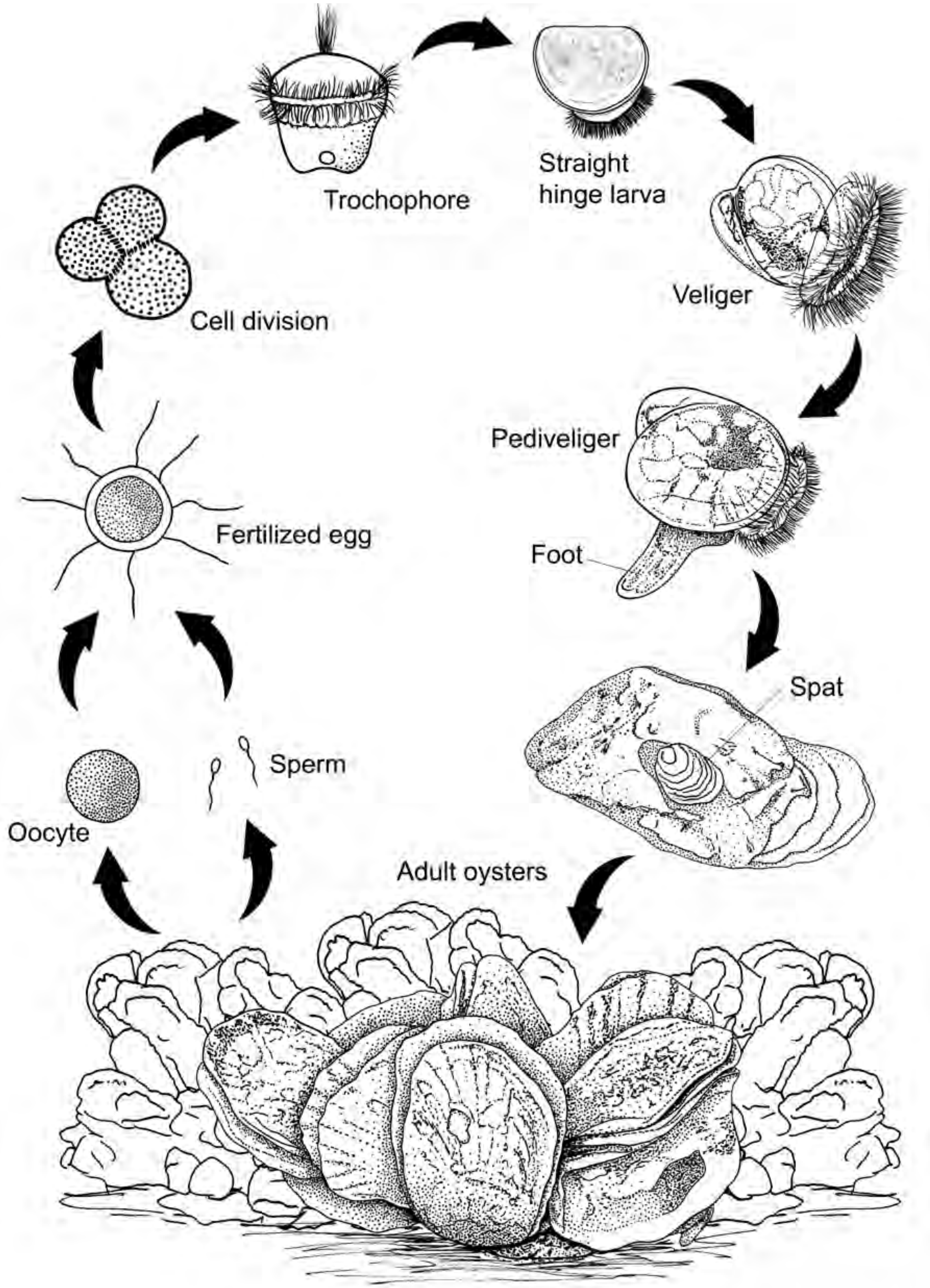


Figure 2. Life cycle of the eastern oyster, Crassostrea virginica.
Virge Kask

Shellfish Cultivation Systems

Figure 3. Hatchery
Tessa Getchis

Figure 4. Upweller
Dale Leavitt

Figure 5. Taylor float
John Supan

Figure 6. Rack and bag
Robert Rheault

Figure 7. Floating bag
Tessa Getchis

Figure 8. Pearl/Lantern net
Jon Rowley

Figure 9. Oyster Gro system
Robert Rheault

Figure 10. Stacked tray
Tessa Getchis

Figure 11. Bottom cage
Diane Murphy

Figure 12. Bottom plant/predator netting
Joseph Buttner

Figure 13. Bottom cultivation and dredging
Stacey Salce

Figure 14. Bottom harvesting
Robert Rheault

Potential Shellfish Production Hazards

Environmental Conditions
Biofouling Organisms
Predators
Diseases and Parasites
Invasive Species
Operational Procedures

Environmental Conditions

Environmental conditions play a significant role in the productivity of the farm. Therefore, it is important for the farmer to understand the role that key environmental parameters may play in crop production, and the potential risks when those parameters fall outside of acceptable limits for the cultivated species.

Much of the information included in this section also applies to hatcheries, but the tolerance ranges in the early life stages of shellfish may be less than for adults.

The environmental variables listed are important to consider when locating and managing a farm. When outside of acceptable limits, these are considered "hazards" to the aquaculture operation.

Often, the local aquaculture extension program has the capacity to measure environmental parameters. See Appendix 4 for contact information.

Potential hazards:

temperature
dissolved oxygen
salinity
pH
metabolic waste products
turbidity
harmful algal blooms
adverse weather

TEMPERATURE

Shellfish are ectothermic, meaning that their body temperature is essentially the same as the temperature of their environment. Because their metabolism is governed to a large degree by temperature, so is their growth and survival. In open water systems, the farmer cannot control the ambient temperature. However, there are numerous farm strategies noted below that can be employed to minimize the risk of extreme environmental temperatures.

Why is it a Potential Hazard?

All ectothermic animals have an temperature range within which they function and grow at their optimal rate. If the temperature increases or decreases to levels outside those tolerated by the animals, stress will ensue and the animals may die. The ranges of optimal and extreme temperature tolerances are unique to each species (see Tables 1-5) and are influenced by the normal temperature range that the animal experiences, e.g. southern oysters can function in a higher temperature range than northern oysters of the same species.

Some shellfish can withstand extended periods (days or weeks) at near-freezing temperatures. Few species are tolerant to subzero temperatures unless insulated by snow or some sort of temporary storage cover. Ice flows in intertidal or shallow subtidal areas can also severely damage aquaculture crops and gear (Figure 1).

While processing is not a focus of this document, it seemed pertinent to include a brief overview of how suboptimal temperatures can also present a hazard when shellfish product is harvested and processed for market. While water quality in shellfish harvesting areas is highly regulated, naturally occurring bacteria (e.g. *Vibrio* spp.) can still present a human health risk. These bacteria multiply more rapidly in warm water and moderate salinity, and in the northeastern U.S.; *Vibrio parahaemolyticus* in particular tends to be present in higher numbers in late spring through fall. Therefore, if the harvested shellfish are exposed to high temperatures, e.g. left to sit in the sun between harvest and transport to the processing facility, the bacteria can proliferate at a rapid rate and result in increased risk of the shellfish pathogens reaching levels that may cause human illness.

How to Measure/Monitor for the Hazard:

Water and air temperature can be monitored using a thermometer and inexpensive continuous temperature monitoring devices are also available. Temperature measurements should be entered into the daily logbook. It is important that any equipment be calibrated.

Hazard Management:

For land-based hatcheries, the use of alarm systems that will notify the farmer of any fluctuations in water temperature is the simplest way to prevent problems.

For open water operations, temperature fluctuations can be addressed either through site selection and handling strategies for the shellfish. Site selection involves gathering information on the temperature range of the proposed growing area and reconciling that with the temperature ranges for the species of interest.

If the site is prone to extreme low water temperatures or ice in the winter, the farmer should consider either moving the shellfish to deeper waters or removing them from the flats to an alternative holding area (e.g. “pitting” oysters). The same is true on the opposite end of the temperature spectrum. When transplanting product during extreme temperatures, handling and exposure should be kept to a minimum to avoid further stress to the animal.

There are some simple ways to manage temperature during the harvest and processing of shellfish. Keeping harvested shellfish out of direct sunlight using tents or tarps can reduce heat stress. It is important to keep adequate airflow above and between bags or baskets of product to allow for even cooling. A good alternative is to have ice available for packing shellfish until they can be transported to the dealer or to the processing facility where they should be refrigerated and reduced to an internal temperature less than 50°F. The time allowed to get product into refrigeration and cooled to the appropriate temperature varies according to state/federal risk assessments. In recent years, an increase in illness outbreaks in some states due to certain strains of *Vibrio parahaemolyticus* has resulted in more stringent time-temperature controls to reduce the risk of human illness.

Due to the potential for human health risk, all states follow federally recommended guidelines for handling shellfish from harvest to processing, based on the National Shellfish Sanitation Program (NSSP) Model Ordinance of the U.S. Food and Drug Administration[1]. It is incumbent upon the farmer to be aware of, and adhere to, their state regulations for handling/monitoring shellfish and following these guidelines as they are handling their shellfish. Consult with the State Aquaculture Coordinator for this information (See Appendix 3).

Figure 1. Ice can severely damage aquaculture product and gear.
John Lowell

[1] U.S. Food and Drug Administration, 2011. National Shellfish Sanitation Program Guide for the Control of Molluscan Shellfish, 2011 Revision.

DISSOLVED OXYGEN

Oxygen is dissolved in seawater, and the solubility changes with water temperature and salinity such that there is less oxygen in the water with increasing temperature and salinity.

Shellfish remove oxygen from the seawater by pumping seawater across their mantle and gills. Some shellfish can extract oxygen directly from the air, and many species can survive for extended periods without oxygen by reverting to anaerobic metabolism. These abilities, however, are species-specific and highly dependent upon environmental conditions.

Why is it a Potential hazard?

Exposure to low oxygen or anoxic conditions may stress the shellfish, and prolonged exposure may result in depressed growth rates or death.

How to Measure/Monitor for the Hazard:

It is important to be aware of oxygen levels on the farm and to measure them regularly so that the farmer can manage for daily (e.g. related to tidal cycle, diel changes) and seasonal fluctuations. Measuring dissolved oxygen in seawater is relatively simple. Test kits are available from marine aquarium suppliers. Inexpensive continuous dissolve oxygen monitoring devices are also available.

Hazard Management:

If an area is prone to hypoxia, it should be avoided. Often, there can be a vertical stratification of the water column with surface waters having higher levels of dissolved oxygen than at depth. If that is the case, bringing the shellfish to the surface, if feasible, is another possible strategy to counter chronic hypoxia. If hypoxia is confined to a small embayment or cove then it may be possible to install large aerators to counter nighttime low oxygen events.

SALINITY

Salinity is a measure of the total salts in the water and is reported as a dimensionless unit. Historically it has been measured in parts per thousand (ppt) or reported as practical salinity units (psu).

Freshwater = salinity of less than 0.5

Brackish = salinity typically between 0.5 and 17 (but up to 30)

Seawater = salinity average of 35

Shellfish are osmoconformers and will adjust the salinity of their body fluids to mimic external conditions, often very rapidly, as they are exposed to variations in environmental salinity. Tolerance of environmental salinity fluctuations is species-specific and is also dependent upon concomitant exposure to other stressors, e.g. temperature and hypoxia.

Why is it a Potential Hazard?

If shellfish are exposed to suboptimal salinities for prolonged periods, they may not be capable of adjusting their internal salinity properly, resulting in cellular damage and mortality. Even short-term exposure may result in some stress, depending upon the species.

As is true of temperature and dissolved oxygen, the response of an animal to changing environmental salinity varies around some level that is considered optimal. Normal ranges of salinity tolerance for major shellfish species are listed in Tables 1-5.

How to Measure/Monitor for the Hazard:

Most farmers measure salinity with an inexpensive hand-held refractometer. Units should be calibrated on occasion (i.e. use freshwater to ensure a salinity of zero).

Hazard Management:

Larval shellfish are highly sensitive to suboptimal changes in environmental parameters. If the hatchery is located on a water body that may have transitory drops in salinity, farmers can restrict flow to the hatchery during those times of low salinity. Another strategy is to adjust the salinity by addition of synthetic sea salts, commonly available from aquarium suppliers.

Adult shellfish typically exhibit a wider range of environmental tolerances than larvae and juveniles, and these are species specific.

pH

The pH of seawater is usually within the range of 7.6 – 8.4, and can vary depending upon temperature, salinity, freshwater source, and other factors.

Why is it a Potential Hazard?

Acidification in the marine environment has received considerable attention, particularly with reference to marine shellfish. Recent evidence suggests that increasing levels of CO_2 will induce significant changes in the marine environment. The pH of both the interstitial waters and those at the sediment-water interface may be impacted by increased loading of organic material and the subsequent decomposition of that material.

The potential threat of decreasing pH (acidification) is primarily associated with the ability of the shellfish to calcify their shell, although it can also have deleterious impacts on the overall physiological function of the animals. Normal shell is formed when the animal manufactures a protein matrix and the mantle lays down new shell along the growing edge. The formation of the crystalline shell structure is highly dependent on the pH of the surrounding environment, as the shellfish needs to extract calcium and carbonate ions from the seawater and chemically combine them during shell formation.

Recent research has demonstrated that a decrease in seawater pH impacts shell formation in larval shellfish resulting in malformed shells leading to failed metamorphosis and death. Documenting this phenomenon in the field has, however, proven more difficult.

How to Measure/Monitor for the Hazard:

Specialized protocols for monitoring pH in hatcheries have been developed and utilize commercially available pH meters.

Hazard Management:

While it is possible to adjust the pH of incoming water in a hatchery by the addition of soda ash, it is not a simple adjustment. Suboptimal pH is a relatively uncommon issue in the northeast.

METABOLIC WASTE PRODUCTS

Shellfish produce soluble nitrogenous waste products, mainly in the form of ammonia, into the surrounding waters.

Why is it a Potential Hazard?

Ammonia (NH_3) and nitrite (NO_2^-), and to a much lesser degree nitrate (NO_3^-), can be very toxic to marine animals if they build up in a static system, e.g. in a hatchery or recirculating system. Excess levels of nitrogenous wastes can be lethal to larval shellfish.

How to Measure/Monitor for the Hazard:

There are a number of simple inexpensive kits available for monitoring for ammonia, nitrite and nitrate in seawater.

Hazard Management:

The nitrogen levels should be monitored regularly. The immediate response to excess levels is to undertake a large volume water change in the system. If the high nitrogen levels are a recurring problem, more frequent water changes may be necessary.

TURBIDITY

Turbidity is a measure of the clarity of the water and can be influenced by a number of factors including the concentration of phytoplankton and suspended organic and inorganic particles in the water (seston).

High turbidity may be beneficial if it is primarily a result of high chlorophyll concentrations which provide nutrition for the shellfish.

Why is it a Potential Hazard?

High turbidity due primarily to suspended sediment may have a negative impact on shellfish. The greatest hazards associated with excess turbidity are potential clogging of gills, a reduction in food quality, potential burial of epifaunal shellfish, and smothering of siphons of infaunal shellfish.

How to Measure/Monitor for the Hazard:

A simple and common method to measure for turbidity is with a Secchi Disc, a weighted plastic disc with black and white quarters (Figure 2). The disk is lowered into the water until the user loses sight of the disk. It is then slowly raised until it comes back into sight. The depth at which the user regains sight of the disc is termed the “Secchi depth” and is dependent upon the turbidity of the water.

One thing to keep in mind when measuring turbidity is that the measurement indicates the overall density of suspended particles in the water column, but it does not inform the user as to the composition of the suspended material.

Hazard Management:

- Avoid culture in areas known to have high inorganic sediment loads and sedimentation rates.
- If adverse weather or other events result in extreme sedimentation, excess silt should be washed from cultivation gear and animals. Planted oyster beds may need to be dredged to remove excess silt.
- Hatcheries should employ use of mechanical filtration to remove suspended solids.

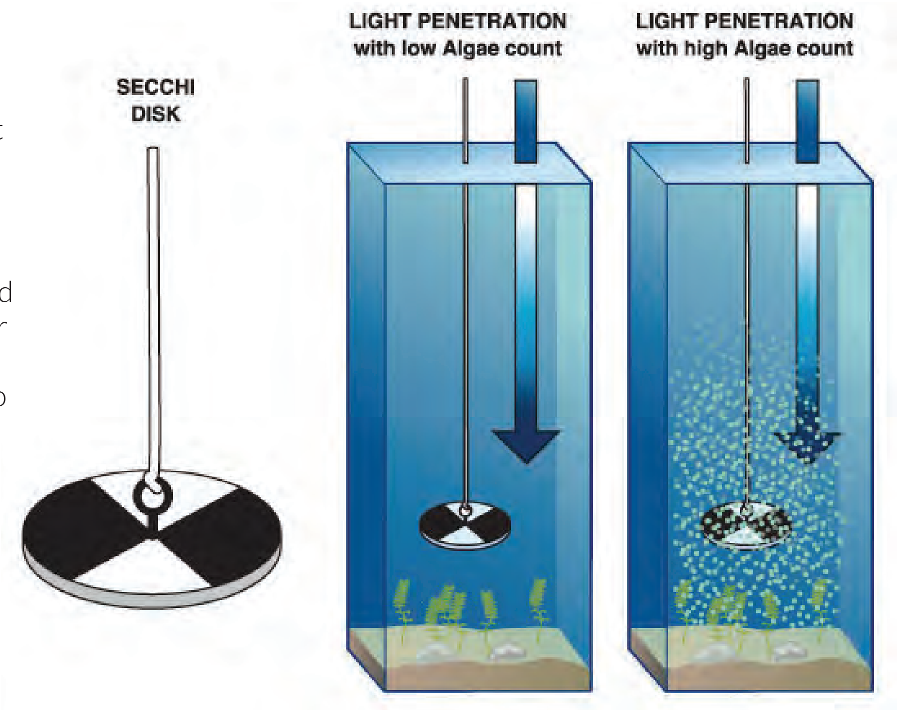


Figure. 2. The Secchi Disk.
U.S. Army Corps of Engineers, Albuquerque District

HARMFUL ALGAL BLOOMS

Microalgae (phytoplankton) are typically single-celled algal species that are very small (3-50µm) and are the primary food source for shellfish. In the wild, numerous microalgal species exist together in the phytoplankton community. Certain environmental conditions sometimes trigger one species of microalgae to reproduce to a level that outcompetes the other algal cells in the community. When this happens, the exceedingly high density of algae is referred to as a bloom. Some bloom species also produce potent neurotoxins that may impact the shellfish themselves and can render them as vectors of toxins when consumed by higher trophic level species including humans. If the algal species is one that causes harm either to the ecosystem or humans it is termed a harmful algal bloom (HAB). Impacts of harmful algal species on shellfish are species specific and dependent upon the algal species-shellfish species pairing in question.

Mode of Action: HAB affect bivalves in several ways. Algae can produce toxins that cause destruction and necrosis of the tissue, e.g. gills and mantle, or digestive and vascular tissues. Some bivalve species can close their valves for extended periods of time to prevent exposure to the toxic algae. Though harmful algae may be edible, they are not necessarily nutritious which may result in the bivalve starving, again, a species-specific phenomenon.

HAB in the Northeast usually occur over a limited time period of several days to weeks, and there are a few immediately noticeable signs that cultured shellfish are affected. Again, impacts vary depending on the HAB species and shellfish species in question.

Why is it a Potential Hazard?

A breakdown of the potential consequences of HAB that exist in this region include:

1) HAB with adverse affects on humans:

- a) Diatoms
  - i) Amnesic Shellfish Poisoning – caused by domoic acid produced by some species of the diatom *Pseudo-nitzschia* spp., it can cause gastrointestinal and neurological problems, including short-term memory loss, in humans. Can be fatal.
- b) Dinoflagellates
  - i) Paralytic Shellfish Poisoning – caused by a suite of potent neurotoxins (saxitoxins and derivatives) that are produced in the Northeast by the dinoflagellate *Alexandrium fundyense*). This neurotoxin can lead to death within 24 hours due to respiratory arrest.
  - ii) Diarrhetic Shellfish Poisoning – caused by okadaic acid produced by *Dinophysis* spp. This causes severe gastrointestinal distress.

2) HAB with adverse effects on culture organism or culture environment:

- a) Eutrophication
  - i) Not attributable to any specific group of microalgae. If environmental conditions are such that it supports a rapid expansion of the population of a single or multiple species of microalgae, the end result is often a substantial scale die-off (bloom crash) delivering large amounts of organic material to the sediment surface. The resulting decomposition of the organic material can result in reduction or depletion of dissolved oxygen on the water overlying the area.
  - ii) A crash of an algal bloom can affect shellfish through low dissolved oxygen as described above.
  - iii) If the bloom is intensive, the high density of microalgal particles can physically clog or irritate gills.
- b) Impairment of shellfish
  - i) Some species of microalgae that are not health hazards to humans have negative direct impacts on the shellfish. Brown Tide (caused by *Aureococcus anophagefferens*) and Rust Tide (caused by *Cochlodinium polykrikoides*) have been implicated in suppressing shellfish growth and mortalities when present in high densities in coastal waters.

How to Measure/Monitor for the Hazard:

For areas with a history of HAB that can result in human health risks, states are required to implement a regular HAB monitoring program.

These microalgae can reach levels resulting in human health impairment without being visible in the water. Stations are routinely sampled and analyzed by state natural resource programs.

For those HAB that are not a direct human health risk, monitoring is not regularly undertaken so the first indication that there is a problem will probably be noticed by the farmer as a reduction or cessation of growth in their shellfish or even mortalities. Sometimes, the bloom is dense enough to be noticeable in coastal waters. Often, even with that information, it is problematic to assign reduced growth to a HAB. Only after close monitoring is it possible to assign growth problems to microalgal dynamics. Therefore, if the farmer suspects that there is a HAB problem then the aquaculture health professional and extension personnel should be consulted (Appendices 4, 5).

Hazard Management:

There are no means available to control algal blooms, nor can they be easily predicted. The scale of impact of a HAB is much larger than the location of the shellfish farm, thus, there is little a shellfish farmer can do to prevent or adjust for an HAB event in the vicinity of their farm other than to mitigate around the presence. Farmers should also avoid relocating shellfish from affected areas as harmful algae can be transported with the shellstock.

ADVERSE WEATHER

Weather is an uncontrollable aspect of farming that requires the farmer to think ahead as to what to expect, and to act on that knowledge by designing and managing the farm to minimize the impacts of the weather.

Why is it a Potential Hazard?

Many weather-related factors have been addressed in descriptions of other potential hazards in this manual, e.g. temperature and salinity. The one aspect not mentioned is the physical intensity of weather. The energy contained in wind-driven waves or the weight of a mass of ice being pushed by the tidal current are forces that will far exceed the farmer's ability to protect any structures that are placed in the water or on the tidal flats. Nonetheless, the farmer must adopt strategies that minimize the risk of damage or loss to the crop from adverse weather conditions.

How to Measure/Monitor for the Hazard:

Developing an awareness of the range of weather conditions at the farm site is a necessary first step in establishing the farm. The primary weather factors that can impact the crop include wind force and direction, temperature range, and extent of rainfall. Historical records of these factors are routinely available from a number of sources, including an array of weather buoys maintained by the National Oceanographic and Atmospheric Administration (NOAA) with other agencies and the National Weather Service (another NOAA Office). The agency websites provide records of weather conditions at local stations so farmers can fine-tune their weather awareness to those sites nearest to the farm site.

Monitoring upcoming weather conditions is an essential component to managing the farm. Short-term management strategies can be implemented to protect farms from damaging weather conditions if those conditions are anticipated. While weather forecasting is an inexact science, the overall capacity of weather forecasters is steadily improving and the current attitude is to have the populace prepare for the worst.

NOAA Weather Radio continually broadcasts marine weather information with forecasts up to 7 days in advance. In addition, there are numerous other sources of weather forecasting that will alert farmers to upcoming adverse weather conditions either through public media (radio or television) or web-based.

Hazard Management:

Advanced planning is primarily associated with initial site selection. Consideration of the prevailing weather conditions, e.g. the fact that winter storms primarily come from the northeast, or summer wind patterns are primarily from the southwest, when selecting and setting up the farm site is essential to minimizing the overall risk from weather. Farmers should seek areas that provide some degree of protection from severe wind conditions. Farmers should understand the normal patterns of ice development and movement within the local water body. Long-term weather patterns are available from NOAA or local environmental agencies. Having a contingency plan for short-term weather events is necessary to minimize risks. If possible, prior to predicted storms or the formation of major ice flows, vulnerable structures should be moved to deeper water or provided additional anchorage. Development of a written emergency preparedness plan and regular review of that plan is the best approach to protecting the farm from adverse weather conditions.

References and Further Reading:

Bayne, B.L. (Ed.), 1976. Marine mussels: their ecology and physiology. Cambridge University Press, New York.

Gosling, E.M., (Ed.), 1992. The Mussel *Mytilus*: Ecology, Physiology, Genetics and Culture. Elsevier, Amsterdam.

Grizzle, R., Shumway, S.E., Bricelj, V.M., 2001. Physiology and Bioenergetics of *Mercenaria mercenaria*, in: Castagna, M., Kraeuter, J.N., (Eds.), Biology of the Hard Clam. Developments in Aquaculture and Fisheries Science – 31, Elsevier Scientific Publishers, New York. pp. 305-382.

Landsberg, J. H., Shumway, S.E., 1998. Harmful algal blooms and their effects on marine and estuarine animals. Proceedings 3rd International Symposium on Aquatic Animal Health. pp. 58-63.

Matsuyama, Y., Shumway, S.E., 2009. Impacts of harmful algal blooms on shellfisheries aquaculture, in: Allan, G., Burnell, G. (Eds.), Shellfish Aquaculture. Woodhead Publishing in Food, Science, and Technology, Oxford. pp. 560-609.

Rice, M.A., Pechenik, J.A., 1992. A review of the factors influencing the growth of the northern quahog, *Mercenaria mercenaria* (Linnaeus, 1758). Journal of Shellfish Research. 11(1), 279-287.

Shumway, S.E., 1990. A review of the effects of algal blooms on shellfish and aquaculture. Journal of the World Aquaculture Society. 21, 65-104.

Shumway S.E., 1996. Natural environmental factors, in: Kennedy, V.S., Newell, R.I.E., Eble, A.F., (Eds.), The eastern oyster, *Crassostrea virginica*. Maryland Sea Grant Publication. pp. 467-513.

Shumway, S.E., Parsons, G.J., (Eds.), 2006. Scallops: Biology, Ecology and Aquaculture, 2nd Edition. Developments in Aquaculture and Fisheries Science Volume 35. Elsevier, San Diego, California.

Table 1. Environmental parameters to consider for eastern oyster (*Crassostrea virginica*) culture.

TEMPERATURE Culture Stage	Temperature Range			Optimal Temperature		
	°F	°C	References	°F	°C	References
Gonad Development	60.4 - 86.0	15.8 - 30.0	Loosanoff & Davis 1963			
Spawning	>68.0	>20.0	Nelson 1928	rapid temperature rise once gonads are ripe		Galtsoff 1964
	>61.0	>16.1	Loosanoff 1969			
Larval rearing	68.0 - 86.0	20.0 - 30.0	Galtsoff 1964	86.0 - 90.5	30.0 - 32.5	Davis & Calabrese 1964
	>63.5	>17.5	Hofstetter 1977	77.0	25.0	MacInnes & Calabrese 1979
				68.0 - 90.5	20.0 - 32.5	Calabrese & Davis 1970
Juvenile to Adult	33.8 - 96.8	1.0 - 36.0	Galtsoff 1964	77.0 - 78.8	25.0 - 26.0	Galtsoff 1964
	28.9 - 96.8	-1.7 - 36.0	EOBRT 2007	68.0 - 86.0	20.0 - 30.0	Sellers & Stanley 1984
	28.4 - 96.8	-2 - 36	Shumway 1996	59.0 - 77.0	15.0 - 25.0	Collier 1954

SALINITY Culture Stage	Salinity Range		Optimal Salinity	
	Salinity	References	Salinity	References
Reproductive development	>6.0	Butler 1949	19.3 - 35.1	Anemiya 1926
	18.0 - 41.0	Anemiya 1926		
Spawning	>7.5	Loosanoff 1948		
Larval rearing			17.5	Calabrese & Davis 1970
	10.0 - 27.0	Calabrese & Davis 1970	26.0	MacInnes & Calabrese 1979
	22.0 - 33.0	Anemiya 1926	24.5 - 29.8	Anemiya 1926
	3.1 - 30.6	Carriker 1951	related to ambient salinity experienced by parents	Davis 1958
	>7.5	Davis 1958		
Juvenile to Adult	Highest limit of tolerance: 34 - 40	Galtsoff 1964	14.0 - 28.0	Quast et al. 1988, Shumway 1996
	5.0 - 30.0	Galtsoff 1964	15.0 - 22.0	Chanley 1957
	5.0 - 27.0	Davis 1958, Loosanoff 1953	10.0 - 28.0	Loosanoff 1965
	0.0 - 42.0	Quast et al. 1988, Shumway 1996	16.0 - 27.0	Butler 1949

DISSOLVED OXYGEN Culture Stage	Dissolved Oxygen	
	DO (mg/l or ppm)	References
Larval rearing	>20% sat minimum	Baker & Mann 1992
Juvenile to Adult	~20 - 100% saturation	EOBRT 2007
	* tolerant of hypoxic conditions for extended time periods	

pH Culture Stage	pH range		Optimal pH range	
	pH	References	pH	References
Larval rearing	6.75 - 8.75	Calabrese & Davis 1966	8.25 - 8.50	Calabrese & Davis 1966

TURBIDITY Culture Stage	Turbidity	
	Turbidity (mg/l)	References
Larval rearing	<750 maximum	Davis & Hidu 1969

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References

Amemiya, I., 1926. Notes on experiments on the early developmental stages of the Portuguese, American, and English native oysters, with special reference to the effect of varying salinity. Journal of the Marine Biological Association (U.K.). 14, 161-175.

Baker, S., Mann, R., 1992. Effects of hypoxia and anoxia on larval settlement, juvenile growth, and juvenile survival of the oyster *Crassostrea virginica*. Biological Bulletin. 182, 265-269.

Butler, P.A., 1949. Gametogenesis in the oyster under conditions of depressed salinity. Biological Bulletin. 96(3), 263-269.

Calabrese, A., Davis, H.C., 1966. The pH tolerance of embryos and larvae of *Mercenaria mercenaria* and *Crassostrea virginica*. Biological Bulletin. 131, 427-436.

Calabrese, A., Davis, H.C., 1970. Tolerances and requirements of embryos and larvae of bivalve molluscs. Helgolander Wissenschaftliche Meeresuntersuchungen. 20, 553-564.

Carriker, M.R., 1951. Ecological observations on the distribution of oyster larvae in New Jersey estuaries. Ecological Monographs. 21(1), 19-38.

Chanley, P.E., 1957. Survival of some juvenile bivalves in water of low salinity. Proceedings of the National Shellfisheries Association. 48, 52-65.

Collier, A., 1954. A study of the response of oysters to temperature and some long-range ecological interpretations. Proceedings of the National Shellfisheries Association. 42, 13-38

Davis, H.C., 1958. Survival and growth of clam and oyster larvae at different salinities. Biological Bulletin. 114, 296-307.

Davis, H.C., Calabrese, A., 1964. Combined effects of temperature and salinity on development of eggs and growth of larvae of *M. mercenaria* and *C. virginica*. U.S Fish and Wildlife Service Fishery Bulletin. 63(3), 643-655.

Davis, H.C., Hidu, H., 1969. Effects of turbidity-producing substances in seawater on eggs and larvae of three genera of bivalve mollusks. Veliger. 11, 316-323.

EOBRT (Eastern Oyster Biological Review Team), 2007. Status review of the eastern oyster (*Crassostrea virginica*). Report to the National Marine Fisheries Service, Northeast Regional Office.

Galtsoff, P.S., 1964. The American oyster *Crassostrea virginica* (Gmelin). U.S. Fish and Wildlife Service Fishery Bulletin. 64, 1-480.

Hofstetter, R.P., 1977. Trends in population levels of the American oyster, *Crassostrea virginica* (Gmelin) on public reefs in Galveston Bay, Texas. Technical Series No. 10. Texas Parks and Wildlife Department, Coastal Fisheries Branch, Austin, Texas.

Loosanoff, V.L., 1948. Gonadal development and spawning of oysters (*C. virginica*) in low salinities. The Anatomical record. 101, 55.

Loosanoff, V.L., 1953. Behavior of oysters in water of low salinities. Proceedings of the National Shellfisheries Association. 43, 135-151.

Loosanoff, V.L., 1965. The American or eastern oyster. U.S. Fish and Wildlife Service, Circular No. 205.

Loosanoff, V.L., 1969. Maturation of gonads of oysters, *Crassostrea virginica*, of different geographical areas subjected to relatively low temperatures. Veliger. 11(3), 153-163.

Loosanoff, V.L., Davis, H.C., 1963. Rearing of bivalve molluscs. Advances in Marine Biology. 1, 1-136.

MacInnes, J.R., Calabrese, A., 1979. Combined effects of salinity, temperature, and copper on embryos and early larvae of the American oyster, *Crassostrea virginica*. Archives of Environmental Contamination and Toxicology. 8(5), 553-562.

Nelson, T.C., 1928. On the distribution of critical temperatures for spawning and for ciliary activity in bivalve molluscs. Science. 67, 220-221.

Quast, W.D., Johns, M.A., Pitts Jr., D.E., Matlock, G.C., Clark, J.E., 1988. Texas oyster fishery management plan. Fishery Management Plan Series No. 1. Texas Parks and Wildlife Department, Coastal Fisheries Branch, Austin, Texas.

Sellers, M.A., Stanley, J.G., 1984. Species profiles: life histories and environmental requirements of coastal fishes and invertebrates (North Atlantic) -- American oyster. U.S. Fish and Wildlife Service FWS/OBS-82/11.23. U.S. Army Corps of Engineers, TR EL-82-4.

Shumway, S.E., 1996. Natural environmental factors, in: Kennedy, V.S., Newell, R.I.E., Eble, A.F. (Eds.), The eastern oyster *Crassostrea virginica*. Maryland Sea Grant College, University of Maryland, College Park, Maryland, pp. 467-513.

Table 2. Environmental parameters to consider for Northern quahog (*Mercenaria mercenaria*) culture.

TEMPERATURE Culture Stage	Temperature Range			Optimal Temperature		
	°F	°C	References	°F	°C	References
Spawning	71.6 - 86.0	22.0 - 30.0	Carriker 1961	78.3	25.7	Carriker 1961
	69.8 - 77.0	21.0 - 25.0	Kennish & Olsen 1975	78.8	26.0	Whetstone et al. 2005
	68.0 - 73.4	20.0 - 23.0	Roegner & Mann 1991			
Larval rearing	54.5 - 91.4	12.5 - 33.0	Roegner & Mann 1991	63.5 - 86.0	17.5 - 30.0	Davis & Calabrese 1964
	59.0 - 91.4	15.0 - 33.0	Rice 1992	78.8	26.0	Whetstone et al. 2005
				72.5 - 79.9	22.5 - 26.6**	Roegner & Mann 1991
				68.0	20.0	Rice 1992
				72.5 - 97.7	22.5 - 36.5	Lough 1975
Juvenile to Adult	32.0 - 95.0	0.0 - 35.0	Stanley & DeWitt 1983	69.8 - 87.8	21.0 - 31.0	Roegner & Mann 1991
	33.8 - 93.2	1.0 - 34.0*	Rice 1992	68.0 - 73.4	20.0 - 23.0	Ansell 1968; Pratt & Campbell 1956
	* Growth ceases at < 9°C or > 31°C (Ansell 1968)			** At 21.5 - 30.0 salinity		
	Pumping ceases at < 6°C (Ansell 1968)					
	Activity stops <1°C and >34°C; growth stops at <9°C or >31°C					

SALINITY Culture Stage	Salinity Range		Optimal Salinity	
	Salinity	References	Salinity	References
Reproductive development	15.0 - 35.0	Roegner & Mann 1991		
Egg Development	20.0 - 35.0	Roegner & Mann 1991	27	Roegner & Mann 1991
Larval rearing	15.0 - 32.0	Davis 1958	20.0 - 27.0	Rice 1992
	17.5 - 35.0	Rice 1992		
Metamorphosis	17.5 - 20.0	Castagna & Chanley 1973		
Juvenile to Adult	10.0 - 35.0*	Stanley & DeWitt 1983	24.0 - 32.0	Rice 1992
	10.0 - 40.0	Baker et al. 2007		
	18.0 - 32.0	Rice 1992		
	>12.0	Roegner & Mann 1991		
	* May be differences based on geographical site of origin			

DISSOLVED OXYGEN Culture Stage	Dissolved Oxygen		Optimal Dissolved Oxygen	
	DO (mg/l or ppm)	References	DO (mg/l or ppm)	References
Larval rearing	>0.5 minimum	Rice 1992		
Juvenile to Adult	>4.0* minimum	Rice 1992	>5.0	Roegner & Mann 1991
	> 5.0 minimum	Hamwi 1969	>4.0	Whetstone 2005
	>2.4 minimum	Baker et al. 2002		
	* tolerant of hypoxic conditions for extended time periods			

pH Culture Stage	pH range		Optimum pH	
	pH	References	pH	References
Larval rearing	6.75 - 8.50	Roegner & Mann 1991	7.50 - 8.50	Roegner & Mann 1991

TURBIDITY Culture Stage	Turbidity	
	Turbidity (mg/l)	References
Larval rearing	<400	Davis 1960
Juvenile growth affected	>44	Bricelj et al. 1984

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References:

Ansell, A.D., 1968. The rate of growth of the hard clam *Mercenaria mercenaria* (L.) throughout the geographical range. ICES Journal of Marine Science. 31, 364-409.

Baker, S.M., Heuberger, D., Phlips, E., Sturmer, L. 2002. Water quality and its role on hard clam production. Institute of Food and Agricultural Sciences, University of Florida Cooperative Extension, Technical Bulletin No. FLSGP-T-02-003.

Baker, S., Hoover, E., Sturmer, L., 2007. The role of salinity in hard clam aquaculture. Institute of Food and Agricultural Sciences, University of Florida Cooperative Extension, Circular No. 1500.

Bricelj, V.M., Malouf, R.E., de Quillfeldt, C., 1984. Growth of juvenile *Mercenaria mercenaria* and the effect of resuspended bottom sediments. Marine Biology. 84, 167-173.

Carriker, M.L., 1961. Interrelations of functional morphology, behavior, and autecology of the bivalve *Mercenaria mercenaria*. Journal of the Elisha Mitchell Science Society. 77(2), 168-241.

Castagna, M., Chanley, P., 1973. Salinity tolerance of some marine bivalves from inshore and estuarine environments in Virginia waters on the Western Mid-Atlantic coast. Malacologia. 12, 47-96.

Davis, H.C., Calabrese, A., 1964. Combined effects of temperature and salinity on development of eggs and growth of larvae of *M. mercenaria* and *C. virginica*. U.S. Fish and Wildlife Service Fishery Bulletin. 63(3), 643-655.

Davis, H.C., 1958. Survival and growth of clam and oyster larvae at different salinities. Biological Bulletin. 114(3), 296-307.

Davis, H.C., 1960. Effects of turbidity- producing materials in seawater on eggs and larvae of the clam (*Venus-(Mercenaria mercenaria*). Biological Bulletin. 118, 48-54.

Hamwi, A., 1969. Oxygen consumption and pumping rate of the hard clam *Mercenaria mercenaria* L. PhD. Thesis, Rutgers University, New Brunswick, New Jersey.

Kennish, M.J., Olsson, R.K., 1975. Effects of thermal discharges on the microstructural growth of *Mercenaria mercenaria*. Environmental Geology. 1, 41-64.

Lough, R.G., 1975. A reevaluation of the combined effects of temperature and salinity on the survival and growth of bivalve larvae using response surface techniques. U.S. National Marine Fisheries Service Fishery Bulletin. 73(1), 86-94.

Pratt, D.M., Campbell, D.A., 1956. Environmental factors affecting growth in *Venus mercenaria*. Limnology and Oceanography. 1(1), 2-17.

Rice, M.A., 1992. The northern quahog: The biology of *Mercenaria mercenaria*. Rhode Island Sea Grant, Narragansett, Rhode Island.

Roegner, G.C., Mann, R., 1991. Hard clam, in: Funderburk, S., Mihursky, J., Jordan, S., Riley, D., (Eds.) Habitat requirements for Chesapeake Bay Living Resources, 2nd edition. Living Resources Subcommittee, Chesapeake Bay Program. Annapolis, Maryland.

Stanley, J., DeWitt, R., 1983. Species profiles: life histories and environmental requirements of coastal fishes and invertebrates (North America)—hard clam. U.S. Fish and Wildlife Service, FWS/OBS-82/11.18.

Whetstone, J.M., Sturmer, L.N., Oesterling, M.J., 2005. Biology and culture of the hard clam (*Mercenaria mercenaria*). USDA Southeastern Regional Aquaculture Center, Publication No. 433.

Table 3. Environmental parameters to consider for softshell clam (*Mya arenaria*) culture.

TEMPERATURE Culture Stage	Temperature Range °F	Temperature Range °C	Temperature Range References	Optimal Temperature °F	Optimal Temperature °C	Optimal Temperature References
Gonad Development				59 - 60.8	15-16	Buttner et al. 2010
Spawning	>53.6	>12	Lawson 1966	> 50 - 53.6	>10 - 12	Christian et al. 2010
				71.6 - 75.2	22 - 24	Buttner et al. 2010
Larval rearing	<93.9	<34.4	Kennedy & Mihursky 1972	62.6 - 73.4	17 - 23	Christian et al. 2010
				69.8 - 75.2	21 - 24	Buttner et al. 2010
Juvenile to Adult	<82.4	<28	Pfitzenmeyer 1972	62.6 - 73.4	17 - 23	Stickney 1964
	>5 - 14	> -10 - -15	Christian et al 2010	42.8 - 57.2	6 - 14	Christian et al. 2010
	<90.5	<32.5	Kennedy & Mihursky 1972	68	20	Newell & Hidu 1983
	28.4 - 82.4	-2 - 28	Weston & Buttner 2010			

SALINITY Culture Stage	Salinity Range: Salinity	Salinity Range: References	Optimal Salinity: Salinity	Optimal Salinity: References
Larval rearing			16 - 32	Stickney 1964
Juvenile to Adult	>5	Christian et al. 2010	25 - 35	Christian et al. 2010
			20 - 32 (Maine)	Gilfillan et al. 1976
			10 - 33 (Massachusetts)	Belding 1909
			4 - 15 (Maryland)	Pfitzenmeyer 1972

DISSOLVED OXYGEN Culture Stage	Dissolved Oxygen: DO (mg/l or ppm)	Dissolved Oxygen: References
Juvenile to Adult	>2.8 minimum	van Dam 1935

TURBIDITY Culture Stage	Turbidity: Turbidity (mg/l)	Turbidity: References
juvenile growth	<100	Grant & Thorpe 1991

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References:

Belding, D.L., 1909. The soft-shelled clam fishery of Massachusetts. Commonwealth of Massachusetts Department of Conservation, Division of Fish and Game, Marine Fisheries Service, Publication No. 1. (republished in 1916 and 1930).

Buttner. J.K., Weston, S., Beal, B.F., 2010. Softshell clam culture: hatchery phase, broodstock care through seed production. USDA Northeastern Regional Aquaculture Center, Publication No. 202-2010.

Christian, J.R., Grant, C.G.J., Meade, J.D., Noble, L.D., 2010. Habitat Requirements and Life history Characteristics of Selected Marine Invertebrate Species Occurring in the Newfoundland and Labrador Region. Canadian Manuscript Report of Fisheries and Aquatic Sciences No. 2925.

Gilfillan, E.S., Mayo, D., Hanson, S., Donovan, O., Jiang, L.C., 1976. Reduction in carbon flux in *Mya arenaria* caused by a spill of No. 6 fuel oil. Marine Biology. 37, 115-123.

Grant, J., Thorpe, B., 1991. Effects of suspended sediment on growth, respiration, and excretion of the soft-shelled clam (*Mya arenaria*). Canadian Journal of Fisheries and Aquatic Sciences. 48, 1285–1292.

Kennedy, R.S., Mihursky, J.A., 1972. Effects of temperature in the respiratory metabolism of three Chesapeake bivalves. Chesapeake Science. 13, l-22.

Lawson, D., 1966. The genus *Mya* in the Arctic region. Malacologia. 3, 399-418.

Newell, C.R., Hidu, H., 1986. Species profiles: life histories and environmental requirements of coastal fishes and invertebrates (North Atlantic) – soft shell clam. U.S. Fish and Wildlife Service Biological Report No. 82(11.53). U.S. Army Corps of Engineers, TR EL-82-4.

Pfitzenmeyer, H.T., 1972. Tentative outline for inventory of molluscs: *Mya arenaria* (soft-shelled clam). Chesapeake Science Supplement. 13, 182-184.

Stickney, A.P., 1964. Feeding and growth of juvenile soft-shell clams, *Mya arenaria*. U.S. Fish and Wildlife Service Fishery Bulletin. 63, 635-642.

van Dam, L., 1935. On the utilization of oxygen by *Mya arenaria*. Journal of Experimental Biology. 12, 86-94.

Weston, S., Buttner, J.K., Beal, B.F., 2010. Softshell clam culture: basic biology and general culture conditions. USDA Northeastern Regional Aquaculture Center, Publication No. 201-2010.

Table 4. Environmental parameters to consider for blue mussel (*Mytilus edulis*) culture.

TEMPERATURE Culture Stage	Temperature Range °F	°C	References	Optimal Temperature °F	°C	References
Gonad Development	>41	>5	Newell 1989	64.4	18	Pronker 2007
Spawning	50 - 53.6	10 - 12	Christian et al. 2010			
Larval rearing	41 - 68	5 - 20	Christian et al. 2010	68	20	Hrs-Brenko & Calabrese 1969
	46.4 - 64.4	8 - 18	Bayne 1965	50 - 60.8	10 - 16	Bayne 1965
	59 - 68	15 - 20	Hrs-Brenko & Calabrese 1969			
Metamorphosis				60.8 - 62.6	16 - 17	Christian et al. 2010
Juvenile	>10.4	>-12	Bourget 1983			
stops growing	>41	> 5	Bayne 1965			
stops growing	<50, >77	<10, >25	Hrs-Brenko & Calabrese 1969			
	<37.4, >77	<3 > 25	Williams 1970, Read & Cumming 1967			
Adult	<80.6 - 84.2	<27-29	Bayne et al. 1977	50 - 68	10 - 20	Couthard 1929, Lutz & Porter 1977
	>104	> 40	Henderson 1929			
SALINITY Culture Stage	**Salinity Range: Salinity**		**References**	**Optimal Salinity: Salinity**		**References**
Spawning	> 15		Christian et al. 2010			
Larval rearing	20 - 40		Christian et al. 2010	25 - 30		Hrs-Brenko & Calabrese 1969
	15 - 35		Hrs-Brenko & Calabrese 1969	30 - 33		Bayne 1965
	30 - 40		Bayne 1965	25 - 35		Newell 1989
	5 - 34		Bayne 1976			
Metamorphosis				28 - 29		Christian et al. 2010
Juvenile to Adult	0 - 31		Christian et al. 2010	26		Mallet & Myrand 1995
	5 - 34		Bayne et al. 1976			
reduced growth	< 5 - >40		Jamieson et al. 1975			
DISSOLVED OXYGEN Culture Stage	**Dissolved Oxygen: DO (mg/l or ppm)**		**References**			
Larval survival						
Juvenile to Adult	>0.01 minimum		Theede et al. 1969			
	> 60% saturation		Newell 1989			
TURBIDITY Culture Stage	**Turbidity: Turbidity (mg/l)**		**References**			
All	>250 filtration stops		Widdows et al. 1979			

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References:

Bayne, B.L., 1965. Growth and the delay of metamorphosis of the larvae of *Mytilus edulis* L. Ophelia. 2, l-47.

Bayne, B.L., 1976. Marine mussels: their ecology and physiology. Cambridge University Press, New York.

Bayne, B.L., Thompson, R.J., Widdows, J., 1976. Physiology I., in: Bayne, B.L. (Ed.) Marine mussels: their ecology and physiology. Cambridge University Press, New York. pp. 121-206.

Bayne, B.L., Widdows, J., Worrall, C., 1977. Some temperature relationships in the physiology of two ecologically distinct bivalve populations, in: Vernberg, F.J., Calabrese, A., Thurnberg, F., Vernberg, W. (Eds.) Physiological responses of marine biota to pollutants. Academic Press, New York. pp. 379-400.

Bourget, E., 1983. Seasonal variations and cold tolerance in intertidal mollusks and their relationship to environmental conditions in the St. Lawrence estuary. Canadian Journal of Zoology. 61, 1193-1201.

Christian, J.R., Grant, C.G.J., Meade, J.D., Noble, L.D., 2010. Habitat Requirements and Life History Characteristics of Selected Marine Invertebrate Species Occurring in the Newfoundland and Labrador Region. Canadian Manuscript Report of Fisheries and Aquatic Sciences, No. 2925.

Couthard, H.S., 1929. Growth of the sea mussel. Contributions to Canadian Biology and Fisheries. 4, 121-136.

Henderson, J.T., 1929. Lethal temperatures of Lamellibranchiata. Contributions to Canadian Biology and Fisheries. 4, 397-412

Hrs-Brenko, M., Calabrese, A., 1969. The combined effects of salinity and temperature on larvae of the mussel *Mytilus edulis*. Marine Biology. 4, 224-226.

Jamieson, G.S., Neish, I.C., Clark, C.S.L., 1975. Perspective and development prospects of mussel cultivation in the Maritime Provinces of Canada. Marine Ecology Laboratory Nova Scotia, Ref. AMRL74-11.

Lutz, R.A., Porter, B., 1977. Experimental culture of blue mussels *Mytilus edulis* in heated effluent waters of a nuclear power plant. Proceedings of the World Mariculture Society. 8, 427-445.

Mallet, A.L., Myrand, B., 1995. The culture of the blue mussel in Atlantic Canada. In: Boghen, A.D. (Ed.). Cold-water Aquaculture in Atlantic Canada. The Canadian Institute for Research on Regional Development. Sackville, NB. pp. 255-296.

Newell, R.I.E., 1989. Species profiles: life histories and environmental requirements of coastal fishes and invertebrates (North and Mid-Atlantic)--blue mussel. U.S. Fish and Wildlife Service Biological Report No. 82(11.102). U.S. Army Corps of Engineers, Technical Report No. EI-82-4.

Pronker, A.E., Nevejan, N.M., Peene, F., Geijsen, P., Sorgeloos, P., 2007. Hatchery broodstock conditioning of the blue mussel *Mytilus edulis* (Linnaeus 1758). Part I. Impact of different micro-algae mixtures on broodstock performance. Aquaculture International. 16(4), 297-307.

Read, K.R.H., Cumming, K.B., 1967. Thermal tolerance of the bivalve molluscs *Modiolus modiolus* L., *Mytilus edulis* L. and *Brachidontes demissus* Dillwyn. Comparative Biochemistry and Physiology. 22, 149-155.

Theede, H., Ponat, A., Hiroki, K., Schlieper, C., 1969. Studies on the resistance of marine bottom invertebrates to oxygen deficiency and hydrogen sulphide. Marine Biology. 2, 325-337.

Widdows, J., Bayne, B.L., Livingstone, D.R., Newell, R.I.E., Donkin, P., 1979. Physiological and biochemical responses of bivalve molluscs to exposure to air. Comparative Biochemistry and Physiology. 62, 301-308.

Williams, R.J., 1970. Freezing tolerance in *Mytilus edulis*. Comparative Biochemistry and Physiology. 35, 145-161.

Table 5. Environmental parameters to consider for bay scallop (*Argopecten irradians*) culture.

TEMPERATURE Culture Stage	Temperature Range			Optimal Temperature		
	°F	°C	References	°F	°C	References
Gonad Development	>68	>20	Castagna 1975			
Spawning				>61.5	>16.4	Belding 1010
Larval rearing						
trochophore	71.6 - 82.4	22 - 28	Wright et al. 1983			
larvae	66.2 - 82.4	19 - 28	Fay et al. 1983; Lin et al. 1989	77.0	25.0	Fay et al. 1983, Lin et al. 1989
				78.8 - 82.4	26 - 28	Castagna 1975
Metamorphosis	55.4 - 89.6	13 - 32	Lin et al. 1989			
Juvenile to Adult	20 - 89.6	-6.6 - 32	Marshall 1960			
	32 - 86	0 - 30	Gutsell 1931, Kirby-Smith 1969			
growth	>45	>7.2	Belding 1910	71.6	22.0	Lin et al. 1991
SALINITY Culture Stage	**Salinity Range**			**Optimal Salinity**		
	Salinity		**References**	**Salinity**		**References**
Larval rearing	>22.5		Castagna 1975	25 - 32		He & Zhang 1990
growth limits	19.3 - 31		Lin et al. 1989			
Metamorphosis	11.7 - 35		Lin et al. 1989			
Juvenile	> 14		Belding 1910, Gutsell 1930	31.2		Lin et al. 1991
lethal	11 - 43		Lin et al. 1991			
growth limits	19 - 34		Lin et al. 1991			
DISSOLVED OXYGEN Culture Stage	**Dissolved Oxygen**					
	DO (mg/l or ppm)		**References**			
Larval survival	>1.38 minimum		Wang & Zhang 1995			
Juvenile to Adult	>1.5 minimum		van Dam 1954			
TURBIDITY Culture Stage	**Turbidity**					
	Turbidity (mg/l)		**References**			
Larval rearing	<0.5 maximum		Stone & Palmer 1973, Moore 1978			

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References:

Belding, D.L., 1910. The scallop fishery of Massachusetts. Marine Fisheries Service No. 3, Division of Fisheries & Game, Commonwealth of Massachusetts, Boston, Massachusetts.

Castagna, M., 1975. Culture of the bay scallop, *Argopecten irradians*. Virginia Marine Fisheries Review. 37(l), 19-24.

Fay, C.W., Neves, R.J., Pardue, G.B, 1983. Species profiles: life histories and environmental requirements of coastal fishes and invertebrates (mid-Atlantic) - bay scallop. U.S. Fish and Wildlife Service, Division of Biological Services, FWS/OBS-82/11.12. U.S. Army Corps of Engineers, Technical Report No. EL-82-4.

Gutsell, J.S., 1930. Natural history of the bay scallop. Bulletin of the U.S. Bureau of Fisheries. 46, 569-632.

He, Y., Zhang, F., 1990. The influence of environmental salinity on various development stages of the bay scallop *Argopecten irradians Lamarck*. Oceanology and Limnology. 21, 197-204.

Kirby-Smith, M.W., 1969. Growth of the bay scallop, *Aequipecten irradians* (Lamarck), as influenced by food concentration and temperature. American Zoologist. 9(4), 1144-1145.

Lin, R., Chen, M., Lin, B., 1989. Effects of temperature and salinity on attachment and metamorphosis of bay scallop larvae *Argopecten irradians*. Journal of Oceanography in Taiwan Strait/Taiwan Haixia. 8, 60-67.

Lin, R., Lin, B., Chen, M., 1991. Study on the effects of temperature and salinity on migrating behavior, growth and survival of juvenile bay scallop *Argopecten irradians* (Lamarck). Journal of Oceanography in Taiwan Strait/Taiwan Haixia. 10, 133-138.

Marshall, N., 1960. Studies of the Niantic River, Connecticut, with special reference to the bay scallop. Limnology and Oceanography. 5, 86-105.

Moore, P.G., 1978. Inorganic particulate suspensions in the sea and their effects on marine animals. Oceanography and Marine Biology Annual Review. 15, 225–363.

Stone, R., Palmer, R., 1973. Effects of turbidity on the bay scallop. Northeastern University Marine Science Institute, Contribution No. 38.

van Dam, L., 1954. On the respiration in scallops. Biological Bulletin. 107, 192-202.

Wang, C-D., Zhang, F-S., 1995. Effects of environmental oxygen deficiency on embryos and larvae of bay scallop, *Argopecten irradians*. Chinese Journal of Oceanology and Limnology. 13, 362-369.

Wright, D.A., Kennedy, V.S., Roosenburg, W.H., Castagna, M., Mihursky, J.A., 1983. Temperature tolerance of embryos and larvae of five bivalve species under simulated power entrainment conditions. Marine Ecology Progress Series. 77, 271-278.

Potential Shellfish Production Hazards

Environmental Conditions

Biofouling Organisms

Predators

Diseases and Parasites

Invasive Species

Operational Procedures

Biofouling Organisms

The term biofouling refers to numerous plant and colonizing animal species that create a nuisance when attached to aquaculture gear and shellfish. Often referred to as fouling organisms, pests such as algae, barnacles, tubeworms, tunicates, and even oyster and mussel set can have adverse impacts on the target aquaculture species. Their effects are usually physical in nature, with impacts ranging from heavy algal mats that smother shellfish to competition for space and resources especially by tunicates and barnacles. The presence of biofouling organisms can interfere with 1) shellfish growth - causing reduced growth rates and shell deformities; 2) feeding activity - restricting water flow and food available to shellfish; and 3) resources - creating competition for space. Additionally, shellfish that are destined to be sold live and in shell should be aesthetically appealing and relatively free of extraneous material. These fouling organisms have a much shorter shelf life than the cultured shellfish, and may spoil or decrease the value of the target product.

Potential hazards:

macroalgae
sponges
bryozoans
hydrozoans
molluscs
polychaetes
crustaceans
ascidians

The presence of fouling organisms, and the time and labor cost to control them, can impose a significant economic burden on a production facility. Commonly used strategies to manage biofouling involve: (1) manual removal (e.g. drying, washing, scraping), (2) mechanical means (e.g. tumbling, aeration), or (3) chemical control (e.g. brine, bleach, lime, or acetic acid dips, and antifouling paints and coatings). Few of these methods have proven effective for the management of biofouling organisms, and some means of chemical control have been shown to have significant adverse environmental effects. Biological control using natural predators has not been demonstrated as effective in managing biofouling organisms. Other strategies can involve the timing of gear deployments to avoid or minimize time in the water during an expected 'bloom' or recruitment (settling period) of organisms such as barnacles, mussels, or tunicates, but this is not always feasible and other mitigation measures involves rotating gear - trading old gear for new.

Routine cleaning and maintenance is key to minimizing the effects of biofouling. Hatchery inflow water should be filtered or disinfected, and regular cleaning and disinfection of equipment, pipes, tanks, and netting can help prevent colonization and ensure adequate flow and system operation.

Marine biofouling organisms have similar affects on shellfish, finfish, and seaweed aquaculture operations. To avoid repetition, this section includes information on potential biofouling hazards for all groups and notes what species or groups are affected. Biofouling reference material for finfish and seaweed aquaculture appear in those respective chapters.

Figure 1: Biofouling diatoms on a kelp line.
Sarah Redmond

MICROALGAE

Name:

- various species of diatoms

Mode of Action: Diatoms, when present in high concentrations, can smother juvenile plants and coat frond surfaces, impacting growth and quality.

Species/Systems Affected:

- kelp (*Saccharina latissima*)
- *Gracilaria tikvahiae*

Hazard Management:

- Optimize density and environmental conditions to maximize growth rates of cultured species.
- Select sites with good current flow or high turbidity.
- Outplant small juveniles well before spring diatom blooms to avoid smothering of small fronds.

Figure 2a. Cage fouled with various species of seaweeds.
Tessa Getchis

Figure 2b. Seaweed long line fouled with non-target seaweeds and other epiphytes.
Sarah Redmond

MACROALGAE

Name: various species of seaweeds including:

- green seaweeds (*Ulva lactuca*, *Ulva* spp., *Chaetomorpha* sp., *Cladophora* sp., *Codium fragile**)
- brown seaweeds (*Pylaiella* sp., *Ectocarpus* spp., *Colpomenia peregrine**)
- red seaweeds (*Ceramium* spp., *Palmaria palmata*, *Agardhiella* sp., *Neosiphonia harveyii*, *Heterosiphonia japonica*, *Gracilaria vermiculopllya**, *Grateloupia turuturu** *Gracilaria tikvahiae* [if not target species])

Mode of Action: These algae form, depending on the species, clumps or mats, which can smother aquaculture gear; these algal mats can lead to anoxic conditions underneath. The algae may also be carried by currents and end up wrapped around cages which can block the flow of water through the gear and increase drag. Other types of unwanted seaweeds can settle and develop on gear or cultured product (in the case of shellfish and seaweeds) and compete for space and resources.

Species/Systems Affected:

- eastern oyster (*Crassostrea virginica*)
- northern quahog (*Mercenaria mercenaria*)
- bay scallop (*Argopecten irradians*)
- blue mussel (*Mytilus edulis*)
- marine finfish
- kelp (*Saccharina latissima*)
- *Gracilaria tikvahiae*

Hazard Management:

- Keep nets clear of algae through regular removal (brushing) or periodically exchange fouled nets with clean gear.
- Remove biofouling by manually scrubbing or power washing gear.

* = non-native

Figure 3. The boring sponge, Cliona celata*.*
Larry Williams

SPONGES

Name:

- boring sponge (*Cliona celata*)
- red beard sponge (*Microciona prolifera*)

Mode of Action: *Cliona* is a common marine sponge that erodes the valves of shellfish leaving many large holes. In heavy infestations, this impacts the integrity of the valves leaving the shellfish vulnerable to other predators. Shellfish may compensate by depositing new shell, but may come at an expense to other normal metabolic processes. Damaged shells can crush easily when shucked and render product unfit for consumption. *Microciona* is commonly found attached to hard surfaces, including shellfish, but as opposed to *Cliona*, only affects the shell superficially.

Species Affected:

- eastern oyster (*Crassostrea virginica)*
- northern quahog (*Mercenaria mercenaria*)

Hazard Management:

- Keep aquaculture gear and shellfish clean through regular removal of sponges or periodically exchange fouled gear with clean gear.
- Remove sponges by manually scrubbing.
- Expose shellfish to air-dry or brine dip (only species that can completely close their valves) as appropriate.

Figure 4a: The lacy crust bryozoan, Membranipora membranacea*, on juvenile oyster shell.*
Tessa Getchis

Figure 4b: Spreading of a colony of M. membranacea *on a kelp blade.*
Sarah Redmond

BRYOZOANS AND HYDROZOANS

Name:

- lacy crust bryozoan* (*Membranipora membranacea*)
- bushy bryozoans (*Bugula turrita*, *B. simplex*, *B. neritina**, *Cryptosula pallasiana*, *Alcyonidium* sp.)
- hydroids (*Tubularia* spp., *Cordylophora* sp., *Campanularia* spp., *Sertularella* spp., *Stylactaria* spp., *Obelia* spp., others to a limited degree)

Mode of Action: All of these species can foul firm surfaces such as aquaculture gear and cultured shellfish and seaweeds. They cause an unsightly appearance and can be difficult to remove once established. Membranipora membranacea can develop a flat crust on kelp blade and stipe tissue that reduces spore output and growth, and contributes to weakened tissue. Other types of bryozoans and hydroids are bushy in appearance. These epiphytes can affect seaweed blade quality, appearance, and strength.

Species Affected:

- eastern oyster (*Crassostrea virginica*)
- bay scallop (*Argopecten irradians*)
- blue mussel (*Mytilus edulis*)
- marine finfish
- kelp (*Saccharina latissima*)
- *Gracilaria tikvahiae*

Hazard Management:

- Remove biofouling by manually scrubbing or power washing gear, preferably off-site and on land if possible to avoid/reduce further spread of non-natives.
- Periodic rotation of gear – exchange fouled gear with clean or new gear.
- Expose shellfish to air-dry or brine dip (only species that can completely close their valves) as appropriate.
- For seaweed culture, select high energy sites over low energy sites to reduce opportunity for settlement.
- For kelp, harvest before onset of fouling period.
- Optimize growth rates and current flow around seaweed.
- Expose *Gracilaria* to freshwater dips as appropriate.

* = non-native

Figure 5a: **Crepidula fornicata** *populations can overwhelm shellfish operations.*
Tessa Getchis

Figure 5b: **Crepidula fornicata** *stacked upon each other.*
Tessa Getchis

GASTROPOD MOLLUSC

Name:

- common slippersnail (*Crepidula fornicata*)

Mode of action: These common gastropods attach to shellfish and aquaculture gear throughout the intertidal and shallow subtidal waters. Individuals stacked together and can quickly foul gear.

Species Affected:

- eastern oyster (*Crassostrea virginica*)
- bay scallop (*Argopecten irradians*)
- blue mussel (*Mytilus edulis*)

Hazard Management:

- Periodic rotation of gear – exchange fouled gear with clean or new gear.
- *Crepidula* are difficult to remove and powerwashing can be successful if done imediately after snails have settled onto gear.

Figure 6: Oyster overset on gear.
Diane Murphy

BIVALVE MOLLUSC

Name:

- blue mussel (*Mytilus edulis*)
- ribbed mussel (*Geukensia demissa*)
- eastern oyster (*Crassostrea virginica*)
- jingle shell (*Anomia simplex*)

Mode of action: Non-target shellfish compete with other shellfish for resources, and therefore interfere with feeding efficiency of the cultured organism. The production of byssal threads (beard) in mussels can bind together shellfish and gear, which can reduce water flow. The additional weight of non-target shellfish can make cultivation gear too heavy to service. Spat from both wild and farmed shellfish can create a problem referred to as overset. The presence of wild set can reduce market value of farmed product if not removed.

Species affected

- eastern oyster (*Crassostrea virginica*)
- bay scallop (*Argopecten irradians*)
- blue mussel (*Mytilus edulis*)

Hazard Management:

- Manual gear scrubbing to remove mussels as early as possible to minimize over growth of byssal fibers.
- Tumbling oysters can remove small overset.
- Manual gear scrubbing to remove oyster set as early as possible to minimize over growth of shell.
- Periodic rotation of gear – exchange fouled gear with clean or new gear.
- Expose shellfish to brine dip to control overset.

Figure 7a: Oyster seed encrusted with Hydroides *tubes.*
Tessa Getchis

Figure 7b: Spirorbis *tubes on bay scallop shells.*
Diane Murphy

POLYCHAETE WORM

Name:

- carnation worm, tube worm (*Hydroides dianthus*)
- coiled worm, tube worm (*Spirorbis borealis, S. spirillum*)
- plume worm (*Diopatra* sp.)

Mode of action: The segmented carnation worm grows up to 7.5 cm long and builds a long, twisted white shell (tube). *Spirorbis* worms grow up to 2 mm long and build tiny, white, coiled shell-like tubes. These worms commonly attach to hard objects such as shellfish and aquaculture gear. Their calcareous tubes create an unsightly nuisance particularly on oyster and scallop shells and their appearance may compromise marketability of shellfish. *Diopatra* sp. has been identified as a problem in upwellers where it may clog mesh and cause mortalities when the screens become strapped tightly to the bottom of worm tubes.

Species affected:

- eastern oyster (*Crassostrea virginica*)
- bay scallop (*Argopecten irradians*)
- blue mussel (*Mytilus edulis*)

Hazard Management:

- Tube worms must be addressed when they first form, or they are very difficult to remove.
- Remove by manually scrubbing or power washing.
- Periodic rotation of gear – exchange fouled gear with clean or new gear.

Figure 8a: The tube building amphipod, Ampelisca *spp.*
Diane Murphy

Figure 8b: Ampelisca *tubes visible after removal of quahog nets.*
Diane Murphy

CRUSTACEAN

Name:

- four-eyed amphipod, tube-building amphipod (*Ampelisca* spp.)

Mode of action: The four-eyed amphipod can be found in lower intertidal zones in muddy or sandy sediments. This small (up to 20mm long) amphipod is easily recognized by the clumps of flexible parchment-like tubes it constructs. The tubes are flattened in appearance and extend up to 0.5" above the sediment surface. When abundant, the network of extensive tubes can bind the sediment creating inhospitable conditions for shellfish, particularly northern quahog and oyster seed.

Species affected:

- northern quahog (*Mercenaria mercenaria*)
- eastern oyster (*Crassostrea virginica*)

Hazard Management:

- Keep shellfish nets clear of amphipod tubes through regular removal (brushing) or periodically exchange fouled nets with clean gear.
- In areas where this organism occurs in abundance, rake or till the sediment to break up tube structures.
- Remove biofouling by manually scrubbing or power washing, preferably off-site and on land if possible to avoid/reduce further spread of this organism.
- Air dry or brine dip shellfish as appropriate.

Figure 9: The skeleton shrimp, Caprella mutica.
Wikipedia Commons

CRUSTACEANS

Name: skeleton shrimp, ghost shrimp including:

- Japanese skeleton shrimp *Aeginella longicornis* (Labrador to the Mid Atlantic U.S.)
- Japanese skeleton shrimp *Caprella septentrionalis* (Greenland to New England)
- Japanese skeleton shrimp *Caprella mutica**

Mode of action: Skeleton shrimp are not true shrimp. Long and thin, they have an appearance similar to walking stick insects. They are commonly found clinging to seaweed fronds and aquaculture gear where they attach themselves to feed. *Caprella mutica* is an invasive species to New England, and can be found in dense colonies on any kind of submerged structure. They have a wide range of environmental tolerances, surviving at temperatures of -1.8°C to 29°C and salinities of 16 to greater than 40.

Species affected:

- kelp (*Saccharina latissima*)
- *Gracilaria tikvahiae*

Hazard Management:

- Harvest before appearance of fouling (Kelp).
- Freshwater dip as appropriate (*Gracilaria*).
- Optimize growth rates and current flow around seaweed.

* = non-native

Figure 10: Barnacle set covering blue mussels.
Diane Murphy

CRUSTACEAN

Name:

- acorn barnacles (*Balanus* spp.),
- northern rock barnacle (*Semibalanus balanoides*)

Mode of action: Barnacles cement themselves to most hard substrates (including aquaculture gear and shellfish) throughout intertidal and subtidal zone creating an unsightly nuisance on farms. Spat mortality from overgrowth of barnacles can be significant. Their growth can interfere with shellfish feeding efficiency; if not removed they can affect marketability of product due to their short shelf life.

Species affected:

- eastern oyster (*Crassostrea virginica*)
- bay scallop (*Argopecten irradians*)
- blue mussel (*Mytilus edulis*)

Hazard Management:

- Barnacles must be addressed when they first form, or they are very difficult to remove.
- Remove biofouling by manually scrubbing or power washing.
- Periodic rotation of gear – exchange fouled gear with clean or new gear.

Figure 11a: Mussels covered with Botrylloides *spp.*
Stephan Bullard

Figure 11b: Oyster seed covered with colonial tunicates.
Tessa Getchis

Figure 11c: Oyster seed covered with colonial tunicates.
Tessa Getchis

COLONIAL ASCIDIANS OR SEA SQUIRTS

Name:

- carpet tunicate* (*Didemnum vexillum*, *Didemnum candidum*, *Didemnum* spp.)
- golden star tunicate* (*Botryllus schlosseri*)
- Pacific colonial tunicate, orange or red sheath tunicate (*Botrylloides violaceus*, *B. diegensis*)
- light bulb tunicate* (*Clavelina lepadiformis*)

Mode of action: These form a rubbery layer over surfaces, including aquaculture gear and shellfish in shallow subtidal waters. Colonies are comprised of dense clusters of individual animals (zooids). Their growth can overtake shellfish, limit feeding efficiency, and negatively affect appearance (=marketability) of shellfish. The additional weight of biofouling organisms can make cultivation gear too heavy to service.

Species affected:

- eastern oyster (*Crassostrea virginica*)
- bay scallop (*Argopecten irradians*)
- blue mussel (*Mytilus edulis*)
- marine finfish
- kelp (*Saccharina latissima*)
- *Gracilaria tikvahiae*

Hazard Management:

- Remove biofouling by manually scrubbing or power washing, preferably off-site and on land if possible to avoid/reduce further spread of this organism.
- Periodic rotation of gear – exchange fouled gear with clean or new gear.
- Expose shellfish to air-dry or brine dip as appropriate.
- Expose seaweed to freshwater dip for 15 minutes once per week; increase to twice a week if no difference is seen (freshwater dips will effect growth of seaweed; further analysis is underway).
- Native and invasive tunicates may be vectors of harmful algal species and should not be discarded back to marine waters.

* = non-native

Figure 12a: The pacific rough sea squirt, Styela clava.
Stephan Bullard

Figure 12b: The sea grape, Molgula manhattensis.
Nancy Balcom

Figure 12c: The sea vase, Ciona intestinalis.
Patrick van Moer

SOLITARY ASCIDIANS OR SEA SQUIRTS

Name:

- Pacific rough sea squirt, clubbed tunicate* (*Styela clava*)
- sea grapes* (*Molgula manhattensis*, *Molgula* spp.)
- sea vase* (*Ciona intestinalis*)

Mode of action: Sea squirts attach to aquaculture gear and shellfish and compete for space and food. The additional weight of these biofouling organisms can make cultivation gear too heavy to service.

Species affected:

- eastern oyster (*Crassostrea virginica*)
- bay scallop (*Argopecten irradians*)
- blue mussel (*Mytilus edulis*)
- marine finfish
- kelp (*Saccharina latissima*)
- *Gracilaria tikvahiae*

Hazard Management:

- Remove biofouling by manually scrubbing or power washing, preferably off-site and on land if possible to avoid/reduce further spread of this organism.
- Periodic rotation of gear – exchange fouled gear with clean or new gear.
- Expose shellfish to air-dry or brine dip as appropriate.
- Cover seed (oysters and clams) with fresh water for an hour or so on a routine basis every week (once they are large enough to withstand the treatment) to control *Molgula*.
- Expose seaweed to freshwater dip for 15 minutes once per week; increase to twice a week if no difference is seen (freshwater dips will effect growth of seaweed; further analysis is underway).
- Native and invasive tunicates can be vectors of harmful algal species and should not be discarded back to marine waters.

* = non-native

References and Further Reading:

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

Arakawa, K., 1980. Prevention and removal of fouling on cultured oysters: a handbook for growers. Maine Sea Grant, Technical Report No. 56, Walpole, Maine.

Arens, C.J., Paetzold, S.C., Ramsay, A., Davidson, J., 2011. Pressurized seawater as an antifouling treatment against the colonial tunicates *Botrylloides violaceus* and *Botryllus schlosseri* in mussel aquaculture. Aquatic Invasions. 6, 465–476.

Blum, J.C., Chang, A.L., Liljesthrom, M., Schenk, M.E., Steinberg, M.K., Ruiz, G.M., 2007. The non-native solitary ascidian *Ciona intestinalis* (L.) depresses species richness. Journal of Experimental Marine Biology and Ecology. 342, 5–14.

Boothroyd, F.A., MacNair, N.G., Landry, T., Locke, A., Davidson, T.J., 2002. Dealing with an aquatic invader: the clubbed tunicate (*Styela clava*) in Prince Edward Island waters. Bulletin of the Aquaculture Association of Canada. 102, 98–99.

Bourque, D., Landry, T., Davidson, J., McNair, N., 2003a. Impact of an invasive tunicate in Atlantic Canada: recruitment and competition. Journal of Shellfish Research. 22, 320.

Bourque, D., LeBlanc, A., Landry, T., 2003b. Inter-specific competition between an invasive filter-feeding tunicate (*Styela clava*) and cultivated mussels (*Mytilus edulis*) in Prince Edward Island, Canada. Aquaculture Canada 2003, Victoria, British Columbia.

Bullard, S.G., Whitlatch, R.B., 2004. A guide to the larval and juvenile stages of common Long Island Sound ascidians and bryozoans. Connecticut Sea Grant, Publication No. CTSG-04-07.

Bullard, S., Whitlatch, R.B., Shumway, S.E., Osman, R.W., 2005. Scientists Crying "Foul": Sea Squirts Invade Long Island Sound. Wrack Lines. CTSG-05-01.

Carman, M.R., Allen, H.M., Tyrrell, C., 2009. Limited value of the common periwinkle snail Littorina littorea as a biological control for the invasive tunicate *Didemnum vexillum*. Aquatic Invasions. 4, 291-294.

Carver, C.E., Chisholm, A., Mallet, A.L., 2003. Strategies to mitigate the impact of *Ciona intestinalis* (L.) biofouling on shellfish production. Journal of Shellfish Research. 22, 621-631.

Carver, C.E., Thériault, I., Mallet, A.L., 2010. Infection of cultured eastern oysters Crassostrea virginica by the boring sponge *Cliona celata*, with emphasis on sponge life history and mitigation strategies. Journal of Shellfish Research. 29(4), 905-915.

Concepcion, A., 2010. Managing the Muck: What You Can Do To Control Biofouling. The Dredge Newsletter. Connecticut Sea Grant, Publication No. CTSG-10-07.

Daigle, R.M., Herbinger, C.M., 2009. Ecological interactions between the vase tunicate (*Ciona intestinalis*) and the farmed blue mussel (*Mytilus edulis*) in Nova Scotia, Canada. Aquatic Invasions. 4, 177–187.

DeBrosse, G.A, Allen Jr., S.K., 1993. Control of overset on cultured oysters using brine solutions. Journal of Shellfish Research. 12(1), 29-33.

Dürr, S., Watson, D.I., 2010. Biofouling and anti-fouling in aquaculture, in: Dürr, S., Thomason, J. (Eds.), Biofouling. Blackwell Publishing, Ames, Iowa, pp. 267-287.

Enright, C., 1993. Control of fouling in bivalve aquaculture. World Aquaculture. 24, 44–46.

Finnie, A.A., Williams, D.N., 2010. Paint and coatings technology for the control of marine fouling, in: Dürr, S., Thomason, J. (Eds.), Biofouling. Blackwell Publishing, Ames, Iowa, pp. 185-206.

Fitridge, I., Dempster, T., Guenther, J., de Nys, R., 2012. The impact and control of biofouling in marine aquaculture: a review. Biofouling. 28, 649–669.

Fralick, R.A., 1970. An ecological study of *Codium fragile* subsp. *tomentosoides* in New England. Masters Thesis, University of New Hampshire, Durham.

Getchis, T.L, Rosa, M., Shumway, S.E., 2012. Sea squirts as potential vectors of harmful algal introductions. Connecticut Sea Grant, Publication No. CTSG-12-07.

Hodson, S.L., Lewis, T.E., Burke, S.M., 1997. Biofouling of fish-cage netting: efficacy and problems of *in situ* cleaning. Aquaculture. 152, 77-90.

Howell, D., Behrends, B., 2010. Consequences of antifouling coatings – the chemist's perspective, in: Dürr, S., Thomason, J. (Eds.), Biofouling. Blackwell Publishing, Ames, Iowa, pp. 226-242.

LeBlanc, N., Davidson, J., Tremblay, R., McNiven, M., Landry, T., 2007. The effect of anti-fouling treatments for the clubbed tunicate on the blue mussel, *Mytilus edulis*. Aquaculture. 264, 205-213.

Lordeiros, C., Garcia, N., 2004. The use of sea urchins to control fouling during suspended culture of bivalves. Aquaculture. 231, 293-298.

MacKenzie Jr., C.L., 1970. Causes of oyster spat mortality, condition of oyster setting beds, and recommendations for oyster bed management. Proceedings of the National Shellfisheries Association. 60, 59-67.

MacKenzie Jr., C.L., 1981. Biotic potential and environmental resistance in the American oyster (*Crassostrea virginica*) in Long Island Sound. Aquaculture. 22, 229-268.

MacKenzie Jr., C.L., 2005. Removal of sea lettuce, Ulva spp., in estuaries to improve the environments for invertebrates, fish, wading birds, and eelgrass, *Zostera marina*. Marine Fisheries Review. 67(4), 1-8.

Malinowski, K.C., Ramus, J., 1973. Growth of the green alga *Codium fragile* in a Connecticut estuary. Journal of Phycology. 9, 102–110.

Morse, D., Rice, M.A., 2010. Mussel aquaculture in the Northeast. USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 211.

Osman, R.W., Whitlatch, R.B., Zajac, R.N., 1989. Effects of resident species on recruitment into a community: larval settlement versus post-settlement mortality in the oyster *Crassostrea virginica*. Marine Ecology Progress Series. 54, 61-73.

Paetzold, S.C., Davidson, J., 2010. Viability of golden star tunicate fragments after high-pressure water treatment. Aquaculture. 303, 105-107.

Pederson, J., 2004. Hitchhikers: MIT Sea Grant guide to exotic species. Massachusetts Institute of Technology Sea Grant College Program, Publication No. MTSG-04-3.

Railkin, A.I., 2004. Marine Biofouling: Colonization Process and Defenses. CRC Press, Boca Raton, Florida.

Ramus, J., 1971. *Codium*, the invader. Discovery. 6, 59–68.

Reinhardt, J.F., 2012. Discovering the light bulb tunicate. Wrack Lines. Connecticut Sea Grant, Groton, Connecticut, pp. 9-10.

Rosa, M., Holohan, B.A., Shumway, S.E., Bullard, S.G., Wikfors, G.H., Morton, S., Getchis, T.L., 2013. Biofouling ascidians on aquaculture gear as potential vectors of harmful algal introductions. Harmful Algae. 23,1-7.

Sievers, M., Fitridge, I., Dempster, T., Keough, M.J., 2013. Biofouling leads to reduced shell growth and flesh weight in the cultured mussel *Mytilus galloprovincialis*. Biofouling. 29(1), 97-107.

Switzer, S.E., Therriault, T.W., Dunham, A., Pearce, C.M., 2011. Assessing potential control options for the invasive tunicate *Didemnum vexillum* in shellfish aquaculture. Aquaculture. 318 (1-2), 145-153.

Van Patten, M.P., 2006. Seaweeds of Long Island Sound. Connecticut Sea Grant, Publication No. CTSG-06-01.

Voulvoulis, N., Scrimshaw, M.D., Lester, J.N., 2002. Comparative environmental assessment of biocides used in antifouling paints. Chemosphere. 47, 789–795.

Walton, W., 2005. Pests of concern to New England shellfish growers. Woods Hole Sea Grant/Cape Cod Cooperative Extension, Brochure No. WHOI-H-05-003.

Warburton, F.E., 1958. Control of the boring sponge on oyster beds. Fisheries Research Board of Canada, Progress Report of the Atlantic Coast Station. 69, 7–11.

Watson, D., Shumway, S.E., Whitlatch, R.B., 2009. Biofouling and the shellfish industry, in: Shumway, S.E., Rodrick, G.E. (Eds.), Shellfish Quality and Safety. Woodhead Publishing, Cambridge, United Kingdom, pp. 317-336.

Zajac, R.N., Whitlatch, R.B., Osman, R.W., 1989. Effects of inter-specific density and food supply on survivorship and growth of newly settled benthos. Marine Ecology Progress Series. 56, 127-132.

Potential Shellfish Production Hazards

Environmental Conditions
Biofouling Organisms
Predators
Diseases and Parasites
Invasive Species
Operational Procedures

Predators

Predators are a significant problem with major economic consequences in shellfish aquaculture. Regardless of the species or cultivation practice, any high densities of shellfish will attract predators. Predators may cause damage that can result in an unsightly shell appearance or shortened crop shelf life, as well as significant mortality. Juvenile shellfish and species that cannot fully close their valves (e.g. softshell clams, scallops) are more vulnerable to predators.

Predator avoidance, removal, exclusion, and deterrents are the major methods of control regularly employed by industry. Predator eradication is usually not a viable option for practical and economic reasons, as the cumulative costs over time may outweigh the actual cost of product losses due to predation. Biological control has been utilized in the field, but data on the effectiveness and cost-efficiency of these measures are lacking. Chemical control was proven effective at controlling boring snails or drills; however, it is now known that dispersal of chemicals in the marine environment can have acute and chronic impacts on habitats and non-target species, and many compounds used previously are now banned.

In the hatchery, predators may be introduced via the source water or broodstock. Source water must be properly filtered to avoid the introduction of predators in either algal or shellfish culture vessels. Seed coming into the hatchery should always be accompanied by a health certificate, and should be quarantined and examined for the presence of predators before deploying them in culture vessels. Standard operating procedures should include the sterilization of all culture vessels, equipment, and tools (e.g. sieves, tubing), and proper and frequent hand washing so as to avoid contamination of cultures.

In the nursery and grow-out stages, predators may be introduced with the seed source, may naturally occur at the cultivation site, or may be attracted to the site following planting. While avoiding sites with large numbers of shellfish predators is advisable, it is not always practical.

Shellfish bottom cultivation areas can be prepared using rakes, dredges, or mops to reduce certain predators (e.g. starfish), but predators will likely recolonize cultivation areas and thus necessitate continuous monitoring and removal. In some cases, predator-prey interactions can be avoided by siting cultivation gear off the bottom; however, this will only reduce the effect of benthic dwelling predators. Exclusion devices can prevent further intrusion of mobile predators such as snails. Predator netting can exclude crabs and fish but must be maintained regularly. The predator control method or device chosen should be appropriate for the target culture species. Factors to consider include the mesh material and mesh size, seed size, and time of planting. Generally, planting larger seed (>25mm) later in the season is recommended. Larger animals are less vulnerable to predators such as crabs and snails, and colder temperatures mean lower metabolic rates of predators, and therefore less feeding activity. Purchasing large seed is more costly, as is maintaining shellfish for long durations in containers rather than bottom planting.

Cultivation gear such as cages and bags and devices such as boards and netting are designed to exclude mobile predators. Containment does not, however, guarantee higher survival of the crop. Many shellfish predators such as crabs and starfish begin their life cycle as planktonic larvae and these larvae can settle on or in shellfish cultivation gear, grow rapidly, and prey on juvenile shellfish. Farmers who do not routinely monitor gear and remove small predators increase the risk of significant crop mortality. Effects of large predators such as fish are difficult to prevent. Deterrents such as decoys or motion-detecting alarms can be used to thwart attacks by birds and seals; however, the best deterrent is having a human presence on site.

Potential hazards:

worms
molluscs
crustaceans
echinoderms
birds
finfish
mammals

Farmers should develop and implement an integrated pest and predator management plan that includes: a) the use of prevention and avoidance techniques that have been field-tested; b) routine monitoring and record keeping of gear and animals for the presence of pests, predators, and damage resulting from them; c) tolerance levels for numbers of pests and predators or level of acceptable damage to shellfish.

Figure 1: Stylochus ellipticus *with egg mass.*
Dean Janiak

PLATYHELMINTH WORM

Name:

- flatworm, oyster leech (*Stylochus ellipticus*)

Mode of action: *Stylochus* is a relatively large (up to 25mm) worm. Both juvenile and adult worms attack shellfish. The oyster leech can slide into narrow valve openings and consume shellfish resulting in gaping shells devoid of tissue. *Stylochus* causes extensive mortality especially among juvenile oysters, but can eat oysters as large as 6 cm. Feeding activity decreases markedly below a temperature of 10°C, but *Stylochus* tolerates a wide salinity range.

Species affected:

- eastern oyster (*Crassostrea virginica*)

Hazard Management:

- Regular visual inspection and removal of predators is essential.
- Control flatworms using a freshwater or brine dip, which immediately eliminates the worm.
- Infestations may reoccur, so dips may need to be performed multiple times.

Figure 2: The milky ribbon worm, Cerebratulus lacteus.
Lauren Pudvha

NEMERTEAN WORM

Name:

- milky ribbon worm (*Cerebratulus lacteus*)

Mode of action: The ribbon worm attacks infaunal bivalves by inserting its proboscis into the shell, injects a paralyzing toxin, and then digests the tissue. The presence of gaping shells with tissue may indicate ribbon worm predation. It is believed that juvenile clams are most vulnerable to predation by *Cerebratulus*. Another nemertean, *Malacobdella grossa* is a minor predator. This parasitic worm invades the mantle cavity of bivalve molluscs, especially clams. Peak infestation rates occur in spring, and in offshore rather than near shore areas.

Species affected:

- softshell clam (*Mya arenaria*)

Hazard Management:

- Regular visual inspection and removal of predators is essential.

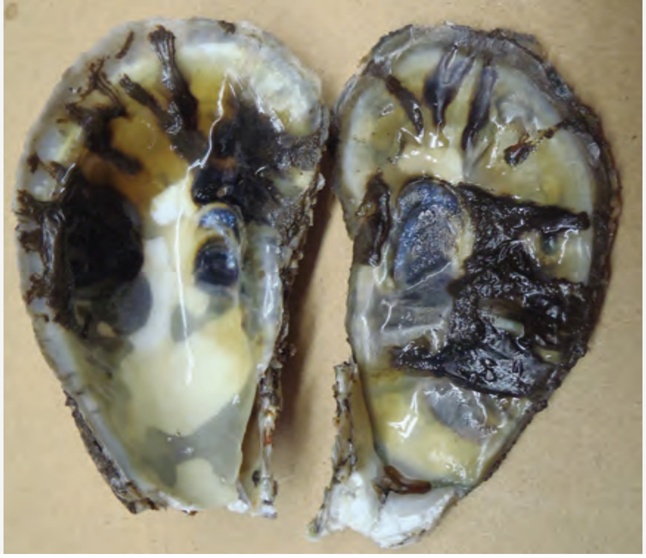


Figure 3a: Mud blisters on oyster shells caused by Polydora *sp.*
Josh Reitsma

Figure 3b: The sand worm, Alitta virens.
Joseph Buttner

POLYCHAETE WORMS

Name:

- mud blister worm (*Polydora websteri*, *P. cornuta*)
- sand worm (*Alitta virens*) - formerly classified as *Nereis virens*
- clam worm (*A. succinea*) – formerly classified as *Neanthes succinea*
- blood worm (*Glycera dibranchiata*)

Mode of action: Metamorphosing *Polydora* larvae can burrow into the shellfish valves. The bivalve repairs the hole, forming a tube referred to as a "blister" that fills with mud or fecal material. This blister results in weakening of the shell, as well as exposure and potential damage to tissue. The presence of mud blisters can weaken the host which tries to cover the burrowing worm in periostracum and nacre. Several worms in one host will cause a marked reduction in Condition Index resulting in slower growth. *Polydora* spp. has been detected in northern quahogs, but in all documented cases the clams were exposed above the sediment surface.

While not major predators, *Alitta virens*, *A. succinea* and *Glycera dibranchiate* may prey on post-metamorphic shellfish.

Species affected:

- eastern oyster (*Crassostrea virginica*)
- softshell clams (*Mya arenaria*)
- bay scallop (*Argopecten irradians*)

Hazard Management:

- Regular visual inspection is essential.
- Submerge oysters in a brine dip lasting 10-15 minutes, followed by at least 15-30 minutes of air drying. Periodic dips may be necessary as new infestations occur.
- Power wash to remove the mud tubes of *P. cornuta*.
- Store oysters in a cold room or cold area (36-38°F) for several weeks to kill worms. This is effective in the late fall and winter when the oysters are conditioned to the cooler temperatures, however, summer treatments may kill the oysters as well.
- Lower planting densities or transplant oysters to a high salinity (>30) area.

Figure 4a: The knobbed whelk consuming an eastern oyster.
Josh Reitsma

Figure 4b: The knobbed whelk, Busycon carica*, consuming a northern quahog.*
Diane Murphy

Figure 4c: The moon snail, Euspira heros*.*
Joseph Buttner

Figure 4d: The rapa whelk, Rapana venosa*, consuming a northern quahog.*
Juli Harding

GASTROPOD MOLLUSC

Name: snails

- knobbed whelk (*Busycon carica*)
- channeled whelk (*Busycotypus canaliculatum*)
- veined rapa whelk* (*Rapana venosa*)
- moon snail (*Neverita duplicatus*)
- moon snail (*Euspira heros*)

Mode of action: An adult whelk pries shellfish valves apart or chips away at the valves, inserts its proboscis, and then digests the soft tissue. The whelks *B. carica* and *B. canaliculatum* are problematic throughout the region. Currently, *R. venosa* is only a major predator south of New England. These species affect mainly quahogs and oysters. The moon snail is a benthic predator that bores a hole through one valve near the shell hinge. These snails consume both seed and adult quahogs and softshell clams.

Species affected:

- northern quahog (*Mercenaria mercenaria*)
- eastern oyster (*Crassostrea virginica*)
- softshell clam (*Mya arenaria*)

Hazard Management:

- Regular visual inspection and removal of predators is essential.
- Use predator exclusion devices (where and when appropriate).
- Remove egg cases.
- Plant hard clams on mud bottom which will greatly reduce predation by Naticid gastropods.

* = non-native

Figure 5a: An oyster drill with a northern quahog. Image shows hole caused by drill predation.
Josh Reitsma

Figure 5b: Egg cases of the oyster drill.
Alison Varian

Figure 5c: Close up of oyster drill egg cases.
Diane Murphy

GASTROPOD MOLLUSC

Name: oyster drills

- Atlantic oyster drill *Urosalpinx cinerea*
- Thick-lip drill *Eupleura caudata*

Mode of action: Drills are benthic predators that bore a small hole in the prey shell. Drills consume both seed and adult shellfish.

Species affected:

- eastern oyster (*Crassostrea virginica*)
- northern quahog (*Mercenaria mercenaria*)
- softshell clam (*Mya arenaria*)
- blue mussel (*Mytilus edulis*)

Hazard Management:

- Regular, visual inspection and removal of predators is essential.
- Drill activity is limited to salinities >12, and temperatures >10°C.
- Seed clams >20mm in shell length to reduce predation in areas where drills are prevalent.
- Plant clams on mud bottom which will reduce predation by drills.
- Remove all drill egg cases from the site.

Figure 6a: The blue crab, Callinectes sapidus.
Tessa Getchis

Figure 6b: The lady crab, Ovalipes ocellatus.
Diane Murphy

Figure 6c: Green crabs are voracious predators of juvenile shellfish, especially eastern oysters.
Tessa Getchis

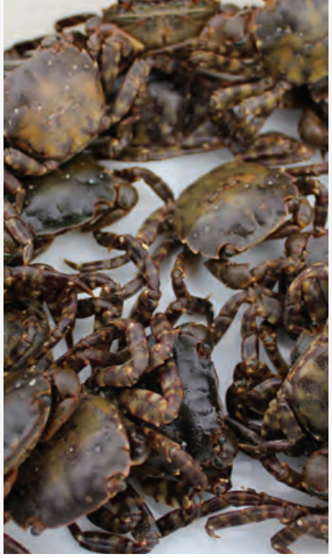


Figure 6d: The Asian shore crab, Hemigrapsus sanguineus.
Kierran Broatch

CRUSTACEAN

Name: crabs

- blue crab (*Callinectes sapidus*)
- lady crab (*Ovalipes ocellatus*)
- green crab* (*Carcinus maenas*)
- Asian shore crab* (*Hemigrapsus sanguineus*)
- Atlantic rock crab (*Cancer irroratus*)
- Jonah crab (*Cancer borealis*)

Mode of action: Mature crabs can crush juvenile shellfish and will attack adult shellfish by chipping away at the valve edge and then prying the valves apart. Feeding activity ceases when temperatures are <13°C. *H. sanguineus* now occupies the niche of another non-native, *C. maenas*, though its affects on species other than *Mytilus* are not documented.

Species affected:

- eastern oyster (*Crassostrea virginica*)
- northern quahog (*Mercenaria mercenaria*)
- softshell clam (*Mya arenaria*)
- bay scallop (*Argopecten irradians*)
- blue mussel (*Mytilus edulis*)

Hazard Management:

- Regular visual inspection and removal of predators is essential.
- Use predator exclusion devices (where and when appropriate). Mesh screens are very effective for controlling these predators if the screens are properly installed and maintained.
- Seed clams >25mm in shell length to reduce predation in areas where swimming crabs (e.g. *C. sapidus*, *O. ocellatus*) are prevalent.
- Seed clams >15 mm in shell length to reduce predation in areas where rock crabs are prevalent.
- Seed oysters >30 to 35 mm in shell length to reduce predation in areas where rock crabs are prevalent.

* = non-native

Figure 7: Mud crabs have large claws that are used to crush shells.
Tessa Getchis

CRUSTACEAN

Name: mud crabs

- *Dyspanopeus sayi*
- *Panopeus herbstii*
- *Rhithropanopeus harrisii*

Mode of action: Mud crabs prey upon small bivalves up to nearly an inch in size. They chip away at or crush the shells with their large, powerful claws. These species are not migratory, tolerating wide temperature and salinity ranges, and thus are a year-round threat to molluscs.

Species affected:

- eastern oyster (*Crassostrea virginica*)
- northern quahog (*Mercenaria mercenaria*)
- softshell clam (*Mya arenaria*)
- bay scallop (*Argopecten irradians*)

Hazard Management:

- Regular visual inspection and removal of predators is essential.
- Seed quahogs >7mm in shell length to reduce predation in areas where mud crabs are prevalent.
- Seed oysters >30 to 35 mm in shell length to reduce predation in areas where rock crabs are prevalent.

Figure 8a: Horseshoe crabs, Limulus polyphemus.
Paige Palmer

Figure 8b: The American lobster, Homarus americanus.
Connecticut Department of Energy and Environmental Protection, Marine Fisheries Division

CRUSTACEAN

Name:

- horseshoe crab (*Limulus polyphemus*)
- American lobster (*Homarus americanus*)

Mode of action: The horseshoe crab, more closely related to spiders than true crabs, preys mainly on benthic infaunal shellfish. They plow through the sediment searching for shellfish and are able to consume both juvenile and adults. Lobster predation is well documented.

Species affected:

- eastern oyster (*Crassostrea virginica*)
- northern quahog (*Mercenaria mercenaria*)
- softshell clam (*Mya arenaria*)
- blue mussel (*Mytilus edulis*)

Hazard Management:

- Regular visual inspection and removal of predators is essential.

Figure 9a: The sea star, Asterias forbesi*, consuming mussels.*
Joseph Buttner

Figure 9b: Starfish mop used to control predation on shellfish beds.
NOAA Fisheries Service

ECHINODERM

Name:

- starfish or sea star (*Asterias forbesi*, *A. rubens*, *A. vulgaris*)

Mode of action: Starfish force open their prey by attaching to the shellfish and prying the shellfish valves apart. They insert their stomach into the shellfish to digest and absorb the shellfish tissue.

Species affected:

- eastern oyster (*Crassostrea virginica*)
- blue mussel (*Mytilus edulis*)
- northern quahog (*Mercenaria mercenaria*)
- bay scallop (*Argopecten irradians*)

Hazard Management:

- Regular visual inspection and removal of predators is essential.
- If oysters and clams are grown in areas where the salinity drops for periods of time, or in areas of high turbidity, starfish will not become a problem.
- For oyster bottom culture, the periodic use of star mops to entangle the sea stars is effective; however, the starfish must afterward be destroyed. This can be achieved by submersion in brine bath.
- Starfish should not be cut up and returned to the water as they can survive and regenerate new arms.
- Where permitted by law, quicklime can be spread over oyster beds to combat starfish.
- Hydrated lime has been used as an immersion treatment against starfish on mussel culture lines.
- Starfish will not tolerate low salinities so sites with freshwater inputs are less prone to predation.
- Mussel seed can be soaked in fresh water overnight or dried in the sun on a hot day to kill starfish.

Figure 10a: The Eider is well-known for adverse impacts to mussel culture operations.
Wikipedia Commons

BIRDS

Name:

- oystercatcher (*Haematopus palliates*)
- gulls (*Larus* spp.)
- red knot (*Calidris canutus*)
- loon (*Gavia* spp.)
- ducks (especially *Somateria mollissima*, *Ana rubripes*, *Aythya* spp., *Mergus* spp., *Oidemia nigra*, *Clangula hyemalis*, *Fulica americana*)

Mode of action: Many seabirds can crush or consume shellfish whole.

Species affected:

- eastern oyster (*Crassostrea virginica*)
- northern quahog (*Mercenaria mercenaria*)
- softshell clam (*Mya arenaria*)
- bay scallop (*Argopecten irradians*)
- blue mussel (*Mytilus edulis*)

Hazard Management:

- Having a person, as well as decoys on site is the best deterrent.
- Other approaches include the use of acoustical visual deterrents such as guns and lasers that can be used both above and under water. They are best used in rotation and combination.
- Use predator exclusion devices such as predator nets (where and when appropriate).
- Eider ducks breed between May and September so seed collection without protection may be carried out during that period[1].

[6]Carter Newell, personal communication

Figure 11: Bottom feeding fish can nip at shellfish siphons.
Tessa Getchis

FINFISH

Name: various fish species including:

- black drum (*Pogonias cromis*) – affects shellfish in southern part of region
- cownose ray (*Rhinoptera bonasus*) – affects shellfish in southern part of region
- summer flounder (*Paralichthys dentatus*)
- northern puffer (*Sphoeroides maculatus*)
- cunner (*Tautogolabrus adspersus*)
- scup, porgy (*Stenotomus chrysops*)

Mode of action: Species such as rays stir up the sediment exposing shellfish and crushing them with their plate-like teeth, while other bottom feeders can nip at siphons or consume juvenile shellfish whole.

Species affected:

- eastern oyster (*Crassostrea virginica*)
- northern quahog (*Mercenaria mercenaria*)
- softshell clam (*Mya arenaria*)
- bay scallops (*Argopecten irradians*)
- blue mussel (*Mytilus edulis*)

Hazard Management:

- Regular visual inspection and removal of predators is essential.
- Exclusion is essential in areas of high predator density.

References and Further Reading:

Arnold, W.S., 1984. The effects of prey size, predator size, and sediment composition on the rate of predation of the blue crab, *Callinectes sapidus* Rathbun, on the hard clam, *Mercenaria mercenaria* (Linné). Journal of Experimental Marine Biology and Ecology. 80(3), 207–219.

Belding, D.L., 1909. A report on the soft-shell clam fishery of Massachusetts including the life history, growth and cultivation of the soft-shell clam (*Mya arenaria*). Commonwealth of Massachusetts Department of Conservation, Division of Fish and Game, Marine Fisheries Service No. 1. (republished in 1916 and 1930).

Belding, D.L., 1910. A report upon the quahaug and oyster fisheries of Massachusetts including the life history, growth and cultivation of the quahaug (*Venus mercenaria*), and observations on the set of oyster spat in Wellfleet Bay. Commonwealth of Massachusetts Department of Conservation, Division of Fish and Game.

Belding, D.L., 1910. The scallop fishery of Massachusetts including an account of the natural history of the common scallop. Commonwealth of Massachusetts Department of Conservation, Division of Fish and Game, Marine Fisheries Service No. 3.

Bisker, R., Castagna, M., 1987. Predation on single spat oysters *Crassostrea virginica* (Gmelin) by blue crabs Callinectes sapidus Rathbun and mud crabs Panopeus herbstii Milne-Edwards. Journal of Shellfish Research. 6, 37-40.

Blundon, J.A., Kennedy, V.S., 1982. Refuges for infaunal bivalves from the blue crab *Callinectes sapidus* (Rathbun). Journal of Experimental Marine Biology and Ecology. 65(1), 67-81.

Botton, M.L., Haskin, H.H., 1984. Distribution and feeding of the horseshoe crab, *Limulus polyphemus*, on the continental shelf off New Jersey. Fishery Bulletin. 82, 383-389.

Bourque, D., Miron, G., Landry, T., 2001. Predation on softshell clams (*Mya arenaria*) by the nemertean *Cerebratulus lacteus* in Atlantic Canada: implications for control measures. Hydrobiologia. 456(1-3), 33-44.

Bower, S.M., 2004. Synopsis of Infectious Diseases and Parasites of Commercially Exploited Shellfish: Shell-boring Polychaetes of Oysters. Department of Fisheries and Oceans Canada.

Carriker, M.R., 1951. Observations on the penetration of tightly closing bivalves by *Busycon* and other predators. Ecology. 32, 73-83.

Carriker, M.R., 1955. Critical review of biology and control of oyster drills *Urosalpinx* and *Eupleura*. U.S. Fish and Wildlife Special Scientific Report–Fisheries 148.

Carriker, M.R., 1981. Shell penetration and feeding by naticacean and muricacean predatory gastropods: a synthesis. Malacologia. 20, 403-422.

Colton, H.S., 1908. How *Fulgar* and *Sycotypus* eat oysters, mussels and clams. Proceedings of the Academy of Natural Sciences of Philadelphia. 60, 3-10.

Commito, J.A., 1982. Effects of *Lunatia heros* predation on population dynamics of *Mya arenaria* and *Macoma balthica* in Maine, USA. Marine Biology. 69, 187-193.

Congleton Jr., W.R., Vassiliev, T., Bayer, R.C., Pearce, B.R., Jacques, J., Gillman, C., 2006. Trends in Maine softshell clam landings. Journal of Shellfish Research. 25(2), 475-480.

Edwards, D.C., 1974. Preferred prey of *Polinices duplicatus* in Cape Cod inlets. Bulletin of the American Malacological Union. 40, 17-20.

Elner, R.W., Lavoie, R.E., 1983. Predation on American oysters (*Crassostrea virginica* [Gmelin]) by American lobsters (*Homarus americanus* [Milne-Edwards]), rock crabs (Cancer irroratus Say), and mud crabs (*Neopanope sayi* [Smith]). Journal of Shellfish Research. 3, 129-134.

Flagg, P.J., Malouf, R.E., 1983. Experimental plantings of juveniles of the hard clam *Mercenaria mercenaria* (L.) in the waters of Long Island, New York. Journal of Shellfish Research. 3, 19-21.

Flimlin, G.E., Beal, B.F., 1993. Major predators of cultured shellfish. USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 180.

Ford, S.E., 2001. Pests, parasites, diseases and defense mechanisms of the hard clam, *Mercenaria mercenaria*, in: Kraeuter, J.M., Castagna, M. (Eds.), Biology of the Hard Clam. Elsevier Science, Amsterdam, pp. 591-649.

Galtsoff, P.S., 1964. The American oyster, *Crassostrea virginica* Gmelin. Fishery Bulletin. 64, 1-480.

Glude, J.B., 1955. The effects of temperature and predators on the abundance of the soft-shell clam, *Mya arenaria*, in New England. Transactions of the American Fisheries Society. 84(1), 13-26.

Gryder, D.K., 2005. Control of mud blister formation in oysters. Virginia Fishery Resource Grant Commission, Final Report No. FRG-00-05.

Haigler, S.A., 1969. Boring mechanism of Polydora websteri inhabiting *Crassostrea virginica*. American Zoologist. 9, 821-828.

Haven, D., Castagna, M., Chanley, P., Wass, M., Whitcomb, J., 1966. Effects of the treatment of an oyster bed with Polystream and Sevin. Chesapeake Science. 7, 179-188.

Haven, D.S., Andrews, J.D., 1957. Survival and growth of *Venus mercenaria*, *Venus campechiensis*, and their hybrids suspended in trays and on natural bottoms. Proceedings of the National Shellfisheries Association. 47, 43-49.

Hoagland, P., Jin, D., 2006. Science and economics in the management of an invasive species. BioScience. 56(11), 931-935.

Jory, D.E., Carriker, M.R., Iverson, E.S., 1984. Preventing predation in molluscan mariculture: an overview. Journal of the World Mariculture Society. 15, 421-432.

Kalin, R.J., 1984. Observations of a feeding method of the Atlantic ribbon worm, *Cerebratulus lacteus*. Estuaries. 7, 179-180.

Karlsson, J.D., 1991. Parasites of the bay scallop, *Argopecten irradians* (Lamarck, 1819), in: Shumway, S.E., Sandifer, P.A. (Eds.), International Compendium of Scallop Biology and Culture. World Aquaculture Society, Baton Rouge, Louisiana, pp. 180-190.

Kent, B.W., 1983. Patterns of coexistence in Busyconine whelks. Journal of Experimental Marine Biology and Ecology. 66, 257-283.

Kraeuter, J.N., 2001. Predators and predation, in: Kraeuter, J.N., Castagna, M. (Eds.), Biology of the Hard Clam. Elsevier, New York, pp. 441-589.

Kraeuter, J.N., Castagna, M., 1980. Effects of large predators on the field culture of the hard clam, *Mercenaria mercenaria*. Fishery Bulletin. 78, 538-541.

Kranz, G.E., Chamberlin, J.V., 1978. Blue crab predation on culchless oyster spat. Proceedings of the National Shellfisheries Association. 68, 38-41.

Landers, W.S., 1954. Notes on predation of the hard clam, *Venus mercenaria*, by the mud crab, *Neopanope texana*. Ecology. 35, 422.

Landers, W.S., Rhodes, E.W., 1970. Some factors influencing predation by the flatworm, *Stylochus ellipticus* (Girard), on oysters. Chesapeake Science. 11(1), 55-60.

Landry, T., Sephton, T.W., Jones, D.A., 1993. Growth and mortality of the northern quahog, *Mercenaria mercenaria* (Linnaeus, 1758) in Prince Edward Island. Journal of Shellfish Research. 12, 321–327.

Leavitt, D., Karney, R., Surier, A., 2010. Biology of the bay scallop. USDA Northeastern Regional Aquaculture Center, Publication No. 213-2010.

Leavitt, D.F., Burt, W.P., 2000. Control of predators of cultured shellfish: exclusion strategies. USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 00-007.

Leibovitz, L., Schott, E.F., Karney, R.C., 1984. Diseases of wild, captive, and cultured scallops. Journal of the World Mariculture Society. 15, 269-283.

Littauer, G., 1990. Avian predators: frightening techniques for reducing bird damage at aquaculture facilities. USDA Southern Regional Aquaculture Center, SRAC Publication No. 401.

Littauer, G.A., Glahn, J.F., Reinhold, D.S., Brunson, M.W., 1997. Control of bird predation at aquaculture facilities: strategies and cost estimates. USDA Southern Regional Aquaculture Center, SRAC Publication No. 402.

Loosanoff, V.L., 1956. Two obscure oyster enemies in New England waters. Science. 123, 1119-1120.

Loosanoff, V.L., 1961. Biology and methods on controlling the starfish, *Asterias forbesi* (Desor). U.S. Fish and Wildlife Service, Fisheries Leaflet No. 520, Washington, D.C.

Loosanoff, V.L., Engle, J.B., 1943. *Polydora* in oysters suspended in the water. Biological Bulletin. 85(1), 69-78.

Loosanoff, V.L., MacKenzie Jr., C.L., Shearer, L.W., 1960. Use of chemicals to control shellfish predators. Science. 131, 1522-1523.

MacKenzie Jr., C.L., 1970a. Oyster culture in Long Island Sound 1966-69. Commercial Fisheries Review. 32(1), 27-40.

MacKenzie Jr., C.L., 1970b. Causes of oyster spat mortality, condition of oyster setting beds, and recommendations for oyster bed management. Proceedings of the National Shellfisheries Association. 60, 59-67.

MacKenzie Jr., C.L., 1970c. Control of oyster drills, *Eupleura caudata* and *Urosalpinx cinerea*, with the chemical Polystream. Fishery Bulletin. 68(2), 285-297.

MacKenzie Jr., C.L., 1977. Predation on hard clam (*Mercenaria mercenaria*) populations. Transactions of the American Fisheries Society. 106(6), 530-537.

MacKenzie Jr., C.L., 1981. Biotic potential and environmental resistance in the American oyster (*Crassostrea virginica*) in Long Island. Aquaculture. 22, 229-268.

MacKenzie, C.L., Jr., 1970. Causes of oyster spat mortality, condition of oyster setting beds, and recommendations for oyster bed management. Proceedings of the National Shellfisheries Association. 60, 59-67.

MacKenzie, C.L., Jr., Pikanowski, R., 1999. A decline in starfish, *Asterias forbesi*, abundance and a concurrent increase in northern quahog, *Mercenaria mercenaria*, abundance and landings in the northeastern United States. Marine Fisheries Review. 61(2), 66-71.

MacKenzie, C.L., Shearer, L.W., 1959. Chemical control of *Polydora websteri* and other annelids inhabiting oyster shells. Proceedings of the National Shellfisheries Association. 50, 105-111.

Mackin, J.G., 1959. Mortalities of oysters. Proceedings of the National Shellfisheries Association. 50, 105-111.

MacPhail, J.S., Lord, E.I., Dickie, L.M., 1955. The green crab—a new clam enemy. Atlantic Progress Reports. 63, 3–12.

Malinowski, S.M., 1985. The population ecology of the hard clam, *Mercenaria mercenaria*, in eastern Long Island Sound. Ph.D. Dissertation, University of Connecticut.

Malinowski, S.M., Whitlatch, R.B., 1984. Natural survivorship of young hard clams, *Mercenaria mercenaria*, in eastern Long Island Sound. Ph.D. Dissertation, University of Connecticut.

Martin, A.C., Uhler, F.M., 1939. Food of game ducks in the United States and Canada. U.S. Department of Agriculture, Technical Bulletin No. 634.

McDermott, J.J., Fowler, F.B., 1951. Preliminary studies of the common mud crabs on oyster beds of Delaware Bay. National Shellfisheries Association Convention Address 1952, pp. 147-150.

McGladdery, S.E., Bower, S.M., Getchell, R.G. 2006. Diseases and parasites of scallops, in: Shumway, S.E., Parsons, G.J. (Eds.), Scallops: Biology, Ecology, and Culture, Second Edition. Elsevier, Developments in Aquaculture and Fisheries Science No. 35, Amsterdam, The Netherlands, pp. 595-639.

Medcof, J.C., Thurber, L.W., 1958. Trial control of the greater clam drill (*Lunatia heros*) by manual collection. Journal of the Fisheries Research Board of Canada. 15, 1355-1369.

Meire, P., 1993. The impact of bird predation on marine and estuarine bivalve populations: a selective review of patterns and underlying causes, in: Dame, R.F. (Ed.), Bivalve Filter Feeders in Estuarine and Coastal Ecosystem Processes. Springer, New York, pp. 197-243

Merriner, J.V., Smith, J.W., 1979. Gear feasibility study for the cownose ray, *Rhinoptera bonasus*. Virginia Institute of Marine Science, Special Reports in Applied Marine Science and Ocean Engineering No. 227.

Morgan, D.E., Goodsell, J., Matthiessen, G.C., Garey, J., Jacobsen, P., 1980. Release of hatchery-reared bay scallops (*Argopecten irradians*) onto a shallow coastal bottom in Waterford, Connecticut. Proceedings of the World Mariculture Society. 11, 247-261.

Morse, D., Rice, M.A., 2010. Mussel aquaculture in the northeast. USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 211.

Moulton, J.M., Gustafson, A.H., 1955. Green crabs and the redistribution of quahogs. Science. 123, 992.

Murton, R.K., Wright, E.N., 1968. The Problem of Birds as Pests. Academic Press, London.

Ordzie, C.J., Garofalo, G.C., 1980a. Behavioral recognition of molluscan and echinoderm predators by the bay scallop, *Argopecten irradians* (Lamark), at two temperatures. Journal of Experimental Marine Biology and Ecology. 43, 29-37.

Ordzie, C.J., Garofalo, G.C., 1980b. Predation, attack success, and attraction to the bay scallop, *Argopecten irradians* (Lamark) by the oyster drill, *Urosalpinx cinerea* (Say). Journal of Experimental Marine Biology and Ecology. 47, 95-100.

Peterson, C.H., 1982. Clam predation by whelks (*Busycon* spp.): experimental tests of the importance of prey size, prey density, and seagrass cover. Marine Biology. 66, 159-170.

Peterson, C.H., Fodrie, F.J., Summerson, H.C., Powers, S.P., 2001. Site-specific and density-dependent extinction of prey by schooling rays: generation of a population sink in top-quality habitat for bay scallops. Oecologia. 129, 349-356.

Pohle, D.G., Bricelj, V.M., Garcia-Esquivel, Z., 1991. The eelgrass canopy: an above-bottom refuge from benthic predators for juvenile bay scallops *Argopecten irradians*. Marine Ecology Progress Series. 74, 47-59.

Prince Edward Island Department of Fisheries, Aquaculture and Environment, 2001. Removal of marine worms from the surface of oyster shells using salt brine. Prince Edward Island Aqua Info Aquaculture Notes, No. 03-2001.

Provenzano Jr., A.J., 1959. Effects of the flatworm *Stylochus ellipticus* (Girard) on oyster spat in two salt water ponds in Massachusetts. Proceedings of the National Shellfisheries Association. 50, 83-88.

Ropes, J.W., 1988. The feeding habits of five crab species at Pettaquamscutt River, Rhode Island. Fishery Bulletin. 87, 197-204.

Ross, B.P., Lien, J., Furness, R.W., 2001. Use of underwater playback to reduce the impact of eiders on mussel farms. ICES Journal of Marine Science. 58, 517-524.

Rowell, T.W., Woo, P., 1990. Predation by the nemertean worm, *Cerebratulus lacteus* Verill, on the soft-shell clam, *Mya arenaria* Linneaus, 1758, and its apparent role in the destruction of a clam flat. Journal of Shellfish Research. 9, 291-297.

Russell, H.J., 1973. An experimental seed bay scallop stocking of selected Rhode Island waters. Rhode Island Division of Fish and Wildlife.

Shearer, L.W., MacKenzie Jr., C.L., 1959. The effects of salt solutions of different strengths on oyster enemies. Proceedings of the National Shellfisheries Association. 50, 97-103.

Shuster Jr., C.N., 1950. Observations on the natural history of the American horseshoe crab, *Limulus polyphemus*, in: Turner Jr., H.J. (Ed.) Third report on investigations of methods of improving the shellfish resources of Massachusetts. Department of Conservation, Commonwealth of Massachusetts, pp. 18-23.

Smith, O., Chin, E., 1951. The effects of predation on soft clams, *Mya arenaria*. National Shellfisheries Association Convention Address, pp. 37-44.

Smith, O.R., 1953. Notes on the ability of the horseshoe crab, *Limulus polyphemus* to locate soft-shell clams, *Mya arenaria*. Ecology. 34, 636-637.

Smith, O.R., Baptist, J.P., Chin, E., 1955. Experimental farming of the soft-shell clam, *Mya arenaria*, in Massachusetts, 1949-1953. Commercial Fisheries Review. 17(6), 1-16.

Stickney, A.P., Stringer, L.D., 1957. A study on the invertebrate bottom fauna of Greenwich Bay, Rhode Island. Ecology. 38, 111-122.

Strieb, M.D., Bricelj, V.M., Bauer, S.I., 1995. Population biology of the mud crab, *Dyspanopeus sayi*, and important predator of juvenile bay scallops in Long Island (USA) eelgrass beds. Journal of Shellfish Research. 14, 347-357.

Turner Jr., H.J., 1948. Report on investigations of the propagation of the soft-shell clam, *Mya arenaria*. Woods Hole Oceanographic Institution. Cont. 462, Woods Hole, Massachusetts, pp. 3-10.

Turner Jr., H.J., 1950. Investigations on the soft-shell clam, *Mya arenaria*, in: Turner Jr., H.J. (Ed.), Third report on investigations of methods of improving shellfish resources of Massachusetts. Department of Conservation, Commonwealth of Massachusetts, pp. 5-15.

Turner Jr., H.J., Hanks, J.E., 1959. Infestation of *Pecten irradians* by Polydora. Nautilus. 72(4), 109-111.

Virnstein, R.W., 1977. The importance of predation by crabs and fishes on benthic infauna in Chesapeake Bay. Ecology. 58: 1199-1217.

Walton, W., 2005. Predators of concern to New England shellfish growers. Woods Hole Sea Grant/Cape Cod Cooperative Extension, Brochure No. WHOI-H-05-003.

Warren, S., 1916. The feeding habits of *Busycon*. Nautilus. 30, 66-68.

Webster, J.R., Medford, R.Z., 1959. Flatworm distribution and associated oyster mortality in Chesapeake Bay. Proceedings of the National Shellfisheries Association. 50, 89-95.

Weston, S., Buttner, J.K., 2010. Softshell clam culture: basic biology and general culture considerations. USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 201.

Whetstone, J.M., Eversole, A.G., 1978. Predation on quahogs, *Mercenaria mercenaria*, by mud crab, *Panopeus herbstii*. Proceedings of the National Shellfisheries Association. 68, 42-48.

White, M.E., Wilson, E.A., 1996. Predators, pests, and competitors, in: Kennedy, V.S., Newell, R.I.E., Eble, A.F. (Eds.), The eastern oyster *Crassostrea virginica*. Maryland Sea Grant College, University of Maryland System, College Park, Maryland, pp. 559-575.

Wiltse, W., 1978. Effects of predation by *Polinices duplicatus* on community structure. Ph.D. Dissertation, University of Massachusetts, Amherst.

Woelke, C.E., 1957. The flatworm, *Pseudostylochus ostreophagus* Hyman, a predator of oysters. Proceedings of the National Shellfisheries Association. 47, 62-67.

Wood, L., Robert, B.A., 1963. Differentiation of effects of two pesticides upon *Urosalpinx cinerea* Say from the Eastern Shore of Virginia. Proceedings of the National Shellfisheries Association. 54, 75-8.

Zachary, A., Haven, D.S., 1973. Survival and activity of the oyster drill *Urosalpinx cinerea* under conditions of fluctuating salinity. Marine Biology. 22, 45-52.

USDA, 1997. Bird predation and its control at aquaculture facilities in the northeastern United States. USDA Animal and Plant Health Inspection Service No. 11-55-009.

Potential Shellfish Production Hazards

Environmental Conditions
Biofouling Organisms
Predators
Diseases and Parasites
Invasive Species
Operational Procedures

Diseases and Parasites

Bivalves are susceptible to infections from organisms and to metabolic processes that cause disease and other physiological responses. Several reviews on the effects of diseases and parasites on cultivated shellfish have been published and are referenced at the end of this chapter.

Disease in all animals is the result of the interactions between the host tissues (the bivalve), the infectious agent (parasites, bacteria, viruses, fungi, etc.), and the environment (marine or fresh waters, intertidal or subtidal) in which both the infected and infective organisms live. Bivalves are especially affected by environmental conditions because they are ectothermic and thus, unlike mammals and birds, cannot control their body temperature. The innate immune system of a bivalve works most effectively at the same temperature range at which the bivalve grows best. Temperatures higher or lower than those that produce the best growth are also less effective at promoting innate immune responses to infectious agents. Changes in environmental factors such as temperature or salinity can either promote or inhibit infection and proliferation of the disease-causing agent in the host.

Listed here are several important diseases, parasites, and pests of commercially important bivalves in the northeastern U.S. Ongoing research is improving detection methods and understanding of bivalve disease, but unfortunately, there is still much that is unknown about infectious agents, parasites, and pests and the bivalve response to these organisms.

Signs of disease are difficult to detect before mortalities begin, so careful attention should be paid to reductions in growth and other abnormalities. Routine testing can help and the sooner a disease issue is identified the more likely something can be done to minimize or eliminate losses. Most diagnoses require complex technical protocols involving microscopic examination of histological samples of preserved tissues, microbiological assays for pathogens, or molecular genetic tests (e.g., PCR). Therefore, any increase in mortality should immediately signal the farmer to contact an aquatic health professional (Appendix 3).

Few treatment methods are available; therefore prevention is key to avoiding disease hazards. It is critical that prior to being moved among bodies of water, shellfish be screened for disease. Contact the aquatic animal health professional to determine laws and policies regarding the import/export of shellfish..

Potential hazards:

Infectious diseases caused by protozoans, bacteria and viruses
Non-infectious diseases caused by genetic disorders or unknown causes
Parasites (e.g. trematodes, pea crabs)

Diseases Listed by Species

General

- Vibriosis

Eastern oyster (*Crassostrea virginica*):

- Dermo
- MSX (Multinucleated Sphere Unknown)
- SSO (Seaside organism)
- *Bonamia exitosa*
- Xenomas
- ROD (Roseobacterium oyster disease)
- Oyster Herpes Virus
- Digenean trematodes
- Pea crabs

Northern quahog (*Mercenaria mercenaria*):

- QPX (Quahog Parasite Unknown)
- Hard clam neoplasia
- Gonadal neoplasia
- Pea crabs

Bay scallop (*Argopecten irradians*):

- Hinge ligament disease
- Chlamydiosis
- Pea crabs

Softshell clam (*Mya arenaria*):

- Clam *Perkinsus*
- Hemocytic neoplasia (=disseminated sarcoma)
- Gonadal neoplasia

Blue mussel (*Mytilus edulis*):

- Mussel egg disease
- Hemocytic neoplasia (=disseminated sarcoma)
- Digenean trematodes
- Pea crabs

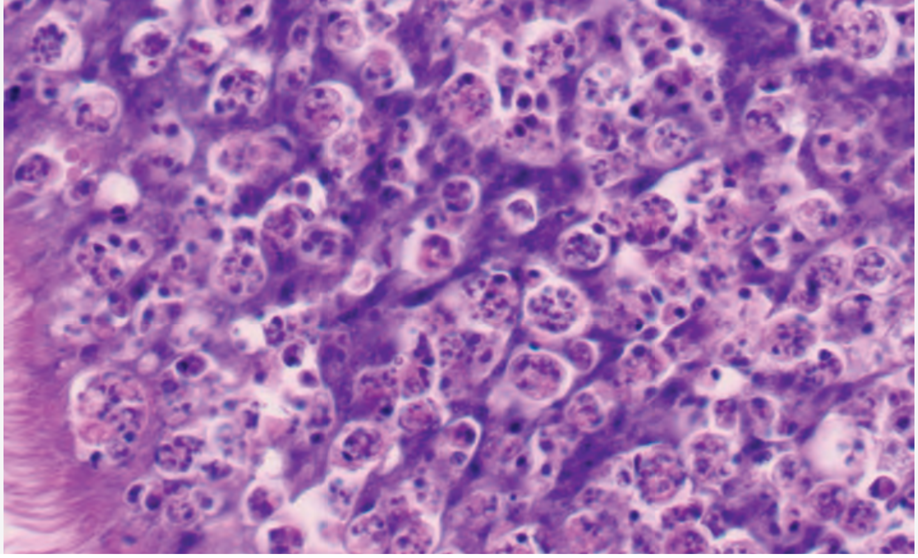


Figure 1: Photomicrograph of a 6 μm section of paraffin embedded gastric mucosa (stomach epithelium) stained with hematoxylin and eosin stain. Abundant Dermo organisms are present in hemocytes migrating between and around epithelial cells of mucosa.
Roxanna Smolowitz

DERMO (*PERKINSUS MARINUS*)

Method of infection: The parasite enters the oyster primarily through the digestive gland epithelium but also through the mantle and gills. The parasite is phagocytized by circulating hemocytes (blood cells of the oyster) and is able to proliferate in the hemocyte, first forming unicellular trophonts and eventually forming meronts. Lysis of hemocytes occurs releasing *Perkinsus* organisms into the surrounding tissues. *Perkinsus* forms infect other hemocytes or float in the hemolymph.

Gross (observable) signs of disease: Severely diseased animals may show thin watery tissues upon shucking. No other observable signs are usually noted.

Microscopic Signs of Disease: Infected hemocytes are present either within the gastric and digestive gland epithelium or circulating in throughout the oyster's body. Disease caused by the infection can result in gut epithelial ulceration/digestive gland necrosis. Hemocyte proliferation is noted in early stages of disease, but the numbers of hemocytes decrease in end stage of disease. Degeneration and atrophy of all tissues is noted in moderately to severely affected individuals resulting in death of the animal. Death of the oyster lags behind infection and occurs approximately 8 months to 1.5 years post-infection. Highest mortality in northeastern U.S. populations of eastern oysters is usually noted in the September/October time period.

Method of Transmission: Dermo organisms are directly infective. Infectious organisms released from dead oysters are spread through the water column to actively feeding oysters. Live oysters can also shed infectious organisms in the feces, which are carried through the water column to another oyster or may drift to the adjacent sediment. The Dermo organisms can be carried from one oyster to another by way of oyster drills.

Potentiating environmental conditions:

- Temperature: 18 to 30°C
- Salinity: 12 to 32 (but does not die at lower salinities)
- Other: Increased oyster stress may increase the severity of the disease.

Hazard Management:

- Maintain animals at low salinity through the first summer.
- Use tolerant/resistant seed.
- Use seed free of infection.
- Do not move infected animals from areas of high infection rates/severities to areas of low infection rates/severities.
- If necessary, quarantine and destroy infected animals.
- Cure shell used as cultch for at least three months to eliminate Dermo in attached tissues.

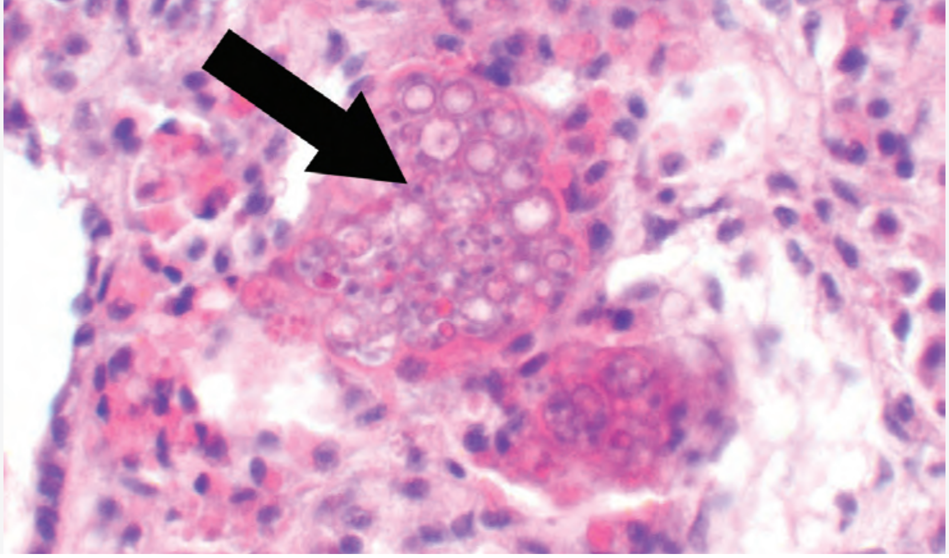


Figure 2: Photomicrograph of a 6 μm section of paraffin embedded tissue with hematoxylin and eosin stain. Numerous hemocytes are attempting to encapsulate the aggregations of parasites.
Roxanna Smolowitz

CLAM PERKINSUS (*PERKINSUS CHESAPEAKI*)

Method of Infection: Suspected to infect the gill primarily via flagellated zoospores, but can be found in connective tissues throughout the body. The disease presents as a mild infection in most animals and currently has been identified in softshell clams in the Chesapeake Bay, in Delaware Bay, and in some other species.

Gross (observable) Signs of Disease: Slight nodular white swelling may be noted in the gill lamellae or palps.

Microscopic Signs of Disease: Aggregations of parasites are surrounded by a homogenous eosinophilic parasitic secretion. Hemocytes surround and encapsulate the foci forming small nodules in the tissue. Moderate to severe infections result in fusion of gills and destruction of the infected gill and other infected organ tissues.

Method of Transmission: Zooporulation is noted in the dead tissues and in culture, and zoospores are suspected to transfer the infection to naive clams.

Potentiating environmental conditions:

- Temperature: Peak prevalence occurs in the fall
- Salinity: 7 to 25

Hazard Management:

- Do not import softshell clams from the Chesapeake Bay.
- Test seed and adults before movement to another body of water.

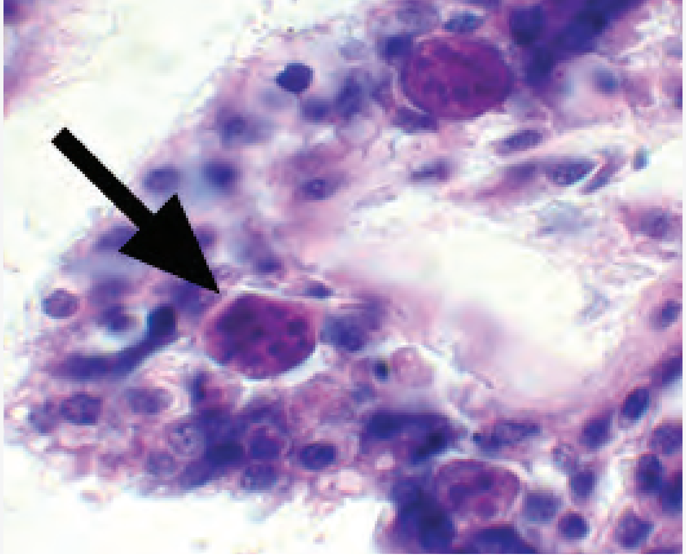


Figure 3: Photomicrograph of a 6 µm section of paraffin embedded gill stained with hematoxylin and eosin stain. MSX plasmodia (arrow) are present between epithelial cells of the gill. **Roxanna Smolowitz**

MSX, MULTINUCLEATED SPHERE UNKNOWN (*HAPLOSPORIDIUM NELSONI*)

Method of infection: Uninucleate forms lodge between epithelial cells of the gill (R. Smolowitz, personal observation), develop into plasmodia (multinucleated cells), then invade the vascular system of the oyster. Plasmodia replicate within the vascular system of the animal and use nutrients that would normally be used by the oyster tissues. Hemocytes (blood cells) do not appear able to effectively destroy the MSX organisms. Highest mortality in the northeastern U.S. populations of oysters is usually noted in the August/September time period.

Gross (observable) signs of disease: Severely diseased (moribund) animals show thin watery tissues upon shucking. No other observable signs are usually noted.

Microscopic Signs of Disease: Early in the infection, plasmodia are noted in the epithelium or sinusoids (vascular system) of the gill. In mid to late stage infections, plasmodia can be found throughout the body's vascular system. In early infections a moderate to severe hemocytic response is noted. Tissue atrophy and degeneration accompanied by a lack of hemocyte response is usually noted in tissues in late stages of the disease.

Method of Transmission: It is widely accepted that the infectious agent is transferred to oysters from another host that has yet to be identified (often termed an intermediate host). In juvenile oysters, less than 18 months old and less than 42 mm in shell height, sporulation of plasmodia can occur in the digestive gland epithelium. Observed occurrences of sporulation (noted histologically) are sporadic and low-level mortality of juvenile oysters has been associated with sporulation. Several studies have shown that spores are not infectious for other oysters; supporting the notion that the parasite needs a second host. In two-host infections, the organism must undergo specific life stages in one host before it becomes infective for the other host. This type of infection results in two major patterns of disease. First, infection and resulting disease can be sporadic; present in one year and not in the next. Second, the disease can be present every year, but can vary greatly in prevalence and severity from year to year. Work continues on identifying the other host(s) in the life cycle. Interestingly, molecular information indicates the parasite was introduced to the east coast with *C. gigas* oysters from another location in the U.S. or a foreign country.

Potentiating environmental conditions:

- Temperature: 5 to 20°C
- Salinity: 10 to 35 (infectious organisms die at lower salinities)
- Presence of the unidentified, infected, intermediate host in the surrounding waters/sediments

Hazard Management:

- Maintain animals at low salinity through the first summer.
- Use tolerant/resistant seed.
- Use seed free of infection.
- Do not move infected animals
- Hold animals at low salinities and high temperature for a period of time to reduce the parasite load.

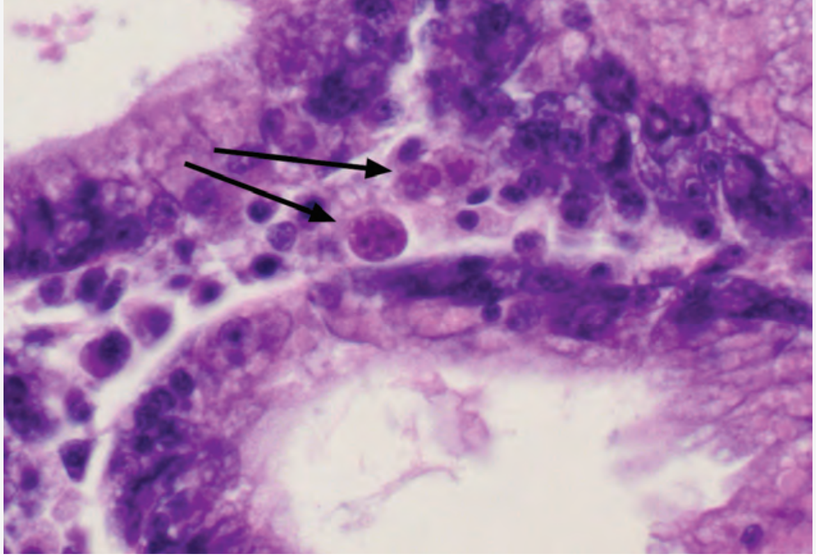


Figure 4: Photomicrograph of a 6 μm section of paraffin embedded tissue with hematoxylin and eosin stain. SSO plasmodia are present in the sinusoidal connective tissues between adjacent tubules of the digestive gland of this eastern oyster.
Roxanna Smolowitz

SSO, SEASIDE ORGANISM (*HAPLOSPORIDIUM COSTALE*)

Method of infection: Infectious agents (spores) invade through the gastric and digestive gland epithelium (Roxanna Smolowitz, unpublished data). Plasmodia (multinucleate cells) are first identified in sinusoids underlying the gastrointestinal tract and subsequently proliferate throughout the sinusoids of the body.

Gross (observable) signs of disease: Severely diseased (moribund) animals show thin watery tissues upon shucking. "graininess" of the watery oyster tissues has also been reported. SSO rarely causes mortality > 30% of the animals in a population, but it can occasionally cause higher mortality, and in animals that do not die it can result in growth checks during the late spring.

Microscopic Signs of Disease: Plasmodia are identified in sinusoids surrounding the gastric and digestive gland tissue early in the infection. As the disease progresses, plasmodia are found in sinusoids throughout the body. Hemocytes (blood cells) respond to the invading plasmodia, but the inflammatory reaction does not appear to adequately prevent proliferation of the plasmodia and spread throughout the body. Plasmodia increase in numbers in the early to late spring in individual animals. In intense infections before sporulation occurs, connective tissue atrophy, digestive gland atrophy/necrosis, and deceased numbers of hemocytes in the tissues are noted. Plasmodia within the vascular system undergo synchronous sporulation in the May/June time period. During synchronous sporulation, all plasmodia throughout the body form sporangia containing developing spores. Mature spores are released into the tissues from the sporangia upon death of the animal. After sporulation, in surviving animals, moderate to high numbers of residual (brown cells) are noted in the connective tissues and no plasmodia or spores/sporangia can be identified. In light infections, the oyster appears to be able to survive the sporulation. In heavy infections, the oyster dies before or during sporulation. Plasmodia, genetically identified as SSO have also been observed to increase in number in the tissues and undergo sporulation in the fall of the year in Connecticut and Massachusetts. Whether this new time period for disease is a result of a new strain of SSO or is a result of changes in the environment is unknown.

Method of Transmission: Method of transmission is unknown, but it is probable that an intermediate host is necessary for the life cycle. This hypothesis is based on the lack of infection in the tissue post-sporulation and variable amounts of time (often 6 to 8 weeks) that occur before plasmodia can again be found again in the tissues using both molecular and histological identification of plasmodia. A potential intermediate host has not been determined.

Potentiating environmental conditions:

- Temperature: proliferates in tissues of oysters during early to late spring in the northeastern U.S.
- Salinity: 28 and greater
- Presence of an unknown intermediate host

Hazard Management:

- Use seed free of infection.
- Do not move infected animals.
- Hold animals at low salinities for a period of time to reduce the parasite load.

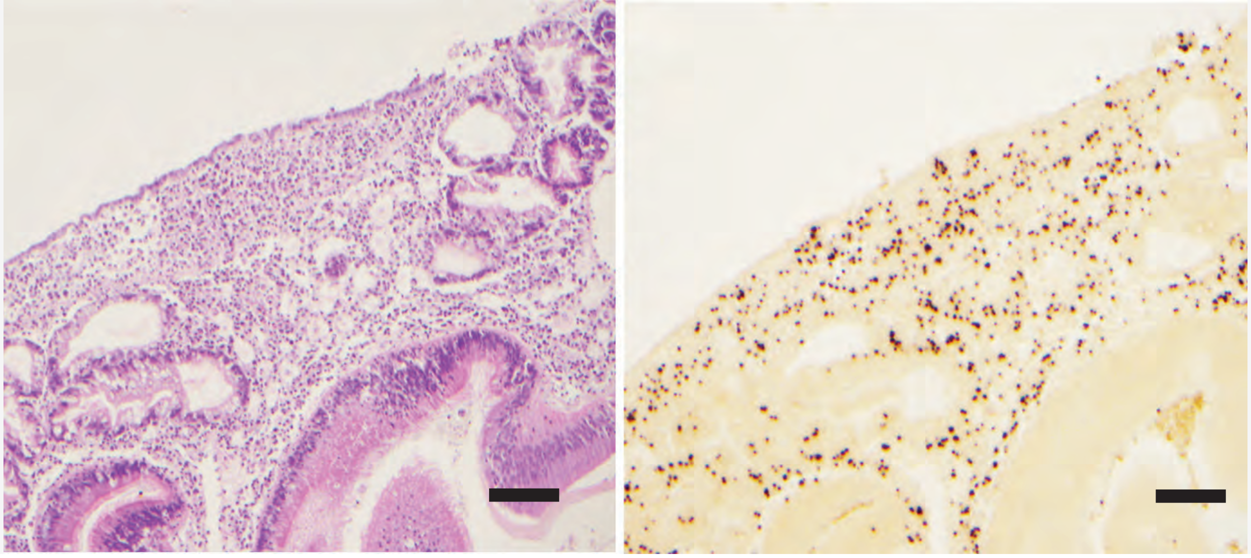


Figure 5: Heavy **Bonamia exitiosa** *infection of* **Crassostrea virginica** *from North Carolina. A. Standard histology, revealing intense hemocytosis throughout the visceral mass and disruption of oyster connective tissues. B. In situ hybridization to the same section using* **B. exitiosa**-*specific DNA probes. Tiny dark spots represent probe hybridization to abundant individual* **Bonamia** *cells. Scale = 50 microns.*
Nancy Stokes

BONAMIA EXITIOSA

Bonamia exitiosa infections were detected at high prevalence (> 90%) in *Crassostrea virginica* oyster seed from a hatchery in North Carolina in 2012. In 2013, there was a similar report of infection in nursery seed from Cape Cod. These are the first reports of *B. exitiosa* affecting *C. virginica*. *Bonamia* species are protistan parasites that can cause lethal infections in some species of oysters. These were the first instances that any *Bonamia* infections were confirmed in eastern oysters[1]. The parasite is also known to infect the native crested oyster, *Ostrea equestris* or *stentina*, in Atlantic coast waters south of Cape Hatteras. Experimental work in North Carolina with the non-native *Crassostrea ariakensis* in the 2000s revealed this oyster to be highly susceptible. Eastern oysters appear to be relatively resistant to *Bonamia*, though heavy infections have occasionally been reported in some individuals.

Method of infection: Infections are presumably acquired through oyster gill, mantle, or gut (possibly all three) during filtration and feeding.The pathology is unclear. Some infections reach high intensities in *C. virginica* (Figure 5), suggesting that a reduction in growth and condition, and possibly some mortality, could result, based on current knowledge of *Bonamia* effects on other oyster hosts. These effects have not been documented thus far in *C. virginica*, which is regarded as relatively resistant to *Bonamia*.

Gross (observable) signs of disease: None.

Microscopic Signs of Disease: Elevated levels of circulating oyster hemocytes (blood cells), particularly in infected organs and tissues: around the gut and digestive gland, or in the gills or mantle. Tiny *B. exitiosa* "microcells" (just 2-3 microns in size) are observed in and among oyster hemocytes that they infect.

Method of Transmission:

- *B. exitiosa* is directly transmissible from oyster to oyster and may be transmissible from or to other host species.
- Potentiating environmental conditions:
- *B. exitiosa* is most pathogenic under euhaline conditions, where salinity exceeds 30. It rarely detected in natural systems at lower salinities. Pathogenicity in *C. ariakensis* in experimental treatments was sharply reduced below salinity of 20. In *C. ariakensis* in North Carolina, *B. exitiosa* was a pathogen of the warmer summer months, falling to very low levels in the winter. Its seasonal cycle in *C. virginica* remains unknown.

Hazard Management:

- Because the impact of *B. exitiosa* on *C. virginica* (as well as other species) is uncertain, care must be taken to avoid spreading it, particularly to areas where it is not known to exist.
- Given the present uncertainty about this parasite's distribution, it is strongly recommended that pathologists specifically include screening for *Bonamia* as a matter of routine examination before health certifications are issued for oyster transfers.

[1]Ryan Carnegie and David Bushek, 2013, personal communication

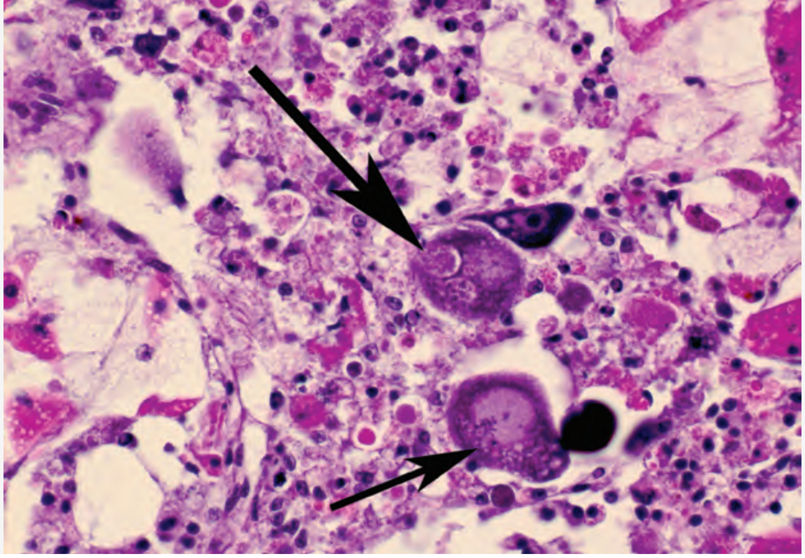


Figure 6: Photomicrograph of a 6 µm section of paraffin embedded blue mussel mantle stained with hematoxylin and eosin stain. Two different stages of Steinhausia *sp. microsporidia are noted in an egg (large and small arrows) within the disrupted and inflamed gonadal tubule.*
Roxanna Smolowitz

MUSSEL EGG DISEASE, *STEINHAUSIA MYTILOVUM*

Method of infection: unknown

Gross (observable) signs of disease: pinpoint, white pits in the mantle

Microscopic signs of disease: localized to diffuse infection and destruction of eggs in the gonadal tubules of the bivalve causing parasitic castration. If inflammation is moderate to severe, tubules can be destroyed and hemocytic inflammation and tissue necrosis will extend into surrounding connective tissues.

Method of transmission: Transmission may occur when loose spores are released along with intact eggs or through hemocytic phagocytosis and subsequent diapediasis of hemocytes into the water column from the animal's tissues.

Potentiating environmental conditions:

- Temperature: an annual cycle appears to occur; and infection may be more common at lower temperatures;

Hazard Management:

- Do not relocate infected shellfish.

Figure 7a,b,c: Gross examination of oyster gill reveals multiple xenomas of varying sizes (a, arrows) as well as scalloped gills (b, arrows). C is a histological section showing water tube occluded by xenomas

Emily Scarpa McGurk

XENOMAS

Species Affected:

- most species, but only abundant in eastern oysters *Crassostrea virginica* from Great Bay, New Hampshire

Method of infection: Xenomas are accumulations of greatly enlarged epithelial cells in water tubules of the gills. Enlargement is caused by ciliates that invade the epithelium and proliferate with the cell causing its hypertrophy (enlargement). Enlarged cells are sloughed into the water tubule and many together can be seen as white foci in the gills. Ciliates causing xenomas in bivalve mollusks belong to the genus *Sphenophyra*.

Gross (observable) signs of disease: In heavily affected individuals, xenomas appear as white nodules of varying size located in the gill tissue (Figure 7a), measuring up to 3 mm in diameter. In many cases, scalloped or otherwise damaged gills may be observed in oysters with either no visible xenomas or xenomas located along the outermost edge of the gill (Figure 7b).

Microscopic Signs of Disease: In histological sections, xenomas are most often located in the gill water tubes. In heavy infestations, they can be large enough to occupy the entire cross sectional area of water tubes and may interfere with filtration (Figure 7c). The number of ciliates within any single xenoma varies from a few to thousands and the diameter of xenomas ranges from about 30 to 3,000 µm.

Method of Transmission: The infectious agents are considered opportunistic and presumably transmitted directly through the water column as oysters feed.

Potentiating environmental conditions:

- Temperature: infections tend to increase from spring to fall
- Salinity: all salinities in which oyster grow
- Other: heavy infestations are routinely documented in oysters from Great Bay, New Hampshire, but are rare elsewhere. Prevalence tends to be greatest in oysters 70-90 mm.

Hazard Management:

- Avoid moving oysters from Great Bay, New Hampshire to other bodies of water.

Figure 8a: High mortality associated with QPX infection.
Roxanna Smolowitz

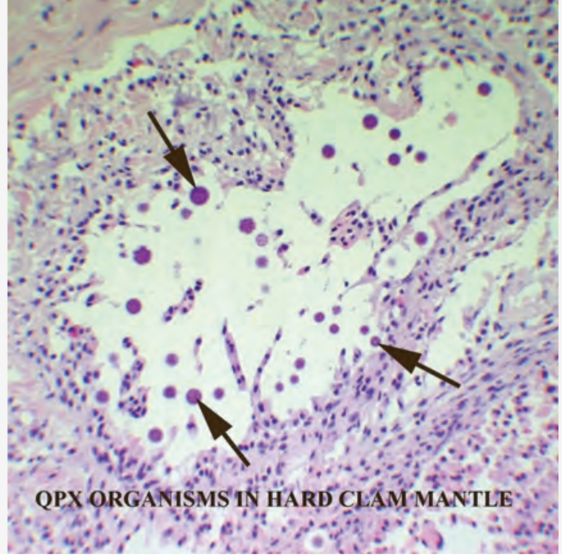


Figure 8b: Photomicrograph of a 6 µm section of paraffin embedded tissue with hematoxylin and eosin stain. QPX organisms (arrows) are embedded in clear spaces.
Roxanna Smolowitz

QUAHOG PARASITE UNKNOWN (QPX), PHYLUM *LABRINTHULOMYCOTA*, FAMILY *THRAUSTOCHYTRIDAE*

Method of infection: Initial infection occurs in the mantle adjacent to the base of the incurrent siphon in the approximate location of pseudofeces storage. Infection through the gill tissues is the second most common area of initial infection. Most commonly after organisms have established a site of infection in the mantle, they circulate in the vascular system and secondarily infect other tissues of the body of the clam.

Gross (observable) signs of disease: Irregular, small to large nodules and swelling of the mantle, most often located at the base of the siphon or in adjacent mantle tissues, inhibit proper functioning of the mantle and result in affected clams inability to maintain their position in the sediment. A common finding is large numbers of live clams lying on the surface at the sediment/water column interface (when in sandy sediment) especially in the spring and fall time period. These animals show a slight gap between the free edge of their valves and upon shucking exhibit nodules and swellings. The disease usually results in mortality in cultured two-year-old animal that are just under market size.

Microscopic Signs of Disease: QPX organisms occur in the tissues as rounded thalli that mature to form sporangia containing numerous endospores. Endospores are released from the sporangia and invade surrounding sinusoids of the mantle and other tissues. Mucus proliferation varies by strain of QPX, but is effective in inhibiting phagocytosis by the hemocytes. QPX incites a significant inflammatory reaction by the clam in the summer, forming relatively solid, yellow-tan nodules/swellings. In the spring and fall, infected foci are characterized by abundant organisms nested within abundant mucoid material with a light to moderate hemocytic reaction. Severely affected animals show QPX invasion in the sinusoids and associated inflammation throughout the animal's body.

Method of Transmission: QPX appears to be a water-borne, opportunistic pathogen. It is present in the environment and only under certain circumstances appears to cause infection. Once infection has occurred; however, it is very likely that the infection will continue to occur annually if clams continue to be planted in the lease. It is likely that selection for pathogenic forms of QPX occurs, and those forms seed the growing locations (Roxanna Smolowitz, unpublished data). Seed spawned from broodstock originating from the southern U.S. are more susceptible to the disease than seed spawned from local broodstock. Research has shown that the immune system of the clams is not able to successfully destroy the QPX parasite in the spring and fall time periods.

Potentiating environmental conditions:

- Temperature: 13 to 30°C; 32° C and higher will kill QPX
- Salinity: 28 to 40

Hazard Management:

- Use seed from local broodstock.
- Do not transfer seed or adults from infected plots.
- Remove infected animals from aquaculture plots.

Figure 9: Valves of two eastern oyster juveniles who died as a result of ROD infections. The deposition of conchiolin is noted on the inner surfaces of the valves.
Roxanna Smolowitz

ROSEOBACTERIUM OYSTER DISEASE (ROD), *ROSEOVARIUS CRASSOSTREAE*, (PREVIOUSLY CLASSIFIED AS JUVENILE OYSTER DISEASE OR JOD)

Method of infection: *R. crassostreae* bacteria primarily colonized the inner surface of the valves external to the mantle epithelium (shell epithelium).

Gross (observable) signs of disease: Animals less than 25 mm in shell height are the most severely affected by the disease. Visual signs of ROD are poor growth, deep cupping of the left valve and shortening of the right valve. Mantle retraction is noted as is a deposition of proliferative, flaky, brown/ yellow, loosely layered, moist conchiolin between the mantle and the inner shell surface.

Microscopic Signs of Disease: Lesions range from small areas of hemocyte infiltration in the affected epithelium of the mantle and underlying sinusoidal connective tissue to severe ulceration of the affected mantle with abundant tissue debris and layers of abnormal conchiolin deposited in the extrapallial space between the shell and the mantle.

Method of Transmission: *R crassostreae* is present in the environment, but abundance, seasonality, or pathogenicity of strains is not known.

Potentiating environmental conditions:

- Temperature: >20° C
- Salinity: 18 to 30
- Other: Stress may increase severity of the disease

Hazard Management:

- Use oyster seed that will reach a size >25 mm in shell height before the critical temperature occurs in the culture site.
- Use tolerant/resistant seed.
- During an outbreak, decrease density and increase water flow.

Figure 10: Small scallop seed are vulnerable to hinge-ligament disease.
Tessa Getchis

HINGE-LIGAMENT DISEASE, CAUSED BY GLIDING BACTERIA (*FLEXIBACTER* SPP. *CYTOPHAGA* SPP.)

Method of infection: Gliding bacteria invade and destroy the ligament that holds the two valves together. These bacteria do not infect the soft tissue of the bivalve, only tissues of the ligament. Bivalves up to 1 cm are especially vulnerable, but larger juveniles can also become infected.

Gross (observable) signs of disease: Affected juvenile bay scallops (and other juvenile bivalves) have loose or dislocated hinges preventing them from effective closing and opening their valves in order to respire and collect food. Eventually the adductor (and other) muscles fatigue from attempting to open and close the valves without a hinge and the death of the animal occurs.

Microscopic Signs of Disease: Gliding bacteria (*Flexibacter spp.* and *Cytophaga* spp.) are noted in large numbers in and on the degenerating ligament, specifically the part of the ligament termed the resilium.

Method of Transmission: The infectious agents are opportunistic bacteria that are commonly present in the water column and colonize the surface of structures in and on the sediment.

Potentiating environmental conditions:

- Temperature: 5 to 20° C (perhaps higher)
- Salinity: varies but usually 15 to 32
- Other: density and water flow strongly affect the occurrence of this disease.

Hazard Management:

- Maintain low densities and high water flow over the juveniles.
- Keep surfaces in the hatchery and nursery clean of debris and films.

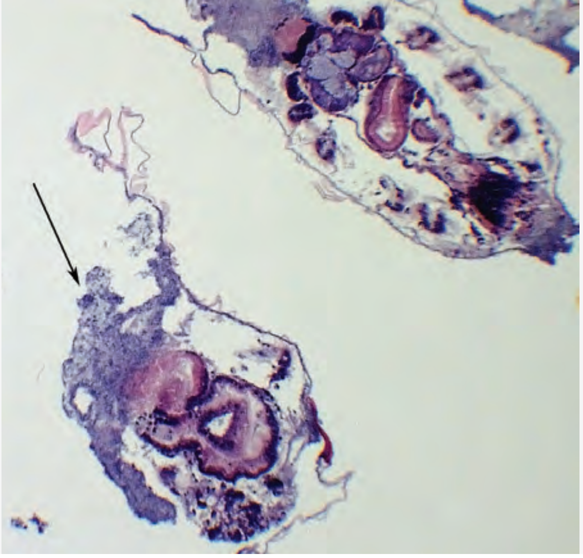


Figure 11: Vibriosis in Ensis directus.
Roxanna Smolowitz

VIBRIOSIS, BACILLARY NECROSIS (*VIBRIO* SPP.)

Method of infection: A disease of very young bivalves (only a few days to weeks old). The disease occurs almost exclusively in the hatchery, but may occur in up/downwellers as well. The bacteria are opportunistic. If high numbers of bacteria are present in the system, they will infect the larval/juvenile bivalves resulting in several, immediate (hours/not days) mortality, usually of all animals in the tank.

Bacteria attach to the velum of pre-metamorphic animals or infect the developing gut and other tissues of both pre and post-metamorphic animals causing destruction and necrosis of these tissues. Additionally, some *Vibrio* species are also thought to produce a toxin that may indirectly destroy the tissues.

Gross (observable) signs of disease: This disease and resultant mortality often occurs so fast it is not noticed until all area dead. A sudden lethargy of many of the larvae with loss of swimming and abnormal swimming, and settling to the bottom, should alert the culturist to the possibility of a *Vibrio* sp. infection.

Microscopic Signs of Disease: Examination of the tissue and shells of freshly dead larvae will demonstrate swarming bacteria in and around the dead/dying tissues. Tissues can be examined histologically to demonstrate the infection and cultures of the dying animals will often yield a reliable identification of the bacteria.

Method of Transmission: *Vibrio* sp. bacteria are a part of the saltwater environment. There are a few of these bacteria in every gallon of water, although most *Vibrio* sp. bacteria prefer to live on the surfaces of rocks, etc. in the "slime" layer. Many different *Vibrio* sp. have been identified as the cause of the disease in different hatcheries.

Potentiating environmental conditions:

- Temperature: Spring/Summer
- Salinity: estuarine to full strength sea water

Hazard Management:

- Good hatchery management such as clean water sources, pipes, and algal cultures.
- Remove infected animals from the facility and clean tanks, nets, pipes, floors, etc.

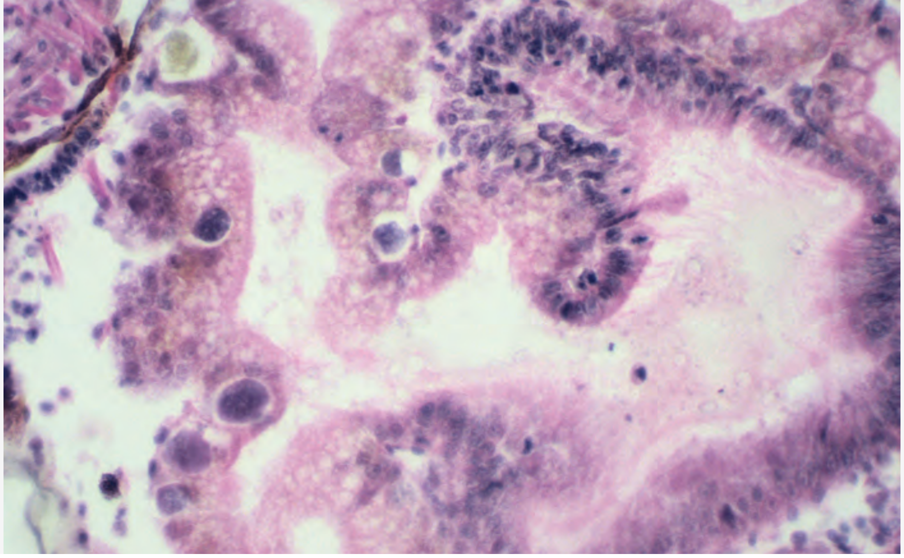


Figure 12: Photomicrograph of a 6 μm section of paraffin embedded tissue stained with hematoxylin and eosin stain. Large, granular, blue, Chlamydial inclusions are noted in the gastric epithelium of this larval bay scallop.
Roxanna Smolowitz

CHLAMYDIAL AND RICKETSIAL INFECTIONS

Species Affected:

- larval and juvenile bay scallops (*Argopecten irradians*), as well as adults of most species of commercially important shellfish

Method of infection: unknown

Gross (observable) signs of disease: In larval and post-metamorphic bay scallops, sudden mortality of the majority of the animals in the spawn occurs. No morbidity or mortality is associated with infections in other bivalves or in adult scallops.

Microscopic Signs of Disease: Chlamydial inclusions are noted in the digestive gland epithelial cells of the larval and juvenile animals and are associated with acute necrosis of the cells. In other bivalves, inclusions noted as Chlamydial/Rickettsial-like are noted in the digestive gland, gill and mantle epithelium but do not appear to cause necrosis of cells or inflammation, and thus are considered incidental findings.

Method of Transmission: probably through the water column

Potentiating environmental conditions:

- High densities of bay scallop larvae and juveniles combined with low water flow are thought to promote the disease.
- Chlamydial/Rickettsial inclusions are routinely noted in adult bay scallops and do not cause disease in adults. Some consider this a "childhood" disease of larval bay scallops.

Hazard Management:

- Separate adult scallops from larval and juvenile animals.

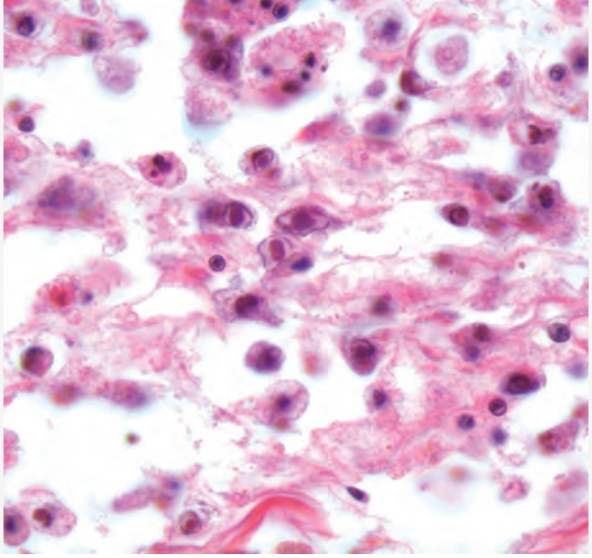


Figure 13a: Herpesvirus in CV 9 - 1000X.
Mary Stephenson

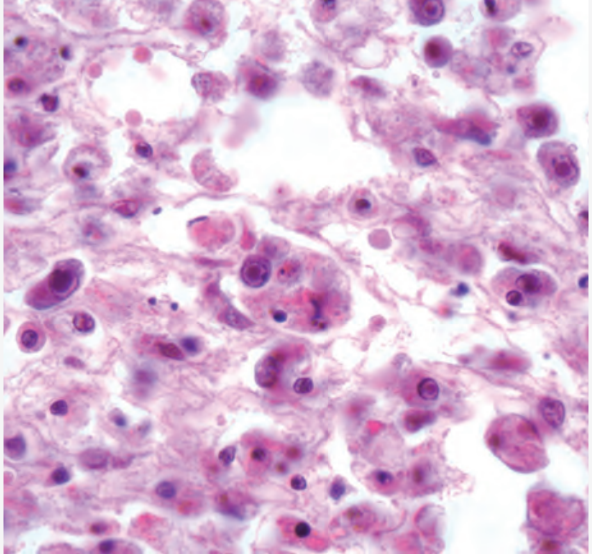


Figure 13b: Herpesvirus in CV 10 - 1000X.
Mary Stephenson

OYSTER HERPES VIRUS (OSHV)

Species Affected: *Crassostrea gigas*, potentially *Crassostrea virginica*

Method of infection: OsHV has been found in several bivalves but causes mortality in only a few. The eastern oyster is not currently noted to be infected by virulent forms of this virus (OsHV-1), but mortality of *C. gigas*, the pacific oyster, has been noted in the western U.S. and in Europe. The virus infects the oysters via the water column. In *C. gigas*, the hemocytes, connective tissue, and epithelial cells become infected resulting in cell destruction in several organs of the oyster's body. Larval and juvenile *C. gigas* are the most likely to experience high mortality. Adult *C. gigas* are considered carriers of the virus, but will develop disease if they have not been previously infected (i.e., resistance/tolerance does not appear to occur). Note that other bivalves may be carriers of the disease.

Gross (observable) signs of disease: No specific signs are noted.

Microscopic Signs of Disease: Intranuclear, Cowdry type A inclusions are noted in infected cells. Necrosis of cells, causing destruction of tissues, results in death of the oyster.

Method of Transmission: Directly transmitted from infected *C. gigas* seed or adults. OsHV is carried by many kinds of bivalves, although most do not show disease associated with infection. Work is ongoing to identify the potential for transmission of the virus to susceptible *C. gigas* from other bivalve sources and virulence of difference strains of OsHV.

Potentiating environmental conditions:

- Temperature: following temperature spikes > 25° C (Washington State)
- Salinity: 28 to 32
- Other: Some strains of cultured *C. gigas* oysters may be more susceptible. Alternately, more pathogenic strains of the virus may occur in various locations in the world. This disease has currently not been identified in *C. virginica* in the northeastern U.S.

Hazard Management:

- Do not transfer seed, water, or adults from areas of infection.

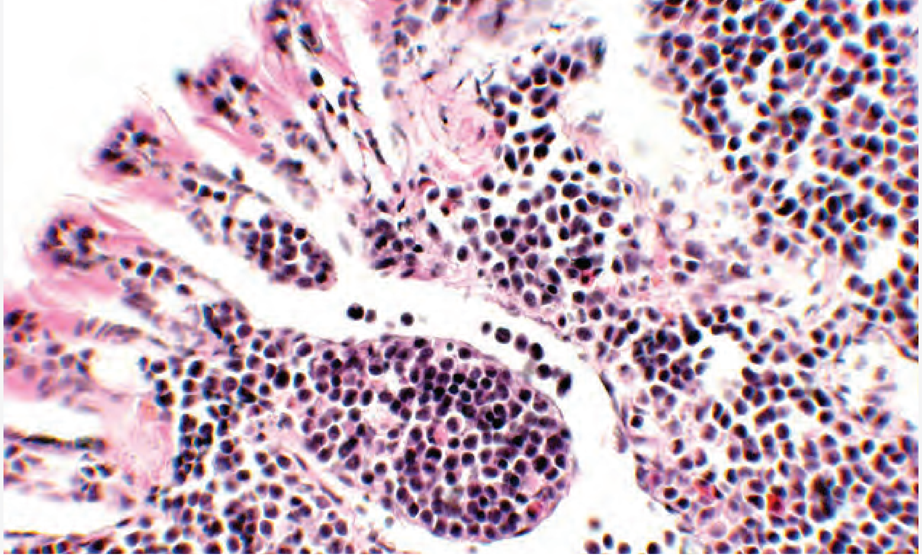


Figure 14: Photomicrograph of a 6 μm section of paraffin embedded gill with hematoxylin and eosin stain. Abundant neoplastic cells with large, blue nuclei are filling and obstructing the vascular space.
Roxanna Smolowitz

HEMOCYTIC NEOPLASIA (=DISSEMINATED SARCOMA)

Species Affected:

- softshell clam (*Mya arenaria*) and rarely in other bivalves such as *Mytilus edulis* and *M. trossulus*

Method of infection: unknown; but a viral etiology or genetic predisposition is possible

Gross (observable) signs of disease: In *M. arenaria*, mortality is usually the first observation. Poor growth combined with milky hemolymph is characteristic of infected animals. In areas with low prevalence of the disease (<13%), mortality is highest in the fall. In areas with high levels of disease (>30%) mortality and prevalence peaks in the fall and again in late winter to early spring. Two to three year old animals (40 to 80 mm in shell length) are the most severely affected. Recently, a similar disease has been identified in northern quahogs (*Mercenaria mercenaria*) in some estuaries on Cape Cod, Massachusetts. Prevalence of this disease and presentation of the disease in the animals is similar to findings in *M. arenaria*. Very rarely, a similar neoplastic condition is noted in other types of bivalves along the east coast of the U.S.

Microscopic Signs of Disease: Large oval to round tumorous cells fill the vascular tissues of the animal causing obstruction of the normal flow of hemolymph through the tissues.

Method of Transmission: Unknown at this time, but a genetic or viral component is suspected. Pollution has not been demonstrated to be a cause of this neoplasia in *M. arenaria*, but may potentiate development of the neoplasia.

Potentiating environmental conditions:

- Temperature: It is unknown whether temperature plays an important part in the transmission or severity of the disease, but the prevalence is noted to be higher in the fall and spring.

Hazard Management:

- Do not use affected seed (may want to monitor broodstock as well as seed).
- Consider removing infected clams to prevent transmission or spread by reproduction of the clam.

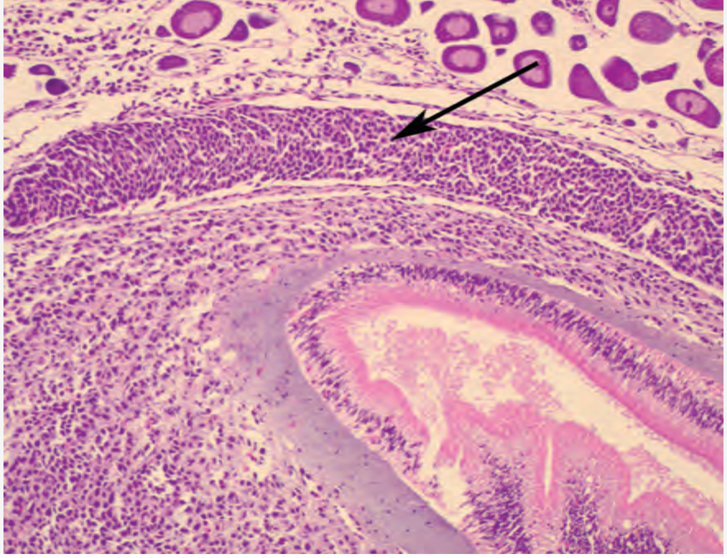


Figure 15: Clam neoplastic cells filling the sinusoids and a vessel.
Roxanna Smolowitz

HARD CLAM NEOPLASIA

Method of infection: unknown

Gross (observable) signs of disease: Animals cannot keep themselves buried in the sandy sediment, and so they rise to the surface and lay on the interface of the sediment and the water column. Shells usually are not gaping. Mortality occurs in the spring. Little to no disease is detected in surviving animals in the fall time period. Poor springtime growth and milky hemolymph are characteristics of the disease. The disease appears to primarily affect primarily 1 ½ to 2-year-old northern quahogs, and to date has been observed in Massachusetts.

Microscopic Signs of Disease: Large oval to round tumous cells fill the vascular tissues of the animal causing obstruction to flow of hemolymph.

Method of Transmission: unknown

Potentiating environmental conditions:

- Temperature: unknown; but most mortality is noted in the spring
- Salinity: 12 to 32

Hazard Management:

- Do not use affected seed or seed from broodstock selected from populations expressing the disease.
- Consider removing infected populations of clams to prevent spawning.

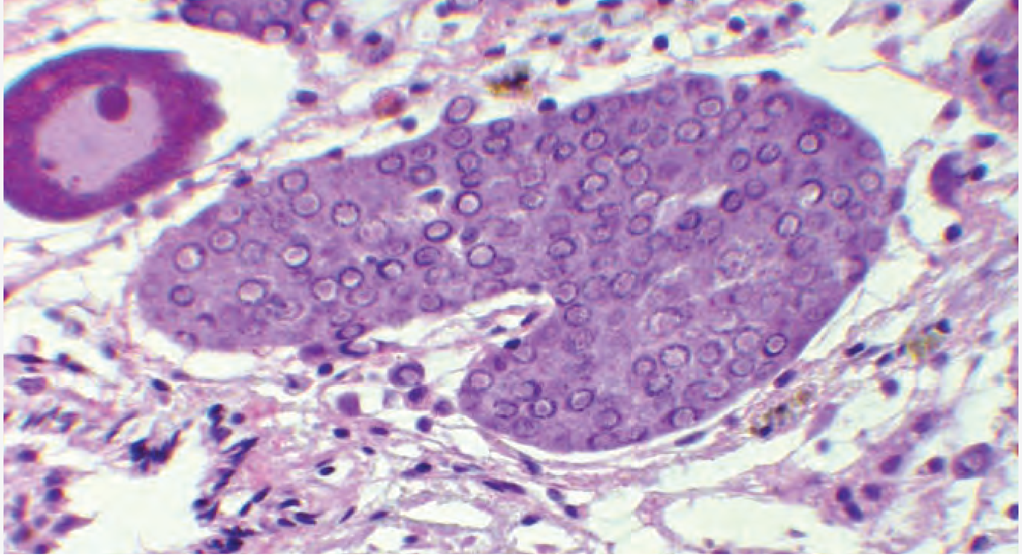


Figure 16: Photomicrograph of a 6 µm section of paraffin embedded tissue with hematoxylin and eosin stain. Tumorous cells fill some of the gonadal tubules of this clam. A maturing egg is noted in the adjacent gonadal tubule.
Roxanna Smolowitz

GONADAL NEOPLASIA (=DYSGERMINOMA OR GERMINOMA)

Species affected:

- softshell clam (*Mya arenaria*) and other commercially important bivalves

Method of infection: Probable genetic predisposition, increased incidence in late winter and early spring of the year in Maine populations of *M. arenaria* coinciding with normal gonadal maturation.

Gross (observable) signs of disease: Increased mortality in late winter and early spring.

Microscopic Signs of Disease: Large undifferentiated germ cells proliferate and fill the gonadal tubules. In *M. arenaria*, rupture of gonadal tubules occurs with spread of the tumorous cells into the surrounding connective tissue and vascular spaces. In other bivalves, tumor is usually identified only in the tubules and does not appear to invade surrounding tissues.

Method of Transmission: Probably genetic in origin. Research has not identified a confirmed association with pollution.

Potentiating environmental conditions:

- unknown

Hazard Management:

- Do not use affected seed or seed from broodstock selected from populations expressing the disease.
- Consider removing infected populations of clams to prevent spawning.

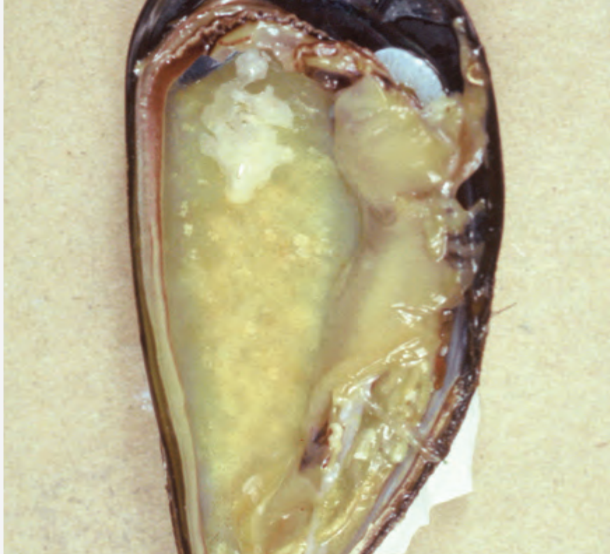


Figure 17a: Partially shucked blue mussel. Pock marks resulting from trematode infection are present in the mantle tissues.
Roxanna Smolowitz

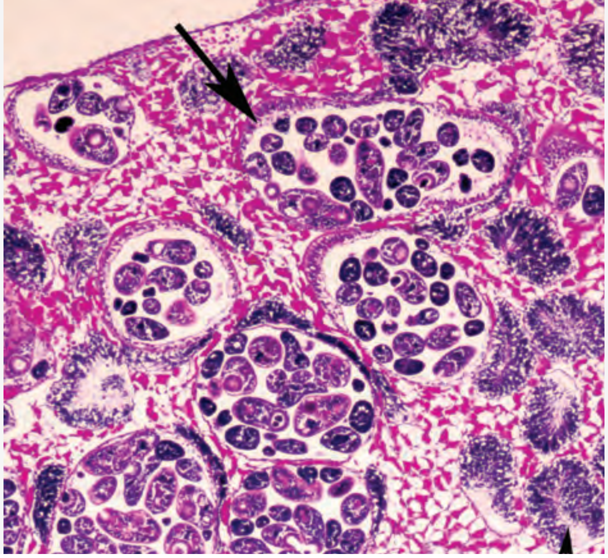


Figure 17b: Photomicrograph of a 6 µm section of paraffin embedded blue mussel mantle stained with hematoxylin and eosin stain. Abundant trematodes (large arrow) are developing in the sporocysts within the male gonadal tubules. Some sperm can still be identified in some gonadal tubules (smaller arrow).
Roxanna Smolowitz

DIGENEAN TREMATODES (*PROCTOECES MACULATUS*, *PROSORHYNCHUS SQUAMATUS*, *BUCEPHALUS* SP.)

Species Affected:

- blue mussel (*Mytilus edulis*), eastern oyster (*Crassostrea virginica*), and other bivalves

Method of infection: Myracidia infect tissues, probably through the digestive tract. Sporocysts develop daughter sporocysts, redia, and cercaria in the gonadal tubules.

Gross (observable) signs of disease: Orange yellow "pocks" in the blue mussel mantle may be noted in severe infections.

Microscopic Signs of Disease: Few to many of the gonadal tubules are filled with various forms of the trematode. Tubules containing parasites are usually greatly dilated. Eggs and sperm are destroyed resulting in parasitic castration. In moderate to severe infections, parasites can be identified in the sinusoids of the gills and body. Inflammation is mild when parasites are confined to the gonadal tubules but can be severe and diffuse when organisms rupture the tubules and spread into the sinusoids. It is not known when and how the cercaria (the infectious form of the parasite) leaves the molluscan tissues.

Method of Transmission: As with most digenean parasites another host is involved in the life cycle; however, some authors suggest the entire life cycle may occur in blue mussels. Several species of fish have been identified as the final hosts (the host in which the adult form of the parasite is found) including scup, cunners, and other mollusc eating fishes. Eggs are produced by adult trematodes in the final host (most likely a fish). Eggs produced in the final host by the adult trematodes are released with feces and develop into myracidia that infect filter feeding bivalves.

Potentiating environmental conditions:

- Temperature: disease may be associated with high summer mortality of blue mussels
- Salinity: high salinity waters

Hazard Management:

- Eliminate the final host from the culture environment.

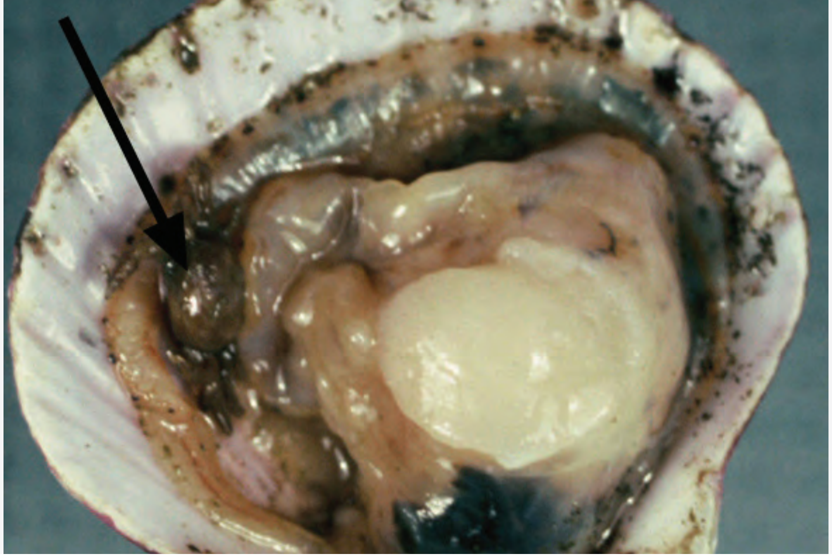


Figure 18: Bay scallop infected by a pea crab (arrow). Although a rare occurrence, significant traumatic damage to the gill, mantle and adjacent soft body has occurred in this animal.
Roxanna Smolowitz

PEA CRABS (*TUMIDOTHERES MACULATES, T. OSTREUM*)

Mode of Action: Pea crabs inhabit the gills of certain bivalves, where they can affect nutrition by disrupting feeding and competing with shellfish for food. Adult pea crabs can occasionally cause significant traumatic damage to the gill, mantle, and body tissues. The highest infection occurs sub tidally. Their presence can decrease the marketability of shellfish.

Species affected:

- blue mussels (*Mytilus edulis*)
- eastern oyster (*Crassostrea virginica*)
- bay scallops (*Argopecten irradians*)

Hazard Management:

- If possible, avoid establishing culture operations in areas with prior pea crab infestations.

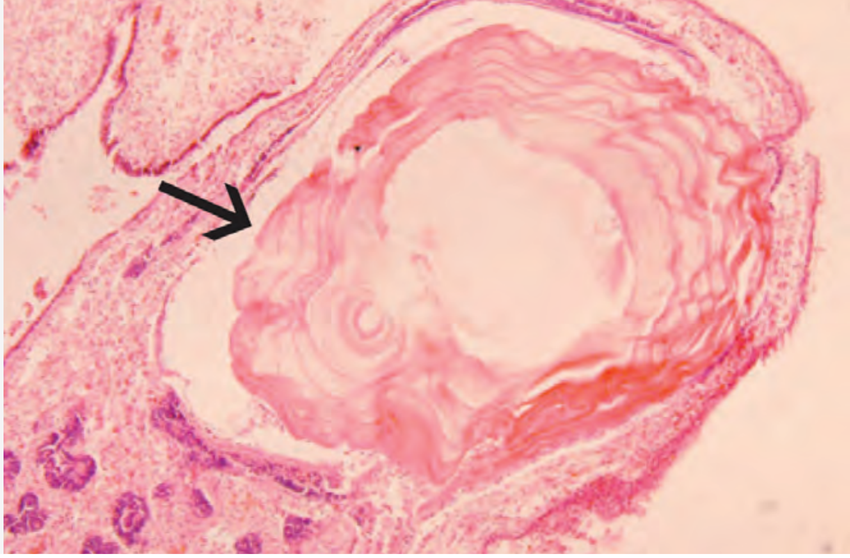


Figure 19: Histological section of a pearl (arrow) with concentric layers in the mantle tissue of a blue mussel.
Meyers and Burton 2009, Alaska Department of Fish and Game

PEARLS

Mode of Action: Pearls are formed when infectious organisms or grit, such as fine sand granules, occur between the shell epithelium of the mantle and the inner surface of the adjacent valve. Hemocyte reaction to the infection organism kills the infectious organism. The dead organisms or grit then become the nidus for pearl formation. In reaction to the "irritation" the shell epithelium produces successive layers of nacre around the inciting dead agent/grit. Over time, layers of nacre build up resulting in the formation of a pearl. Pearls can take various shapes and sizes depending on the bivalve and the inciting cause. Their presence (when small and grit like) can decrease the marketability of shellfish or when large and iridescent can be an amazing find! Pearls of commercially edible shellfish are, however, not typically of gem quality.

Species affected:

- Any bivalve; however, some bivalves such as blue mussels are more prone to containing "grit" like proliferations of pearls in their tissue

Hazard Management:

- If possible, avoid areas with a history of grit-like pearl formation.
- Harvest mussels before they reach 3 years of age.

References and Further Reading:

Allen, S. K., Gaffney, P.M., Ewart, J.W., 1993. Genetic Improvement of the eastern oyster for growth and disease resistance in the northeast. USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 210.

Andrews, J.D., 1988. Epizootiology of the disease caused by the oyster pathogen *Perkinsus marinus* and its effects on the oyster industry, in: Fisher, W.S. (Ed.), Disease Processes in Marine Bivalve Molluscs. American Fisheries Society Special Publication No. 18, Bethesda, Maryland, pp. 47-63.

Audemard, C., Carnegie, R.B., Bishop, M.J., Peterson, C.H., Burreson, E.M., 2008. Interacting effects of temperature and salinity on *Bonamia* sp. parasitism in the Asian oyster *Crassostrea ariakensis.* Journal of Invertebrate Pathology. 98(3), 344-350

Barber, B.J., 1996. Effects of gonadal neoplasms on oogenesis in softshell clams, *Mya arenaria*. Journal of Invertebrate Pathology. 67, 161-168.

Bierbaum, R.M., Ferson, S., 1986. Do symbiotic pea crabs decrease growth rate in mussels? Biological Bulletin. 170, 51-61.

Boardman, C.L., Maloy, A.P., Boettcher, K.J., 2008. Localization of the bacterial agent of juvenile oyster disease (*Roseovarius crassostreae*) within affected eastern oysters (*Crassostrea virginica*). Journal of Invertebrate Pathology. 97, 150-158.

Bower, S.M., 1997. Synopsis of Infectious Diseases and Parasites of Commercially Exploited Shellfish: Chlamydiosis of Scallops. Fisheries and Oceans Canada.

Bower, S.M., 2006. Synopsis of Infectious Diseases and Parasites of Commercially Exploited Shellfish: Gonadal Neoplasia of Clams. Fisheries and Oceans Canada.

Bower, S.M., 2007a. Synopsis of Infectious Diseases and Parasites of Commercially Exploited Shellfish: *Haplosporidium costale* (SSO) of Oysters. Fisheries and Oceans Canada.

Bower, S.M., 2007b. Synopsis of Infectious Diseases and Parasites of Commercially Exploited Shellfish: *Haplosporidium nelsoni* (MSX) of Oysters. Fisheries and Oceans Canada.

Bower, S.M., 2009a. Synopsis of infectious diseases and parasites of commercially exploited shellfish: juvenile disease of eastern oysters. Fisheries and Oceans Canada.

Bower, S.M., 2009b. Synopsis of Infectious Diseases and Parasites of Commercially Exploited Shellfish: *Proctoeces maculatus* Trematode Disease of Mussels. Fisheries and Oceans Canada.

Bower, S.M., 2011.a Synopsis of Infectious Diseases and Parasites of Commercially Exploited Shellfish: *Perkinsus marinus* ("Dermo" Disease) of Oysters. Fisheries and Oceans Canada.

Bower, S.M., 2011.b Synopsis of Infectious Diseases and Parasites of Commercially Exploited Shellfish: *Perkinsus* of Clams and Cockles. Fisheries and Oceans Canada.

Bower, S.M., 2012. Synopsis of Infectious Diseases and Parasites of Commercially Exploited Shellfish: *Steinhausia mytilovum* (Mussel Egg Disease). Fisheries and Oceans Canada.

Brothers, C., Marks III, E., Smolowitz, R., 2000. Conditions affecting the growth and zoosporulation of the protistan parasite QPX in Culture. Biological Bulletin. 199, 200-201.

Burge, C.A., Griffin, F.J., Friedman, C.S., 2006. Mortality and herpesvirus infections of the Pacific oyster *Crassostrea gigas* in Tomales Bay, California, USA. Diseases of Aquatic Organisms. 73, 31-43.

Burge, C.A., Strenge, R.E., Friedman, C.S., 2011. Detection of the oyster herpesvirus in commercial bivalves in northern California, USA: conventional and quantitative PCR. Diseases of Aquatic Organisms. 94, 107-118.

Burreson, E.M., 1994. Further evidence of regular sporulation by Haplosporidium nelsoni in small oyster, *Crassostrea virginica*. Journal of Parasitology. 80(6), 1036-1038.

Burreson, E.M., Ford, S.E., 2004. A review of recent information on the Haplosporidia, with special reference to *Haplosporidium nelsoni* (MSX disease). Aquatic Living Resource. 17(4), 499-517.

Burreson, E.M., Stokes, N.A., Carnegie, R.B., Bishop, M.J., 2004. *Bonamia* sp. (*Haplosporidia*) found in non-native oysters, *Crassostrea ariakensis*, in Bogue Sound, North Carolina. Journal of Aquatic Animal Health. 16(1), 1-9.

Bushek, D., Landau, B.J., Scarpa, E.M., 2008. *Perkinsus chesapeaki* in stout razor clams *Tagelus plebeius* from Delaware Bay. Diseases of Aquatic Organisms. 78(3), 243-247.

Calvo, L.M.R., Ford, S.E., Kraeuter, J., Smolowitz, R., Burreson, E.M., 2007. Influence of host genetic origin and geographical location on QPX disease in northern quahogs (= hard clams), *Mercenaria mercenaria*. Journal of Shellfish Research. 26(1), 109-119.

Carnegie, R.B., Stokes, N.A., Audemard, C., Bishop, M.J., Wilbur, A.E., Alphin, T.D., Posey, M.H., Peterson, C.H., Burreson, E.M., 2008. Strong seasonality of *Bonamia* sp. infection and induced *Crassostrea ariakensis* mortality in Bogue and Masonboro Sounds, North Carolina, USA. Journal of Invertebrate Pathology. 98(3), 335-343

Cheng, T.C., 1967. Marine molluscs as hosts for symbiosis. Advances in Marine Biology. 5, 1-424.

Comtet, T., Garcia, C., Le Coguic, Y., Joly, J., 2004. First record of the microsporidian parasite *Steinhausia mytilovum* in *Mytilus* sp. (Bivalvia: Mytilidae) from France. Diseases of Aquatic Organisms. 58, 261-264.

Dungan, C.F., Elston, R.A., Schiewe, M.H., 1989. Evidence for colonization and destruction of hinge ligaments in cultured juvenile Pacific oysters (*Crassostrea gigas*) by *Cytophaga*-like bacteria. Applied Environmental Microbiology. 55(5), 1128-1135.

Elston, R.A., 1990. Mollusc Diseases: Guide for the Shellfish Farmer. Washington Sea Grant Program, University of Washington, Seattle, Washington.

Elston, R.A., 1999. Health Management, Development and Histology of Seed Oysters. World Aquaculture Society. Baton Rouge, Louisiana.

Ercolini, C., Giorgi, I., Prearo, M., Serracca, L., Gallo, F., Ceschia, G., 2008. Presence of *Steinhausia mytilovum* in mussel (*Mytilus galloprovincialis*) by La Spezia Gulf (Eastern Ligurian Sea, Italy). Ittiopatologia. 5(2), 81-86.

Ewart, J. W., Ford, S.E., 1993. History and impact of MSX and Dermo disease on oyster stocks in the Northeast Region. Northeastern Regional Aquaculture Center, NRAC Publication No. 200.

Ford, S.E., 2001. Pests, parasites, diseases and defense mechanisms of the hard clam, *Mercenaria mercenaria*, in: Kraeuter, J.M., Castagna, M. (Eds.), Biology of the Hard Clam. Elsevier Science, Amsterdam, pp. 591-649.

Ford, S.E., Tripp, M.R., 1996. Diseases and Defense Mechanisms, in: Kennedy, V.S., Newell, R.I.E., Eble, A.F., (Eds.), The eastern oyster (*Crassostrea virginica*). Maryland Sea Grant College, College Park, Maryland, pp. 581-660.

Gast, R., Moran, D., Audemard, C., Maille, L.M., DeFavari, J., Reese, K.S., Leavitt, D., Smolowitz, R., 2008. Environmental distribution and persistence of Quahog Parasite Unknown (QPX). Diseases of Aquatic Organisms. 81(3), 219-229.

Haven, D.S., 1958. Effects of pea crabs *Pinnotheres ostreum* on oysters *Crassostrea virginica*. Proceedings of the National Shellfisheries Association. 49, 77-86.

Hughes, F.M., Foster, B., Grewal, S., Sokolova, I.M., 2010. Apoptosis as a host defense mechanism in *Crassostrea virginica* and its modulation by *Perkinsus marinus*. Fish and Shellfish Immunology. 29, 247-257.

Jones, J.B., Creeper, J., 2006. Diseases of pearl oysters and other molluscs: a Western Australian perspective. Journal of Shellfish Research. 25, 233-238.

Karlsson, J.D., 1991. Parasites of the bay scallop, *Argopecten irradians* (Lamarck, 1819), in: Shumway, S.E., Sandifer, P.A. (Eds.), International Compendium of Scallop Biology and Culture. World Aquaculture Society, Baton Rouge, Louisiana, pp. 180-190.

Karney, R.C., Blake, J.C., 1999. Developments in the private aquaculture industry on Martha's Vineyard. Journal of Shellfish Research. 18(1), 269-270.

Kryczynski, W.L., 1972. The effect of the pea crab, *Pinnotheres maculatus* Say, on the growth of the bay scallop, *Argopecten irradians concentricus* Say. Chesapeake Science. 13, 218-220.

La Peyre, M., Casa, S., La Peyre, J., 2006. Salinity effects on viability, metabolic activity and proliferation of three *Perkinsus* species. Diseases of Aquatic Organisms 71 (1): 59-74.

Leavitt, D.F., McDowell Capuzzo, J., Smolowitz, R.M., Miosky, D.L., Lancaster, B.A., Reinisch, C.L., 1990. *Hematopoietic neoplasia* in *Mya arenaria*: prevalence and indices of physiological condition. Marine Biology. 105, 313-321.

Leibovitz, L., 1989. Chlamydiosis: a newly reported serious disease of larval and postmetamorphic bay scallops, *Argopecten irradians* (Lamarck). Journal of Fish Diseases. 12, 125-136.

Leibovitz, L.E., Schott, F., Karney, R.C., 1984. Disease of wild, captive, and cultured scallops. Journal of the World Mariculture Society. 15, 269-283.

Maloy, A.P., Ford, S.E., Karney, R.C., Boettcher, K.J., 2007. *Roseovarius crassostreae*, the etiological agent of juvenile oyster disease (now to be known as roseovarius oyster disease) in *Crassostrea virginica*. Aquaculture. 269, 71-83.

McGladdery, S.E., Bower, S.M., Getchell, R.G., 2006. Diseases and parasites of scallops, in Shumway, S.E. (Ed.), Scallops: Biology, Ecology and Aquaculture. Elsevier, Amsterdam, Netherlands, pp. 632.

McGladdery, S.E., Stephenson, M.F., McArthur, F., 1999. *Prosorhynchus squamatus* (Digenea: Platyhelminthes) infection of blue mussels, *Mytilus edulis* in Atlantic Canada. Journal of Shellfish Research. 18, 297.

McGurk, E.S., 2013. Ciliate Xenomas In *Crassostrea virginica* From Great Bay, New Hampshire, A thesis submitted to the Graduate School – Camden, Rutgers, The State University of New Jersey. 58 pp.

McLaughlin, S.M., Faisal, M., 1998. Histopathological alterations associated with *Perkinsus* spp. infection in the softshell clams *Mya arenaria*. Parasite. 5(3), 263-271.

McLaughlin, S.M., Faisal, M., 2000. Prevalence of *Perkinsus* spp. in Chesapeake Bay softshell clams, *Mya arenaria* Linnaeus, 1758 during 1990-1998. Journal of Shellfish Research. 19(1), 349-352.

Meyers, T., Burton, T., 2009. Common Diseases of Wild and Cultured Shellfish in Alaska. Alaska Department of Fish and Game.

Morse, D., Rice, M.A., 2010. Mussel aquaculture in the Northeast. USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 211.

Perkins, F.O., 1988. Parasite morphology, strategy and evolution, in: Fisher, W.S. (Ed.), Disease Processes in Marine Bivalve Molluscs. American Fisheries Society Special Publication No. 18, Bethesda, Maryland, pp. 93-111.

Peters, E.C., 1988. Recent investigations on the disseminated sarcomas of marine bivalve molluscs. American Fisheries Society Special Publication. 18, 74-92.

Reese, K.S., Dungan, C.F., Burreson, E.M., 2008. Molecular epizootiology of *Perkinsus marinus* and *P. chesapeaki* infections among wild oysters and clams in Chesapeake Bay, USA. Diseases of Aquatic Organisms. 82, 237-248.

Smolowitz, R., 2013. A review of current state of knowledge concerning *Perkinsus marinus* effects on *Crassostrea virginica* (Gmelin) (the eastern oyster). Veterinary Pathology. 50(3), 404-411.

Smolowitz, R., Leavitt, D., Perkins, F., 1998. Observations of a protistan disease similar to QPX in *Mercenaria mercenaria* (hard clams) from the coast of Massachusetts. Journal of Invertebrate Pathology. 71, 9-25.

Stunkard, H.W., Uzmann, J.R., 1959. The life cycle of the digenetic trematode, *Proctoeces maculatus* (Looss, 1091) Odhner, 1911 [Syn. P. Subtenuis (Linton, 1907) Hanson, 1950] and description of cercaria andranocerca N. SP. Biological Bulletin. 116(1), 184-193.

Sunila, I., Stokes, N.A., Smolowitz, R., Karney, R.C., 2002. *Haplosporidium costale* (seaside organism), a parasite of eastern oyster, is present in Long Island Sound. Journal of Shellfish Research. 21(1), 113-118.

Sunila, I., Williams, L., Russo, S., Getchis, T., 2004. Reproduction and pathology of blue mussels, *Mytilus edulis* (L.) in an experimental longline in Long Island Sound, Connecticut. Journal of Shellfish Research. 23, 731-740.

Taraska, N.G., Böttger, S.A., 2012. Selective initiation and transmission of disseminated neoplasia in the soft shell clams *Mya arenaria* dependent on natural disease prevalence and animal size. Journal of Invertebrate Pathology. 112, 94-101.

Tubiash, H.S., Colwell, R.R., Sakazaki, R., 1970. Marine Vibrios associated with bacillary necrosis, a disease of larval and juvenile bivalve mollusks. Journal of Bacteriology. 103(1), 271-272.

Walton, W., Murphy, D., 2008. Managing Quahog Parasite Unknown (QPX) disease. Marine Extension Bulletin. Woods Hole Sea Grant and Cape Cod Cooperative Extension.

Potential Shellfish Production Hazards

Environmental Conditions

Biofouling Organisms

Predators

Diseases and Parasites

Invasive Species

Operational Procedures

Invasive Species

An increasing number of invasive species have been documented in marine waters of the northeastern U.S, and many of these species pose a threat to aquaculture, either as predators, competitors, or potential vectors of disease or harmful algal bloom transfer. The potential for additional invasive species is also increasing as species expand their range as a result of climate change. Shellfish farmers, in particular, spend an inordinate amount of time, effort, and capital dealing with invasive fouling organisms and predators. Farmers should be cognizant of the risks and dangers of introducing invasive species and take extra precautions to avoid becoming a "vector" for the next environmental calamity.

Several species of macroalgae, or seaweed species considered non-indigenous species are known to exist in the region. *Codium fragile* ssp. *tomentosoides*, commonly known by names such as "oyster thief," "green fleece," and "dead man's fingers," is believed to have been introduced to Boothbay Harbor, Maine in the 1960s through oyster beds, and has had adverse impacts on shellfish and other marine life, aquaculture, and recreation. It has increased its range and is now present throughout the region. In the last few years, other non-native, and potentially invasive species of marine algae have also been documented.

Over the past several decades there has also been an increase of non-native ascidians, also known as tunicates or sea squirts, in the coastal waters of the northeast. Several species of tunicates including colonial forms (e.g. *Didemnum vexillum*, *Botryllus schlosseri*, and *Botrylloides violaceus*) and solitary forms (e.g. *Styela clava*, *Ciona intestinalis*), have adversely affected aquaculture operations across the region and in Canada. Some species have been found on large swaths of the ocean floor on Georges Bank off Massachusetts and in New England tidal lagoons and estuaries.

While the invasive European green crab (*Carcinus maenas*) has inhabited the region for over a century, several other species of non-indigenous crabs have been increasing in range, including the Chinese mitten crab (*Eriocheir sinensis*) and the Asian Shore crab (*Hemigrapsus sanguineus*). Little is known or documented about the effects of these invasives on cultured shellfish populations.

While these are only a few examples of invasive species effecting aquaculture operations, many threats exist. As such, there are state, federal, and international regulations which restrict the movement of non-native species. Many states regulate the sources of imported seedstock to reduce the potential for disease transmission, but minimizing inadvertent invasive species introductions is also an important consideration. To reduce the risk of invasive transfer, shellfish farmers should inspect seed and product entering and leaving their facility. Shipments containing non-native species should be rejected. Biofouling organisms should be removed to the extent possible, and properly disposed of using a land-based facility. If a farmer is buying used gear or boats from outside his or her immediate watershed, it is worth taking some care to ensure that fouling organisms and undesirable non-natives are all dead and removed.

Because a good portion of the workload for shellfish farmers is cleaning the field gear, farmers should be diligent in inspecting their shellfish and their gear for invasive species. Observations out of the ordinary should be brought to the attention of state shellfish officials or aquaculture extension personnel. Since it is virtually impossible to eradicate a marine invasive once it becomes established, it makes sense to do everything to prevent introduction in the first place.

References and Further Reading:

Carlton, J.T., 2001. Introduced Species in U.S. Coastal Waters: Environmental Impacts and Management Priorities. Pew Oceans Commission, Arlington, Virginia.

International Council on Exploration of the Seas (ICES), 2005. ICES code of practice on the introductions and transfers of marine organisms.

Padilla, D.K., Shumway, S.E., 2011. Invasive species: in: Shumway, S.E. (Ed.), Shellfish Aquaculture and the Environment. Wiley Science Publishers, Ames City, Iowa, pp. 395-424.

McKindsey, C.W., Landry, T., O'Beirn, F.X., Davies, I.M., 2007. Bivalve aquaculture and exotic species; a review of ecological considerations and management issues. Journal of Shellfish Research. 26, 281–294.

Potential Shellfish Production Hazards

Environmental Conditions

Biofouling Organisms

Predators

Diseases and Parasites

Invasive Species

Operational Procedures

Operational Procedures

It important for farmers to recognize what operational factors influence farm productivity. There are some basic steps that a farmer can take to ensure that the facility/cultivation area is adequately prepared for propagating, receiving, containing, grading, growing, handling, and processing shellfish. Operational conditions that are unsanitary or stressful to the animals may present a hazard to production.

Not all strategies are relevant to every shellfish farm location and situation. Therefore, it is the responsibility of the farmer to consider strategies that are relevant to their business and then to optimize them based on the observations of conditions at their location.

This chapter includes tips on important issues to consider:

facility maintenance
water flow
substrate
animal health and condition
animal grading and containment
animal stocking densities
animal handling and transport

FACILITY MAINTENANCE:

The facility of cultivation should be adequately prepared for propagating, receiving, containing, grading, growing, handling, and processing shellfish. Aside from proper design, the next most important step is to ensure the cleanliness of the facility, equipment, supplies, vessels, etc.

In the hatchery and, to a lesser degree, the nursery, cleanliness focuses on the hygiene of the culture systems, as microalgae cultures and larval/early post-set rearing systems are prone to microbial and protozoan infestations that can lead to extremely high larval/early juvenile mortality. In the grow-out stage, cleanliness primarily focuses on predator control and antifouling management. The latter is to ensure proper water flow through the culture apparatus.

What is the potential hazard?

Microbial infections during early shellfish rearing can lead to high mortality. There are many naturally occurring opportunistic microbes, e.g. various *Vibrio* species of bacteria, which will flourish under the high nutrient loads frequently encountered in both micro-algal culture and larval/early juvenile rearing systems. As the microbial populations increase, the larvae and early juveniles are an easy host to attack and the microbes can become pathogenic should the levels increase above a certain threshold. It is not uncommon to experience greater than 90% mortality of shellfish larvae within 24 hours following contamination with *Vibrio* spp.

In addition, the potential for protozoan infestation in larval and early juvenile systems can be problematic. Protozoans, such as ciliates, are naturally occurring single-celled animals in seawater that can flourish in the hatchery. They can foul the shells of larval and juvenile shellfish as well as consume the soft tissue of the larvae and juveniles resulting in significant mortality.

At the grow-out stage, adequate water flow is essential. Water flow delivers dissolved oxygen and food to the growing animals. Therefore, anything that obstructs water flow through the holding system will impair growth and may lead to mortality.

Hazard management:

- Washing and sanitizing of equipment should be part of the normal cleaning routine. Depending on the culture operations, the larval or juvenile culture systems must be cleaned and sanitized routinely, e.g. static larval culture systems are normally maintained on a two-day water change cycle where the holding tank and its contents are drained, cleaned, and sanitized every other day. In addition, water and nutrients used in the culture process should be filtered and sterilized. For example, culture media used for microalgae culture is normally either heat or chemically sterilized prior to the inoculation of the media with new stock cultures.
- A developing method to maintain a healthful environment for growing shellfish larvae is through the use of probiotics to the culture system. Probiotics include beneficial bacteria that, when added to the culture system, reduce the ability of potentially pathogenic microbial proliferation by either limiting competing microbial growth by exudate release or outcompeting for space or resources. Although there are reports of the use of antibiotic additions to larval culture systems as a means to keep microbial populations down, it must be emphasized that there are no approved antibiotic treatments for shellfish hatchery applications in the United States and it is illegal to use these compounds as a treatment.

- Observation of the culture conditions and the status of the larvae during the culture interval should be routine practice. Sub-sampling microalgae, larvae, or post-set juveniles and examining them under magnification on a daily basis are essential to ensuring that the cultures are clean. Microbial problems are manifest as accumulations of bacteria around the vicinity of the velum in larvae or along the ventral margin of the shell in post-set juveniles. Ciliates and other protozoans are readily apparent under the microscope as they are often highly mobile or attached to juvenile shells. If a bacterial problem is suspected, contact the local aquatic animal health professional (Appendix 3).
- In the field under grow-out conditions, routine observation of the culture gear is required to monitor biofouling and sedimentation, among other hazards. Gear should be washed or rotated as necessary to prevent flow restrictions.

WATER FLOW

At the nursery and grow-out levels, the primary consideration to maintain productive environmental conditions is to ensure adequate flow through the aquaculture gear.

What is the potential hazard?

Reduced oxygen can inhibit growth and may cause mortality under extreme low oxygen conditions, as is described in the Environmental Hazards Section Reduced food across the feeding apparatus of the shellfish will also result in reduced growth and a longer time to market.

Hazard management:

- Follow species-appropriate guidelines (if available) for water flow.
- Reduce holding densities in shellfish aquaculture gear.

SUBSTRATE

With those species that are set on substrate in the hatchery or are planted either in the substrate (clams) or directly on the substrate (free-planted oysters) during grow-out, the substrate type is also an important environmental consideration.

What is the potential hazard?

Without proper substrate in the hatchery, the overall proportion of larvae setting can be dramatically impacted, e.g. setting larval oysters on microcultch.

Planting nursery seed in the grow-out stage requires the seed to be placed in sediment that is appropriate for the species. For example, quahogs require sandy mud that is not so densely compacted that the seed have difficulty digging in. For those shellfish that are reared in some form of containment device that is elevated above the sediment, then substrate type is of less consequence. The one caveat to that statement is if the substrate is of a type that is easily re-suspended during wind/wave events, in which case the resulting increase in turbidity can have some impact on the productivity of the system.

Hazard management:

- Follow species-appropriate guidelines (if available) for substrate

ANIMAL HEALTH AND CONDITION

Animal health and condition is key to the success of the farm. Unless an animal is relatively stress free and has adequate resources to support growth, the overall farm performance can suffer.

What constitutes condition in a shellfish species? Condition Index (CI) is a measurable quantity that describes the appearance of a normal, healthy shellfish. Numerous clues are available that allow for a general assessment of condition and all are predicated on the experience of the farmer as to what is "normal" on their farm. Some of the signs used to assess condition include:

- **Mortality:** Note signs that may indicate illness or observing mortality events as they begin, there may be an opportunity to correct the situation. Contact the local aquatic animal health professional at the onset of a problem (Appendix 3).
- **Shell growth:** New growth is readily apparent along the outer margin of the shell. If it stops for an unexplained reason, then something is amiss. Awareness of what normal growth patterns can be expected through the year is a necessary knowledge base to allow for assessment of growth patterns as an indicator of a problem. Growth is not always seen in the shell. Often in spring animals are devoting their energy to gonadal development and shell growth may lag temperature and food. Sometimes temperatures are too high or food is absent or inadequate (or of low quality) and growth will pause just when conditions appear optimal.
- **Overall appearance of tissue (meat):** As they approach market size, shellfish go through seasonal cycles of robust plump meats followed by thin watery meats right after a spawn. This is almost entirely dependent on the annual reproductive cycle of the animal, where the meat increases in size (and quality) as the individual approaches sexual maturity with ripe gonads. Following spawning, the meats are dramatically reduced and appear watery and thin, almost translucent to the light. Then, they gradually build back up again as the animal feeds and gains back tissue structure in preparation for the next spawning event.
- **Appearance of key anatomical features (gill, mantle, gonad):** There are a few anatomical features that are often the first to be compromised should a problem arise. Knowing what a normal gill, mantle, and gonad looks like is a necessary to monitoring these anatomical features for potential problems. Observation of these structures as a follow up to the observation of poor growth or poor meat appearance may allow a better diagnosis of the problem. Some features to look for include malformed structure, inconsistent or aberrant coloration, swelling, or size reduction. While there is considerable variability in the formation of an individual shellfish organ, it is often possible to discern potential problems if the disrupted feature is observed on repeated individuals within the farm.

What is the potential hazard?

Shellfish in poor condition, outside of their normal seasonal changes, indicate that the growing environment is inadequate for maintaining the vitality of the animal. While poor condition is not a hazard in itself, it is an indicator of suboptimal growing conditions or other negative factors on the farm. Poor condition may result in lower growth rates and, often, increased mortality. Recognizing the reduced condition of the farmed shellfish provides an early warning of things to come and allows the farmer time to analyze the problem and apply a corrective action before losing some proportion of the crop.

Hazard management:

- Measure growth on a regular basis. Use calipers to measure length of a few dozen animals, or one can use a calibrated bucket to measure volume increases to compare growth between treatments or to assess the effects of different gear types or stocking densities.

- Evaluate the condition of the crop on a regular basis. Shuck a few animals and observe meat appearance to assess the overall condition of the animal, provided the farmer has an awareness of what the normal production cycle is on their farm. For example, thin watery meats in September are often a giveaway that there may be a disease problem on the farm. Thin meats when they should be full could indicate overstocking, poor fouling control, poor food conditions or disease.
- When in doubt, contact the local aquatic animal health professional (Appendix 3)

ANIMAL GRADING AND CONTAINMENT

In hatchery, nursery, and growout culture, shellfish are separated using simple sieves or other grading equipment. In the hatchery, shellfish are often retained in a culture unit with a mesh bottom that allows for simultaneous animal retention and water flow. These devices must be designed to retain the appropriate size shellfish, maintained to ensure it functions as required, and cleaned to ensure the appropriate flow.

Various mesh sizes are available for shellfish aquaculture. Mesh sizes are often described as the length of one side of a mesh square or the diameter if the mesh configuration is round. In the case of square or diamond meshes, it is important to remember that the length of a side does not ultimately describe the size of the animal retained on the mesh as the diagonal dimension is larger than the side measurement. Therefore, measurement of the mesh size with a ruler or calipers is always recommended if there is any doubt as to the actual dimension of the mesh.

What is the potential hazard?

If shellfish are not matched to the retention gear (mesh) size appropriately, the potential for individuals to pass through the mesh or to retain too broad a range of sizes on the screen, in the case of size grading, can negate the reason for the activity. If the seed are too small for the mesh, they can pass through the mesh and be lost. If too broad a size range of individuals is retained in a culture gear, uneven growth due to competition for food or space resources may result. If the mesh is too small, then biofouling will be more prevalent, flow will be impeded and food limitation may result. Sieve screen sizes for larval retention based on shellfish species and development stage are noted in Table 1.

Another consideration is that within a population of shellfish, there is a range of sizes around the descriptive mean size. Therefore, if 2 mm clam seed is purchased, the actual sizes may range from 0.75 mm to 3.25 mm, depending on how carefully the seed provider size-graded the lot before it was sold. Also, if the size of the shellfish seed is too closely matched to the size of the mesh, the seed may actually grow into the mesh making the two inseparable forcing the farmer to either kill the shellfish or tear the mesh (or both).

Hazard management:

- Inspect and repair all gear on a routine basis.
- Ensure that the containment mesh size is appropriate to retain the size of seed targeted for holding. Follow guidelines when available (Table 1) and measure the actual mesh prior to use.
- Inspect the seed and measure to ensure that they are matched to the mesh size.
- Ask the seed supplier what size sieve, or sieves, the seed was graded on in the hatchery.
- Sieve the seed going into a container on a sieve size that is larger than the mesh of the container it is going into. This will greatly reduce fall throughs in the containers (mesh bags).

Table 1. Screen sizes for larval retention based on shellfish species and development stage.

Generic			Eastern Oyster (*Crassostrea virginica*)		Northern Quahog (*Mercenaria mercenaria*)	
Screen Size		Larval size	Larval size	Development	Larval size	Development
side (µm)	diagonal (µm)	(µm)	(µm)	Stage	(µm)	Stage
35	49	50	45-62	egg	70-73	egg
50	71	75	68	D-stage	90-140	D-stage
80	113	120				
100	141	145			140-220	umboned
120	170	170	180	5 days	170-230	pediveliger
150	212	210			200-250	metamorphosis
180	255	255				
200	283	280	275-315	metamorphosis		
210	297	300			300	post-set
Reference:			Reference:		Reference:	
modified from Helm et al. 2004			Loosanoff & Davis 1963		Loosanoff & Davis 1963	
			Dupuy et al. 1977		Hadley & Whetstone 2007	

Bay Scallop (*Argopecten irradians*)		Softshell Clam (*Mya arenaria*)		Blue Mussel (*Mytilus edulis*)	
Larval size	Development	Larval size	Development	Larval size	Development
(µm)	Stage	(µm)	Stage	(µm)	Stage
50-65	egg	68-73	egg	68-70	egg
100	D-stage; 2 day	93	D-stage	70-110	trochophore
125	D-stage; 4 day	109.5	2 days	110-185	D-stage
150	umboned	120	5 days		
175	pediveliger	170-228	metamorphosis	185-260	umboned
200	metamorphosis				
				260	metamorphosis
Reference:		Reference:		Reference:	
Loosanoff & Davis 1963		Loosanoff & Davis 1963		Loosanoff & Davis 1963	
Widman et al. 2001		Buttner et al. 2010		Newell 1989	

References:

Buttner. J.K., Weston, S., Beal, B.F., 2010. Softshell clam culture: hatchery phase, broodstock care through seed production. USDA Northeastern Regional Aquaculture Center, Publication No. 202-2010.

Dupuy, J.L., Windsor, N.T., Sutton, C.F., 1977. Manual for design and operation of an oyster hatchery. Spec. Rep. Appl. Mar. Sci. Ocean Eng., No. 142. Virginia Institute of Marine Science, Gloucester Point, Virginia.

Hadley, N.H., Whetstone, J.M. 2007. Hard clam hatchery and nursery production. USDA Southern Regional Aquaculture Center, Publication 4301.

Helm, M.M., Bourne, N., Lovatelli, A., 2004. Hatchery culture of bivalves. A practical guide. FAO Fisheries Technical Paper. No. 471. Rome, FAO.

Loosanoff, V.L., Davis, H.C., 1963. Rearing of bivalve mollusks. Advances in Marine Biology, 1, Academic Press Ltd, London: 1–136.

Newell, R.I.E., 1989. Species profiles: life histories and environmental requirements of coastal fishes and invertebrates (North and Mid-Atlantic)--blue mussel. U.S. Fish and Wildlife Service Biological Report No. 82(11.102). U.S. Army Corps of Engineers, Technical Report No. EI-82-4.

Widman, J.C., Choromanski, J., Robohm, R.A., Stiles, S., Wikfors, G.H., Calabrese, A., 2001. Manual for Hatchery Culture of the Bay Scallop. Connecticut Sea Grant, Publication No. CTSG-01-03.

STOCKING DENSITY

One of the primary decisions a shellfish farmer must make for their nursery or grow-out system is how many seed should be stocked in the culture unit. The number of animals in a culture unit (stocking density) is a crucial decision because if stocked at too low a density then the number of culture units required increases (i.e. increased gear cost); however, if stocked too densely the growth rate will be compromised (i.e. reduced production). This results in a decrease in profitability for the farm.

What is the problem?

There are two mechanisms by which stocking density may affect production, food limitation, or stress from overcrowding.

Shellfish growth is directly related to food availability, which in turn is a function of food particle concentration in the water and the flow rate of the water past the animal (i.e. food flux). Should too many animals be stocked into culture gear, water flow through the unit will be disrupted. In addition, having too many feeding individuals in the unit, they may reduce the food particle concentration for those animals downstream within the gear. Either or both events result in less food availability per individual animal in the culture unit thereby impacting growth rate of all of the animals. Typical stocking densities for larval rearing and seed are listed in Table 2 and Table 3, respectively.

The other factor associated with overcrowding is physical interference among the animals because they are held too closely. Overcrowding can also increase the potential for disease to spread through a population. The end result is a crop that does not perform as well as a less crowded cohort leading to reduced growth or increased susceptibility to other stressors.

Hazard Management:

The primary means to detect lower productivity due to high stocking density is through careful observation, a familiarity with expected growth rates, and the capacity to evaluate different stocking densities. One of the first signs of food limitation is increased variance in growth rates leading to a wide range of sizes. Starting with locally acceptable stocking rates, the farmer should set up small experiments to evaluate variations above and below the initial stocking rate to measure the local site's capacity to support growth. Small differences in bottom topography will impact current speed that will have a large impact on food availability and optimal stocking density. Different sized seed will need to be evaluated separately because they have different metabolic needs and growth rates. Because not all locations support the same stocking density, it is important for the farmer to adjust their farming operations to accommodate the conditions on their farm.

Table 2. Common stocking densities of bivalve larvae in a hatchery culture system (individual larvae/mL).

Development Stage	Eastern Oyster (*Crassostrea virginica*)	Northern Quahog (*Mercenaria mercenaria*)	Bay Scallop (*Argopecten irradians*)	Softshell Clam (*Mya arenaria*)	Blue Mussel (*Mytilus edulis*)
Static System					
fertile egg	15-20	20-30	15-30	15-25	15-25
trochophore	15-20	15-25		15-25	15-25
D-stage	10-20	5-10	10-20	10-20	10-20
umboned	5-10		5-10		
pediveliger	2-5	1-2	2-5	3-6	
setting	100/cm^2 with microcultch in a downweller or 100/shell for spat on shell		1-2/ml using spat bags or 100-500/cm^2 in downweller		
post-set		575/cm^2 @ 600 µm		100/cm^2 @ 500 µm	
References:	Helm et al. 2004	Helm et al. 2004	Helm et al. 2004	Helm et al. 2004	Helm et al. 2004
	Wallace et al. 2008	Castagna & Kraeuter 1981	Widman et al. 2001	Buttner et al. 2010	
		Hadley & Whetstone 2007	Surier et al. 2010		
Flow-through System					
Generic	15-100				
4 time's static density	20-50				
References:	Helm et al. 2004				
	Reiner 2011				

Note: The stocking densities listed here are a general starting point. These numbers may require adjustment as the farmer gains more experience with their specific site.

References:

Buttner. J.K., Weston, S., Beal, B.F., 2010. Softshell clam culture: hatchery phase, broodstock care through seed production. USDA Northeastern Regional Aquaculture Center, Publication No. 202-2010.

Castagna, M., Kraeuter, J.N., 1981. Manual for growing the hard clam *Mercenaria*. Special Report No. 249, Virginia Institute of Marine Science, Gloucester Point, Virginia.

Hadley, N.H., Whetstone, J.M. 2007. Hard clam hatchery and nursery production. USDA Southern Regional Aquaculture Center, Publication 4301.

Helm, M.M., N. Bourne, and A. Lovatelli. 2004. Hatchery culture of bivalves. A practical Guide. FAO Fisheries Technical Paper. No. 471. Rome, FAO.

Reiner, S.L., 2011. Evaluating the use of flowthrough larval culture for the Eastern oyster, *Crassostrea virginica*. Virginia Institute of Marine Resources. Masters Thesis.

Surier, A., Karney, R., Leavitt, D., 2010. Hatchery culture of the bay scallop. USDA Northeast Regional Aquaculture Center, Publication No. 214-2010.

Widman, J.C., Choromanski, J., Robohm, R.A., Stiles, S., Wikfors, G.H., Calabrese, A., 2001. Manual for Hatchery Culture of the Bay Scallop. Connecticut Sea Grant, Publication No. CTSG-01-03.

Wallace, R.K., Waters, P., Rickard, F.S., 2008. Oyster hatchery techniques. USDA Southern Regional Aquaculture Center, Publication No. 4302.

Table 3. Common stocking densities of bivalves in nursery and growout systems (individuals/ft^2).

Culture Stage	Eastern Oyster (*Crassostrea virginica*)	Northern Quahog (*Mercenaria mercenaria*)	Bay Scallop (*Argopecten irradians*)	Softshell Clam (*Mya arenaria*)
Nursery				
Land-based Raceway		1,400 @ 8mm		
		700 @ 10-12mm		
Field Raceway		500-625 @ 7-8mm		
Elevated off bottom in bags	4,500 @ 2.5mm	5,000 - 15,000 @ 1-2mm	50 @ 25mm	
	930 @ 5mm			
	370 @ 8mm			
Suspended culture		n/a	100 @ <28	
Floating culture		n/a	3,000 @ 2-3mm	
			300 - 500 @ 25mm	
Upweller	1,600 @ 25mm	312,000 @ 1mm		
		125,000 @ 1.5mm		
		35,000 @ 2.5-3.3mm		
		27,000 @ 3.9mm		
		19,500 @ 6mm		
		9,000 @ 8.3mm		
Growout				
Free plant on bottom	8 - 15	75 @ 8-50mm (This number represents and average; some farmers plant small seed (~8mm under predator netting at much higher densities ~275 but then they are thinned out in year two and seeded at a density of ~40-50.		31 - 62 @ 8-10mm
				20 - 50 @ 10-15 mm
bottom bags	222 @ 23-49mm	125 (soft bag)	25 - 50	
	46 - 70 @ 49-63mm	67 - 111 (ADPI suitcase)		
	46 @ 63-75mm			
bottom tray	75 @ 40mm			
	64 @ 50mm			
	56 @ 63mm			
	50 @ 75mm			
Elevated off bottom in bags	44 - 55 @ 75mm			
Suspended culture			25 @ <28mm	
Floating culture in bags	225 @ <31mm		30 - 50	
	110 @ 31-50mm			
	55 @ 51-65mm			
	44 @ >65mm			

Note: The planting densities listed here are a general starting point. These numbers represent a range based on various farm field trials and may require adjustment as the farmer gains more experience with their specific site.

HANDLING AND TRANSPORT

Handling and transport are routine for a shellfish farm, ranging from inoculating micro-algal culture tubes, to importing seedstock, to harvesting and transporting the final shellfish crop to market. Any activity that removes the organism from its normal environment coupled with the physical manipulation of the organism during transport will induce stress in the population.

The majority of shellfish are sedentary organisms that do not react well to being handled. Both growth and survival can be impacted. For example, moving a crop of bay scallops can cause the animal to lay down a noticeable growth ring check mark (indicative of a disruption in shell growth).

Care must be exercised to reduce those levels of stress associated with handling and transport activities.

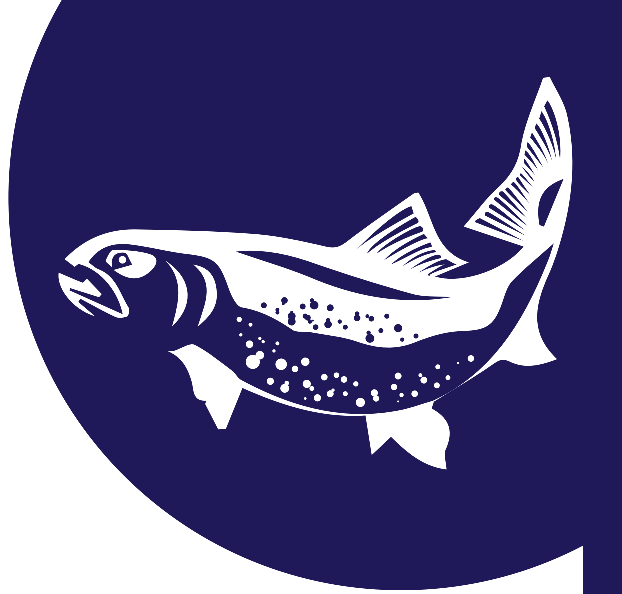
Why is it a Problem?

This combination of deleterious actions heightens the level of stress and makes plants and animals more prone to physical or stress induced damages, such as shell damage from improper handling, reduced growth due to disruption in feeding or removal from oxygenated water, heightened susceptibility to opportunistic or obligatory pathogens that may be lurking in the environment, and other responses in the organism. The end result is a loss of productivity that may impact the crop long after the actual handling or transport event took place.

Hazard Management:

Minimize factors that may cause stress during handling/transport:

- extended holding periods out of the water
- overcrowding
- rapid changes in temperature (to the degree allowable considering state and federal guidelines)



CHAPTER 4

Finfish Aquaculture in the Northeastern U.S.

Overview

Various species of freshwater and marine finfish are cultivated in the northeastern U.S. The major commercial finfish species include:

- salmonids (various species of trout and salmon)
- tilapia (*Oreochromis niloticus)*
- bass (the largemouth bass *Micropterus salmoides*, striped bass *Morone saxatilis* and hybrids)
- various species of perch (especially yellow perch *Perca flavescens*)
- minnows (the common shiner *Luxilus cornutus*, the fathead *Pimephales promelas*, the golden shiner *Notemigonus crysoleucas*)
- white sucker (*Catostomus commersonii*)
- koi (*Cyprinus carpio*)

Marine finfish culture is relatively new and the industry is currently focused on grow-out of the:

- Atlantic cod (*Gadus morhua)*
- black sea bass (*Centropristis striata)*
- striped bass (*Morone saxatilis)* and hybrids (also grown in freshwater)

This chapter includes production-related risks for all of these species with the exception of the barramundi Lates calcarifer, which is currently produced by a single, large-scale farmer, but is not predicted at this time to expand to other farmers.

This chapter includes generalized finfish morphology and the life cycle (Figure 1-2), as well as images of cultivation (Figures 3-7). Detailed information on hatchery production and growout practices are documented in a number of references listed at the end of this manual (Appendix 5).

Finfish Morphology and Life Cycle

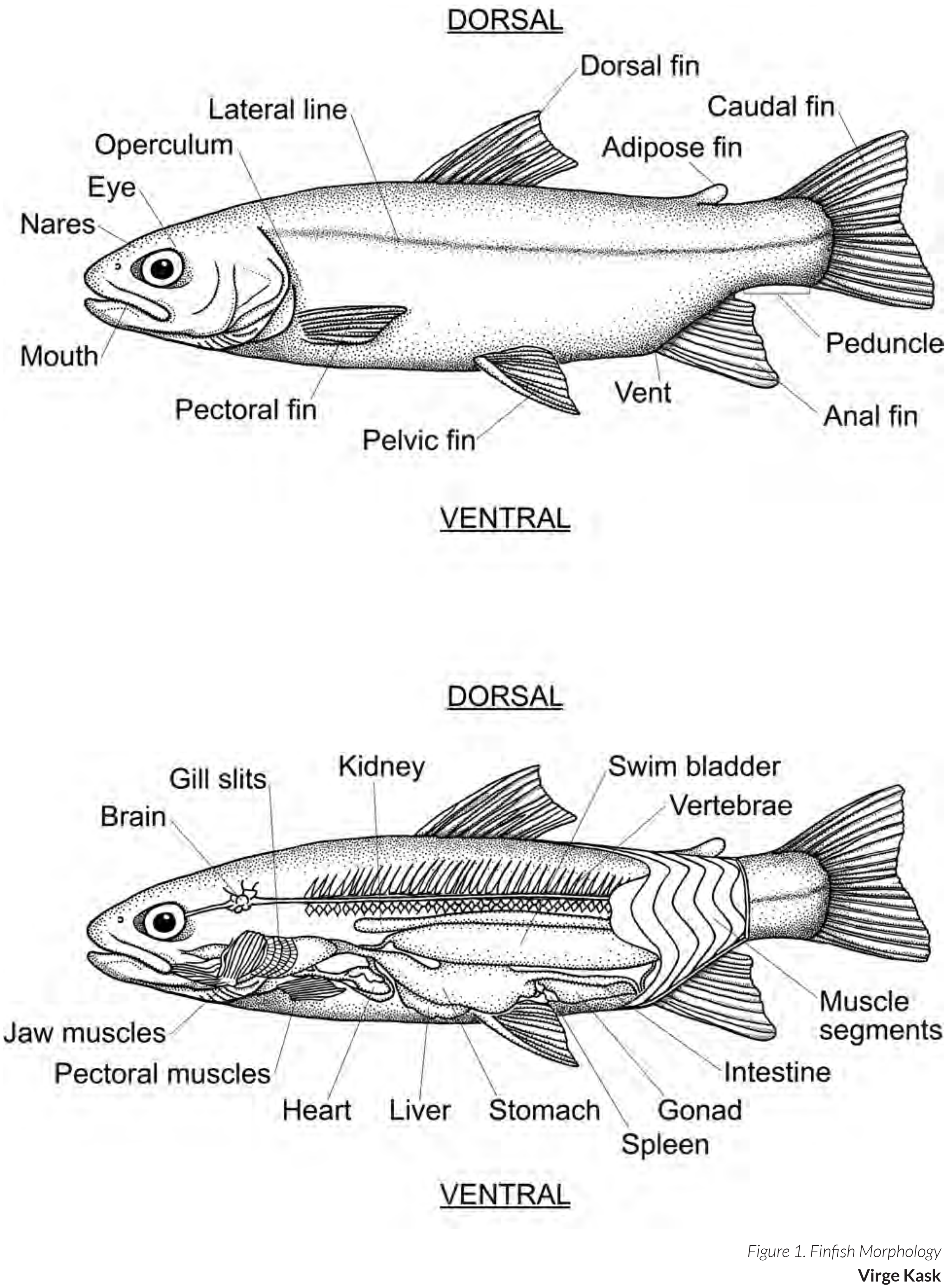


Figure 1. Finfish Morphology
Virge Kask

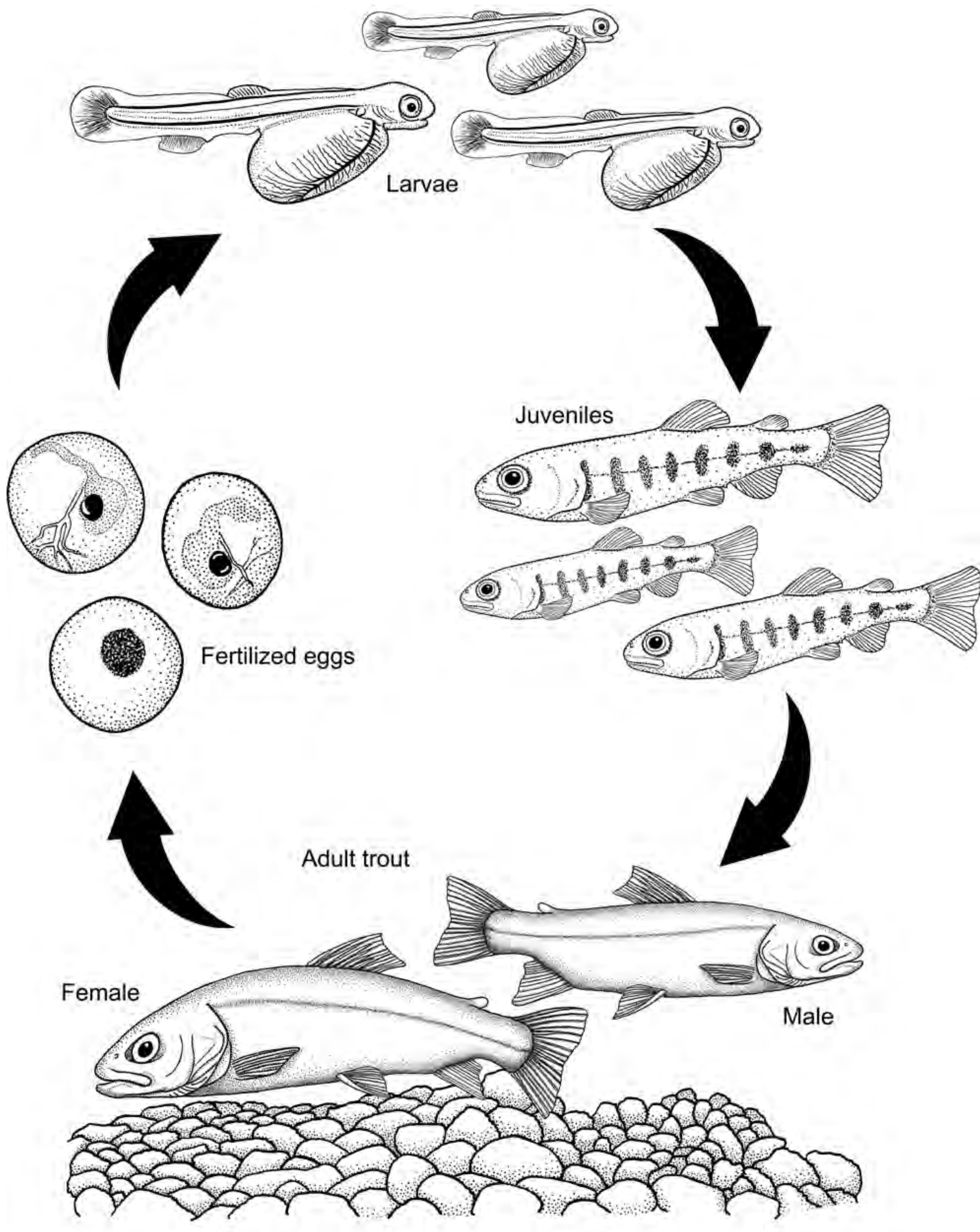


Figure 2. Finfish Life Cycle
Virge Kask

Finfish Cultivation Systems

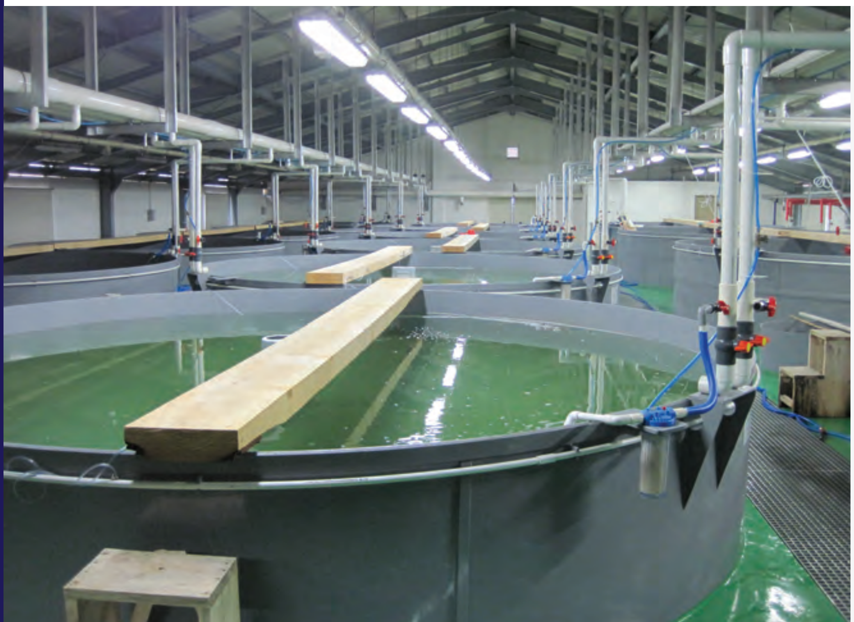


Figure 3. Flow through tanks
Michael Pietrak

Figure 4. Raceways
Michael Pietrak

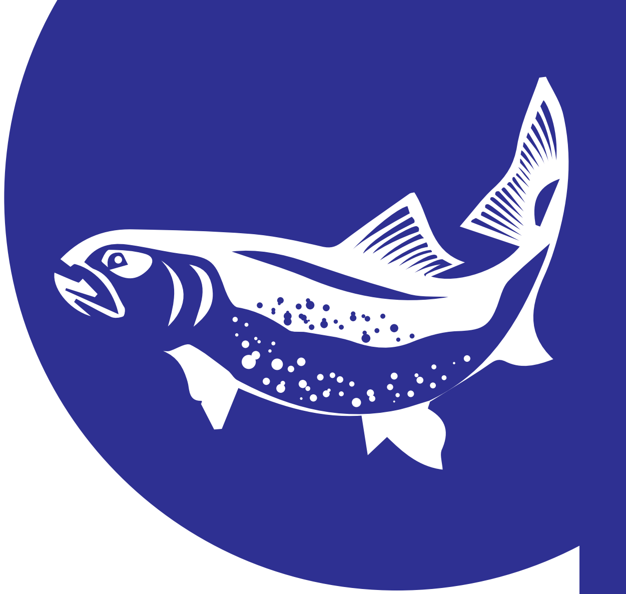
Figure 5. Recirculating systems
Michael Pietrak

Figure 6: Net Pens
Michael Pietrak

Figure 7. Fish ponds
Michael Pietrak

Potential Finfish Production Hazards

Environmental Conditions
Biofouling Organisms
Predators
Diseases and Parasites
Invasive Species
Operational Procedures



Environmental Conditions

The quality of the water that fish live in defines the quality of their environment. Not only does water quality impact fish, the fish impact water quality. Poor water quality leads to environmental conditions that stress fish and can result in significant losses, either directly or indirectly. While the drivers that result in a fish loss will likely change from event-to-event, the underlying concerns are often the same. Fish rely upon their environment to supply them with oxygen, to maintain their body temperature, to provide physical support, for food, to facilitate osmoregulation, and to mitigate their waste products. Fish modify their aquatic environment by removing oxygen and venting wastes (i.e., feces, ammonia, carbon dioxide). As the number of fish maintained in a system increases, their impact on water quality is magnified, as is the need to manage the system actively. Therefore, maintaining the aquatic environment within accepted tolerance limits is critical to the success of any aquaculture operation.

Management of the environmental conditions within a production system will vary depending upon the type of production system in question, stocking density, and environmental parameter in question. Critical issues to keep in mind include:

- Systems should be designed and constructed to facilitate monitoring and management of water quality;
- Identify, monitor, and manage critical water quality parameters at appropriate intervals and locations within the system;
- Check for and prevent the introduction of contaminants (e.g., pesticides and herbicides in ponds, leachates from new tanks, heavy metals);
- Minimize water quality fluctuations and maintain suitable limits to avoid stressing or killing fish;
- Integrate corrective measures into the system and management protocols to facilitate remediation when needed;
- Maintain good detailed records to document conditions and aid with improved management strategies

Environmental parameters that should be considered for farming major aquaculture species are found in Tables 1-7.

Potential hazards:

temperature
salinity
dissolved oxygen
pH
carbon dioxide
metabolic wastes
contaminants
harmful algal blooms

Figure 1: Routine monitoring and management of water quality can prevent many environmental problems.
Joseph Buttner

TEMPERATURE

Mode of Action: direct, lethal temperature; indirect, suboptimal temperature

Description of Hazard: All biological and chemical processes in an aquaculture operation are influenced by temperature. Fish adjust their body temperature and metabolic rate by moving into cooler or warmer water. Each species has a preferred or optimum temperature range where it grows best. At temperatures above or below optimum, fish growth is reduced and mortalities may occur at extreme temperatures. Similarly, the nitrification rate mediated by bacteria in the biological filter is impacted by temperature, increasing as temperature increases within biological limits.

Hazard Management:

- Avoid temperature related problems by proper site selection
- Select species that will tolerate the anticipated culture temperatures: warm, cool, cold.
- Select appropriate system for the species. Closed systems (e.g., RAS) offer more opportunity for control and management than do semi-closed (e.g., cages, flow-through) or open (e.g., ponds) systems.
- Consider costs associated with temperature management and the needs of bacteria in RAS biofilters. Temperature is directly correlated with the ability of the bacteria to convert nitrogenous wastes. Heaters and chillers can be used to adjust temperatures, but their use will increase operational expenses and size limitations preclude larger scale application.

SALINITY

Mode of Action: direct; stressful or lethal

Description of Hazard: The total concentration of dissolved ions in the water is its salinity. Freshwater fish exhibit a range in salinity tolerance. Many commercially important freshwater species (e.g., channel catfish, *Ictalurus punctatus*; largemouth bass, *Micropterus salmoides*; tilapia, *Oreochromis* sp.) survive and grow well in slightly saline water. After they smolt, salmon and trout can tolerate salt water. Marine fishes also exhibit a range in salinity tolerance. Flounder (*Paralichthys dentatus*; *Pseudopleuronectes americanus*), striped bass (*Morone saxatilus*) and other estuarine species tolerate salinity shifts; other marine fishes are less tolerant such as post smolt salmonids and Atlantic cod (*Gadus morhua*). Salinity not only affects osmoregulation, it also influences the toxicity of un-ionized ammonia. Increasing the salinity of the water can decrease the toxicity of ammonia.

Hazard Management:

- Adjust system design, operation, and water supply to maintain optimal or acceptable salinities.
- Measure salinity and determine the appropriateness of the water source during the planning stage of the operation. Farmers should consider the potential effects of storm events that may dramatically change salinity levels.
- Consider closed systems which offer more opportunity for manipulation than do semi-closed or open systems.
- Select species that will tolerate the anticipated culture salinity: fresh, brackish or salt water.
- Adjust salinity to suit the needs of the culture species.

DISSOLVED OXYGEN

Mode of Action: extremely low or high levels can cause stress and can be lethal

Description of Hazard: Dissolved oxygen (DO) in a culture system must be maintained above levels considered stressful to fish. Warm water fish can tolerate lower DO concentrations than cold water fish. DO tolerance for cool water fish is between that exhibited by warm and cold water fishes. Excessively low DO concentrations, less than 1 - 2 mg/L (milligram per liter, used interchangeably with parts per million or ppm), will kill fish. Prolonged exposure to low, nonlethal levels of DO constitutes a chronic stress and will cause fish to stop feeding, reduce their ability to convert ingested food into fish flesh, and make them more susceptible to disease. In northern climes, iced-over ponds with deep snow cover can experience winter kill as oxygen is depleted since diffusion and photosynthesis can't replenish DO used by fish and other organisms in winter.

Super saturation of oxygen (and nitrogen) can cause gas bubble disease, which is the accumulation of gas in the blood or tissues. Gas bubble disease can result in erratic behavior, stress, and death.

Hazard Management:

- Design system and operation to maintain optimal or acceptable DO concentrations.
- Maintain DO >3.0 mg/L and >5.0 – >7.0 mg/L for warm and cold water fish, respectively. This is species-specific (see Tables 1-7).
- Maintain supplemental aeration in open and semi-closed systems and oxygenation in closed systems to avoid oxygen deficiencies and resultant stress/mortalities.
- Winter kill in northern ponds may be avoided with a wind mill connected to a compressor, which maintains a refuge of iceless area.

pH

Mode of Action: direct, lethal high or low pH; indirect, increased solubility of heavy metals and increased toxicity of metabolic wastes

Description of Hazard: The concentration of bases and acids in the water determines its pH. A low pH is acidic and a high pH is basic; a pH of 7 is neutral. Fish survive and grow best in waters with a pH between 6 and 9. If pH is outside this range, fish growth is reduced. At a pH level below 4.5 or above 10, mortalities may occur. As pH increases, the proportion of the Total Ammonia Nitrogen (TAN) in the toxic form, NH_3, increases. As pH decreases, the solubility of heavy metals and, therefore, their toxicity also increases. Portable, hand-held pH meters are reasonably priced and facilitate quick, accurate measurements.

Hazard Management:

- Design system and operation to maintain optimal or acceptable pH levels.
- Low pH is difficult or impossible to correct in open systems.
- In ponds and RAS, pH may be managed by maintaining a suitable alkalinity, the buffering capacity of culture water expressed as mg/L calcium carbonate ($CaCO_3$). As carbon dioxide levels fluctuate, so too does the pH of the water. The magnitude of pH shift is determined by the buffering capacity of the water or its ability to absorb acids and bases. A suitable range for alkalinity is 20 to 300 mg/L.

- Alkalinity >300 mg/L does not adversely affect fish survival, but it does interfere with action of certain commonly used chemicals (e.g., copper sulfate) and reproduction in several fishes, most notably salmonids.
- Alkalinity <20 mg/L fails to provide adequate buffering capacity to maintain a consistent, safe pH.
- Alkalinity and, therefore pH, can be increased by adding agricultural limestone to ponds or sodium bicarbonate to RAS.

CARBON DIOXIDE

Mode of Action: direct, lethal high concentrations of carbon dioxide in freshwater systems

Description of Hazard: An excess of carbon dioxide in the water can cause fish to become fatally sedated. In addition, free carbon dioxide can decrease the pH of the water. Only when using groundwater, transporting fish at high densities, or in RAS with oxygenation (as opposed to aeration) are carbon dioxide problems likely to develop. At high concentrations, carbon dioxide causes fish to lose equilibrium, become disoriented, and possibly die.

Hazard Management:

- Prevention or avoidance of carbon dioxide-related problems is preferable to correction, so system design and operation should be adjusted, if possible, to avoid or reduce carbon dioxide to safe concentrations.
- Test groundwater before use and aerate, if necessary, to reduce carbon dioxide to acceptable levels.
- Careful planning, aeration or oxygenation, and buffering of water will keep carbon dioxide at acceptable levels when large numbers of fish are hauled extended distances.
- Strip carbon dioxide in RAS if necessary and when the system is oxygenated or if stocking densities exceed 0.5 lbs. fish/gallon of water. This can be achieved with a stripping tower, surface or diffused aerator.

METABOLIC (NITROGENOUS) WASTES

Mode of Action: direct, lethal concentrations of ammonia and nitrites; indirect, sublethal levels that are stressful

Description of Hazard: Metabolic wastes include ammonia (NH_3), nitrites (NO_2^-), and nitrates (NO_{3-}). Most fish and freshwater invertebrates excrete ammonia as their principle nitrogenous waste. In culture systems, toxic ammonia (NH_3) co-exists with the nontoxic ammonium ion (NH_4^+) and their collective sum is expressed as Total Ammonia Nitrogen (TAN). The amount of TAN in the toxic form (NH_3) increases dramatically as pH goes above 7.5 and less so as temperature increases. Fish continuously exposed to more than 0.02 mg/L of NH_3 may exhibit reduced growth and increased susceptibility to disease. NH_3 concentrations greater than approximately 0.02 mg/L can be lethal for some species, with cold-water fish generally being more susceptible to NH_3 toxicity than warm water fish. When fish are cultured intensively and fed protein-rich feeds they can produce high concentrations of ammonia in the water. Ammonia and other metabolic wastes are gradually removed by natural processes in ponds or through the use of biological filters in recirculating systems. Ammonia is removed by bacteria that initially convert it into nitrite and subsequently into nitrate. Nitrite (NO_2^-) is toxic to fish at approximately 1 mg/L (though this varies greatly by species, and depending on the species, toxicity may vary with the chloride content of the water) (see Tables 1-7).

Exposure to high nitrite levels causes “brown blood” disease, which interferes with oxygen transport. NO_2^- concentrations of 0.5 mg/L have been shown to reduce growth and adversely affected fish. Fish can tolerate nitrate (NO_3^-) to several hundred mg/L.

Salinity influences the toxicity of un-ionized ammonia. Increasing the salinity of the water can decrease the toxicity of ammonia.

The time that it takes to get the nitrogen in the system in balance is dependent upon temperature, and may take weeks (at high temperature) to months (at low temperature). The only way to be sure is to feed (add ammonia) the system and monitor the nitrogen levels daily. Simple and inexpensive nitrogen test kits are available from most aquarium supply stores or aquaculture vendors.

Hazard Management:

- Prevention or avoidance of excessive nitrogenous wastes in the water is preferable to correction, so system design and operation should be adjusted, if possible, to maintain nitrogenous wastes at or below safe concentrations.
- Removal or detoxification of ammonia is facilitated by providing and maintaining an optimal environment for the appropriate bacteria (pH between 7 and 9; temperature approximately 75 - 85 °F).
- In RAS properly sizing biological filters and paying attention to system maintenance (e.g., cleaning filters, removing dead fish, monitoring feeding rates) are the best ways to avoid the buildup of nitrogenous wastes in the system.
- To mitigate possible NO_2^- toxicity, salt (NaCl) can be added at 10 - 20 mg/L; only agricultural grade salt should be used as table salt destined for direct human use often contains other substances that are detrimental to fish.
- In ponds, the best management approach, should NH_3 concentrations become problematic, is to cease feeding temporarily. If NO_2^- concentrations become a problem, agricultural grade salt can be added at 10 mg/L for each 1 mg/L NO_2^-.
- If a problem arises in an extreme situation, water exchanges can help flush excess nitrogenous wastes from the system, but care must be taken, as replacing a large volume of the system water may expose culture animals to other stresses.

CONTAMINANTS

Mode of Action: direct, lethal concentration; indirect, sublethal stressful concentrations

Description of Hazard: Water can become contaminated with any number of substances that can be harmful to fish. These can include pesticides, hydrocarbons, heavy metals and industrial solvents. Once introduced to an aquaculture system, there is little that can be done to mitigate their effects, so careful attention must be paid to the quality and sources of water used.

Hazard Management:

- Prevention or avoidance of contaminants in aquaculture is preferable to correction, so system design and operation should be adjusted, if possible, to ensure culture water is contaminant-free.
- Water used to culture fish should be obtained from clean sources. In some cases this clean water can dilute concentrations of potentially harmful compounds (e.g., ammonia), add dissolved oxygen, and carry wastes out of the system.

- If biological contamination is suspected, water to RAS and flow-through systems should be filtered and sterilized prior to use.
- Avoid pond designs that allow surface water runoff.

HARMFUL ALGAL BLOOMS (HAB)

Mode of Action: direct, release of toxins; indirect, reduce light penetration and decrease dissolved oxygen in pond, stress fish during harvest as seine becomes clogged

Description of Hazard: Freshwater and particularly marine algae can release toxins into the water that can stress or kill fish. The threat increases as abundance of problematic algae increase. Regardless of algal type, if algae become too abundant, such as in a phytoplankton bloom, light penetration decreases resulting in reduced photosynthetic activity and less dissolved oxygen in the water. Similarly, during seine harvest of ponds if filamentous algae are excessively abundant they can clog the net, complicating harvest and increasing stress to fish.

Hazard Management:

- Prevention or avoidance of harmful algal bloom in aquaculture is preferable to correction, so pond or cage location and management practices should be adjusted to avoid locations where blooms are common and to avoid adding excessive nutrients to a pond.
- If algal abundance increases they can be cropped by adding phytoplanktonic feeding organisms, removed by flushing a pond or relocating a cage, or treated with approved herbicides.
- Water used to culture fish should be obtained from clean sources. In some cases this clean water can dilute concentrations of potentially harmful compounds (e.g., ammonia), add dissolved oxygen, and carry wastes out of the system.
- To avoid biological contamination, water to RAS and flow-through systems should be filtered and sterilized prior to use.

Figure 2. Algae bloom in a fish pond.

Nathan Stone

References and Further Reading:

Boyd, C.E., Tucker, C.S., 1998. Aquaculture Water Quality Management. Boston: Kluwer Academic Publishers.

Brunson, J.W., Stone, N., Hargreaves, J., 1999. Fertilization of fish ponds. USDA Southern Regional Aquaculture Center, SRAC Publication No. 471.

Buttner, J.K., Soderberg, R.W., Terlizzi, D.E., 1993. An introduction to water chemistry in freshwater aquaculture. USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 170.

Durborow, R.M., Crosby, D.M., Brunson, M.W., 1997a. Nitrite in fish ponds. USDA Southern Regional Aquaculture Center, SRAC Publication No. 462.

Durborow, R.M., Crosby, D.M., Brunson, M.W., 1997b. Ammonia in fish ponds. USDA Southern Regional Aquaculture Center, SRAC Publication No. 463.

Good, C., Davidson, J., Welsh, C., Brazil, B., Snekvik, K., Summerfelt, S., 2009. The impact of water exchange rate on the health and performance of rainbow trout *Oncorhynchus mykiss* in water recirculation aquaculture systems. Aquaculture. 294(1-2), 80 - 85.

Hargreaves, J.A., Tucker, C.S., 2004. Managing ammonia in fish ponds. USDA Southern Regional Aquaculture Center, SRAC Publication No. 4603.

Kristensen, T., Atland, A., Rosten, T., Urke, H.A., Rosseland, B.O., 2009. Important influent - water quality parameters at freshwater production sites in two salmon producing countries. Aquacultural Engineering. 41(2), 53-59.

Lewis Jr., W.M., Morris, D.P., 1986. Toxicity of nitrite to fish: a review. Transactions of the American Fisheries Society. 115, 183-195.

Masser, M. P., Murphy, T.R., Shelton, J.L., 2013. Aquatic weed management control: herbicides. USDA Southern Regional Aquaculture Center, SRAC Publication No. 0361.

Morgan, E.R., Brunson, M.W., 2002. Toxicities of agricultural pesticides to selected aquatic organisms. USDA Southern Regional Aquaculture Center, SRAC Publication No. 4600.

Scarfe, D., Cheng-Sheng, L., O'Bryen, P., 2004. Aquaculture Biosecurity: Prevention, Control and Eradication of Aquatic Animal Disease. World Aquaculture Society, Baton Rouge, Louisiana.

Shelton, J.L., Murphy, T.R., 1989. Aquatic weed management: control methods. USDA Southern Regional Aquaculture Center, SRAC Publication No. 360.

Stickney, R.R., 2009. Aquaculture an Introductory Text 2nd Edition. CABI, Cambridge, MA. 304 pp.

Svobodová, Z., Lloyd, R., Máchová, J., Vykusová, B., 1993. Water Quality and Fish Health. EIFAC Technical Paper. FAO No. 54. Rome, Italy.

Swann, L., 1997. A fish farmer's guide to understanding water quality. Illinois - Indiana Sea Grant Program, Publication No. IL-IN-SG-97-2.

Sweilum, M.A., 2006. Effect of sublethal toxicity of some pesticides on growth parameters, hematological properties and total production of Nile tilapia (*Oreochromis niloticus* L.) and water quality of ponds. Aquaculture Research. 37(11), 1079-1089.

Tucker, C.S., D'Abramo, L.R., 2008. Managing high pH in freshwater ponds. USDA Southern Regional Aquaculture Center, SRAC Publication No. 4604.

Tucker, C.S., Hargreaves, J.A., 2008. Environmental Best Management Practices for Aquaculture. World Aquaculture Society, Baton Rouge, Louisiana.

Whyte, J.N.C., Haigh, N., Ginther, N.G., Norma, G., Keddy, L.J., 2001. First record of blooms of *Cochlodinium* sp. (Gymnodiniales, Dinophyceae) causing mortality to aquacultured salmon on the west coast of Canada. Phycologia. 40(3), 298-304.

Wurts, W.A., Durbow, R.M., 1992. Interactions of pH, carbon dioxide, alkalinity and hardness in fish ponds. USDA Southern Regional Aquaculture Center, SRAC Publication No. 464.

Table 1. Environmental parameters to consider for salmonid culture.

Culture Stage	Overall Conditions					
	Temperature (°C)	DO (mg/L)	Salinity	pH	Ammonia (mg/L)	Nitrite (ppm)
Spawning	0 - 26					
Egg Development	0.5 - 12	6 to saturation		>5		
Parr	0 - 26	5 to saturation		>5		
Smolts - Adults	0 - 22	5 to saturation	0-35			

Culture Stage	Optimum Conditions					
	Temperature (°C)	DO (mg/L)	Salinity	pH	Ammonia (mg/L)	Nitrite (ppm)
Spawning	0 - 10					
Egg Development	<8	8 to saturation		6.6 - 6.8		
Parr	15 - 19	8 to saturation		6.7 - 8.5	<0.2	<0.5
Smolts - Adults	5 - 17	8 to saturation	30-35	6.7 - 8.5	<0.2	<0.5

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References

Brannon, E.L., 1991. Rainbow Trout Culture. In Stickney, Robert R. (Ed.) Culture of Salmonid Fishes. CRC Press Inc. Boca Raton, Florida.

Danie, D.S., Trial, J.G., Stanley, J.C., 1984. Species profiles: Life histories and environmental requirements of coastal fish and invertebrates (North Atlantic) -- Atlantic salmon. U.S. Fish and Wildlife Service FWS/OBS-82/11.22. U.S. Army Corps of Engineers, Technical Report No. EL-82-4.

Jones, M., 2004. Cultured Aquatic Species Information Programme. *Salmo salar*. Cultured Aquatic Species Information Programme. In: FAO Fisheries and Aquaculture Department [online]. Rome.

Table 2. Environmental parameters to consider Atlantic cod (*Gadus morhua*) culture.

Culture Stage	Overall Conditions				
	Temperature (°C)	DO (ppm)	Salinity	pH	Ammonia* (ppm)
Spawning	4-6	90-100	33-35	7.9-8.2	
Egg Development	3-8	90-10	33-36	7.9-8.1	
Juveniles	5-14	90-120	18-35	7.5-8.2	<0.03
Adults	3-14	85-125	16-35	7.6-8.2	<0.03

Culture Stage	Optimum Conditions				
	Temperature (°C)	DO (ppm)	Salinity	pH	Ammonia* (ppm)
Spawning	5	35		8-8.2	
Egg Development	5-7	35		8-8.2	
Juveniles	8-12	28-33		7.9-8.1	<0.02
Adults	7-10	28-35	3-10	7.9-8.1	<0.02

*Un-ionized ammonia

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References:

George Nardi, 2013, personal observations.

Table 3. Environmental parameters to consider for hybrid striped bass (*Morone saxatilis* x *M. chrysops*) culture.

Culture Stage	Overall Conditions				
	Temperature (°C)	DO (ppm)	Salinity	pH	Ammonia* (ppm)
Spawning	15-21 (white bass)				
Egg Development					
Juveniles		1.5-12	0-25	4.5-9.5	<0.2
Adults	4-33	1.5-12	0-25	4.5-9.5	

Culture Stage	Optimum Conditions				
	Temperature (°C)	DO (ppm)	Salinity	pH	Ammonia* (ppm)
Spawning	16-18 (white bass)				
Egg Development	16-20				
Juveniles		6-12		7-8.5	<0.1
Adults	27-27	6-12	3-10	7-8.5	<0.1

*Un-ionized ammonia

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References:

Anderson, R.O., Tave, D., 1993. Strategies and Tactics for Management of Fertilized Hatchery Ponds. Food Products Press.

Hodson, R.G., 1989. Hybrid Striped Bass- Biology and Life History, USDA Southern Regional Aquaculture Center, SRAC Publication No. 300.

McGinty, A.S., Hodson, R.G., 2008. Hybrid Striped Bass: Hatchery Phase, USDA Southern Regional Aquaculture Center, SRAC Publication No. 301.

Wood III, L.C., 2005. Striped bass and hybrid striped bass culture, in: Kelly, A.M., Silverstein, J. (Eds.), Aquaculture in the 21st Century, American Fisheries Society, Symposium 46, Bethesda, Maryland, pp. 339-353.

Table 4. Environmental parameters to consider for yellow perch (*Perca flavescens*) culture.

Culture Stage	Overall Conditions					
	Temperature (°C)	DO (mg/L)	Salinity	pH	Ammonia (ppm)	Alkalinity (ppm)
Spawning	6-12		0-2	6-8.5		
Egg Development	7-20		0-2	6-8.5		
Juveniles	10-30	>3.5 mg/L	0-5	6-8.5	<0.0125	>75
Adults	10-30	>3.5 mg/L	0-13	6-8.5	<0.0125	>75

Culture Stage	Optimum Conditions					
	Temperature (°C)	DO (mg/L)	Salinity	pH	Ammonia (ppm)	Alkalinity (ppm)
Spawning			0-2			
Egg Development	14		0-2			
Juveniles	19-24	> 5.0 mg/L	0-5	6.5-8.5	<0.0125	>75
Adults	24.7	> 5.0 mg/L	0-13	6.5-8.5	<0.0125	>75

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References:

Hart, S.D., Garling, D.L., Malison, J.A., (Eds.). 2006. Yellow perch, *Perca flavescens*, culture guide. North Central Regional Aquaculture Center, Culture Series #103. Iowa State University. Ames, Iowa.

Piavis, P.G., 1991. Yellow perch, *Perca flavescens*. Maryland Department of Natural Resources. Report. Tevensville, Maryland.

Table 5. Environmental parameters to consider for shiner, minnow, and white sucker culture.

Culture Stage	Overall Conditions				
	Temperature (°C)	DO (ppm)	Salinity	pH	Ammonia* (ppm)
Spawning	16-24				
Egg Development	16-25				
Juveniles	18-28	> 3		6-8.5	
Adults	16-28	>3	0-6	6-8.5	<1.25 (pH= 7)

Culture Stage	Optimum Conditions				
	Temperature (°C)	DO (ppm)	Salinity	pH	Ammonia* (ppm)
Spawning	20-22				
Egg Development	20-24				
Juveniles	22-26	>5	<1		
Adults	22-26	>5	<1		<0.2

*Un-ionized ammonia

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References:

Clemment, T., Stone, N., 2010. Golden shiner egg production during a spawning season. North American Journal of Aquaculture. 72, 272-277.

Guidice, J.J., Gray, D.L., Martin, J.M., 1982. Manual for baitfish culture in the south. University of Arkansas Cooperative Extension Service, Little Rock, Arkansas, Publication EC550-5M-8-82R.

Melandri, M., Stone, N., Lochmann, R., 2008. Effects of temperature on growth of golden shiners in aquaria. North American Journal of Aquaculture. 70, 452-458.

Murai, T., Andrews, J.W., 1977. Effects of salinity on the eggs and fry of the golden shiner and goldfish. Progressive Fish Culturist. 39, 121-122.

Sink, T.D., 2010. Influence of pH, salinity, calcium, and ammonia source on acute ammonia toxicity to golden shiners, *Notemigonus crysoleucas*. Journal of the World Aquaculture Society. 41, 411-420.

Stone, N., Thomforde, H., 2001. Common farm raised baitfish, USDA Southern Regional Aquaculture Center, SRAC Publication No. 120.

Stone, N., Goodwin, A., Lochmann, R., Phillips, H., Engle, C., Thomforde, H., 2005. Baitfish culture, in: Kelly, A.M., Silverstein, J. (Eds.), Aquaculture in the 21st Century, American Fisheries Society, Symposium 46, Bethesda, Maryland, pp. 339-353.

Stone, N., Park, E., Dorman, L., Thomforde, H., 1997. Baitfish culture in Arkansas: golden shiners, goldfish and fathead minnows. MP 386, Cooperative Extension Program, University of Arkansas at Pine Bluff.

Table 6. Environmental parameters to consider for tilapia (*Oreochromis niloticus*) culture.

Culture Stage	Overall Conditions					
	Temperature (°C)	DO (%)	Salinity (ppt)	pH	Ammonia (ppm)	Alkalinity
Spawning	25-30 (6)	not studied (3)	0 (7)			
Egg Development	25-30 (6)		0-30 (1)			
Juveniles	12-42 (3)	0.1-400 (4)	0-30 (1)	4-11 (3)	< 1.0 (4)	<30 (9)
Adults	12-42 (3)	0.1-400 (4)	0-30 (1)	4-11 (3)	< 1.0 (4)	<30 (9)
Culture Stage	**Optimum Conditions**					
	Temperature (°C)	**DO (%)**	**Salinity (ppt)**	**pH**	**Ammonia (ppm)**	**Alkalinity**
Spawning	25-30 (6)	not studied (3)	<10 (3),0 (7)			
Egg Development	25-30 (6)		<19 (5),0 (7)			
Juveniles	28	>3 (5)	<10 (3),0 (7)	6-9 (4,8)	< 0.08 (4)	100-250 (8)
Adults	28-30 (2)	>3 (5)	<15 (3)	6-9 (4,8)	< 0.08 (4)	100-250 (8)

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References:

[1]Schofield, P.J., Peterson, M. S., Lowe, M. R., Brown-Peterson, N. J., Slack, W.T., 2011. Survival, growth and reproduction of non-indigenous Nile tilapia, *Oreochromis niloticus* (Linnaeus 1758). Physiological capabilities in various temperatures and salinities. Marine and Freshwater Research. 62, 439-449

[2]Rakocy, J.E., McGinty, A.S., 1989. Pond Culture of Tilapia, USDA Southern Regional Aquaculture Center, SRAC Publication No. 280.

[3]El-Sayed, Abdel-Fattah M. 2006. Tilapia Culture. CABI Publishing, Cambridge, Massachusetts.

[4]Popma, T., Masser, M., 1999. Tilapia life history and biology, USDA Southern Regional Aquaculture Center, SRAC Publication No. 283.

[5]Mjoun, K., Rosentrater, K.A., Brown, M. L., 2010. Tilapia: Environmental biology and nutritional requirements. South Dakota State University Cooperative Extension, FS963-02.

[6]Chapman, F., 1992. Culture of hybrid tilapia: a reference file. University of Florida, IFAS Extension, Cir #1051.

[7]Fridmana, S., Brona, J., Ranab, K., 2012. Influence of salinity on embryogenesis, survival, growth and oxygen consumption in embryos and yolk-sac larvae of the Nile tilapia. Aquaculture. 334-337, 182-190.

[8]DeLong, D.P., Losordo, T.M., Rakocy, J., 2009. Tank culture of tilapia, USDA Southern Regional Aquaculture Center, SRAC Publication No. 282.

[9]Sheldon, W.L., Popma, T., 2006. Biology. In: Lim, C., Webster, C.D., (Eds.). Tilapia: biology, culture and nutrition. Haworth Press.

.

Table 7. Environmental parameters to consider for koi (*Cyprinus carpio*) culture.

Culture Stage	Overall Conditions					
	Temperature (°C)	DO (%)	Salinity (ppt)	pH	Ammonia (ppm)	Alkalinity
Spawning	13-27 (5)					
Egg Development	20-29 (1)					
Juveniles						
Adults	0-35 (1)	Low/anoxia tolerant (2)	<10 (4)	5-9 (1), 4.5-10.5 (2)	<0.05 (1)	50-400 (5)

Culture Stage	Optimum Conditions					
	Temperature (°C)	DO (%)	Salinity (ppt)	pH	Ammonia (ppm)	Alkalinity
Spawning	~20 (1), >17 (3)					
Egg Development			0 (2)			
Juveniles	27-34 (3)					
Adults	18-24 (1), 20-28 (3), 6-27 (5)	>5 mg/l (1) to saturation (5)	0-6 (2)	7 (1), 6.8-7.5 (3)	<0.05 (1)	

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References:

[1]Watson, C.A., Hill, J.E., Pouder, D.B., 2004. Species profile: koi and goldfish, USDA Southern Regional Aquaculture Center, SRAC Publication No. 7201.

[2]Nico, L.G., Schofield, P.J., Larson, J., Fusaro, A., 2013. *Carassius auratus*. USGS Nonindigenous Aquatic Species Database, Gainesville, FL.

[3]Burden, D., Lutz, C.G., 2013. Koi or Carp Profile. USDA, Agricultural Marketing Resource Center, Iowa State University.

[4]Schofield, P.J., Brown, M.E., Fuller, P., 2006. Salinity tolerance of goldfish *Carassius auratus*, a non-native fish in the United States. Florida Scientist. 69, 258-268.

[5]Piper, R.G., McElwain, I.B., Orme, L.E., McCraren, J.P., Fowler, L.G., Leonard, J.R., 1982. Fish Hatchery Management. US Fish and Wildlife Service, Washington D.C. Sixth Printing.

[6]Murai, T., Andrews, J.W., 1977. Effects of salinity on the eggs and fry of the golden shiner and goldfish. Progressive Fish Culturist. 39, 121-122.

Potential Finfish Production Hazards

Environmental Conditions

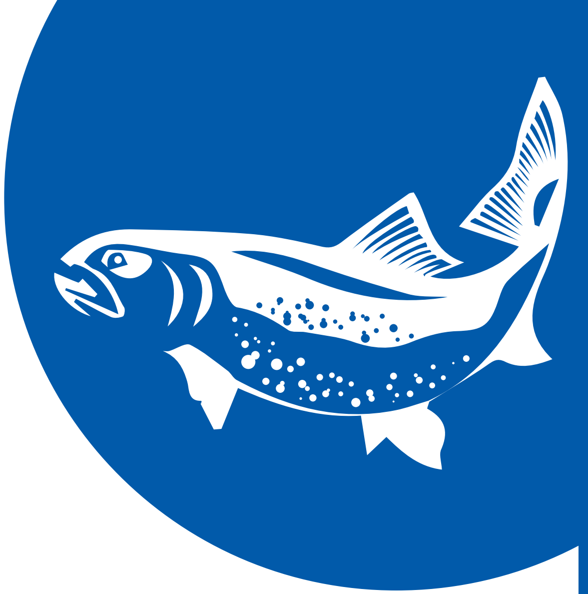
Biofouling Organisms

Predators

Diseases and Parasites

Invasive Species

Operational Procedures



Biofouling Organisms

Biofouling occurs when organisms settle or accumulate on equipment used in the culture of finfish. Organisms such as bacteria, algae, and animals may attain densities sufficient to restrict water flow to and from cultured fish. Restricted flow causes water quality to deteriorate as metabolic wastes accumulate and dissolved oxygen is reduced. Poor water quality, if sufficiently diminished, will cause mortalities directly. Sublethal, chronic exposure to poor water quality stresses fish, which makes them more vulnerable to diseases and leads to indirect losses. Some biofouling organisms can provide a refuge for parasites and pathogens, while others harbor or release substances toxic to fish. Fish cultured in ponds, raceways, net-pens, and recirculating aquaculture systems (RAS) are exposed to many biofouling challenges some common to all systems and others unique to each system.

The type of culture system employed frequently determines biofouling management options, but the following practices should be considered:

- Systems should be designed and constructed to facilitate biofouling management: inspection, access and maintenance.
- In-flow and discharge systems should be constructed in parallel so one system operates while the other is cleaned.
- Plumbing and netting should be of greater than required diameter to permit adequate water flow as biofouling accumulates.
- Water flow, netting material and delivery components should be routinely inspected and cleaned.
- Preventive procedures (e.g. applying anti-fouling coatings) should be employed carefully and as approved to avoid fish loss.
- Biofouling organisms should be properly removed and disposed to prevent further impact within the system or on the surrounding environment.
- Consult with federal, state and local officials concerning restrictions or required permits for preventative measures or fouling removal/disposal.
- Biofouling will occur, so monitoring, management and record-keeping are critical.
- Management plans should be adjusted seasonally and when necessary for continued success.

Potential freshwater biofouling hazards are included in this chapter. A list and description of marine biofouling organisms are provided in Chapter 3, see 'biofouling'.

Figure 1: Pipe with a moderate biofilm.
Joseph Buttner

BACTERIA

Name:

- heterotrophic bacteria

Mode of Action: indirect; reduce water flow, reduce water quality, and can harbor pathogens
Heterotrophic bacteria naturally colonize and accumulate on surfaces. These bacteria when exposed to continuous supply of nutrient-rich water can become excessively abundant, interfering with water flow through plumbing and netting, which can cause water quality to deteriorate. In addition, heavy biofilms may harbor fish pathogens, serving as a reservoir, which can become a problem if the fish within the system become stressed and are more susceptible to infection.

Hazard Management:

- To manage biofouling by heterotrophic bacteria it is recommended that organic loads and especially solids filtration be adequately addressed in system design. In all systems, avoid organic build up by removal of wastes in RAS and raceway systems or maintenance of water flow in cage culture.
- Treatment with UV or ozone can help to manage bacterial loads in RAS.
- Regular cleaning and maintenance of surface area will help to ensure adequate flow and system operation.
- In some cases coating nets or cages with anti-fouling substances may increase the amount of time before cleaning of fouling organisms is needed. Anti-fouling substances should be approved for use with fish culture operations.

Figure 2: Strands of filamentous algae can form and rapidly clog gear and pipes.
Nathan Stone

ALGAE

Name:

- one-celled microscopic to filamentous and macroscopic species; algae can be attached or free floating

Mode of Action: Algae are photosynthetic organisms; they require light and nutrients to survive. If light is eliminated and nutrients are removed, algae cannot survive or become abundant. While these species are beneficial in controlling nitrogenous fish wastes, some blue-green cyanobacteria and dinoflagellates may release substances toxic to fish. Cyanobacteria can release compounds that cause off-flavors in the fish. Microscopic and macroscopic algae attach to substrates such as netting and piping. When nutrient loads are high and water flow is continuous, algal densities may become quite high and reduce water flow. Reduced water flow causes water quality to deteriorate, stressing and potentially killing fish.

Systems affected: marine and freshwater; ponds, cages

Hazard Management:

- Prevention is preferable to correction. System design, location, and operation should be adjusted to preclude conditions that favor colonization by algae.
- Once algae have attached they can be removed mechanically by drying or scrubbing.
- UV or ozone treatments may prove doable in RAS.
- In ponds, barley straw, shading dyes, or herbicides may be used.
- When feasible, relocation of cages can avoid blooms of toxic phytoplankton.
- If off-flavors become an issue, a purging system may be needed before harvest.

Figure 3a: The quagga mussel, Dreissena bugensis *(bottom) and the zebra mussel,* Dreissena polymorpha *(top).*
Northeast Aquatic Nuisance Species Panel

Figure 3b. Zebra mussels can quickly clog pipes and foul aquaculture gear.
Scott Camazine

BIVALVE MOLLUSC

Name:

- Asiatic clam (*Corbicula fluminea*)
- freshwater mussels (*Dreissena* spp.)

Mode of Action: In freshwater systems, Corbicula, zebra and quagga mussels can accumulate in unfiltered piping systems, while mussels may attach to the netting of cages. In these systems, bivalves can become abundant, restricting water flow and indirectly impacting fish by reducing water quality.

Hazard Management:

- Prevention is preferable to correction, so system design and operation should be adjusted, if possible, to preclude conditions that favor entry and colonization by bivalve larvae. Inflow water can be filtered or disinfected.
- Regular cleaning, air-drying, and maintenance to filtration and disinfection equipment, pipes, tanks, and netting can help prevent colonization and ensure adequate flow for system operation.
- In some circumstances, chemical treatments such as salt or formalin may be appropriate.
- Copper-based anti-fouling coatings are routinely used on nets in salmon farming.
- A monitoring protocol should be identified if invasive *Corbicula*, zebra, or quagga mussels are not present but are found within the geographic range.

References and Further Reading:

Chen, S., Malone, R.F., 1991. Suspended solids control in recirculating aquaculture systems. Engineering aspects of intensive aquaculture, in: Proceedings from the Aquaculture Symposium, Cornell University, Ithaca, New York, pp. 170-186

Bartley, D.M., McIntire, H., Volkoff, M., 2008. Guidelines to reduce the risk of quagga/zebra mussels in California aquaculture. California Department of Fish and Game.

Brunson, M.W., Lutz, C.G., Durborow, R.M., 1994. Algae blooms in commercial fish production ponds. USDA Southern Regional Aquaculture Center, SRAC Publication No. 466.

Buttner, J.K., 1986. Biology of *Corbicula* in catfish rearing ponds. Proceedings Second International *Corbicula* Symposium. American Malacological Society Bulletin Special Edition No. 2:211-218.

Jewett, E.B., Lopez, C.B., Dortch, Q., Etheridge, S.M., 2007. National assessment of efforts to predict and respond to harmful algal blooms in U.S. waters. Interim Report. Interagency Working Group on Harmful Algal Blooms, Hypoxia, and Human Health of the Joint Subcommittee on Ocean Science and Technology, Washington, D.C.

Madin, J., Chong, Y.C, Hartstein, N.D., 2010. Effects of water flow velocity and fish culture on net biofouling in fish cages. Aquaculture Research. 41(10), 602-617.

Masser, M.P., Murphy, T.R., Shelton, J.L., 2001. Aquatic weed management: herbicides. USDA Southern Regional Aquaculture Center, SRAC Publication No. 361.

Page, F.H., Hanke, A., Martin, J.L., LeGresley, M., Chang, B., 2005. Characteristics of *Alexandrium fundyense* blooms that affect caged salmon in the Bay of Fundy. Aquaculture Association of Canada, Special Publication No. 9.

Rice, J., 1995. Zebra mussels and aquaculture: what you should know. North Carolina Sea Grant, Publication No. BP-95-01.

Rodgers, J.H., 2008. Algal toxins in pond aquaculture. USDA Southern Regional Aquaculture Center, SRAC Publication No. 4605.

Schrader, K.K., Dennis, M.E., 2005. Cyanobacteria and earthy/musty compounds found in commercial catfish (*Ictalurus punctatus*) ponds in the Mississippi Delta and Mississippi-Alabama Blackland Prairie. Water Research. 39(13), 2807-2814.

Schrader, K.K., Rimando, A.M., 2003. Off-flavors in aquaculture: an overview, in: Rimando, A.M., et al., (Eds.), Off-flavors in aquaculture. American Chemical Society: Washington D.C., pp. 1-12.

Stone, N., Daniels, M., 2006. Algal blooms, scums and mats in ponds. University of Arkansas Cooperative Extension Service, Publication No. FSA9094.

Swistock, B., Hartle, M., Curtis, A., 2006. Filamentous algae. Penn State University Cooperative Extension, Pond Facts No. 15.

Tan, C.K.F., Nowak, B.F., Hodson, S.L., 2002. Biofouling as reservoir of *Neoparamoeba pemaquidensis* (Page, 1970), the causative agent of amoebic gill disease in Atlantic salmon. Aquaculture. 210(1-4), 49-58.

Treasurer, J., Grant, A., Hannah, F., 1999. Monitoring programmes for phytoplankton bloom. Fish Farmer. 13(5), 30-33.

Tucker, C.S., Hanson, T.R., Kingsbury, S.K., 2001. Management of off-flavors in pond-cultured channel catfish with weekly applications of copper sulfate. North American Journal of Aquaculture. 63(2), 118-130.

Tucker, C.S., van der Ploeg, M., 1999. Managing off-flavor problems in pond-raised catfish. USDA Southern Regional Aquaculture Center, SRAC Publication No. 192.

Yanong, R.P.E., 2003. Fish health management considerations in recirculating aquaculture systems – Part 2: pathogens. IFAS Publication Circular No. 121.

Yanong, R.P.E., Pouder, D.B., Falkinham, J.O., 2010. Association of mycobacteria in recirculating aquaculture systems and mycobacterial disease in fish. Journal of Aquatic Animal Health. 22, 219–223.

Potential Finfish Production Hazards

Environmental Conditions

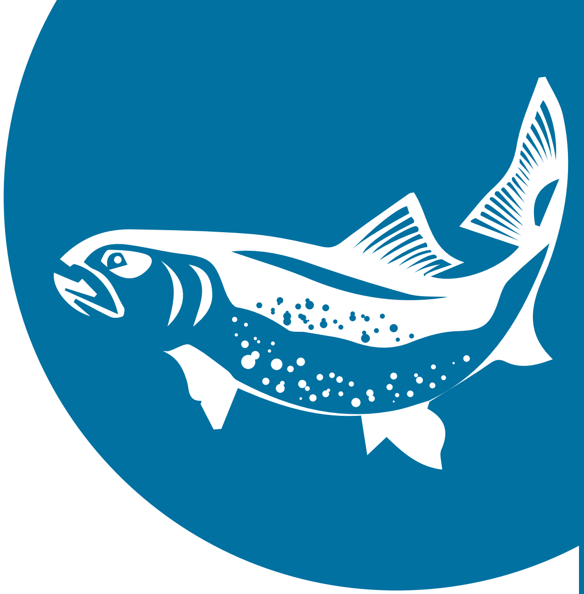
Biofouling Organisms

Predators

Diseases and Parasites

Invasive Species

Operational Procedures



Predators

Predators cause fish losses directly and indirectly, by increasing stress on stocks resulting in mortality. Also, both birds and mammals can introduce pathogens and invasive species into growing systems. Certain species are particularly problematic at aquaculture facilities. Avian predators (cormorant, heron, kingfisher, osprey) feed on stocks primarily from late spring to fall, whereas terrestrial mammals (fisher cat, mink, otter, raccoon) are more prominent in the winter months. Marine mammals (primarily seals) are a problem for ocean net pens.

The type of culture system determines some predator management options but the following practices should be considered.

- Feed containers should be secure and kept out of reach of predators.
- Weed and brush should be reduced around culture areas.
- Where appropriate, trees should be removed to reduce nesting and perching sites for birds.
- Consult with federal, state, and local officials concerning restrictions or required permits with regards to predator control or intervention.
- Problem predator species should be positively identified before corrective actions are taken.
- Physical barriers against predators are the most effective deterrent when practical (e.g. predator nets on salmon pens).
- Deterrent systems should be established before predators establish a feeding routine.
- Deterrent systems should include a variety of devices employed at different locations and different times.
- Success of deterrent methods should be monitored and losses to predation should be recorded.
- Seasonal plans should be adjusted when necessary for continued success.
- If deterrent methods are ineffective, kill permits may be an option for avian or terrestrial predators. For specific information, contact appropriate state and federal wildlife authorities (Appendix 3).

Potential hazards:

Avian predators
Terrestrial mammals
Marine mammals

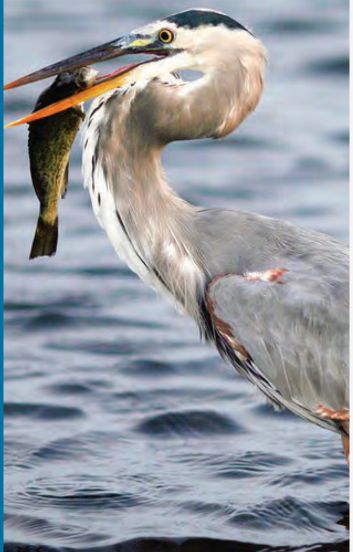


Figure 1a: The great blue heron, Ardea herodias.

Figure 1b: The belted king fisher, Megaceryle alcyon.

Figure 1c: The osprey, Pandion haliaetus.

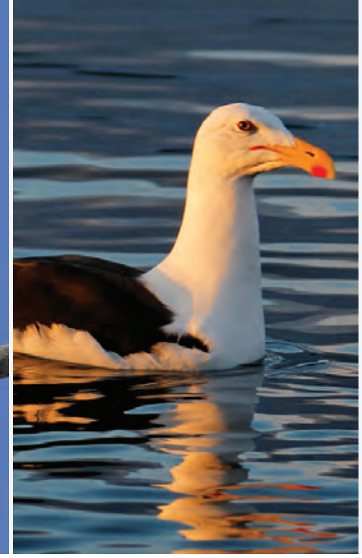


Figure 1d: The American herring gull, Larus smithsonianus.

All photos courtesy of Wikimedia Commons

AVIAN PREDATORS

Wading birds:

- great blue heron (Ardea herodias)

Diving birds:

- kingfisher (*Megaceryle alcyon*)
- osprey (*Pandion haliaetus*)
- gulls (*Larus marinus*, *L. smithsonianus*, *L. delawarensis*)

Hazard Management:

- exclusion, scare devices

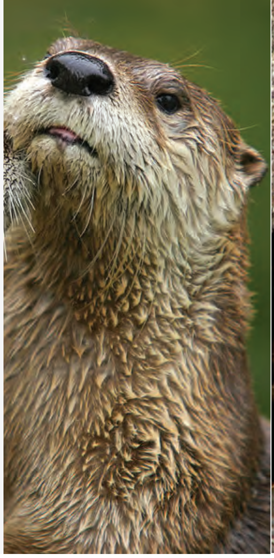


Figure 2a: The North American river otter, Lontra Canadensis.

Figure 2b: The mink, Neovison vison.

Figure 2c: The fisher cat, Martes pennant.

Figure 2d: The gray seal, Halichoerus grypus.

Figure 2e: The harbor seal, Phoca vitulina.

All photos courtesy of Wikimedia Commons

TERRESTRIAL AND MARINE MAMMALS

Species include:

- North American river otter (*Lontra canadensis*)
- mink (*Neovison vison*)
- fisher cat (*Martes pennant*)
- raccoon (*Procyon lotor*)
- gray seal (*Halichoerus grypus*)
- harbor seal (*Phoca vitulina*)

Hazard Management:

- Use trapping (permits may be necessary)
- Use exclusion device such as a barrier pen
- Use sonic/acoustic deterrents

References and Further Reading:

Alberta Agriculture Food and Rural Development, 1999. Predator damage control in cultured fish. Agdex 485/685-1.

Barras, S., 2007. Avian predators at aquaculture facilities in the southern United States. USDA Southern Regional Aquaculture Center, SRAC Publication No. 400.

Bevan, D.J., Chandroo, K.P., Moccia, R.D., 2002. Predator control in commercial aquaculture in Canada. University of Guelph, AEC ORDER No. 02-001.

Curtis, K.S., Pitt, W.C., Conover, M.R., 1996. Overview of techniques for reducing bird predation at aquaculture facilities. Jack Berryman Institute, Utah State University.

De La Croix, R., (no date). Preventing damage to fish by seals and sea lions. White paper. Hydraacoustics, Inc.

Falker, S.T., Brittingham, M.C., 1998. Controlling birds at aquaculture facilities. Pennsylvania State University, Wildlife Damage Control No. 12.

Fjalling, A., Wahlberg, M., Westerberg, H., 2006. Acoustic harassment devices reduce seal interaction in the Baltic salmon-trap, net fishery. ICES Journal of Marine Science. 63, 1751-1758.

Gorenzel, W.P., Conte, F.S., Salmon, T.P., 1994. Bird damage at aquaculture facilities, in: Hygnstrom, S.E., Timm, R.M., Larson, G.E. (Eds.), Prevention and Control of Wildlife Damage. University of Nebraska, Lincoln, pp. E5-E18.

Littauer, G., 1990. Avian predators: frightening techniques for reducing bird damage at aquaculture facilities. USDA Southern Regional Aquaculture Center, SRAC Publication No. 401.

Littauer, G.A., Glahn, J.F., Reinhold, D.S., Brunson, M.W., 1997. Control of bird predation at aquaculture facilities: strategies and cost estimates. USDA Southern Regional Aquaculture Center, SRAC Publication No. 402.

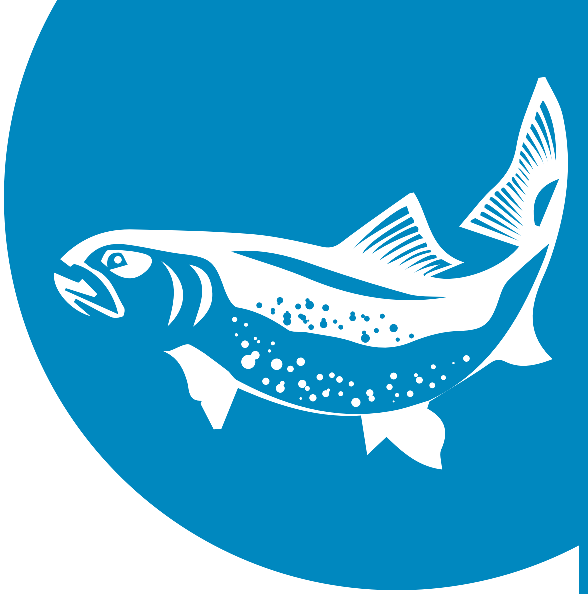
Nelson, M.C., Gilbert, J.R., Boyle, K.J., 2006. The influence of siting and deterrence methods on seal predation at Atlantic salmon (Salmo salar) farms in Maine, 2001–2003. Canadian Journal of Fisheries and Aquatic Sciences. 63(8), 1710-1721.

Quick, N.J., Middlemas, S.J., Armstrong, J.D., 2004. A survey of antipredator controls at marine salmon farms in Scotland. Aquaculture. 230, 169-180.

USDA Animal Plant Health Inspection Service (APHIS), 1997. Bird predation and its control at aquaculture facilities in the northeastern United States. USDA APHIS No. 11-55-009.

Potential Finfish Production Hazards

Environmental Conditions
Biofouling Organisms
Predators
Diseases and Parasites
Invasive Species
Operational Procedures



Diseases and Parasites

Disease can pose a serious risk to any aquaculture operation. Outbreaks of disease can lead to expensive treatments for the animals, loss of animals, regulatory impacts, or decreased marketability of the cultured animals. Fortunately, a range of lower-cost options exists to help reduce the risk of disease. Often an outbreak of disease results from a combination of poor water quality, poor biosecurity, and poor husbandry. The most important step that any farmer can take to reduce their risk of disease is to maintain optimal water quality, practice good biosecurity, and utilize good animal husbandry. In addition, it is important to develop a plan to monitor the health of the animals with the local aquatic animal health professional. They can also help in the development of appropriate biosecurity plans. It is also important to learn the normal behavior of the animals. Often good observation of when animals appear to be normal or they are behaving abnormally can be an early warning that there may be problems in the system.

Potential hazards:

parasites
bacteria
viruses
fungi

Included here is a list of diseases and parasites for most commonly cultured finfish in the region. This list is by no means all-inclusive, but represents either common diseases or diseases/parasites of particular regulatory concern. The challenges, hazards, and risks posed by more common diseases maybe different from those posed by diseases of regulatory concern; however, both can be significant. The diseases have not been classified into common or regulatory concern as it is important for the farmer to develop a relationship with a local fish health professional that can assist in assessing the risks the farmer may face and developing strategies to minimize those risks. This list is designed to help make the farmer aware of some of the important disease risks the farmer may face and common measure to minimize disease risks. Following the list is specific information on each disease and parasite including, if available, prevention and management strategies.

Consult with an aquatic animal health professional if disease is suspected, or treatment is necessary. Some treatments may not be legal for use on fish destined for human consumption, or may be subject to a withdrawal period before the fish can be marketed.

Diseases Listed by Species

These are the diseases that are most likely to occur on the farm. Farmers should recognize, however, that there are many opportunistic diseases may arise. It important to consult the local aquatic animal health professional in order to understand what diseases are important locally (Appendix 3).

Salmonids:

- external ciliated parasites (e.g. *Trichodina* spp.)
- ichthyobodiasis
- ichthyophthiriasis
- diplomonadiasis or hexamitosis
- whirling disease
- parasitic copepods (*Lepeophtheirus salmonis*, *Caligus elongatus*)
- furunculosis
- bacterial gill disease
- cold water disease (caused by *Flavobacterium psychrophylium*)
- columnaris
- bacterial kidney disease
- cold water disease (caused by *Vibrio* spp.)
- enteric red mouth disease
- infectious pancreatic necrosis virus
- infectious salmon anemia virus
- viral hemorrhagic septicemia virus
- saprolegniasis
- branchiomycosis
- *Ichthyophonus hoferi*

Atlantic cod:

- external ciliated parasites (e.g. *Trichodina* spp.)
- parasitic copepods (*Caligus elongates*, *Lernaeocera brachialis*)
- microsporidian parasite (*Loma branchialis*)
- furunculosis
- franciselliosis
- cold water disease (caused by *Vibrio* spp.)
- infectious pancreatic necrosis virus
- viral encephalopathy and retinopathy
- viral hemorrhagic septicemia virus
- *Ichthyophonus hoferi*

Largemouth bass, striped bass and hybrids:

- external ciliated parasites (e.g. *Trichodina* spp.)
- ichthyobodiasis
- ichthyophthiriasis
- velvet disease
- furunculosis
- edwardsiellosis
- columnaris
- streptococcal disease
- cold water disease (caused by *Vibrio* spp.)
- viral hemorrhagic septicemia virus
- large mouth bass virus (largemouth and smallmouth bass only)
- epizootic ulcerative syndrome
- saprolegniasis
- branchiomycosis

Perch:

- external ciliated parasites (e.g. *Trichodina* spp.)
- ichthyobodiasis
- ichthyophthiriasis
- proliferative gill disease
- columnaris
- streptococcal disease
- viral hemorrhagic septicemia virus
- saprolegniasis

Minnows (including common shiner, fathead) and white sucker:

- external ciliated parasites (e.g. *Trichodina* spp.)
- ichthyobodiasis
- ichthyophthiriasis
- parasitic copepods (*Lernaea* spp., especially *Lernaea cyprinacea*)
- furunculosis
- columnaris
- enteric red mouth disease
- viral hemorrhagic septicemia virus
- saprolegniasis
- branchiomycosis

Tilapia:

- external ciliated parasites (e.g. *Trichodina* spp.)
- velvet disease
- furunculosis
- franciselliosis
- edwardsiellosis
- columnaris
- mycobacteriosis
- streptococcal disease
- viral encephalopathy and retinopathy
- branchiomycosis
- saprolegniasis

Koi

- external ciliated parasites (e.g. *Trichodina* spp.)
- ichthyobodiasis
- ichthyophthiriasis
- parasitic copepods (*Lernaea* spp., especially *Lernaea cyprinacea*; *Ergasius* spp.)
- furunculosis
- columnaris
- mycobacteriosis
- carp pox
- koi herpes virus
- spring viremia of carp
- branchiomycosis
- saprolegniasis

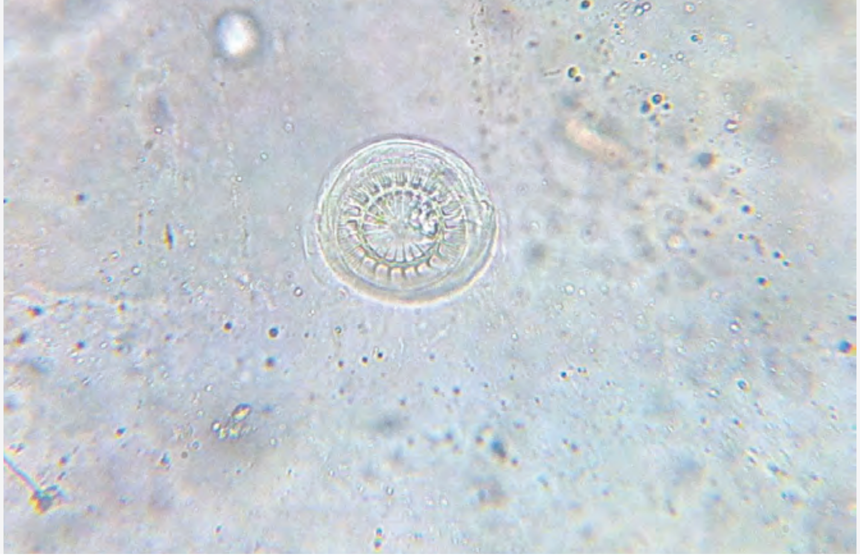


Figure 1: An image of the parasitic copepod, Trichodina *sp.*
Fish Vet Group

PARASITES

Name: external ciliated parasites (e.g. *Trichodina* spp.)

Mode of Action: This is an external parasite commonly found on the gills, scales and skin of fish, appearing as a grayish film or as white, pinhead-size bumps. Parasite can only be seen microscopically. They do not feed on the fish directly, but rather on bacteria. They attach to the fish with a row of hooks that causes irritation and damage to gill and skin.

Major Aquaculture Species Affected in this Region:

Most fish species and all listed in this manual:

- salmonids
- Atlantic cod
- largemouth bass, striped bass and hybrids
- perch
- minnows
- tilapia
- koi

Hazard Management:

- Check water quality, as outbreaks of tricodinids can be a sign of eutrophic water.
- There are effective ways to treat this parasite so consult with an aquatic animal health professional for treatment options and application procedures.

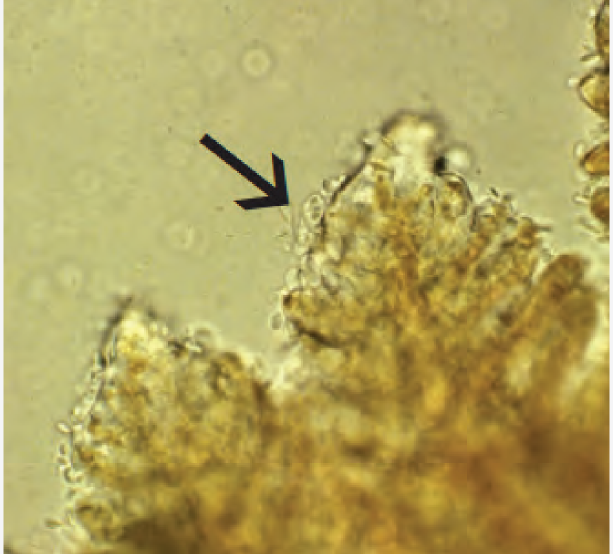


Figure 2a: Ichthyobodiasis is caused by a flagellated protozoan (genus Ichthyobodo*) (arrow) which stick to the gill lamellae. :*
Meyers, et al. 2008, Alaska Department of Fish and Game:

Figure 2b: Close up of gill lamellae
Meyers, el al. 2008, Alaska Department of Fish and Game:

PARASITES

Name: Ichthyobodiasis; caused by: Ichthyobodo (previously classified as Costia) spp., especially *I. pyriformis* and *I. necator*

Mode of Action: This is an obligate external flagellate parasite of fish. Infected fish may flash, scrape against the side of tank or net, or stop feeding. External symptoms vary but can include: excess mucus production, also known as blue slime, removal of the epithelium, and missing pigmentation.

Major Aquaculture Species Affected in this Region:

Most freshwater fish species including:

- salmonids
- largemouth bass, striped bass and hybrids
- perch
- minnows
- koi

Hazard Management:

- Good biosecurity and husbandry are important to help prevent infection.
- *Ichthyobodo* can be treated with potassium permanganate, formalin, copper sulfate, and salt.
- Consult with an aquatic animal health professional for additional treatment options and application procedures.

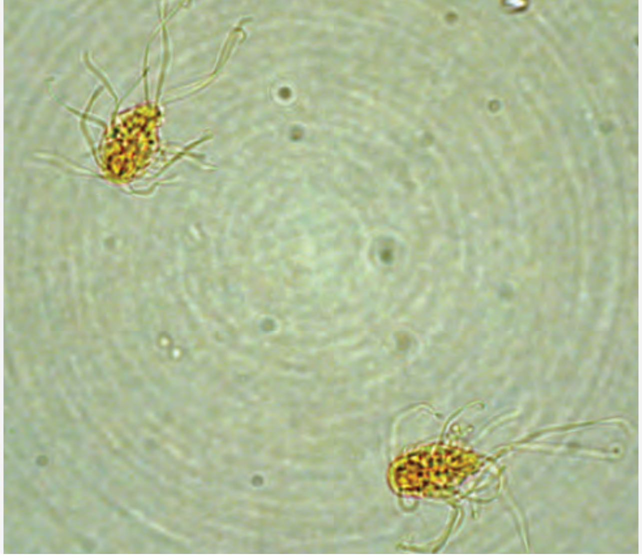


Figure 3: Diplomonadiasis or hexamitosos is caused by a flagellated protozoan.
Meyers et al. 2008, Alaska Department of Fish and Game

PARASITES

Name: diplomonadiasis or hexamitosis; caused by diplomonad flagellates

Mode of Action: Diplomonad flagellates, across at least three genera, cause disease in fish. Some of these flagellates are well described while others are not. The signs and symptoms vary based on the species of flagellates and the species of fish infected. Infections can result in sluggish behavior, decreased appetite, anorexia, and agitation. External symptoms vary but may include: anorexia, little to no growth, abdominal distension, darker pigmentation, red vent, and pale shiny feces. A fecal exam or necropsy will be needed to diagnose this infection.

Major Aquaculture Species Affected in this Region:

- salmonids

Hazard Management:

- Good biosecurity and husbandry are important to help prevent infection.
- There are few options for treatment as the most effective drugs for treating this parasite are not labeled (i.e. not approved) in the U.S. and Europe for use in aquaculture.
- Consult with an aquatic animal health professional for additional treatment options and application procedures.

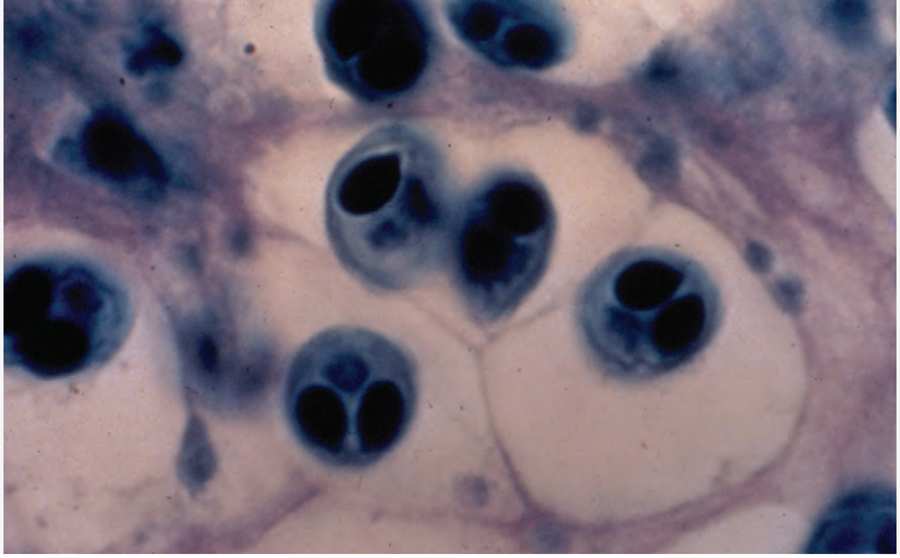


*Figure 4: Whirling disease (*Myxobolus cerebralis*) pathology.*
Wikimedia Commons

PARASITES

Name: whirling disease; caused by *Myxobolus cerebralis*

Mode of Action: Fish infected with this parasite swim in circular patterns or chase their tail. In 3 to 6 month old trout, the tail can be darker in color (called black tail) and often develop a hump, twist, or bend. The parasite can also cause other skeletal deformaties such as a shortened jaw or operculum and gaping mouth. Infection is more common in younger fish than older ones. Adult brown trout can be carriers of the parasite. This parasite requires an oligochaete (an aquatic segmented worm) host, *Tubifex tubifex* to complete its life history.

Major Aquaculture Species Affected in this Region:

- salmonids

Hazard Management:

- Good biosecurity and husbandry are important to help prevent infection.
- While the pathogen is present in most of the region, notably it is not in Maine or Vermont.
- Control of the disease is difficult as the spores can survive in sediments for several years.
- Maintaining clean rearing systems free of the oligochaete hosts.
- Use of well water or disinfected surface water and adhering to movement regulations can control the spread of the pathogen.
- It is best to try and avoid infection with this parasite, as there are no current treatments available.

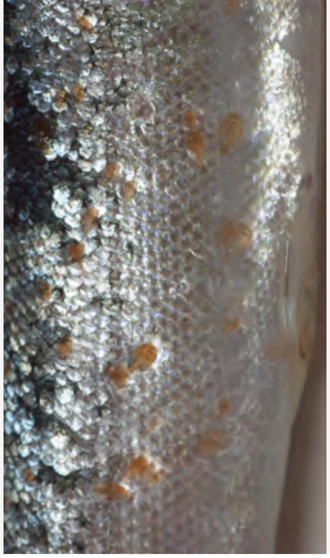


Figure 5a: Numerous chalimus stage lice attached to an Atlantic salmon
Michael Pietrak

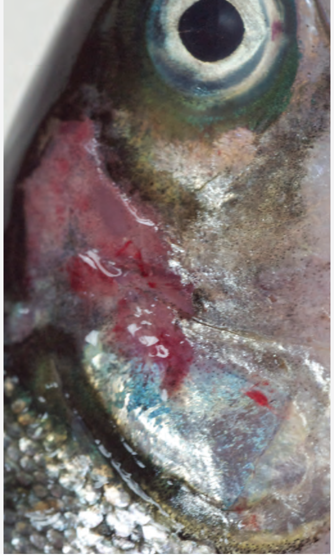


Figure 5b: Severe damage on the head and operculum of a fish due to feeding of lice. Wounds of this nature are prone to secondary infection and cause osmotic stress.
Michael Pietrak

Figure 5c: Adult female and gravid females behind the anal fin. The fish is exhibiting mild damage from the feeding of the lice.
Michael Pietrak

Figure 5d: Sea lice, Caligus elongatus.
Michael Pietrak

PARASITES

Name: parasitic copepods, sea lice, salmon lice (*Lepeophtheirus salmonis, Caligus elongatus*)

Mode of Action: Sessile stages (Chalimus 1-4) and mobile stages (preadult 1-2 and adults) of *L. salmonis* feed on the mucus and skin (epidermis) of the fish. In rare cases and in young of the year, they can cause ulcers deep in to the underlying tissues, even to the bone. Eventually fish die from osmotic imbalance but often succumb to secondary infection first. Sessile stages (Chalimus 1-4) and mobile stages (preadult 1-2 and adults) of *C. elongatus* feed on the mucus and epidermis of the fish. In severe cases they can eat down to the bone. Eventually fish die from osmotic imbalance but often succumb to secondary infection first. The primary source of damage with *C. elongatus* is cosmetic and presence of the parasite is not conducive to live marketed fish. The copepod *C. elongatus* does not typically cause the same severe damage characteristic of *L. salmonis*. Sea lice control is a major area of international research.

Major Aquaculture Species Affected in this Region:

- The copepod *L. salmonis* is primarily found in marine salmonids
- The copepod *C. elongatus* can commonly be found on range of marine fish including salmonids and gadoids (e.g. Atlantic cod).

Hazard Management:

- Lice can sometimes be managed through husbandry practices, such as maintaining lower densities, all-in all-out and single-year class stocking, separation between farms, and fallowing between year classes.
- The copepod *C. elongatus* is known to be very mobile, moving not only from fish to fish on the farm, but frequently moving between farmed and wild fish making it very difficult to treat with drugs, especially bath-based drugs.
- There are chemotheraputants available in the U.S. under Investigational New Animal Drugs (INAD) and veterinarian supervision.
- Consult with an aquatic animal health professional for additional treatment options and application procedures.

Figure 6: Gravid female cod worms, Lernaeocera branchialis*, on the gills of a host.*
Hans Hillewaert

PARASITE

Name: parasitic copepod, cod worm (*Lernaeocera branchialis*)

Mode of Action: The mated female worm attaches to the gills of Atlantic cod and metamorphoses into the gravid female, the typical kidney bean stage. This stage extends mouthparts into the blood vessels and heart to feed on the blood of the fish. Single parasites can reduce growth and multiple infects can have a severe affect on growth and result in mortality

Major Aquaculture Species Affected in this Region:

- Atlantic cod and other gadids (infected by gravid females)

Hazard Management:

- Good biosecurity and husbandry should be practiced.
- Infected animals should be culled.
- Cod and flat fish should never be cultured on the same site in order to avoid completing the life cycle on the farm.

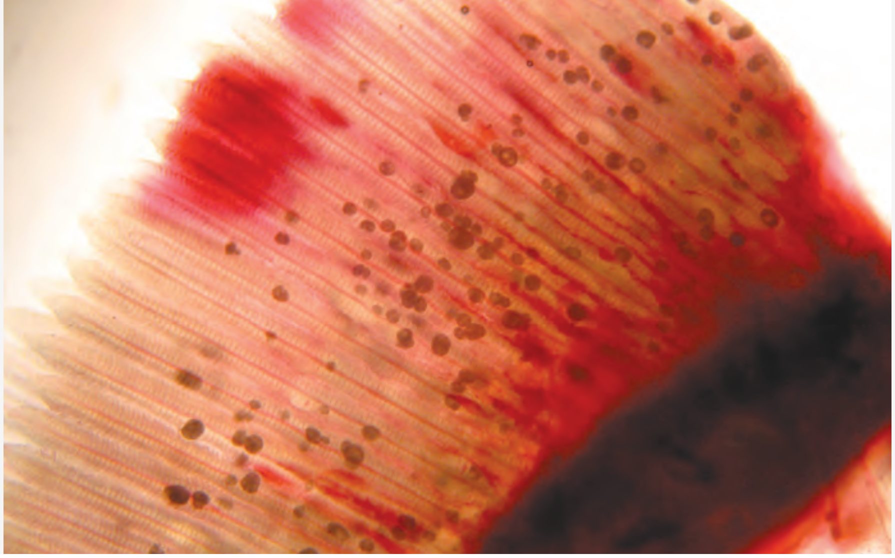


Figure 7: The microsporidan parasite, Loma branchialis.
C. Banner, Oregon Department of Fish and Wildlife

PARASITE

Name: microsporidian parasite (*Loma branchialis*)

Mode of Action: This parasite forms a white, cyst-like structure called xenomas primarily on the gills of fish. Heavy infections have been noted in cultured cod and they can lead to reduced growth, poor condition factor, and death.

Major Aquaculture Species Affected in this Region:

- Atlantic cod

Hazard Management:

- Currently no management strategies exist for the control of *Loma branchialis*.
- Good biosecurity and husbandry should be practiced, and UV or ozone sterilization of incoming water may help prevent infections in land-based systems.
- It has been suggested that rearing fish at temperatures below 6°C may be an effective control.

Figure 8: Whole parasite removed from a host. Approximately the upper 25 % of the animal pictured would be anchored within the tissue of the host.
U.S. Geological Survey

PARASITE

Name: parasitic copepods, anchor worms (*Lernaea* spp., especially *Lernaea cyprinacea*)

Mode of Action: Optimal temperature for infection is between 23 and 40 °C. The parasite goes dormant <15 °C. The lifecycle involves two hosts often these hosts are cyprinids but not always. Nauplii hatch from egg strings attached to the gravid female and are free living in the water while under going 3 molts before attaching to the first host. They can attach to the host along the body surface or gills. After molting they leave the first host and go through 5 copepodid stages. The males and females mate during the 4th stage and the adult female attaches to the second host during the 5th stage. This final stage, gravid female, resembles an anchor tag with the cephalic arms buried in the tissue of the fish while the body and egg sacks are outside of the fish. The gravid female can be up to 15 mm in length but average about 9 mm. They do not survive in waters with salinities >1.5. Infected fish can see secondary infections at the attachment point of the parasite.

Major Aquaculture Species Affected in this Region:

All freshwater fish, but especially:

- minnows
- koi

Hazard Management:

- Good biosecurity and husbandry should be practiced and good water quality maintained to prevent infection.
- Use of a 0.0001% bleach solution in the water to kill free-living stages has been suggested as a control method.

Figure 9: Effects of gill lice, Ergasilus *spp.*
C. Banner, Oregon Department of Fish and Wildlife

PARASITE

Name: parasitic copepods, gill maggots, gill lice (Ergasilus spp.)

Mode of Action: *Ergasilus* are parasitic copepods found in both fresh and marine waters. They are a number of different species in the genus, all of which are parasitic. They are capable of extensive periods of time as free-swimming animals. The females attach primarily to the gills of fish to produce their egg strings.

Major Aquaculture Species Affected in this Region:

Most fresh water and marine fishes, but especially:

- koi

Hazard Management:

- Good biosecurity and husbandry should be practiced and good water quality maintained to prevent infection.

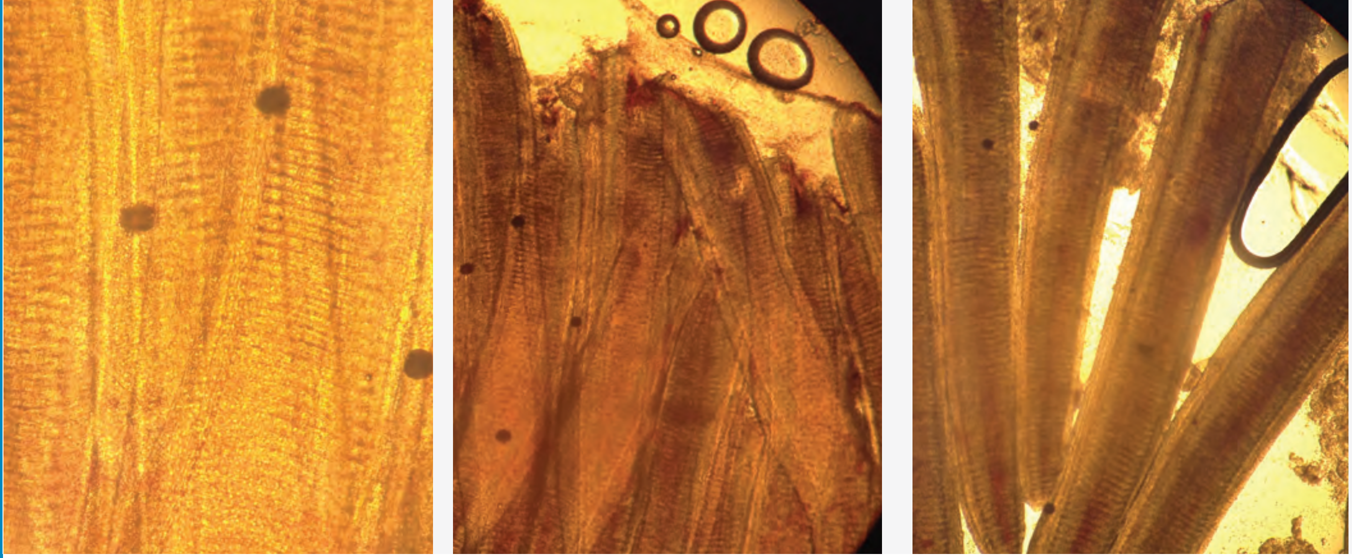


Figure 10: Velvet disease is caused by Amylodinium *sp.*
Fish Vet Group

PARASITE

Name: velvet, rust, or gold dust disease; caused by *Amylodinium* spp.

Mode of Action: This is a dinoflagellate that infects the gills and skin of fish. Symptoms that may be exhibited by fish include loss of appetite, flashing, gold to brown hue on skin, loss of scales, and patchy mucus. It is possible for mortalities to occur with no obvious signs of infection.

Major Aquaculture Species Affected in this Region:

- hybrid striped bass, cultured in brackish or salt water

Hazard Management:

- Good biosecurity and husbandry are important to help prevent infection.
- *Amylodinium* may be managed with treatments such as copper and freshwater dips under an aquatic animal health professional's supervision, but there are no drugs approved for use in food fish.

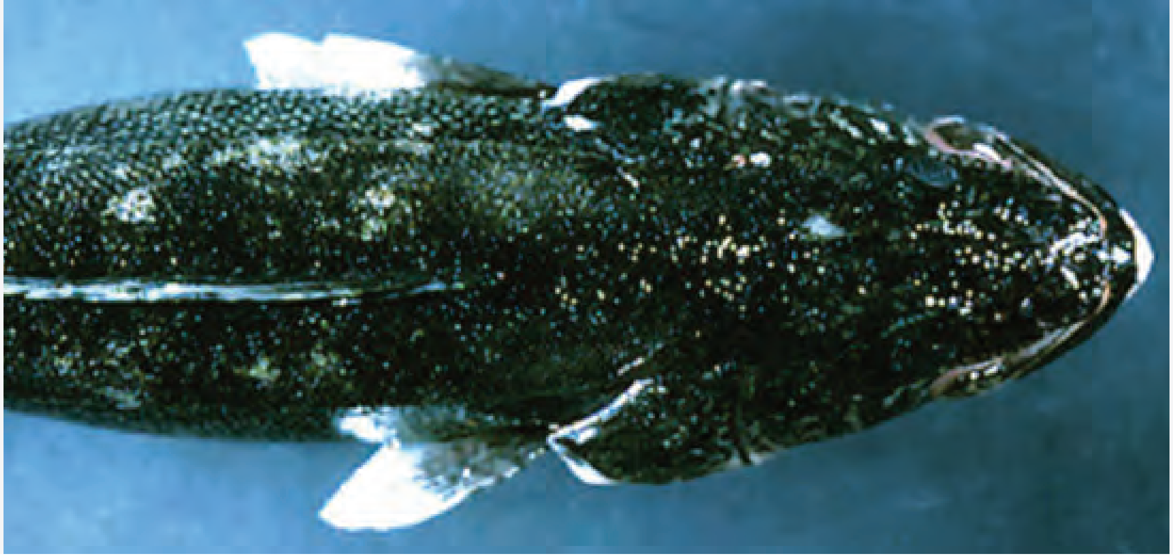


Figure 11: Ich observed on walleye.
P.R. Bowser, Cornell University

PARASITE

Name: ichthyophthiriasis, freshwater white spot, freshwater ich; caused by *Ichthyophthirius multifiliis*

Mode of Action: This protozoan is a fast spreading parasite of most freshwater fish. It infects the skin and gills of the fish and feeds on surface tissue within a nodule formed by the skin. When mature, before dropping off the fish to settle in the sediment, it divides and produce new infective forms that swim up to infect another fish. Infected fish may also display flashing, rubbing on objects on the side of the tank, anorexia, resting on the bottom, and hiding.

Major Aquaculture Species Affected in this Region:

Most freshwater fish species including:

- salmonids
- largemouth bass, striped bass and hybrids
- perch
- minnows
- koi

Hazard Management:

- Good biosecurity and husbandry are important to help prevent infection. In particular, keeping equipment clean and using separate gear for various areas of the facility where possible as the parasite can spread easily on infected gear.
- Disinfection on equipment is possible with heat as parasite does not survive >32 °C.
- Salt and various chemicals including: formalin, copper sulfate and potassium permanganate can be used to treat fish and water systems.
- Most treatments attack only the free-swimming stage and therefore multiple treatments are required to kill the parasite.
- Consult with an aquatic animal health professional for treatment options and application procedures.

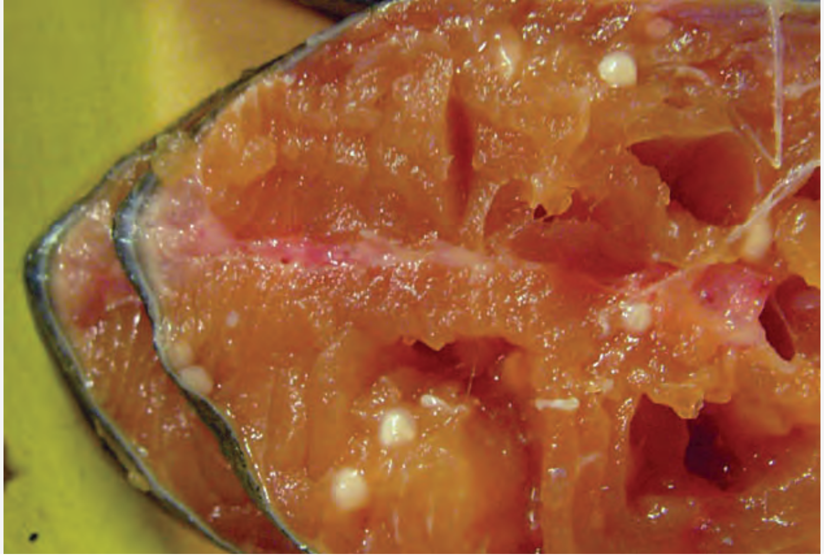


Figure 12: Henneguya *sp. results in white nodules in salmon tissue.*
Michal Mañas

PARASITE

Common Name: proliferative gill disease (PGD), hamburger gill

Scientific Name of Causative Agent: *Henneguya* spp.

Mode of Action: There are several species of *Henneguya* that infect various types of fish. They are myxosporidian parasites that require an oligochaete worm as an intermediate host. In some infections, the parasite is in the flesh of the fish and presents as white nodules. In other species, particularly catfish, the parasite causes significant damage to the gill filaments leading to severe mortality.

Major Aquaculture Species Affected in this Region:

- perch

Hazard Management:

- Good biosecurity and husbandry are important to help prevent infection.
- Fish can be removed from the infected water and will often recover.

Figure 13: Brown trout with furunculosis.
Ian Bricknell

BACTERIA

Name: furunculosis; caused by *Aeromonas salmonicida*

Mode of Action: Aeromonads are very common opportunistic bacteria in fresh and brackish water. Furunculosis is a serious bacterial infection in a range of freshwater and brackish species caused by *Aeromonas salmonicida*. The bacterium *A. salmonicida* is very different from the other Aeromonads. *Aeromonas salmonicida*, the causative organism for furunculosis, is considered an obligate pathogen. This means that it typically is found in its host and can exist in the environment for only a limited period of time. Fish can have acute, chronic, and latent infections. For furunculosis, the furuncle is the typical lesion in the chronic form of the disease. The term "furuncle" is a misnomer because it refers to an infected hair follicle in a mammal. But when the disease was first described, the lesion in the fish appeared to be similar to the mammalian furuncle, thus the name. In the acute form of the disease the typical lesion involves hemorrhage.

Other Aeromonads (e.g. *A. hydrophila*, *A. sobria*, *A. caviae*) are considered environmental opportunistic pathogens and are not associated with furunculosis. They are ubiquitous in the environment and cause disease when fish are stressed. External symptoms vary but may include: weak or lethargic swimming, darkened pigmentation, exophthalmia, bloody spots, distended abdomen, observable furuncles, and petechial (small red to purple discolorations) hemorrhages at the base of fins.

Major Aquaculture Species Affected in this Region:

Most freshwater and brackish water fish, but especially:

- salmonids
- Atlantic cod
- largemouth bass, striped bass, and hybrids
- minnow
- tilapia
- koi

Hazard Management:

- Good biosecurity and husbandry should be practiced. Removal of the environmental stressor responsible for the outbreak.
- There are commercial vaccines available for furunculosis, but not other aeromonads.
- Furunculosis and other aeromonad infections can be treated by veterinarians with antibiotics.
- Consult with an aquatic animal health professional for additional treatment options and application procedures.

Figure 14: Edwardsiella tarda *results in damage to eye of largemouth bass.*
Andy Goodwin

BACTERIA

Name: Edwardsiellosis; caused by *Edwardsiella tarda*

Mode of Action: Edwardsiellosis is an opportunistic bacterial infection in a range of species, predominately in freshwater. *Edwardsiella* spp. are part of the normal gut flora of fish, but can cause disease in fish and occasionally in humans. External signs vary, but can include: bursts of activity, increased food consumption, exophthalmia, cataracts, pale inflamed gills, enlarged organs, hemorrhagic red spots and ulcers on the skin and fins, and erosion of the skin.

Major Aquaculture Species Affected in this Region:

- striped bass
- tilapia

Hazard Management:

- Good biosecurity and husbandry should be practiced and good water quality maintained to prevent infection by this bacterium.
- There are commercial vaccines available for *Edwardsiella*.
- Consult with an aquatic animal health professional for additional treatment options and application procedures.

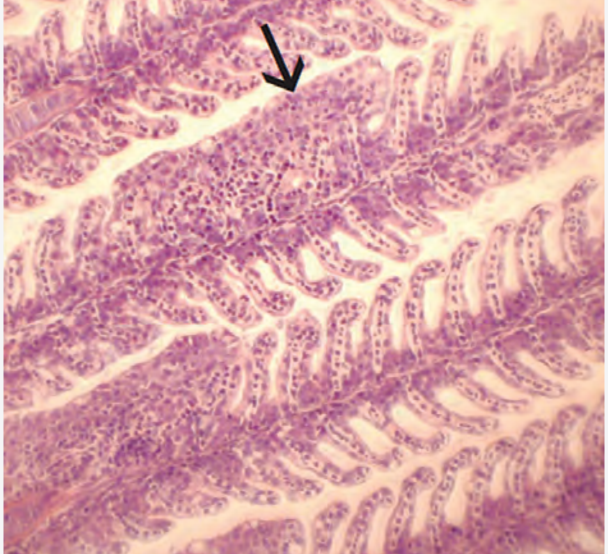


Figure 15a: Histological section of gill lamellar fusion (arrow) caused by Flavobacterium branchiphilum*.*
Meyers, et al. 2008, Alaska Department of Fish and Game

Figure 15b: Higher magnification showing filamentous bacteria (arrow) on gill.
Meyers, et al. 2008, Alaska Department of Fish and Game

BACTERIA

Name: bacterial gill disease; caused by *Flavobacterium branchiphilum*

Mode of Action: Fish <5g are particularly susceptible to this bacterial disease. Infections tend to occur in situations of stress, low dissolved oxygen and high ammonia concentrations, and high amounts of suspended particulates. Infected fish tend to be lethargic, consume less feed, display increased gill activity, flared operculum, and fused gill filaments.

Major Aquaculture Species Affected in this Region:

- salmonids

Hazard Management:

- Good biosecurity and husbandry should be practiced and good water quality maintained to prevent infection by this bacterium.
- Good attention to tank cleaning to remove particulates and avoidance of overfeeding will contribute to avoidance of bacterial gill disease.

Figure 16: This fish was infected with cold water disease caused by Flavobacterium psychrophilum.
Wikimedia Commons

BACTERIA

Name: cold water disease, bacterial coldwater disease, fry mortality syndrome, peduncle disease, or low temperature disease; caused by *Flavobacterium psychrophilum*

Mode of Action: This disease may be spread via vertical transmission (adult to egg) as well as horizontal (fish to fish) means. Infections are typically seen in fish < 1 year old and at water temperatures below 12 °C. Other factors influencing infection are increased handling, malnutrition, increased nitrite levels, and the presence of dead fish. Infected fish initially can display eroding caudal fins and skin ulcerations typically in the peduncle region. In early lesions, fish can have darker pigmentation in the tail and peduncle region. Fish that survive infection can be lethargic and may develop spinal deformities.

Major Aquaculture Species Affected in this Region:

- salmonids

Hazard Management:

- Good biosecurity and husbandry should be practiced and good water quality maintained to prevent infection.
- Deformed and moribund fish should be culled regularly from the population.
- Infections can be treated with several drugs approved for use in aquaculture under the supervision of an aquatic animal health professional.

Figure 17a: Columnaris observed in juvenile yellow perch.
P.R. Bowser, Cornell University

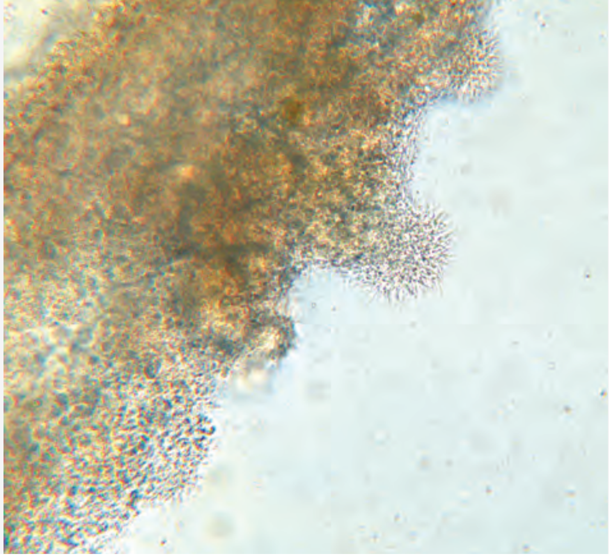


Figure 17b: Skin scrape showing columnaris at 40x.
P.R. Bowser, Cornell University

BACTERIA

Name: columnaris disease; caused by *Flavobacterium columnare*

Mode of Action: These bacteria infect all sizes of freshwater fish causing shallow to deep ulcerations of the skin. Whitish plaques are first noted on the skin. Infections can progress rapidly and often result in significant mortalities. Infections do particularly well in temperatures >14 °C, low dissolved oxygen, and elevated ammonia. When present, a range of external foci and shallow ulcers can be seen.

Major Aquaculture Species Affected in this Region:

- salmonids
- largemouth bass, striped bass and hybrids
- perch
- minnows
- tilapia
- koi

Hazard Management:

- Good biosecurity and husbandry should be practiced and good water quality maintained to prevent infection.
- Infections can be treated with several drugs approved for use in aquaculture under the supervision of an aquatic animal health professional.

Figure 18a Bacterial kidney disease, caused by Renibacterium salmoniarum*, caused petechial hemorrhages such as these seen in a salmonid.*
Meyers, et al. 2008, Alaska Department of Fish and Game

Figure 18b: Exophthalmia or pop-eye is commonly see in fish with BKD.
Meyers, et al. 2008, Alaska Department of Fish and Game

BACTERIA

Name: bacterial kidney disease (BKD); caused by *Renibacterium salmoninarum*

Mode of Action: BKD is a bacterial disease that can be transmitted vertically (adult to egg). Disease outbreaks can result in significant losses that tend to occur over an extended period of time. External signs vary and may not be seen at all, but can include: darkened pigmentation, exophthalmia, pale anaemic gills, and hemorrhaging at the base of the fins.

Major Aquaculture Species Affected in this Region:

- salmonids

Hazard Management:

- Good biosecurity and husbandry should be practiced and good water quality maintained.
- Broodstock, especially the maternal lines, should be screened for BKD at time of spawning.
- Eggs lots should be kept separated until screening results come back as negative.
- Any fish with positive results should be culled from the population.

Figure 19 Atlantic salmon smolt experimentally infected with Vibrio anguillarum.
Sarah Barker

BACTERIA

Name: cold water disease, vibriosis; caused by *Vibrio* spp., especially *V. salmonicida* and *V. anguillarum*

Mode of Action: *Vibrio* spp. are opportunistic bacteria that cause infection in stressed or immunocompromised animals. *Vibrio* spp. are common in marine and brackish water globally. External signs vary and may include: darkened pigmentation, pale gills, hemorrhaging at the base of the fins extensive petechial (small red to purple discolorations) hemorrhaging on the skin, and open lesions in severe cases.

Major Aquaculture Species Affected in this Region:

- salmonids
- Atlantic cod
- striped bass
- tilapia

Hazard Management:

- Good biosecurity and husbandry should be practiced.
- There are commercial vaccines available for vibriosis.
- Infections can be treated with several drugs approved for use in aquaculture under the supervision of an aquatic animal health professional.

Figure 20. Enteric red mouth disease, caused by Yersinia ruckeri, *results in petechial hemorrhages of the liver.*
Meyers, et al. 2008, Alaska Department of Fish and Game

BACTERIA

Name: enteric red mouth disease; caused by *Yersinia ruckeri*

Mode of Action: This is a bacterial infection in marine and freshwater fish that can result in significant mortality. External signs vary and may include: a characteristic reddening of the mouth and throat caused by subcutaneous hemorrhaging. Hemorrhaging may also be seen in gills, fins, and internally. If left untreated it can cause erosion of the jaw and palate.

Major Aquaculture Species Affected in this Region:

- salmonids
- minnows

Hazard Management:

- Good biosecurity and husbandry should be practiced.
- There are commercial vaccines available for enteric red mouth disease.
- Infections can be treated with several drugs approved for use in aquaculture under the supervision of an aquatic animal health professional.

Figure 21a. Close up of Francisella.
Michael Pietrak

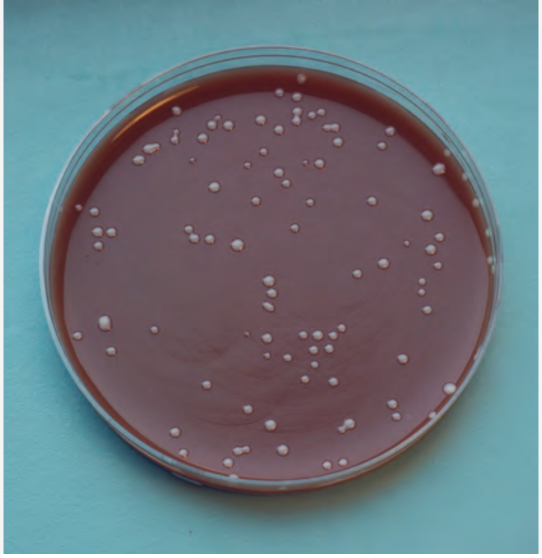


Figure 21b. Francisella noatunensis *colonies growing on culture media.*
Michael Pietrak

BACTERIA

Name: franciselliosis; caused by *Francisella noatunensis*

Mode of Action: A recently discovered disease of concern primarily in cod (*F. noatunensis*) and tilapia (*F. noatunensis* subsp. *orientalis*) culture, this bacterium is an intracellular pathogen. In cod it has been reported to cause approximately 40% - 75% mortality. External signs of disease can include: emaciation and raised hemorrhagic nodules on the skin.

Major Aquaculture Species Affected in this Region:

- Atlantic cod
- tilapia

Hazard Management:

- As a relatively newly reported disease there are few control options for *Francisella*.
- Good biosecurity and husbandry are important to help prevent infection.
- It has been reported to be susceptible to some antibiotics, but is resistant to others.
- Consult an aquatic animal health professional about treatment options.

Figure 22. Streptococcus *has cause exophthalmia in this tilapia.*
John Plumb, Retired, Auburn University

BACTERIA

Name: streptococcal disease; caused by *Streptococcus iniae*

Mode of Action: This bacterial infection is known to spread between fish through cannibalism and coprophagy. Infected fish may display erratic swimming, darkening pigmentation, lethargy, raised hemorrhagic patched of skin, operculum or fins, and exophthalmia. *Streptococcus iniae* is of particular concern as infections can spread to humans through open wounds or being pricked by fin rays.

Major Aquaculture Species Affected in this Region:

- hybrid striped bass
- perch
- tilapia

Hazard Management:

- Good biosecurity and husbandry are important to help prevent infection.
- Streptococcus iniae can be treated with a variety of antibiotics under the supervision of a veterinarian.

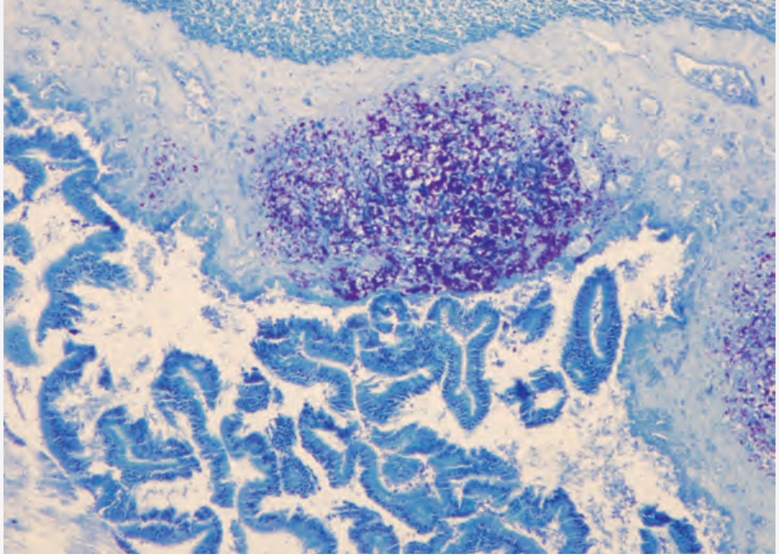


Figure 23. Granulomas, caused by Mycobacterium *spp., in gut tissue of a fish.*
Tim Bowden

BACTERIA

Name: mycobacteriosis; caused by *Mycobacterium spp.*

Mode of Action: This bacterium causes chronic disease in fish that leads to dehabilitation. Small to large nodules are found in the abdominal organs and occasionally hemorrhaging lesions are noted on the skin. Mycobacteriosis often occurs in poor water quality especially under conditions of low dissolved oxygen and low pH. Mycobacterium can be transmitted from mother to offspring (vertical transmission) or fish to fish (horizontal transmission). Mycobacteriosis is considered to be most common in closed recirculation aquaculture systems such as aquaria and other large closed tank systems.

Major Aquaculture Species Affected in this Region:

- All freshwater and marine fish
- Species of particular concern include striped bass and hybrid striped bass, goldfish, koi, zebra fish colonies.

Hazard Management:

- Good biosecurity and husbandry should be practiced and good water quality maintained to prevent infection.
- It is important to source disease free animals or quarantine all incoming animals.

Figure 24. Atlantic salmon infected with IPNV.
Ian Bricknell

VIRUS

Name: infectious pancreatic necrosis virus (IPNV); caused by aquabirnavirus

Mode of Action: IPNV is a non-enveloped aquabirnavirus that can be vertically transmitted. It can cause significant mortality especially in hatcheries and after smoltification and transfer to marine net pens. The virus can cause erratic, corkscrew swimming in fish and acute intestinal enteritis with the shedding of the intestinal lining and mucusa. Chronically infected asymptomatic carriers can exist, especially in areas where the virus in endemic.

Major Aquaculture Species Affected in this Region:

- salmonids
- Atlantic cod
- Striped bass and hybrids can be carriers of this virus.

Hazard Management:

- Good biosecurity and husbandry should be practiced.
- Restrict movement of infected fish.
- Test striped bass and hybrids before transplanting.

Figure 25a. Atlantic salmon showing external signs of ISAV.
Deborah Bouchard

Figure 25b. Internal organs of an Atlantic salmon infected with ISAV.
Deborah Bouchard

VIRUS

Name: infectious salmon anemia virus (ISAV), caused by orthomixovirus

Mode of Action: ISAV is an enveloped orthomixovirus that can cause significant mortality in marine salmonids. The virus can cause anemia and pale gills, along with petechial (small red to purple discolorations) hemorrhaging. There is a non-pathogenic, wild-type strain that can be found in routine screening for pathogenic ISAV. Fish may exhibit reduced feeding.

Major Aquaculture Species Affected in this Region:

- salmonids

Hazard Management:

- Good biosecurity and husbandry should be practiced.
- All infected cages or tanks should be culled and removed promptly.

Figure 26: Viral hemmorhaggic septicemia virus in gizzard shad.
P.R. Bowser, Cornell University

VIRUS

Name: viral hemorrhagic septicemia virus (VHSV); caused by novirhabdovirus

Mode of Action: VHSV is an enveloped novirhabdovirus with four major genotypes found in both marine and freshwater. VHSV can cause significant mortalities and infected fish may be asymptomatic. External symptoms may include: exophthalmia, petechial (small red to purple discolorations) hemorrhaging of the eyes, skin, gills and fins, bloated appearance, and possibly open sores. VHSV is a highly regulated pathogen.

Major Aquaculture Species Affected in this Region:

- salmonids
- Atlantic cod
- bass
- minnows
- perch

Hazard Management:

- Good biosecurity and husbandry should be practiced.
- Care should be taken with feeding of any wild fishmeal that has not been pasteurized as the virus can withstand freezing.
- All infected cages or tanks should be culled and removed promptly.

VIRUS

Name: viral encephalopathy and retinopathy (VER), viral nervous necrosis; caused by nodavirus

Mode of Action: This is a viral disease seen around the globe in warm and cold waters. Nodavirus can be vertically transmitted and is known to cause rapid significant mortalities (40-100% in 48 hours). It predominately affects larval fish but is known to occur in older fish. Fish can exhibit erratic swimming behavior, resting belly up, hyperactivity, poking head above water, and anorexia. External symptoms may include: color change, blindness, emaciation, and over inflated swim bladder.

Major Aquaculture Species Affected in this Region:

- Atlantic cod
- tilapia

Hazard Management:

- Good biosecurity and husbandry should be practiced.

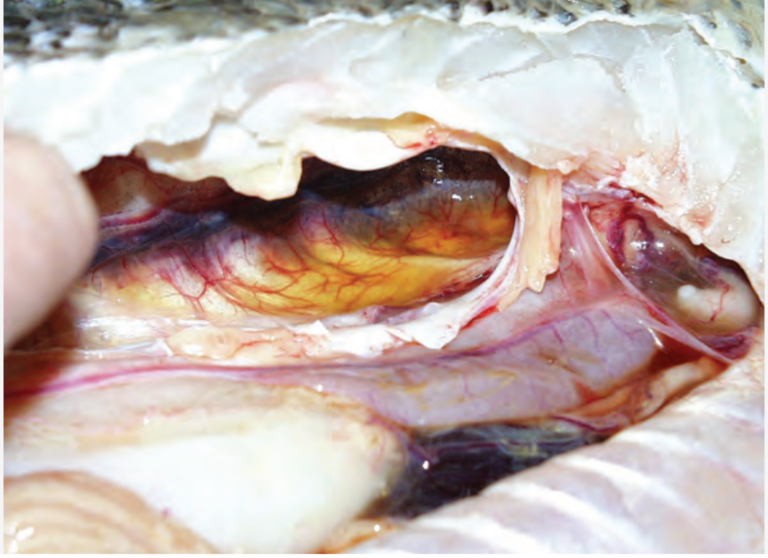


Figure 28: Largemouth bass virus affects the swim bladder of fish.
Andy Goodwin

VIRUS

Name: large mouth bass virus; caused by iridovirus

Mode of Action: Large mouth bass virus (LMBV) is a highly communicable pathogen. Infected fish can swim lethargically or lose equilibrium.

Major Aquaculture Species Affected in this Region:

- largemouth bass, smallmouth bass

Hazard Management:

- Good biosecurity and husbandry should be practiced.
- Animals brought in from the wild should be quarantined.
- Surface water should be disinfected prior to use in culture facilities.

Figure 29: Carp pox infections cause white waxy lumps on fish.
David Cline

VIRUS

Name: carp pox; caused by cyprinid herpesvirus-1 or CyHV-1

Mode of Action: Infection with this virus is often non-lethal except in juvenile fish. Infections are presented as white waxy lumps on the fish, which are considered disfigurements. This virus can be highly infectious in crowded conditions. This virus can be highly infectious in crowded conditions.

Major Aquaculture Species Affected in this Region:

- koi or common carp (*Cyprinus carpio*)

Hazard Management:

- Good biosecurity and husbandry should be practiced and good water quality maintained to prevent infection.
- It is important to source disease free animals or quarantine all incoming animals.
- Water temperatures can be increased to help disfigurements go away naturally.

Figure 30: Koi herpes virus
P.R. Bowser, Cornell University

VIRUS

Name: koi herpes virus disease (KHVD); caused by cyprinid herpesvirus-3 or CyHV-3

Mode of Action: This virus spreads through carrier fish and can cause severe mortality within 7-14 days of infecting susceptible fish. Infected fish may secrete extra mucus clouding the water or lose equilibrium. External signs of infection can include: white patchy appearance in the skin or gills, hemorrhages on the epidermis, and sunken eyes. It can be common for infected fish to develop secondary infections of *Flavobacterium*.

Major Aquaculture Species Affected in this Region:

- koi or common carp (*Cyprinus carpio*)

Hazard Management:

- Good biosecurity and husbandry should be practiced.
- Animals brought in from the wild or untested facilities should be quarantined.
- A vaccine is available.

Figure 31: Spring viremia of carp causes hemorrhaging of the skin.
Andy Goodwin

VIRUS

Name: spring viremia of carp (SVC); caused by *Rhabdovirus carpio*

Mode of Action: This virus has been diagnosed in farmed and wild carp in a selected number of states across the US. The virus typically spreads in the winter when water temperatures are below 10 °C. Mortality starts to occur as the water warms with peak mortality occurring between 15-17 °C. Fish appear to develop immunity at temperatures above 20 °C. Infected fish may exhibit exophthalmia, hemorrhaging of the epidermis, and distended abdomen.

Major Aquaculture Species Affected in this Region:

- koi or common carp (*Cyprinus carpio*)

Hazard Management:

- Good biosecurity and husbandry should be practiced.
- In particular animals should be sourced from certified disease free facilities.
- All surface water should be disinfected prior to use and routine health monitoring should occur.

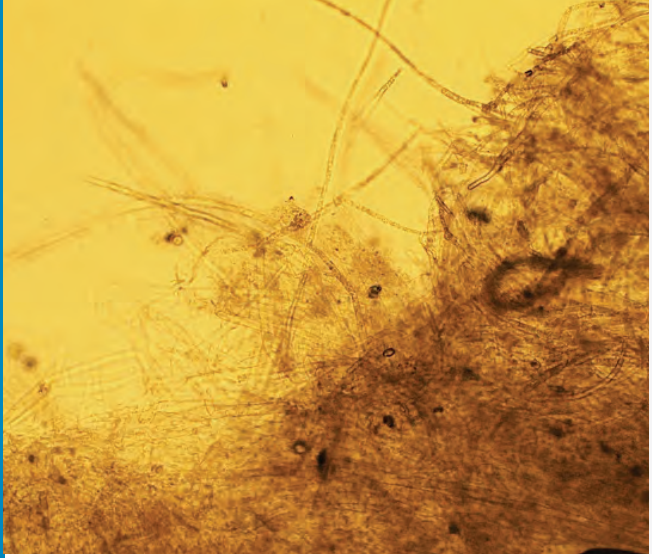


Figure 32a: Close up of fungal hyphae on fish.
Fish Vet Group

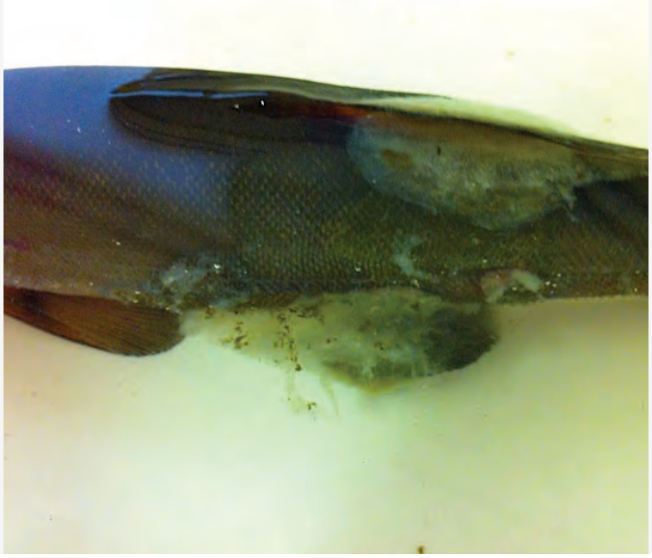


Figure 32b: Fungal hyphae on fish .
Fish Vet Group

FUNGUS

Common Name: saprolegniasis; caused by *Saprolegnia*

Mode of Action: This is an external fungal infection on fish and fish eggs from brackish and freshwater. It is an opportunistic fungus that often appears as grayish white cotton-like growth on the skin, gills, eyes and fins of animals undergoing environmental stress.

Major Aquaculture Species Affected in this Region:

- salmonids
- largemouth bass, striped bass and hybrids
- perch
- minnows
- tilapia
- koi

Hazard Management:

- Good biosecurity and husbandry are important to help prevent infection.
- Excess feed, mortalities and other excess organic materials should be removed frequently from rearing water.
- Saprolegnia can be treated with potassium permanganate, formalin, hydrogen peroxide, and salt under the supervision of an aquatic animal health professional.

Figure 33: Branchiomycosis is largemouth bass.
P.R. Bowser, Cornell University

FUNGUS

Name: branchiomycosis, gill rot; caused by *Branchiomyces sanguinis* and *B. demingrans*

Mode of Action: This fungal disease invades the gills of fish. Infected fish can display gasping at the surface, high mortality and pale or whitish gills.

Major Aquaculture Species Affected in this Region:

- rainbow trout
- largemouth bass, smallmouth bass and striped bass
- minnows
- tilapia
- koi

Hazard Management:

- Good biosecurity and husbandry should be practiced and good water quality maintained to prevent infection.

Figure 34: Ichthyophonus hoferi *is a protozoan parasite that causes granulomas in the tissue.*
Meyers T. et al. 2008. Alaska Department of Fish and Game

FUNGAL-LIKE

Name: *Ichthyophonus hoferi*

Mode of Action: *Ichthyophonus* is a fungal-like parasite of uncertain taxonomy. Spores infect multiple tissues, but particularly the heart causing granulomas to form. Infected tissues become atrophied weakening the overall condition of the fish. There may be few external signs of disease, and they include: rough skin, flat lesions, spongy muscle tissue.

Major Aquaculture Species Affected in this Region:

- salmonids
- Atlantic cod

Hazard Management:

- Good biosecurity and husbandry should be practiced.
- Suspected infected fish should be culled.
- One possible means of infections is thought to be infected feed.
- Moist feeds or feeds made from unpasteurized fish or plankton meal should be avoided.

Fig 35a. Epizootic ulcerative syndrome caused by Aphanomyces invadans.
Yasu Kiryu

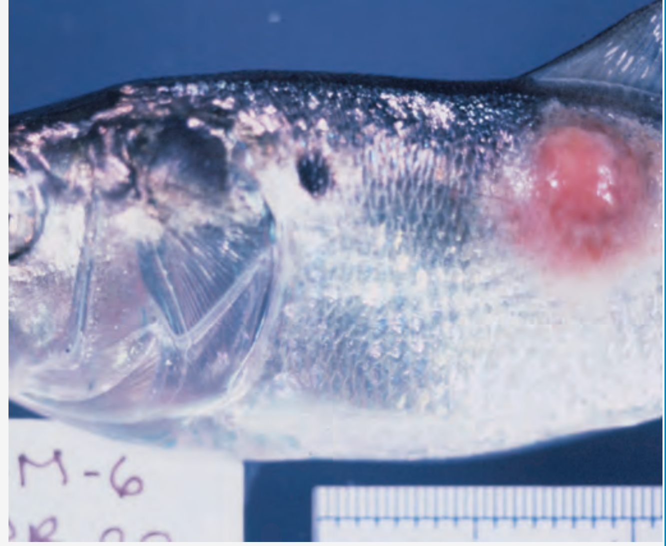


Fig 35b. EUS effects epidermis.
Yasu Kiryu

FUNGAL-LIKE

Common Name: epizootic ulcerative syndrome (EUS), red spot disease (RSD), mycotic granulomatosis (MG) and ulcerative mycosis (UM); caused by *Aphanomyces invadans*

Mode of Action: This is an oomycete or fungal-like disease of freshwater and brackish water fish. *Aphanomyces invadans* is related to *Saprolegnia* but unlike *Saprolegnia* invades deeply into the tissues underlying the skin. Infection occurs when the organism releases swimming zoospores into the surrounding water. If the zoospores do not find a host in a suitable timeframe, then they are capable of encysting. It is unknown how long the zoospores or cysts can remain viable. The organism is known to grow best between 20 – 30 °C. It does not grow above 37 °C or in salinities >2. It generally infects juvenile and young adults and is not known to infect fish fry or larvae. Infected fish may display red spots, small hemorrhagic lesions, or ulcers.

Major Aquaculture Species Affected in this Region:

- bass

Hazard Management:

- Good biosecurity and husbandry should be practiced and good water quality maintained to prevent infection.
- In ponds and tanks liming the water with agricultural lime or adding salt and improving water quality can help to control outbreaks when infected animals are removed.

References and Further Reading:

AFS-FHS (American Fisheries Society-Fish Health Section). 2012. FHS (Fish Health Section) blue book: suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Bailey, T.A., 1994. Branchiomycosis, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Barnes, M.E., Brown, M.L., 2011. A review of *Flavobacterium psychrophilum* biology, clinical signs, and bacterial cold water disease prevention and treatment. The Open Fish Science Journal. 4, 1-9.

Batts, W.N., Winton, J.R., 2010. Viral hemorrhagic septicemia, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Birkbeck, T.H., Feist, S.W., Verner-Jeffreys, D.W., 2011. *Francisella* infections in fish and shellfish. Journal of Fish Diseases. 34, 173-187.

Bowser, P.R., 2009. Fish diseases: Viral Hemorrhagic Septicemia (VHS). USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 201-2009.

Bowser, P.R., 2009. Fish diseases: Mycobacteriosis of fish. USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 202-2009.

Bowser, P.R., 2009. Fish diseases: Spring viremia of Carp (SVC). USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 203-2009.

Bovo, G., 2012. Viral encephalopathy and retinopathy. In OIE Manual for Diagnostic Tests for Aquatic Animals 2012, Section 2.3.11. Last accessed on February 28, 2013 at: http://www.oie.int/fileadmin/Home/eng/Health_standards/aahm/2010/2.3.02_EUS.pdf.

Bruno, D.W., 1996. Cold water vibriosis caused by *Vibrio salmonicida*. The Scottish Office Agriculture, Environment and Fisheries Department, Aquaculture Information Series No. 15.

Bullock, G.L., 1990. Bacterial gill disease of freshwater fishes. U.S. Department of the Interior Fish and Wildlife Service, Fish Disease Leaflet No. 84.

Bullock, G.L., 2003. Columnaris disease, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Bullock, G.L., 2003. Enteric redmouth disease, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Camus, A.C., Durborow, R.M., Hemstreet, W.G., Thune, R.L., Hawke, J.P., 1998. *Aeromonas* bacterial infections – motile aeromonad septicemia. USDA Southern Regional Aquaculture Center, SRAC Publication No. 478.

Crozier, D.M., Molloy, D.P., Bartholomew, J., 2013. Whirling disease – *Myxobolus cerebralis*. Last accessed on January 2013 at: http://el.erdc.usace.army.mil/ansrp/myxobolus_cerebralis.pdf.

Dannevig, B., 2009. Infectious salmon anaemia. In OIE manual for Diagnostic Tests for Aquatic Animals 2012, Section 2.3.5. Last accessed on February 28, 2013 at: http://www.oie.int/fileadmin/Home/eng/Health_standards/aahm/2010/2.3.02_EUS.pdf.

Durborow, R.M., 2003. Protozoan parasites. USDA Southern Regional Aquaculture Center, SRAC Publication No. 4701.

Durborow, R.M., Mitchell, A.J., Crosby, M.D., 1998. Ich (white spot disease). USDA Southern Regional Aquaculture Center, SRAC Publication No. 476.

Durborow, R.M., Thune, R.L., Hawke, J.P., Camus, A.C., 1998. Columnaris disease: a bacterial infection caused by *Flavobacterium columnare*. USDA Southern Regional Aquaculture Center, SRAC Publication No. 479.

Durborow, R.M., Wise, D.J., Terhune, J.S., 2003. Saprolegniasis (winter fungus) and branchiomycosis of commercially cultured channel catfish. USDA Southern Regional Aquaculture Center, SRAC Publication No. 4700.

Ewing, M.S., 2002. *Ichthyophthiriasis*, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Francis-Floyd, R., Reed, P., 1997. *Ichthyophthirius multifiliis* (white spot) infections in fish. University of Florida, Cooperative Extension Service, Circular No. 920.

Ganzhorn, J., 1994. Vibriosis, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Goodwin, A., 2012. Herpes viruses in fish. USDA Southern Regional Aquaculture Center, SRAC Publication No. 4710.

Goodwin, A.E., Winton, J.R., 2004. Spring viremia of carp, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Grizzle, J.M., 2007. Largemouth bass virus disease, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Hartman, K.H., Yanong, R.P.E., Pouder, D.B., Petty, B.D., Francis-Floyd, R., Riggs, R.C., 2008. Koi herpesvirus (KHV) disease. School of Forest Resources and Conservation Program in Fisheries and Aquatic Sciences and the Department of Large Animal Clinical Sciences (College of Veterinary Medicine), Florida Cooperative Extension Service, Institute of Food and Agricultural Sciences, University of Florida, Fact Sheet VM-149.

Hawke, J.P., 2003. *Edwardsiella tarda* septicemia, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Hedrick, R.P., 2004. Koi herpes virus disease, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Holt, R.A., Bertolini, J., Taylor, P.W., 2003. Coldwater disease, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Johnson, K., 1994. Lernaeid parasitism. External infection by ciliated parasites, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Johnson, S., Margolis, L., 1994. Sea lice, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Kanchanakhan, S., 2012. Section 2.3.2 Epizootic Ulcerative Syndrome. In OIE Manual for Diagnostic Tests for Aquatic Animals 2012. Last accessed on February 28, 2013 at: http://www.oie.int/fileadmin/Home/eng/Health_standards/aahm/2010/2.3.02_EUS.pdf.

Keleher, W.R., Bouchard, D.A., Merrill, P.L., 2001. Infectious salmon anemia, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Khan, R.A., 2005. Prevalence and influence of *Loma branchialis* (Microspora) on growth and mortality in Atlantic cod (*Gadus morhua*) in coastal Newfoundland. Journal of Parasitology. 91, 1230-1232.

Klinger, R., Francis-Floyd, R., 2009. Introduction to freshwater fish parasites. Fisheries and Aquatic Sciences Department, Florida Cooperative Extension Service, Institute of Food and Agricultural Sciences, University of Florida, Circular No. 716.

MacConnell, E., 2002. Whirling disease of salmonids, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Mayer, K., 2000. *Saprolegnia*: there is a fungus among us. Last accessed on January 31, 2013 at: http://tnfish.org/FishDiseasesParasites_TWRA/files/Saprolegnia.pdf

McAllister, P.E., 2007. Infectious pancreatic necrosis, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Meyers, T., Burton, T., Bentz, C., Starkley, N., 2008. Common Diseases of Wild and Cultured Fishes in Alaska. Alaska Department of Fish and Game.

Mitchell, A.J., Durborow, R.M., Crosby, M.D., 1998. Proliferative gill disease (hamburger gill disease). USDA Southern Regional Aquaculture Center, SRAC Publication No. 475.

Olesen, N.J., Skall, H.F., 2012. Viral haemorrhagic septicaemia. In OIE manual for Diagnostic Tests for Aquatic Animals 2012, Section 2.3.9. Last accessed on February 28, 2013 at: http://www.oie.int/fileadmin/Home/eng/Health_standards/aahm/2010/2.3.02_EUS.pdf.

Pascho, R.J., Elliott, D.G., 2001. Bacterial kidney disease, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Petty, B.D., Riggs, A.C., Klinger, R., Yanong, R.P, Francis-Floyd, R., 2012. Spring viremia of carp. Program in Fisheries and Aquatic Sciences, School of Forest Resources and Conservation, Florida Cooperative Extension Service, Institute of Food and Agricultural Sciences, University of Florida, Fact Sheet VM-142.

Poynton, S.L., 2003. Diplomonad (Hexamitid) flagellates: diplomonadiasis, hexamitosis, spironucleosis, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Rahimian, H., 1998. Pathology and morphology of *Ichthyophonus hoferi* in naturally infected fishes off the Swedish Coast. Diseases of Aquatic Organisms. 34, 109-123.

Reed, P., Francis-Floyd, R., 1994. *Amylodinium* infections of marine fish. College of Veterinary Medicine, Florida Cooperative Extension Service, Institute of Food and Agricultural Sciences, University of Florida, Fact Sheet No. VM-90.

Roberts, S.D., 1994. Bacterial gill disease, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Rodgers, W., 1994. Ichthyobodiasis, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Schultz, M., May, E.B., Kraeuter, J.N., Hetrick, F., 1984. Isolation of infectious pancreatic necrosis virus from an epizootic occurring in cultured striped bass (*Morone saxatilis*). Journal of Fish Diseases. 7, 505-507.

Shotts Jr., E.B., 1994. Furunculosis, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Shotts Jr., E.B., Plumb, J., 1994. Streptococcal disease, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Smith, S., Schwarz, M., 2009. Dealing with *Trichodina* and *Trichodina-like* species. Virginia Cooperative Extension, Publication No. 600-205.

Taylor, P.W., Goodwin, A.E., 2002. External infection by ciliated parasites, in: American Fisheries Society-Fish Health Section (Ed.), Fish Health Section Blue Book: Suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Tirmizi, H., 2003. Animal Diversity Web: *Lernaea cyprinacea*. Last accessed on February 23, 2013 at: http://animaldiversity.ummz.umich.edu/accounts/Lernaea_cyprinacea/.

Wechsler, S.J., Woods, L.C., Kraeuter, J.N., Hetrick, F.M., McAllister, P.E., 1987. Transmission of infectious pancreatic necrosis virus in striped bass, *Morone saxatilis* (Walbaum). Journal of Fish Diseases. 10, 29-34.

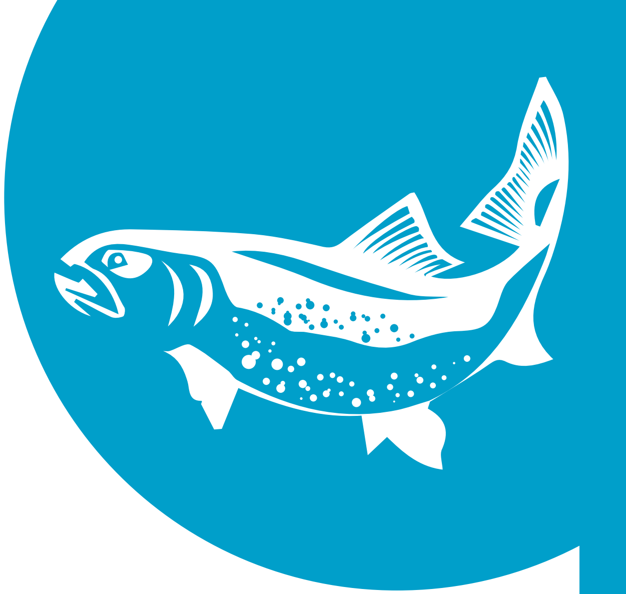
Williams C.F., Lloyd, D., Poynton S.L., Jorgensen A., Millet, C.O.M., Cable, G., 2011. Spironucleus species: Economically-Important Fish Pathogens and Enigmatic Single-Celled Eukaryotes. Journal of Aquaculture Research and Development, Open Access Report S2.

Yanong, R.P.E, Francis-Floyd, R., 2010. Streptococcal infections of fish. Florida Cooperative Extension Service, Institute of Food and Agricultural Sciences, University of Florida, Circular No. 57.

Zubchenko, A.Z., Karaseva, T.A., 2002. *Ichthyophonus hoferi* as one of possible causes of increased marine mortality in post-smolts of Atlantic salmon. North Pacific Anadromous Fish Commission, Technical Report No. 4.

Potential Finfish Production Hazards

Environmental Conditions
Biofouling Organisms
Predators
Diseases and Parasites
Invasive Species
Operational Procedures



Invasive Species

Non-native, exotic, non-indigenous, or foreign are various terms used to describe aquatic organisms that cause harm and thrive outside their natural range. These aquatic invasive species (AIS) can be harmful invaders ranging in size from the microscopic virus to as large as a 120-pound flathead catfish. While some invaders escaped from other continents (i.e. ship ballast water, hitchhikers in shipments of other species), a surprising number have been spread by human activity from other parts of North America.

Why should fish farmers care about AIS? In short, invasives can cause health, environmental, or economic harm including impacts to finfish businesses. Effects from bio-invasions include ecosystem disruptions, increased business production costs, and management costs for the public. When an infestation of New Zealand mud snails closed down a trout farm in Colorado the cost to eradicate these tiny snails from the farm was significant and subsequently a national plan to control these snails was been implemented. Left uncontrolled, the snail population would have exploded, clogging all the pipes. Mud snails also posed a risk of spreading to other waterways and aquaculture facilities. When viral hemorrhagic septicemia (VHS), an infectious disease, spread from freshwater salmonids in Western Europe to North America it caused large-scale die-offs of a wide variety of fish in the Great Lakes region. Most American fish farmers now feel the pinch of tighter regulations on live fish movement designed to reduce the risk of VHS to native fish populations and to protect aquaculture. Asian carp that escaped into the Mississippi drainage caused a trifecta of damage to health, the economy, and the native fisheries. Scientists are working to understand the effect of silver carp on native fish populations. The federal government has identified and is developing management plans for a number of aquatic invasive species.

In most cases, the cost to prevent an introduction is a small fraction of the cost to control an invader once it becomes established. By applying biosecurity principles to aquaculture operations, the risk of costly AIS introductions can be greatly minimized. Knowing whether the activities at a facility present a risk is the first step. Most operations pose a very low risk; however, without adequate assessment of each aquaculture facility's risks, unwanted species may show up or be inadvertently spread. By identifying the ways in which AIS might be introduced to the aquaculture operation - the vectors - a farmer can design operating procedures to minimize the risk of new introductions. For example, movements of fish, fingerlings, eggs, water, boats, equipment, and baitfish could be potential AIS vectors; farmers can develop methods to monitor and minimize the AIS risk for each activity. A vector-based approach can effectively deal with diverse practices and operations and vector management has been shown to prevent and delay the spread of AIS to new areas.

References and Further Reading:

Aquatic Nuisance Species Task Force. 2007. National Management and Control Plan for the New Zealand Mudsnail (*Potamopyrgus antipodarum*). Prepared for the ANS Task Force by the New Zealand Mudsnail Management and Control Plan Working Group.

Benson, A.J., Kipp, R.M., Larson, J., Fusaro, A., 2012. *Potamopyrgus antipodarum*. USGS Nonindigenous Aquatic Species Database, Gainesville, Florida. Revision Date: 6/11/2012.

Freedman, J.A., Butler, S.E., Wahl, D.H., 2012. Impacts of Invasive Asian Carps on Native Food Webs" that points to potential impacts. Illinois-Indiana Sea Grant, Final Report.

Hill, J., 2008. Non-native species in aquaculture: terminology, potential impacts, and the invasion process. USDA Southern Regional Aquaculture Center, SRAC Publication No 4303.

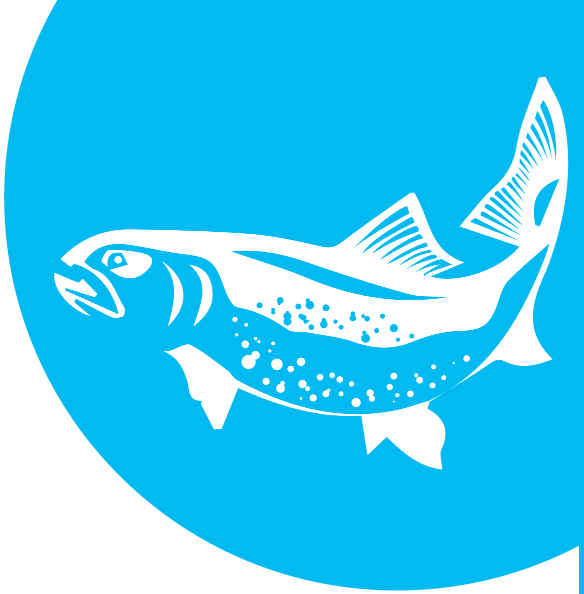
Irons, K.S., Sass, G.G., McClelland, M.A., Stafford, J.D., 2007. Reduced condition factor of two native fish species coincident with the invasion of non-native Asian carps in the Illinois River, USA. Is this evidence for competition and reduced fitness?" Journal of Fish Biology. 71, 258-273.

Pimentel, D., Zuniga, R., Morrison, D., 2005. Update on the environmental and economic costs associated with alien-invasive species in the United States. Ecological Economics. 52(3), 273–288.

Zajicek, P.W., Hill, J.E., Stone, N., Thomforde, H., Ohs, C., Cooper, D., Flimlin, G., Lane, B., Anderson, W.D., 2009. Preventing hitchhiking nonindigenous species in live shipments. USDA Southern Regional Aquaculture Center, SRAC Publication No. 3902.

Potential Finfish Production Hazards

Environmental Conditions
Biofouling Organisms
Predators
Diseases and Parasites
Invasive Species
Operational Procedures



Operational Procedures

In the operation of any business, especially one as technical as aquaculture, hazards can be encountered in operational routine, which can have significant impacts. For this reason, it is important to consider operations and write down standard operating procedures (SOP) for production systems regardless of type or species. The goal of the operational and system hazards section is to outline potential hazards in operational procedures of finfish culture systems which have the potential to cause losses, in terms of livestock or profitability, so that they are considered before they become an issue. These can be critical steps in an operation, routine or occasional, where risk is enhanced and steps can be taken to ensure the risk is minimized. With each potential hazard, strategies for minimizing the risks are discussed.

Procedural operations will vary by farm setup, but the following general guidelines should be considered:

- Record keeping is extremely important on many levels including: production schedules, business planning, regulatory compliance, trouble shooting.
- Establish standard operating procedures (SOP) for every practice or routine to ensure streamlined and thorough operations .
- All staff should be made aware of SOP and the logic behind them.
- Handling of fish should be minimized and well planned to reduce fish stress.
- Eggs or juveniles should be sourced carefully, and quarantined or disinfected if appropriate.
- All-in, all-out stocking is a recommended practice to minimize health risk to naive populations being stocked into the systems.
- Keep a regular inventory of fish size and number.
- Grade fish to separate size classes which can improve feed efficiency and reduce cannibalism.

Potential hazards:

- **introduction of pathogens when sourcing eggs/fry/ juveniles**
- **loss due to system deficiencies**
- **handling stress**
- **variation in fish growth**
- **equipment failure**
- **feed quality**
- **feed storage**
- **feed management**

Potential Hazard: introduction of pathogens when sourcing eggs/fry/juveniles

Mode of Action: Depending on the source of animals there can be significant risk for the introduction of potential pathogens. If a pathogen is carried by incoming animals it can be introduced to the system and become an issue for fish health. Consideration must be made when anything is brought on site with the potential to harbor pathogens.

Systems Affected: **all**

Hazard Management:

- Only bring in animals approved to be pathogen free, whether eggs, fry or juveniles. If in-house hatchery production is performed, similar care should be taken when sourcing brood stock.
- Quarantine systems on arrival are a good idea when bringing in animals, especially larger fish that have greater potential pathogen exposure and may need to be evaluated before further stock out.
- It is also a good practice in open systems such as net pens or ponds, or in situations where pathogens may be suspected, to harvest or transfer the entire lot of fish before a new and naive population is restocked to the system. It also helps minimize variations in size or growth.
- Special considerations must also be made to move fish grown on a farm site to a different site. There are usually regulatory guidelines for this as well. Consult local and state authorities if needed.

Potential Hazard: loss due to system deficiencies

Mode of Action: Fish can be lost due to simple system deficiencies such as improper screen or net sizes, stand pipe height, water flow, aeration level, predator enclosure, etc. A system should be designed to have all the necessary pieces for the production cycle intended, but likewise, as every new cycle is initiated, adjustments may be necessary to ensure it is ready for restocking. An error as small as the position of a valve or standpipe can sometimes lead to large-scale losses.

Systems Affected: **all**

Hazard Management:

- A protocol should be established for everything that requires a check or adjustment before a system is ready for stocking with fish.
- Adjustments made to the system through the course of production should be recorded and communicated to other staff so that they are aware of changes.
- Protocols should also be established for all routine operations so that any question as to what should be done is available for reference.
- Emergency protocols, contacts, and contingency plans should also be established and readily available for all potential situations, such as: storms, floods, power outages, suspected theft, disease, contamination or loss, etc.
- All staff should be aware of these protocols so that, in times of confusion; written guidance is available.

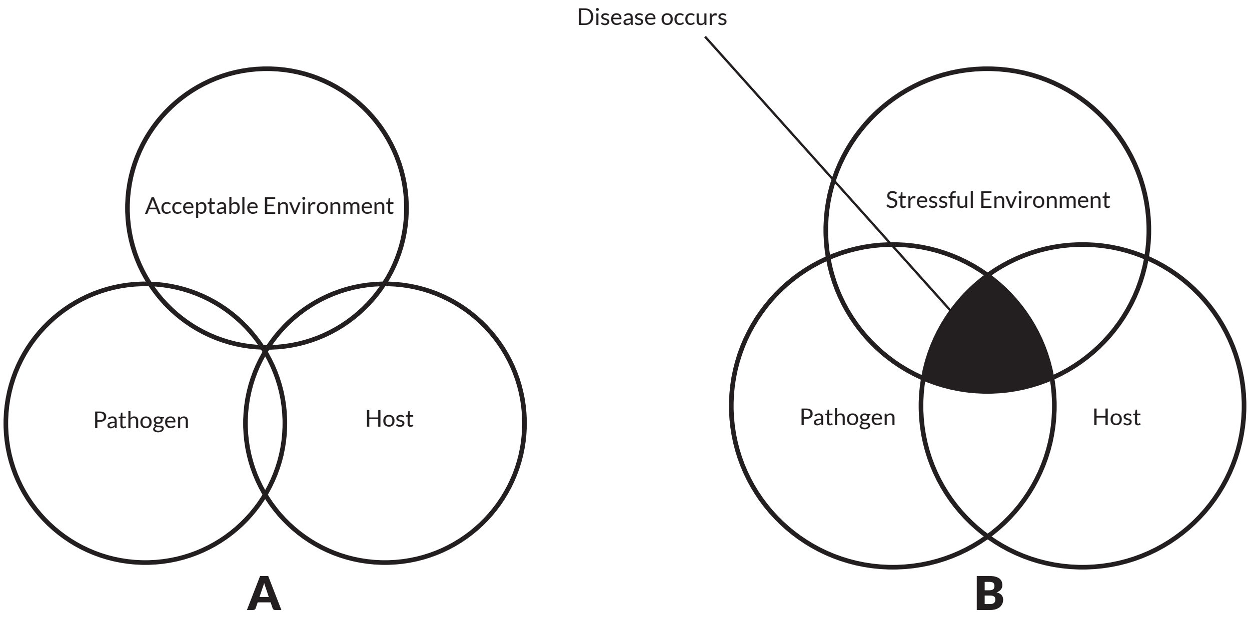


Figure 1:Image showing the influence of stress on fish health
LSU AgCenter

Potential Hazard: handling stress

Mode of Action: Many operations throughout the cycle of finfish production require the fish to be physically handled. These include: moving animals between systems (tanks, ponds, pens, raceways, etc.), vaccinations, grading, stock inventories, and harvest. Any time a fish is handled there is opportunity to inflict direct physical damage or cause stress. As fish become stressed, the overall health of the animals becomes compromised, so stress should be minimized.

Systems Affected: **all**

Hazard Management:

- Only handle animals when necessary and ensure animals are healthy enough for the handling operation.
- Make sure water quality is within the optimal range for all systems involved with handling operations.
- It is good practice to take fish off feed for 1-2 days before handling (depending on water temperature) to allow food to be cleared from the system. This reduces fish oxygen consumption and metabolite production, providing for more stable water quality conditions during handling.
- Handling surfaces should be cleaned and kept moist to minimize mucous sloughing during handling. If an anesthetic or therapeutic is to be used, use only according to manufacturer or veterinarian guidelines, and provide a space for animal recovery.
- Special considerations may be needed for moving animals off-site, or to new systems, including tempering for changes in temperature, salinity, or pH.

Figure 2: Cannibalism can be a major problem with some species of fish like this example with tiger muskellunge.
Brian Richardson, Maryland Department of Natural Resources

Potential Hazard: variation in fish growth

Mode of Action: As a stock of fish grows, individuals rarely grow uniformly, even when starting at the same length or weight. As fish grow at different rates, problems may emerge such as cannibalism by larger fish on smaller fish, reduced feeding efficiency due to larger fish dominance, or the inability to effectively match feed size to the growing stock. A large disparity in size of fish within a stock also can complicate harvest when fish are usually desired at a fairly uniform size.

Systems Affected: **all**

Hazard Management:

- As a stock of fish grows it is important to take inventory routinely of the length and weight of the fish. This allows for an assessment of health and condition, and will also allow for management decisions in terms of feed size, rate, and timing of production steps.
- Knowing the size and number of fish also allows management of the stocking density of the system, so that overstocking can be avoided before it happens.
- Periodic grading is recommended to help prevent cannibalism and to separate size classes within a stock to optimize feed efficiency and growing conditions.
- Record keeping of all inventory data is invaluable to project growth and harvest, feed requirements, and can be important in determining when and where problems may have originated.

Potential Hazard: equipment failure

Mode of Action: The equipment used to provide the optimal conditions for animal growth and production efficiency can fail resulting in all or partial loss of animal stocks. Common reasons for equipment failure include: normal wear and tear, loss of power, loss of control systems, exceeding design capacity and human error.

Systems Affected: **all**

Hazard Management:

- It is important that facility designs and equipment selected is sufficient to handle the maximum load or biomass expected in addition to an appropriate safety buffer.
- All critical systems should be identified and redundant backup systems installed. When selecting backup systems it is important to consider if all components will function in the event there is no power.
- It is also important to establish a routine maintenance plan for all equipment according to the manufacturer's recommendations.
- A system to track equipment maintenance can be extremely useful.
- These measures should be reviewed on a regular basis and whenever new equipment is added or facility procedures change.

Potential Hazard: poor feed quality

Mode of Action: Use of poor quality or inappropriate feed leads to lower profits, increased waste production, and can lead to poor health due to nutritional deficiencies.

Systems Affected: **all**

Hazard Management:

- It is important to seek out and utilize the proper diet for the culture species.
- Feed manufacturers can assist and there are fact sheets available for species-specific nutrition for most currently cultured species.
- Ask feed manufacturers about the ratio of digestible protein to digestible energy. Most fish diets contain some level of fishmeal and fish oil; ask how the fishmeal is processed. A low-temperature pasteurized fishmeal costs more but is more digestible and improves the efficiency with which fish utilize the protein and reduces the waste produced.
- For some species feed manufacturers make low phosphorous diets, which can decrease waste concerns.

Potential Hazard: compromised feed in storage

Mode of Action: Improperly stored feed can attract pests and predators. Rodents can move throughout a facility potentially spreading disease in addition to sanitation issues. Improperly stored feed can also lead to decreased shelf life of feed and increased spoilage. Excessive temperatures can reduce the shelf life of feed and increase the decay rate of some vitamins and nutrients. Feeds that have been stored improperly may become less palatable to the fish or even contain toxic molds. Improper storage of live feeds or algae is a particular risk for loss of nutritional quality or microorganism contamination.

Systems Affected: **all**

Hazard Management:

- Feed should be stored in a designated location that is protected from the elements and preferably climate controlled according to manufacturer's instructions.
- Open feed bags should be stored in closed containers and spilled feed should be regularly cleaned up to reduce rodents and pests.
- A rodent and pest management plan should be establish to help keep these animals away from stored feed.

Potential Hazard: poor feed management or feed efficiency

Mode of Action: Improper feed management can reduce the feed conversion ratios, increase production costs and result in reduced water quality or environmental concerns.

Systems Affected: **all**

Hazard Management:

- Once proper feeds are selected, it is important to select the appropriate delivery method and feeding schedule.
- Feed should be delivered at an appropriate rate for the life stage and species of fish. It is important to have a mechanism for monitoring feed delivery, whether automated or by manual observation.
- Underwater video cameras can be utilized to feed to satiation or various scales to feed a known amount of feed per feeding.
- The appropriate feeding strategy is species and system specific. Regardless of the strategy used it is important to keep accurate records of feeding activities and regularly sample the growth of the fish.
- This data can be used to monitor feed conversion ratios, which are lower with greater efficiency, as well as other production performance measures.
- For some species it is important to distribute the feed evenly across the cage to prevent dominant fish from guarding the feed.

References and Further Reading:

Cain, K., Garling, D., 1993. Trout culture in the north central region. USDA North Central Regional Aquaculture Center, NCRAC Publication No. 108.

Coyle, S.D., Durborow, R.M., Tidwell, J.H., 2004. Anesthetics in aquaculture. USDA Southern Regional Aquaculture Center, SRAC Publication No. 3900.

Creswell, L., 2010. Phytoplankton culture for aquaculture feed. USDA Southern Regional Aquaculture Center, SRAC Publication No. 5004.

Davis, K.B., 2006. Management of physiological stress in finfish aquaculture. North American Journal of Aquaculture. 68, 116-121.

Gatlin III, D.M., 2010. Principles of fish nutrition. USDA Southern Regional Aquaculture Center, SRAC Publication No. 5003.

Grusenmeyer, D., 2003. Developing effective standard operating procedures. Cornell University Department of Animal Science, Publication No. 2003-02-01.

Hinshaw, J.H., 1999. Trout production: feeds and feeding methods. USDA Southern Regional Aquaculture Center, SRAC Publication No. 223.

Jensen, G.L., 1990. Sorting and grading warm water fish. USDA Southern Regional Aquaculture Center, SRAC Publication No. 391.

Killian, H.S., Heikes, D., Van Wyk, P., Masser, M., Engle, C.R., 1998. Inventory assessment methods of aquaculture ponds. USDA Southern Regional Aquaculture Center, SRAC Publication No. 395.

Lochmann, R., Stone, N., Park, E., 2002. Baitfish: feeds and feeding practices. USDA Southern Regional Aquaculture Center, SRAC Publication No. 121

Masser, M., Rakocy, J., Losordo, T., 1999. Recirculating aquaculture tank production systems management of recirculating systems. USDA Southern Regional Aquaculture Center, SRAC Publication No. 452.

McIntosh, D., 2008. Aquaculture risk management. USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 107.

Morris, J.E., Kohler, C.C., Mischke, C.C., 1999. Pond culture of hybrid striped bass in the north central region. USDA North Central Regional Aquaculture Center, NCRAC Publication No. 107.

Pietrak, M., Leavitt, D., Walsh, M., 2010. Biosecurity on the farm – guidelines & resources for developing a biosecurity plan. Northeastern Regional Aquaculture Center, NRAC Publication No. 208.

Pomeroy, R., 2003. Aquaculture record keeping. Connecticut Sea Grant, Publication No. CTSG-03-15.

Rice, M.A., Bengston, D.A., Jaworski, C., 1994. Evaluation of artificial diets for cultured fish. USDA Northeastern Regional Aquaculture Center, NRAC publication No. 222.

Riche, M., Garling, D., 2003. Feeding tilapia in intensive recirculating systems. USDA North Central Regional Aquaculture Center, NCRAC Publication No. 114.

Rottmann, R.W., Francis-Floyd, R., Durborow, R., 1992. The role of stress in fish disease. USDA Southern Regional Aquaculture Center, SRAC Publication No. 474.

Royes Russo, J.A., Yanong, R.P.E., 2002. Molds in fish feeds and aflatoxicosis. Department of Fisheries and Aquatic Sciences, Florida Cooperative Extension Service, Institute of Food and Agricultural Sciences, University of Florida, Circular No. FA-95.

Sadler, J., Goodwin, A., 2007. Disease prevention on fish farms. USDA Southern Regional Aquaculture Center, SRAC Publication No. 4703.

Schwarz, M.H., Delbos, B., Mclean, E., Craig, S., 2009. Intensive marine finfish larviculture. Virginia Cooperative Extension, Publication No. 600-050.

Schwarz, M.H., Jahncke, M.L., Lazur, A.M., 2010. Overview of good aquaculture practices. Virginia Cooperative Extension, Publication No. 600-054.

Yanong, R.P.E., 2003. Fish health management considerations in recirculating aquaculture systems - part 3: general recommendations and problem-solving approaches. Department of Fisheries and Aquatic Sciences, Florida Cooperative Extension Service, Institute of Food and Agricultural Sciences, University of Florida, Circular No. 122.

Yanong, R.P.E., Erlacher-Reid, C., 2012. Biosecurity in aquaculture, part 1: an overview. USDA Southern Regional Aquaculture Center, SRAC Publication No. 4707.

CHAPTER 5

Seaweed Aquaculture in the Northeastern U.S.

Overview

Marine macroalgae (a.k.a. “seaweeds”, “sea vegetables”) and macroalgal extracts have many applications that are utilized in a variety of industries, including food, animal feeds, phycocolloids, ecosystem services, etc. While 99.8% of seaweed aquaculture occurs in China, Korea, Japan, and Chile, market demand for edible seaweeds in North America is estimated at over $35 million. Concerns over biosecurity of food imports and the increasing demand for locally produced food have added to the appeal of cultivating native species of sea vegetables in the U.S.

Seaweed aquaculture is a new and developing industry in the United States. Building upon many decades of research on native species, growers are now gaining invaluable experience in the field as domestic cultivation develops and expands on the North American coast. The summary below covers just a fraction of the information available about macroalgal cultivation and is meant to be a starting point. This chapter will grow, no doubt, along with the seaweed industry. The two species covered in this chapter (*Saccharina latissima* and *Gracilaria tikvahiae*) are two types of seaweed that are currently in cultivation on the East Coast of the U.S., though there are several other native species with great economic potential (e.g. “nori” species [*Porphyra*, *Pyropia*, and *Wildemania*], *Chondrus crispus*, *Palmaria palmata*, etc.)

This chapter includes an overview of morphology and life cycles for the two species, as well as images of growout culture. Nursery culture has been reviewed in detail in recent publications (see reference list on next page), and, thus, is not covered in this manual.

General Hazard Management Strategies:

- Select a culture site with sufficient current flow and nutrient levels;
- Culture only locally sourced, native strains;
- Out-plant during optimal growing conditions to ensure rapid growth, which can inhibit recruitment of predators, biofouling organisms and other epiphytes;
- Maintain optimal densities to prevent epiphytes and fouling epifauna from colonizing target crop; and
- Harvest before predators, fouling, and epiphytes become established.

Sources and Further Reading:

FAO (Food and Agriculture Organization), 2010. The State of World Fisheries and Aquaculture. Rome, FAO.

FAO (Food and Agriculture Organization), 1990. Training manual on *Gracilaria* culture and seaweed processing in China. Training Manual 6.

Flavin, K., Flavin, N., Flahive, B., 2013. Kelp Farming Manual: A Guide to the Processes, Techniques, and Equipment for Farming Kelp in New England Waters. Ocean Approved, Portland, Maine.

Lopez, G., Carey, D., Carlton, J., Cerato, R., Dam Guerrero, H., Digiovanni, C., Elphick, C., Frisk, M., Gobler, C., Hice, L., Howell, P., Jordan, A., Lin, S., Liu, S., Lonsdale, D., McEnroe, M., McKown, K., Mcmanus, G., Orson, R., Peterson, B., Pickerell, C., Rozsa, R., Shumway, S., Talmage, S., Taylor, G., Thomas, D., Van Patten, P., Vaudrey, J., Wikfors, G., Yarish, C., Zajac, R., 2013. Biology and Ecology of Long Island Sound, Chapter 6. In Long Island Sound: Prospects for the Urban Sea. Latimer, J.S., Tedesco, M., Swanson, R.L., Yarish, C., Stacey, P., Garza, C. (Eds.), Springer Publishers, New York.

Redmond, S., L. Green, L., Yarish, C., Kim, J., Neefus, C., 2014. New England Seaweed Culture Handbook. Nursery Systems. Connecticut Sea Grant, CTSG-14-01.

Santelices, B., Doty, M., 1989. A review of *Gracilaria* farming. Aquaculture, 78(2) 95-133.

Yarish, C., Pereira, R., 2008. Mass production of Marine Macroalgae. In: Jørgensen, S.E., Fath, B.D. (Eds.), Ecological Engineering. Vol. [3] of Encyclopedia of Ecology, 5 vols. pp. 2236-2247. Elsevier, Oxford.

Yarish, C., Redmond, S., Kim, J., 2012. *Gracilaria* Culture Handbook for New England. Wrack Lines (report). Paper 72. Last access on August 16, 2013 at: http://digitalcommons.uconn.edu/wracklines/72.

Yarish, C., Redmond, S., Kim, J., 2012. *Gracilaria* Culture Handbook for New England. Wrack Lines (video). Paper 70. Last accessed on August 16, 2013 at: http://digitalcommons.uconn.edu/wracklines/70.

Seaweed Morphology and Life Cycle

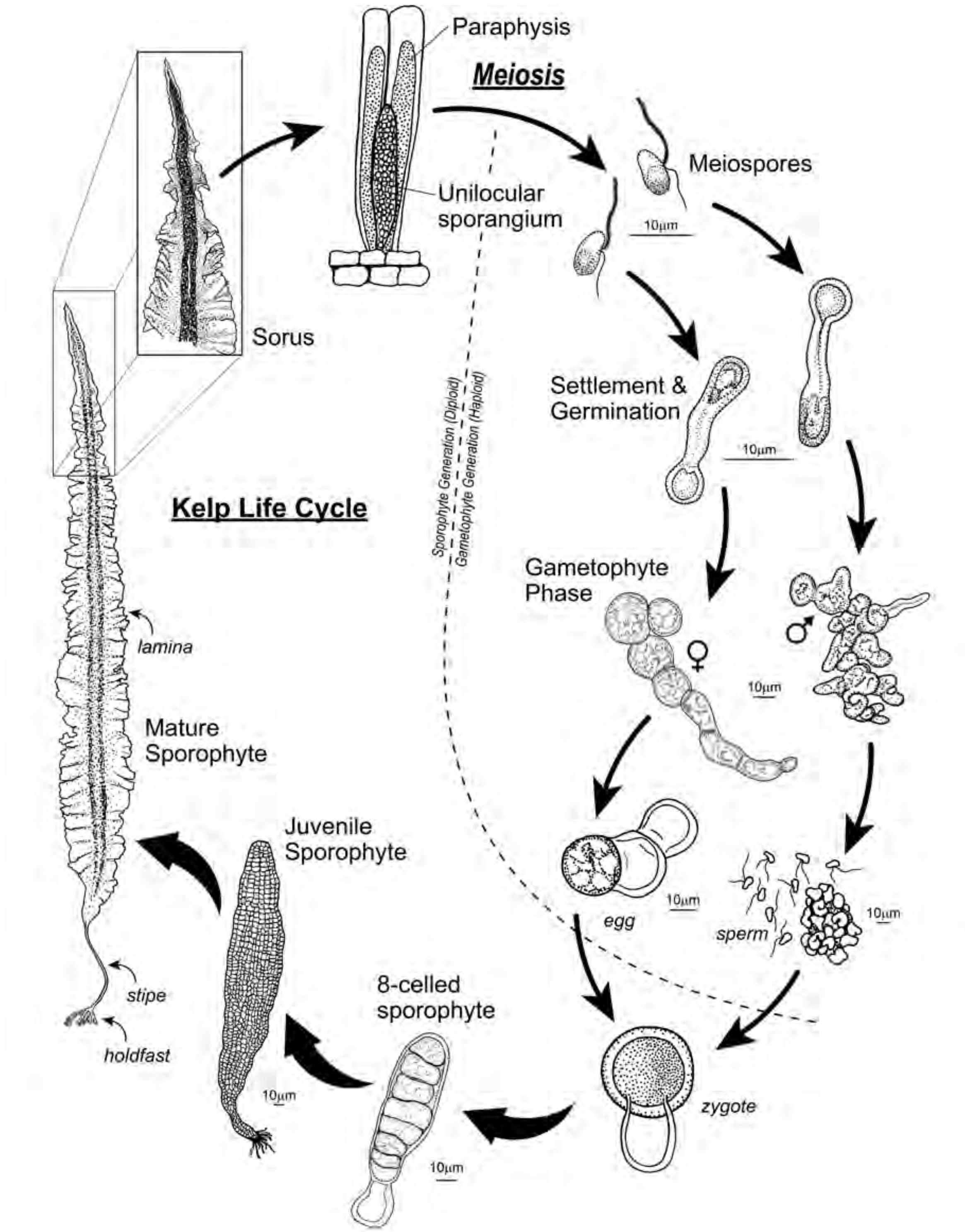


Figure 1: Morphology and life cycle of the kelp, Saccharina latissima.

Virge Kask, Reprinted with permission from: Redmond, S., Green, L., Yarish, C., Kim, J., Neefus, C., 2014. New England Seaweed Culture Handbook Nursery Systems. Connecticut Sea Grant Publication No. CTSG-14-01. 92 pp. http://seagrant.uconn.edu/publications/aquaculture/handbook.pdf.

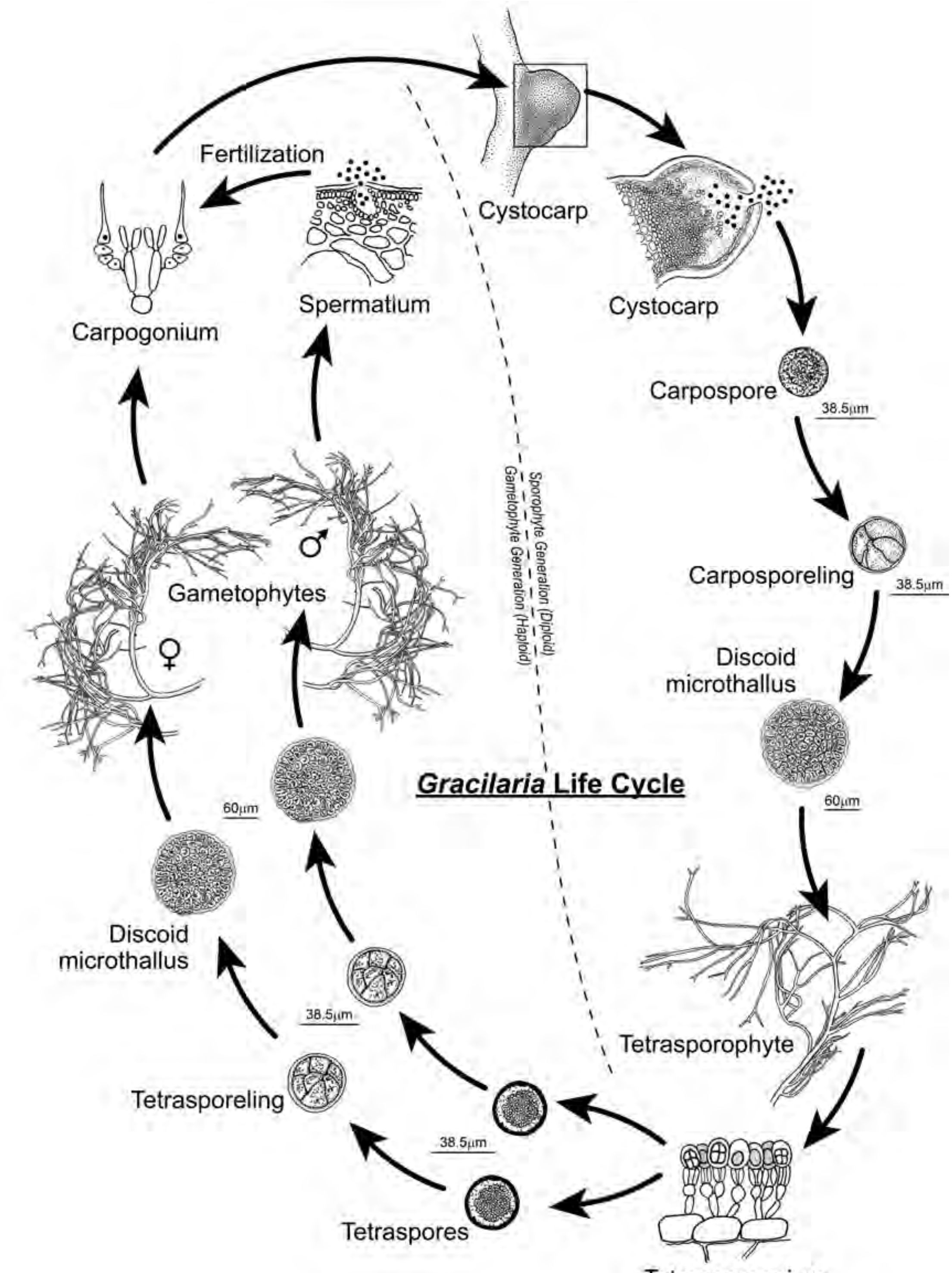


Figure 2: Morphology and life cycle of Gracilaria tikvahiae.

Virge Kask, Reprinted with permission from Yarish, C., Redmond, S., Kim, J., 2012. *Gracilaria* Culture Handbook for New England. Wrack Lines (report). Paper 72. http://digitalcommons.uconn.edu/wracklines/72.

Seaweed Cultivation Systems

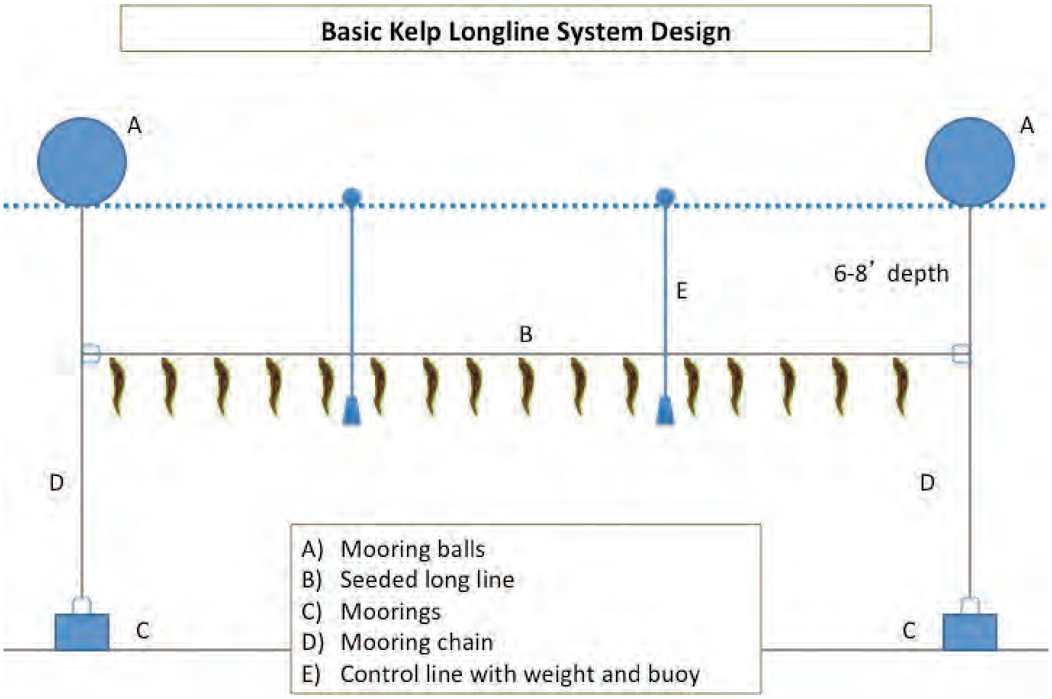


Figure 3. A basic design of the kelp growout system.
Sarah Redmond

Figure 4: Kelp growout system in use.
Holly Turner

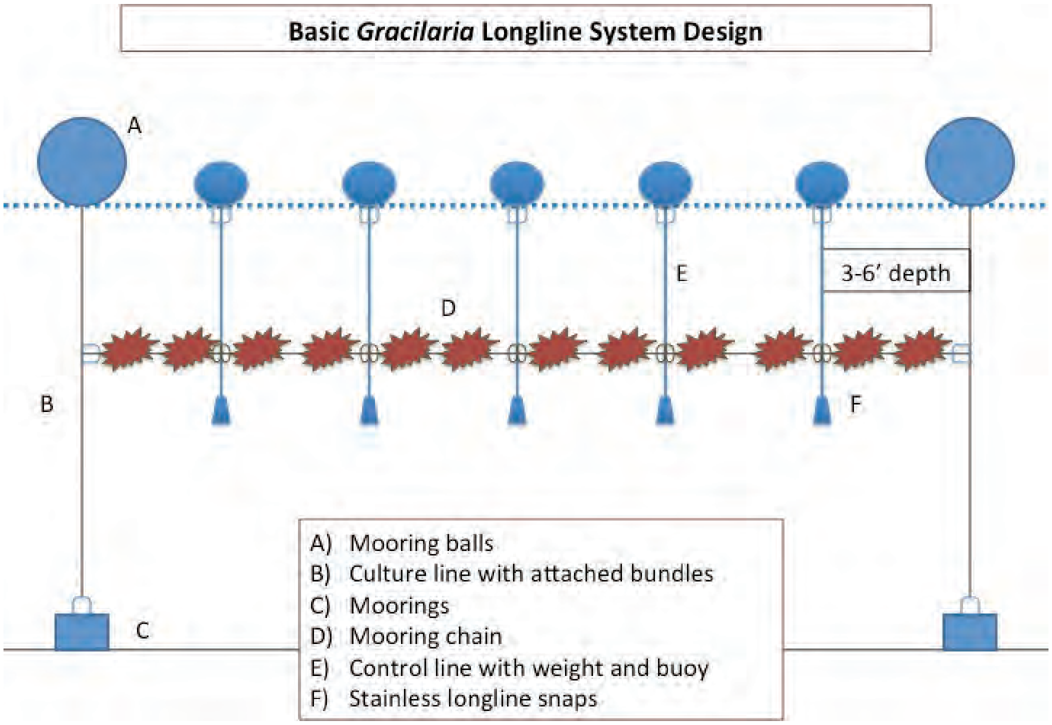


Figure 5. A basic design of the Gracilaria *growout system.*
Sarah Redmond

Figure 6: Gracilaria *growout system in use.*
Jang Kim

Potential Seaweed Production Hazards

Environmental Conditions
Biofouling Organisms
Predators
Diseases and Parasites
Invasive Species
Operational Procedures

Environmental Conditions

Marine macroalgal distribution, growth, and reproductive states are determined by a combination of environmental factors. Environmental conditions of light, day length, temperature, salinity, nutrient levels, turbidity, and water motion (current and wave exposure) are all important factors that vary throughout the year. Many species have seasonal requirements that dictate periods of planting, growth, and harvest, and it is important for the farmer to understand the role that key environmental parameters may play in overall production.

Potential hazards:

Temperature (extremes)
adverse weather

TEMPERATURE

Optimal environmental conditions differ between species, and these can be vastly different in New England, where we experience a wide range of seasonal conditions. Most seaweed species can tolerate a wide range of temperatures, but there is a narrower optimal temperature range that will allow for periods of rapid growth and overall health. Kelps (*Saccharina latissima*, *Alaria esculenta*, and *Laminaria digitata*) are cold-temperate to Arctic species, with a typical "winter" growth pattern, with rapid growth from late winter to late spring, and minimal to no growth in summer though this growth pattern may vary based on nutrient concentration in seawater. *Gracilaria*, on the other hand, are warm temperate species, with a "summer" growth season, from late spring to fall.

Temperature can vary greatly with depth and site, and the farmer should observe temperature throughout the year. There are several options for monitoring temperature. Small reusable underwater temperature data loggers can be placed throughout the water column and will constantly record temperature over time, allowing the farmer to create a record of temperature at a particular site for a range of depths and over seasons. Digital thermometers can also be used for on-site monitoring of temperature. A Niskin bottle can be used to sample water below the surface, or a homemade "water scoop" can be made from a section of PVC pipe attached to a line. Surface seawater temperature data can also be obtained from nearby buoys, found online at the National Oceanic and Atmospheric Administration National Data Buoy Center.

Temperature is one of the most important environmental factors determining the growth of seaweed. It is essential that the seaweed crop is cultivated within appropriate temperature ranges. Extreme or sub-optimal temperatures will stress the crop, leading to greater risk of disease or crop.

ADVERSE WEATHER

Extreme weather events can pose a hazard to the farm by causing damage or loss of farm infrastructure, damage or stress to the crop, or by impacting water quality. Farms should be sited in relatively protected areas and designed to withstand storms or high wind events. Good site selection, proper mooring tackle, and frequent mooring inspections should allow farms to withstand most storms.

Flexibility in the design of a farm can help minimize losses. An example of farm risk management would be the ability to bring *Gracilaria* longlines in for storage in holding tanks during major storms, or being able to adjust depth so that kelp lines could be lowered to avoid extreme wave action or runoff events.

Heavy rainfall could impact water quality at a farm site by creating a stressing reduction in salinity, increasing turbidity, or by causing waters to be closed for bacterial contamination. *Gracilaria* has a wide range of salinity tolerance but drastic changes can be stressful, while kelp have a lower tolerance to long periods of exposure to reduced salinities. Farms should be sited in areas with good mixing and away from major sources of freshwater runoff. Line depth can be increased to avoid surface variations.

An increase in turbidity as a result of freshwater runoff can affect underwater light levels and growth as well as damage fronds through abrasion or sedimentation. Turbidity, however, reduces phytoplankton production and *Gracilaria* could benefit from reduced competition. Kelp is generally found in areas of low turbidity, which allows them to grow at greater depths. Turbidity and light penetration of a site can be monitored with a Secchi disc or photometer, which allows the farmer to estimate optimal line depth. Turbidity can change throughout the year, and each site will be different.

Heavy rainfall and extreme storm events can affect water quality by allowing for an increase in harmful bacterial growth. State waters are continually monitored for water quality throughout the year for shellfish harvesting, so these guidelines should be utilized for any harvest of seaweeds for human consumption.

References and Further Reading:

Bird, C.J., McLachlan, J., 1986. The effect of salinity on distribution of species *Gracilaria* Grev. (Rhodophyta, Gigartinales): an experimental assessment. Botanica Marina. 29, 231-238.

Chapman, A.R.O., Craigie, J.S., 1977. Seasonal growth in *Laminaria longicruris*: relations with dissolved inorganic nutrients and internal reserves of nitrogen. Marine Biology. 40, 197–205.

Galimany, E., Rose, J.M., Dixon, M.S., Wikfors, G.H., 2013. Quantifying feeding behavior of ribbed mussels (*Geukensia demissa*) in two urban sites (Long Island Sound, USA) with different seston characteristics. Estuaries and Coasts. 36, 1265–1273.

Karsten, U., 2007. Salinity tolerance of arctic kelps from Spitsbergen. Phycological Research. 55, 257-262.

Lüning, K., 1990. Seaweeds - Their Environment, Biogeography, and Ecophysiology, In: Yarish, C., Kirkman, H. (Eds.), Edited Translation of the German language edition Meeresbotanik: Verbreitung, Okophysiologie und Nutzung der marinen Makroalgen by Klaus Lüning. John Wiley and Sons, Inc. New York.

Wiencke, C., Bischof, K., (Ed.) 2012. Seaweed biology: Novel insights into ecophysiology, ecology and utilization. Ecological Studies, 219. Springer-Verlag: Berlin, Heidelberg.

Yarish, C., Breeman, A.M., van den Hoek, C., 1984. Temperature, light and photoperiod responses of some Northeast American and west European endemic rhodophytes in relation to their geographic distribution. Helgoländer Meeresunters. 38, 273-304.

Yarish, C., Breeman, A.M., van den Hoek, C., 1986. Survival strategies and temperature responses of seaweeds belonging to different biogeographic distribution groups. Botanica Marina. 24, 215-230.

Table 1. Environmental parameters to consider for kelp (*Saccharina latissima*) and *Gracilaria tikvahiae* culture.

	Optimal Range		
Species	**Temperature**	**Salinity**	**References**
Saccharina latissima	10 - 15 °C	28 - 34 ppt	1, 2
Gracilaria tikvahiae	20 - 28 °C	25 - 33 ppt	3

Note: This table is for guidance only. Parameters will vary with species, strain, and size of organism, and may be dependent on other environmental factors.

References:

[1]Bird, C.J., McLachlan, J., 1986. The effect of salinity on distribution of species *Gracilaria* Grev. (Rhodophyta, Gigartinales): an experimental assessment. Botanic Marina. 29, 231-238.

[2]Egan, B., Yarish, C., 1990. Productivity and life history of *Laminaria longicruris* de la Pyl. At its southern limit in the Western Atlantic Ocean. Marine Ecology Progress Series. 76, 263-273.

[3]Karsten, U., 2007. Salinity tolerance of arctic kelps from Spitsbergen. Phycological Research. 55, 257-262.

Table 2. Stocking strategies for kelp (*Saccharina latissima*) and *Gracilaria tikvahiae* culture.

	Optimal Conditions		
Species	**Density**	**Depth**	**References**
Saccharina latissima	thin blades if crowded*	1 - 3 meters below surface	1, 2
Gracilaria tikvahiae	separate bundles 10 - 20 cm apart or more	0.5 - 1 meter below surface	3

*Crowding may result in reduced growth rate.

References:

[1]Flavin, K., Flavin, N., Flahive, B., 2013. Kelp Farming Manual: A Guide to the Processes, Techniques, and Equipment for Farming Kelp in New England Waters. Ocean Approved, Portland, Maine.

[2]Redmond, S., L. Green, L., Yarish, C., Kim, J., Neefus, C., 2014. New England Seaweed Culture Handbook. Nursery Systems. Connecticut Sea Grant. CTSG-14-01. 92 pp. URL: http://seagrant.uconn.edu/publications/aquaculture/handbook.pdf.

[3]FAO (Food and Agricultural Organization of the United Nations), 1990. Training manual on *Gracilaria* culture and seaweed processing in China. Training Manual 6. Online at http://www.fao.org/docrep/field/003/ab730e/AB730E00.htm#TOC.

Potential Seaweed Production Hazards

Environmental Conditions

Biofouling Organisms

Predators

Diseases and Parasites

Invasive Species

Operational Procedures

Biofouling Organisms

Most fouling organisms have a planktonic phase, which allows them to settle anywhere within the water column. When they do settle, cultured seaweed and supporting lines can become covered with tiny organisms that can significantly decrease product quality. Common epibionts include animals such as tunicates, hydroids, bryozoans, mussels, worms, amphipods, and plants known as epiphytes. Epiphytes may include both microalgal and macroalgal species, usually with seasonal patterns of abundance. Cultured seaweeds with high growth rates may allow fewer opportunities for epiphyte colonization.

Fouling fauna can be a major problem during the summer months, especially for *Gracilaria*. Colonization of filter feeding tunicates (*Molgula*) and hydroids (*Tubularia*) can inhibit growth by removing organic nutrients from the water column and by competing for space on lines. The presence and abundance of epibionts can affected by water temperature, as was observed in Long Island Sound in September 2011 when tunicate biomass was up to 60 gram per one meter of *Gracilaria* longline, but decreased when the water temperature dropped below 20˚C. Besides tunicates and hydroids, other types of animals can utilize *Gracilaria* lines as habitat. A total of eighteen species in fourteen families of fouling organisms were identified at *Gracilaria* open water farms in Long Island Sound and the Bronx River Estuary including *Ampithoe vailida*, *Ampithoe longimana*, *Corophium insidiosum*, *Jassa falcate*, *Unciola irrorata*, *Leptocherius pinguis*, *Caprella penatis*, *Idotea balthica*, *Carcinus maenas*, *Hemigrapsus sanguineus*, *Semibalanus balanoides*, *Balanus improvises*, *Paleomonetes* sp., *Molgula manhattensis*, *Tubularia* sp. *Eubranchus exiguous*, *Phascolopsis gouldii*, and *Nereis pelagica*.

There are several methods available to minimize fouling. These include controlling depth, stocking density, and out-planting or harvest time. Increasing stocking density and maximizing growth rates will allow the fronds to outcompete or exclude potential fouling organisms. Depth can be adjusted, either up or down, to minimize settlement or survival of particular organisms, which tend to be more abundant at a particular depth. By understanding the life cycle of the fouling organism and the specific nature of the fouling community at each farm, the farmer can time the planting and harvest of the crop to potentially avoid these "fouling windows".

A list and description of marine biofouling organisms are provided in Chapter 3, see 'biofouling'.

Figure 1. Biofouling on a seaweed farm site.
Sarah Redmond

References and Further Reading:

Ashton, G.V., Willis, K.J., Burrows, M.T., Cook, E.J., 2007. Environmental tolerance of *Caprella mutica*: Implications for its distribution as a marine non-native species. Marine Environmental Research. 64(3), 305-312.

Brady-Campbell, M.M., Campbell, D.B., Harlin, M.M., 1984. Productivity of kelp (*Laminaria* spp.) near the southern limit in the Northwestern Atlantic Ocean. Marine Ecology Progress Series. 18, 79-88.

Bullard, S.G., Whitlatch, R.B., 2004. A guide to the larval and juvenile stages of common Long Island Sound ascidians and bryozoans, Dept. of Marine Sciences, University of Connecticut, Groton CT 06340, U.S.A. CTSG-04-07.

Egan, B., Yarish, C., 1990. Productivity and life history of *Laminaria longicruris* de la Pyl. At its southern limit in the Western Atlantic Ocean. Marine Ecology Progress Series. 76, 263-273.

Fletcher, R.L., 1995. Epiphytism and fouling in *Gracilaria* cultivation: an overview. Journal of Applied Phycology. 7, 325-333.

Haska, C.L., Yarish, C., Kraemer, G.P., Blaschik, N., Whitlatch, R., Zhang, H., Lin, S., 2011. Assessing Bait Worm Packaging as a Potential Vector of Invasive Species into Long Island Sound, U.S.A. Biological. Invasions 14, 481-493.

Johnson, W., Allen, D. 2012. Zooplankton of the Atlantic and Gulf Coasts: A guide to their identification and ecology. 2nd Ed. John Hopkins University Press, Maryland.

Karlson, R.H., Osman, R.H., 2012. Species composition and geographic distribution of invertebrates in fouling communities along the east coast of the USA: a regional perspective. Marine Ecology Progress Series. 458, 255-268.

Karney, R., Grunden, R., Carman, M., Morris, J., Hoagland, P., 2008. Invasive tunicates at shellfish restoration and aquaculture sites on Martha's Vineyard, Massachusetts. Poster Presentation, 13th International Conference on Shellfish Restoration. Charleston, South Carolina.

Lambert, W.J., Phillip, S.L., Berman, J., 1992. Change in the structure of a New England (USA) kelp bed: the effects of an introduced species? Marine Ecology Progress Series. 88, 303-307.

Lindell, S., 2013. Sea Vegetable Farming in our Coastal Ponds: Producing local food while improving water quality. Presentation at the World Aquaculture Society meeting, Nashville, TN.

Lopez, G., Carey, D., Carlton, J., Cerato, R., Dam Guerrero, H., Digiovanni, C., Elphick, C., Frisk, M., Gobler, C., Hice, L., Howell, P., Jordan, A., Lin, S., Liu, S., Lonsdale, D., McEnroe, M., McKown, K., Mcmanus, G., Orson, R., Peterson, B., Pickerell, C., Rozsa, R., Shumway, S., Talmage, S., Taylor, G., Thomas, D., Van Patten, P., Vaudrey, J., Wikfors, G., Yarish, C., Zajac, R., 2013. Biology and Ecology of Long Island Sound, Chapter 6. In Long Island Sound: Prospects for the Urban Sea. Latimer, J.S., Tedesco, M., Swanson, R.L., Yarish, C., Stacey, P., Garza, C. (Eds.), Springer Publishers, New York.

MaClellan, J., 2005. Long Island Sound Invasive Species List. Compiled for the EPA Long Island Sound Study, 2005.

Molino, P.J., Wetherbee, R., 2008. The biology of biofouling diatoms and their role in the development of microbial slimes. Biofouling. 24(5), 365-379.

Nabivailo, Y., Titlyanov, E., 2006. Competitive relationships in natural and artificial algal Communities. Russian Journal of Marine Biology. 32(1), S21-S31.

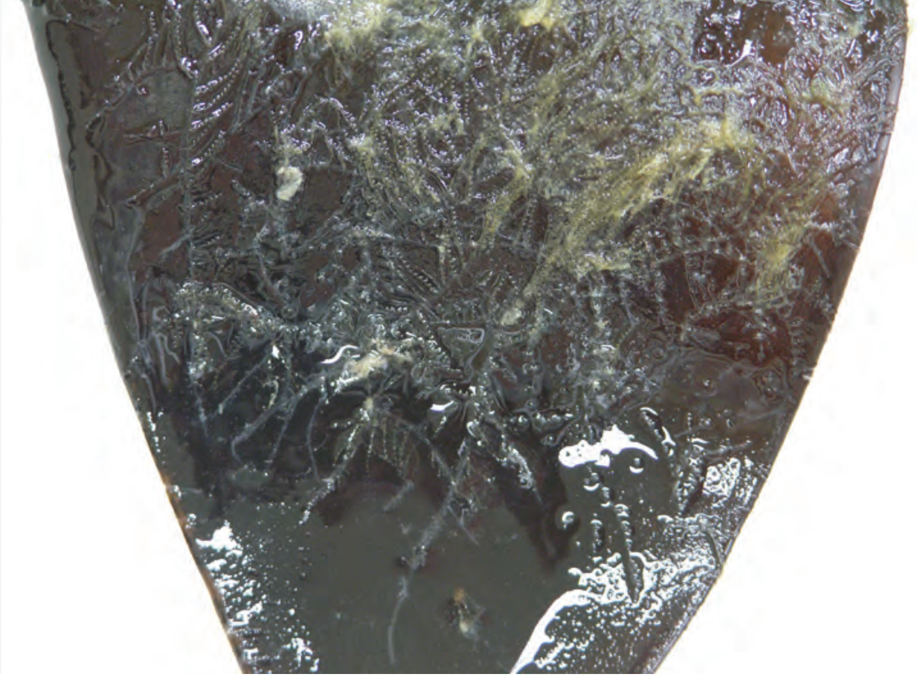


Figure 2. Various biofouling organisms bryozoans and hydroids seen here have the potential to cause rot in seaweeds and affect consumer acceptance.
Dr. E.K. Hwang, National Fisheries Research and Development Institute of Korea

National Park Service, 2013. *Tubularia crocea*, *Ectopleura crocea*. Marine Invasive Species. National Park Service factsheet.

Nettleton, J.C., Mathieson, A.C., Thornber, C., Neefus, C.D., Yarish, C., 2013. Introduction of *Gracilaria vermiculophylla* (Rhodophyta, Gracilariales) to New England, USA: estimated arrival times and current distribution. Rhodora. 115, 28-41.

Newell, C., et al. 2013. Maine Shellfish R&D Assistance to Mollusk Restoration to Increase Habitat and DO Around a Wastewater Treatment Plant. Final Programmatic Report to the National Fish and Wildlife Foundation, 13 May 2013.

Non-native Species Secretariat. Japanese skeleton shrimp factsheet. Online at: http://www.nonnativespecies.org/factsheet/factsheet.cfm?speciesId=647.

Park, C.S., Hwang, E.K., 2012. Seasonality of epiphytic development of the hydroid *Obelia geniculata* on cultivated *Saccharina japonica* (Laminariaceae, Phaeophyta) in Korea. Journal of Applied Phycology. 24(3), 433-439.

Pederson, J., Bullock, R., Carlton, J., Dijkstra, J., Dobroski, N., Dyrynda, P., Fisher, R., Harris, L., Hobbs, N., Lambert, G., Lazo-Wasem, E., Mathieson, A., Miglietta, M.P., Smith, J., Smith III, J., Tyrrell, M., 2003. Marine invaders in the Northeast: rapid assessment survey of non-native and native marine species of floating dock communities, August 2003. Massachusetts Institute of Technology, Sea Grant College Program, Publication No. 05-3.

Saier, B., Chapman, A.S., 2004. Crusts of the alien bryozoan Membranipora membranacea can negative impact spore output from native kelps (*Laminaria longicruris*). Botanica Marina. 47, 265-271.

Schlenk, C., 1983. Characteristics of *Laminaria saccharina* cultivated in Long Island Sound, New York. MS Thesis. Stony Brook University, New York.

Totti, C., Poulin, M., Romagnoli, T., Perrone, C., Pennesi, C., De Stefano, M., 2009. Epiphytic diatom communities on intertidal seaweeds from Iceland. Polar Biology. 32, 1681-1691.

Tiffany, M.A., 2011. Epizoic and Epiphytic Diatoms. In: Seckbach, J., Kociolek, J.P (Eds.), The Diatom World. 195-210.

Yarish, C., Egan, P., 1987. Biological Studies of *Laminaria longicruris* and its aquaculture potential in Long Island Sound. Halasi-Kun, G. (Ed.), Pollution and Water Resources.

University of Connecticut, 2001. Benthic Marine Algal Herbarium searchable database. http://www.algae.uconn.edu/index.html.

Potential Seaweed Production Hazards

Environmental Conditions
Biofouling Organisms
Predators
Diseases and Parasites
Invasive Species
Operational Procedures

Predators

Marine macroalgae provide both food and habitat for many organisms, including herbivorous predators. Herbivores cause physical damage through grazing, the effects of which are unsightly and can result in general weakening of fronds, increasing susceptibility to storm damage. The main herbivores in the Northeast are various types of snails and the green sea urchin, though various types of isopods and amphipods can be found at times grazing on seaweeds.

Herbivores settle directly from planktonic stages onto seaweed fronds and lines or can migrate up into fronds if the crop comes into contact with the sea floor. The farmer should plant or harvest around these “settlement windows” to avoid heavy infestation, and lines should be located at a sufficient depth to avoid touching the sea floor at lowest tide.

Potential hazards:

molluscs
echinoderms

Figure 1a: Lacuna vincta *snails and grazing damage on kelp blade*
Sarah Redmond

Figure 1b: L. vincta *and egg cases on kelp.*
Sarah Redmond

MOLLUSCS

Name: banded chink snail, northern Lacuna (*Lacuna vincta*)

Mode of action: *Lacuna vincta* (Montagu) is a small herbivorous snail that prefers to graze upon kelp species, but can also be found on other red, brown, and green seaweed species in intertidal and subtidal zones, usually in summer and fall. Their extensive grazing can impact blade quality and strength by creating full-thickness perforations or partial-thickness excavations in the blade. Other types of snails can occasionally also be found on kelp, including the common periwinkle (*Littorina littorea*), common tortoise limpet (*Testudinalia testudinalis*), and the lunar dovesnail (*Astyris lunata*), though *A. lunata* is believed to feed upon encrusting ascidians and bryozoans, not on seaweed fronds.

Species affected:

- kelp (*Saccharina latissima*)

Hazard Management:

- Culture kelps during the winter season and harvest in spring before settlement occurs

Figure 2: Young green sea urchin on kelp.
Sarah Redmond

ECHINODERMS

Name: green sea urchin (*Strongylocentrotus droebachiensis*)

Mode of action: Green sea urchins are voracious consumers of kelp and other seaweeds and can cause significant damage if they have access to seaweed lines. Larval urchins can settle onto seaweed from the plankton, or they can climb lines in contact with the sea floor. Green sea urchins are distributed from the Arctic to Cape Cod and are occasionally found south to New Jersey in intertidal and subtidal areas.

Species affected:

- kelp (*Saccharina latissima*)

Hazard Management:

- Keep lines from touching the sea floor.

Sources and Further Reading:

Brady, S., Scheibling, R., 2005. Repopulation of the shallow subtidal zone by green sea urchins (*Strongylocentrotus droebachiensis*) following mass mortality in Nova Scotia, Canada. Journal of the Marine Biological Association of the United Kingdom. 85, 1511-1517.

Chavanich, S., Harris, L., 2002. The influence of macroalgae on seasonal abundance and feeding preference of a subtidal snail, *Lacuna vincta* (Montagu) (littorinidae) in the Gulf of Maine. Journal of Molluscan Studies. 68, 73-78.

Johnson, C., Mann, K., 1986. The importance of plant defense abilities to the structure of subtidal seaweed communities: the kelp *Laminaria longicruris* de la Palaie survives grazing by the snails *Lacuna vincta* (Montagu) at high population densities. Journal of Experimental Marine Biology and Ecology. 97, 231-267.

Mann, K.H., 1973. Seaweeds: Their productivity and strategy for growth. Science. 182(4116), 975-981.

Osman, R.W., Whitlatch, R.B., 1995. Predation on early ontogenetic life stages and its effect on recruitment into a marine epifaunal community. Marine Ecology Progress Series. 117, 111-126.

Scheibling, R., Hatcher, B., 2001. The ecology of *Strongylocentrotus droebachiensis*. In: Lawrence, J.M. (Ed.), Edible sea urchins: Biology and ecology. New York: Elsevier Science. pp. 271-306.

Scheibling, R., Hennigar, A., Balch, T., 1999. Destructive grazing, epiphytism, and disease: the dynamics of sea urchin-kelp interactions in Nova Scotia. Canadian Journal of Fisheries and Aquatic Sciences. 56(12), 2300-2314.

Schlenk, C.G., 1983. Characteristics of *Laminaria saccharina* cultivated in Long Island Sound, New York. M.S. Thesis, State University of New York at Stony Brook.

Schmidt, A.L., Scheibling, R.E., 2006. A comparison of epifauna and epiphytes on native kelps (*Laminaria species*) and an invasive alga (*Codium fragile* ssp. *tomentosoides*) in Nova Scotia, Canada. Botanica Marina. 49, 315-330.

Steneck, R., Graham, M., Bourque, B., Corbett, D., Erlandson, J., Estes, J., Tegner, M., 2002. Kelp forest ecosystems: biodiversity, stability, resilience and future. Environmental Conservation. 29(4), 436-459.

Vadas, R., Steneck, R., 1995. Overfishing and inferences in kelp-sea urchin interactions. In: Skjodahl, H.R., Hopkins, C., Erikstad, K.E., Leinaas, H.P. (Eds.), Ecology of fjords and coastal waters. New York: Elsevier Science.

Potential Seaweed Production Hazards

Environmental Conditions
Biofouling Organisms
Predators
Diseases and Parasites
Invasive Species
Operational Procedures

Diseases and Parasites

Seaweed diseases can be caused by abiotic (chemical or physical) or biotic (pathogenic) factors, and usually include some combination of both. Environmental stress and disease is caused by poor conditions of light, nutrients, and temperature, while pathogenic factors can include bacterial, viral, or fungal infections. Many of the seaweed diseases have been reported from other countries, which may or may not be applicable for US farms. Most disease is caused by physiological stress, so the best management strategies will optimize culture conditions of light, nutrients, density, and temperature.

Potential hazards:

bacteria
endophytes
fungi

Figure 1. Rot disease in seaweed tissue.
Sarah Redmond

ROT DISEASE

Name: white, black, or green rot

Mode of action: Kelp diseases include different types of "rot" characterized by unhealthy discoloring and eventual decay of tissue. "Rots" are most commonly caused by poor environmental conditions, including high light levels, high temperatures, and low nutrients, combined with enhanced microbiological activity. Spot rot, or lesions, can occur in tissue, where fronds have been damaged and colonized by pathogenic bacteria or entire fronds can decay as a result of stress and bacterial growth. While these diseases are usually described for kelp, other types of seaweed can similarly be affected.

"White rot" disease is characterized by a yellowing of the frond, which will eventually fade to white and disappear as the frond decays. Deterioration will start in the apical portion of the blade and spread downward if conditions are not improved. It is caused by high light levels coupled with low available nitrogen. This condition is usually described for juveniles in the nursery stage, and is corrected by discarding the affected culture, thoroughly cleansing the system with bleach and freshwater, and starting again.

"Green rot" occurs on kelp lines in the sea, and is caused by insufficient illumination. Tissue will soften and turn green, signaling the onset of decay. A "black rot" disease has been described for giant kelp (Macrocystis pyrifera), caused by high temperatures. Other general signs of decay and disease in kelp tissues have been observed during the high temperature, low nutrient level conditions of summer months and as a result of excessive sedimentation covering blades.

Species affected:

- various species of kelp

Hazard Management:

- Increase depth of culture lines to reduce light intensity, provide optimal culture conditions (white rot).
- Raise kelp lines to increase illumination, or thin plants to relieve shading (green rot).
- Harvest before excessive summer temperatures, or lower lines to colder depths (black rot).

Figure 2: Twisted stipe disease observed in farmed kelp, Saccharina latissima, *in Maine.*
Sarah Redmond

TWISTING DISEASE (=TWISTED BLADE, TWISTED FROND, OR TWISTED STIPE DISEASE)

Mode of action: There are several types of disease that can cause kelp stipes and blades to develop abnormal twists and bends. Two different types of this disease have been reported from China: the twisted blade disease and the twisted frond disease. The twisted blade disease causes blades to twist and wrinkle, and is thought to be caused by exposure to excessive light or currents.

The twisted frond disease causes swollen stipes, twisted, roughened fronds, and thickened holdfasts. This disease tends to occur in areas with low current flow (less than 10cm/sec) with insufficient nutrient levels, and is caused by a mycoplasma-like organism.

This "twisting" phenomenon has been observed in kelp in Maine, but the actual cause is uncertain. In the north Atlantic, twisted stipes and deformations were caused by brown algal endophytes in Saccharina latissima.

Species affected:

- `kelp (*Saccharina latissima*)

Hazard Management:

- Increase depth of culture lines to reduce light intensity (twisted blade)
- Remove all infected individuals,
- Improve culture conditions

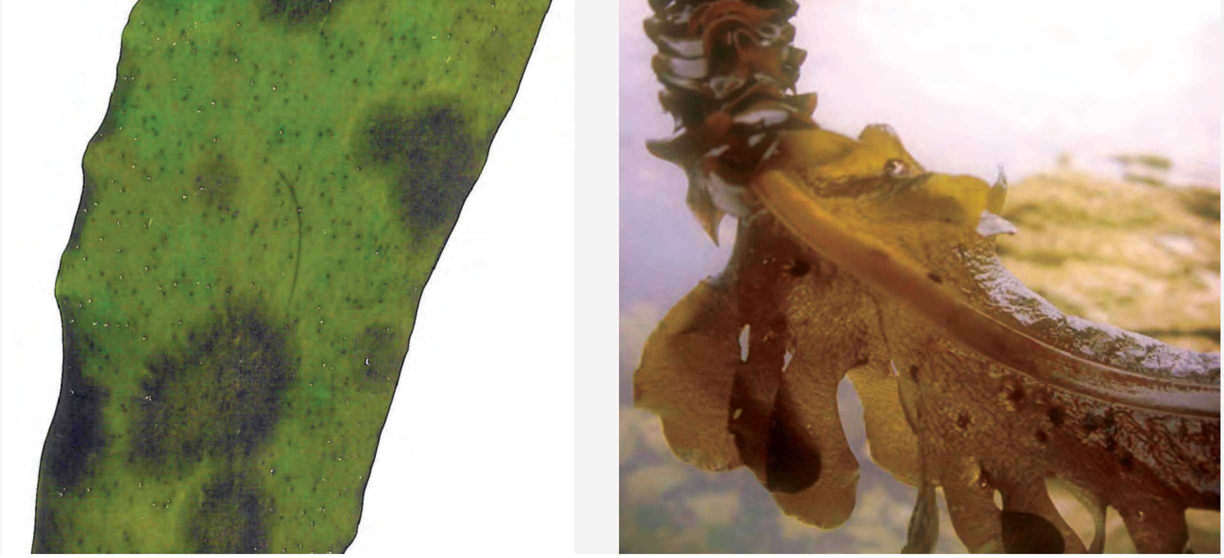


Figure 3a,b. Laminariocolax *spp. cause dark spots and deformation of tissue in various kelp species.*
Ignacio Manuel Bárbara Criado

ENDOPHYTES

Name: Brown algal endophytes, "dark spot disease", *Streblonema* disease caused by:

- *Laminariocolax aecidioides* (*previously Streblonema aecidioides*)
- *L. tomentosoides*
- *L. elsbetiae*

Mode of action: While there have been few studies on the prevalence or role of algal endophytic pathogens in marine algae, endophytes have been observed to cause disease in native Atlantic kelp populations. Endophytic brown algae of the ectocarpalean order can attach and penetrate healthy tissue, and are responsible for deformations and dark spots on the thallus and spiraling and warts on the stipes of kelps.

Species affected:

- kelp (*Saccharina latissima*)

Hazard Management:

- Remove and discard any affected individuals.
- Optimize culture conditions

Figure 4. Blisters on the surface of kelp, Saccharina latissima, *blade.*
Sarah Redmond

BLISTER DISEASE

Mode of action: The subtidal kelps are sensitive to sharp changes in salinity, and the tissue will blister if exposed to freshwater. These blisters affect product quality and can lead to tissue decay.

Species affected:

- kelp (*Saccharina latissima*)

Hazard Management:

- Place culture lines at a sufficient depth to avoid freshwater run-off.

MARINE FUNGI

Name: stipe blotch disease; species reported to infect Laminariales (kelp species) in the Atlantic include:

- Ascomycetes: *Phycomelaina laminariae*, causes "stipe blotch disease". Infects the meristematic tissue of the stipe of *Saccharina latissima*, forming black patches
- Oomycetes: *Petersenia* sp., *Pleotrachelus minutus*, *Labyrinthomyza sauvageaui*
- various unknown species

Mode of action: Marine fungi penetrate algal tissue and can result in overall reduced health, legions, necrotic tissue, blotchiness, blackened patches, and contortions. There are probably many different species that can cause infection, but only a few have been studied. Large *Porphyra* farms in Japan have experienced major losses from fungal infection, called "red wasting disease". A few species have been reported for the Atlantic kelps.

Species affected:

- various species of kelp

Hazard Management:

- Observe crop for signs of infections,
- Remove and discard all infected tissue

References and Further Reading:

Chen, J., 2004. *Laminaria japonica*. Cultured Aquatic Species Information Programme. In: FAO Fisheries and Aquaculture Department, Rome.

Correa, J.A., Craigie, J.S., 1991. Algal pathology. In: Seaweed Cellular Biotechnology, Physiology and Intensive Cultivation. Proceedings of a COST - 48 (Subgroup 1) Workshop. Reina, G.G., Pedersen, M. (Eds.), Universidad de las Palmas de Gran, Canaria, Feb. 8-17.

Craigie, J.S., Shacklock, P.F., 1995. Culture of Irish moss. In: Cold -Water Aquaculture in Atlantic Canada, 2nd edition. Boghen, A.D. (Ed.), Can. Inst. Res. Reg. Devel, University of Moncton, Moncton, New Brunswick.

Craigie, J S., Correa, J.A., 1996. Etiology of infectious diseases in cultivated *Chondrus crispus* (*Gigartinales, Rhodophyta*). In: Lindstrom, S.C., Chapman, D.J. (Eds.), Fifteenth International Seaweed Symposium, Hydrobiologia. 326/327, 97-104.

Goff, L.J., Glasgow, J.C., 1980. Pathogens of marine algae. Special Publication Number 7. Santa Cruz, Centre for Coastal Marine Studies, University of California.

Eggert, A., Peters, A.F., Kupper, F.C., 2010. The potential impact of climate change on endophyte infections in kelp sporophytes, in: Seckbach, J. (Ed.), Seaweeds and their role in globally changing environments. Springer, Berlin, pp. 141-154.

Ellertsdottir, E., Peters, A.F., 1997. High prevalence of infection by endophytic brown algae in populations of *Laminaria* spp. (Phaeophyceae). Marine Ecology Progress Series. 146, 135-143.

FAO (Food and Agricultural Organization of the United Nations), 1998. Chapter 9: Site Selection for Kelp (*Laminaria japonica* Aresch) in Culture of Kelp (*Laminaria japonica*) in China. Fisheries and Aquaculture Department, FAO.

Kawai, H., Tokuyama, M., 1995. Laminarionema elsbetiae gen. et sp. Nov. (*Ectocarpales, Phaeophyceae*), and new endophyte in *Laminaria sporophytes*. Phycological Research. 43, 185-190.

Neill, K., Heesch, S., Nelson, W., 2008. Diseases, pathogens and parasites of *Undaria pinnatifida*. Biosecurity New Zealand, Ministry of Agriculture and Forestry, Technical Paper No. 2009/44.

Neushul, M. Harger, W.W., 1987. Near shore kelp cultivation, yield and genetics. In: Bird KT & PH Benson (Eds.), Seaweed Cultivation for Renewable Resources. Elsevier, Amsterdam, pp. 69-93.

North, J., 1987. Biology of the *Macrocystis* resource in North America, In: Doty, M.S., Caddy, J.F., Santelices, B. (Eds.), Case studies of seven commercial seaweed resources. FAO Fisheries, Technical Paper No. 281.

Porter, D., 1986. Mycoses of marine organisms: an overview of pathogenic fungi. In: Moss, S.T. (Ed.), Biology of Marine Fungi. University Press, Cambridge. pp. 141-154.

Potential Seaweed Production Hazards

Environmental Conditions
Biofouling Organisms
Predators
Diseases
Invasive Species
Operational Procedures

Invasive Species

Non-native and invasive species of non-target seaweeds have been increasing in frequency with climate change. Some invasive species have a similar appearance to target seaweed species and present a risk to aquaculture operations and the natural environment. These species are included in Chapter 3 under “biofouling”. Additional information on non-native and invasive species can be found online at the website of the Northeast Aquatic Nuisance Species Panel, and references can be found on the next page.

Potential hazards:

Codium fragile
Gracilaria vermiculophylla
Grateloupia turuturu
Heterosiphonia japonica

References and Further Reading:

Gavio, B., Fredericq, S., 2002. *Grateloupia turuturu* (Halymeniaceae, Rhodophyta) is the correct name of the non-native species in the Atlantic known as *Grateloupia doryphora*. European Journal of Phycology. 37, 349-359.

Green, L.A., Mathieson, A.C., Neefus, C.D., Traggis, H.M., Dawes, C.J., 2012. Southern expansion of the brown alga *Colpomenia peregrina* Sauvageau (Scytosiphonales) in the Northwest Atlantic Ocean. Botanica Marina. 55(6), 643–647.

Guiry, M., Guiry, M., 2013. AlgaeBase. World-wide electronic publication, National University of Ireland, Galway. http://www.algaebase.org.

Mathieson, A.C., Dawes, C.J., Pederson, J., Gladych, R.A., Carlton, J.T., 2007. The Asian red seaweed *Grateloupia turuturu* (Rhodophyta) invades the Gulf of Maine. Biological Invasions. 10(7), pp. 985-988.

Nettleton, J.C., Mathieson, A.C., Thornber, C., Neefus, C.D., Yarish, C., 2013. Introduction of *Gracilaria vermiculophylla* (Rhodophyta, Gracilariales) to New England, USA: Estimated arrival times and current distribution. Rhodora. 115(961), 28-41.

Newton, C., Bracken, M.E.S., McConville, M., Rodrigue, K., Thornber, C.S., 2013. Invasion of the red seaweed *Heterosiphonia japonica* spans biogeographic provinces in the western North Atlantic Ocean. PLoS ONE. 8(4), e6226.

Provan, J., Murphy, S., Maggs, C.A., 2005. Tracking the invasive history of the green alga *Codium fragile* ssp. tomentosoides. Molecular Ecology. 14, 189-194.

Scheibling, R.E., Gagnon, P., 2006. Competitive interactions between the invasive green alga *Codium fragile* ssp. tomentosoides and native canopy-forming seaweeds in Nova Scotia. Marine Ecology Progress Series. 325, 1-14.

Schneider, C.W., 2010. Report of a new invasive alga in the Atlantic United States *Heterosiphonia japonica* in Rhode Island. Journal of Phycology. 46(4), 653-657.

Thomsen, M.S., Gurgel, C.F.D., Fredericq, S., McGlathery, K.J., 2005. *Gracilaria vermiculophylla* (Rhodophyta, Gracilariales) in Hog Island Bay, Virginia: A cryptic alien and invasive macroalga and taxonomic correction. Journal of Phycology. 42, 139-141.

Thomsen, M.S., McGlathery, K.J., Schwarzschild, A., Silliman, B.R., 2009. Distribution and ecological role of the non-native macroalga *Gracilaria vermiculophylla* in Virginia salt marshes. Biological Invasions. 11, 2303–2316.

Van Patten, M., 2006. Beware of the Red Menace! *Grateloupia* is Invading Long Island Sound. Wrack Lines Magazine. Connecticut Sea Grant Publication. 6(2), Fall/ Winter Issue.

Voyer, C., Hadlock-Seeley, R., Fact Sheet on *Heterosiphonia japonica*. Gulf of Maine Research Institute. Last accessed on August 16, 2013 at: http://vitalsignsme.org/sites/default/files/content/ci_heterosiponia_japonica_112712.pdf.

Yokoya, N.S., Kakita, H., Obika, H., Kitamura, T., 1999. Effects of environmental factors and plant growth regulators on growth of the red alga *Gracilaria vermiculophylla* from Shikoku Island, Japan. Proceedings of the International Seaweed Symposium. 16, 339-347.

Potential Seaweed Production Hazards

Environmental Conditions
Biofouling Organisms
Predators
Diseases
Invasive Species
Operational Procedures

Operational Procedures

The operational procedures of a farm can greatly impact its productivity. Good management practices influence a crop's growth, survival, and marketability. It is important for a farmer to identify, address, and modify (if and when necessary) all operational procedures affecting overall productivity.

Operational procedures will vary by farm setup but the following general guidelines should be considered:

- Record keeping is extremely important on many levels including: Log of activities and observations, environmental conditions, production schedules, business planning, regulatory compliance, and trouble shooting
- Establish standard operating procedures (SOPs) for every practice or routine to ensure streamlined and efficient operations.

Specific hazards:

Maintenance
Handling and transport

MAINTENANCE

The longline system should be checked on a regular basis to ensure that there is no loss or damage from storms, vandalism, or passing boats. Kelp (*Saccharina latissima*) can develop hollow stipes, which will float the line at the surface. If this occurs, weights must be added to move the fronds away from the surface or tissue will be damaged through desiccation and UV exposure.

Hazard Management:

- For kelp, thin blades if lines get too crowded.
- For *Gracilaria*, increase the distance between bundles if lines get too crowded or harvest excess growth by trimming the outer portion of the fronds allowing fronds to re-grow.
- Adjust depth of lines when necessary.

HANDLING AND TRANSPORT

Handling and transport of seed-string from a nursery setting to an open-water cultivation system must be conducted with care and preparation. Out-planting should be conducted during the optimal growing season to ensure success of the crop. Juveniles, or "seed", should be shaded from full sunlight and protected from extreme temperatures.

Hazard Management:

- Kelp seed-string is comprised of juvenile kelp and generally out-planted when plants are 1-2 mm in length.
- Seed spools should be handled with care to protect the small blades from stress or exposure when transferring from the lab to farm site.
- Spools can be moved in small, sealed containers in 10°C seawater, minimizing exposure and movement.
- Blades are sensitive to exposure and should be protected from sunlight, freezing, or drying during transplanting.
- *Gracilaria* is a stress tolerant species, but extreme changes in light, salinity or temperature can stress a culture unit and reduce growth and production.

Sources and Further Reading:

Flavin, K., Flavin, N., Flahive, B. 2013. Kelp Farming Manual: A Guide to the Processes, Techniques, and Equipment for Farming Kelp in New England Waters. Ocean Approved, Portland, Maine.

Pereira, R., Yarish, C., 2010. The role of *Porphyra* in sustainable culture systems: Physiology and Applications. In: Israel, A., Einav, R. (Eds.), Role of Seaweeds in a Globally Changing Environment. Springer. pp. 339-354.

Redmond, S., Green, L., Yarish, C., Kim, J., Neefus, C., 2014. New England Seaweed Culture Handbook Nursery Systems. Connecticut Sea Grant Publication No. CTSG-14-01.

Sahoo, D., Yarish, C., 2005. Mariculture of seaweeds. In: Andersen, R. (Ed.), Phycological Methods: Algal Culturing Techniques. Ch. 15. Academic Press, Elsevier. Pp. 219-237.

Yarish, C., Redmond, S., Kim, J., 2012. *Gracilaria* Culture Handbook for New England. Wrack Lines (report). Paper 72. http://digitalcommons.uconn.edu/wracklines/72.

APPENDIX 1.

Instructions for Aquaculture Hazard Analysis

The following instructions are designed to help the farmer work through the Northeast Aquaculture Management Guide to identify the potential hazards associated with the operation, and to develop a plan to manage for them. The hazard analysis, when completed, will serve as the basis for a business risk management plan.

Before beginning the hazard analysis, the farmer should review the introduction to this manual, and the relevant chapters and appendices that identify hazards and information associated with production of their product. It is important that the hazard analysis process and management plan accurately reflects the business' products and procedures as is practical. Furthermore, it is recommended that the plan be re-evaluated annually to ensure that it continues to be representative of actual on-farm procedures. A large component in any plan to identify and mitigate aquaculture production related hazards is to ensure that correct information is collected at the appropriate times.

To complete the hazard analysis process, use the templates provided in Appendix 2, and follow these steps:

Step 1. Complete the Business Description Form:

Enter business contact information and describe the aquaculture product.

Step 2. Create a Production Flow Chart:

Draw a schematic of the production process.

Step 3. Complete the Hazard Analysis and Production Risk Management Forms

Column 1. List each step of the production process.

Column 2. Select the potential hazards that may occur at each production step.

Column 3. Explain how each particular hazard affect production.

Column 4. Identify management strategies for each hazard.

Column 5. Describe the monitoring plan.

- i. Identify what will be measured/monitored.
- ii. Identify how the parameter will be measured/monitored.
- iii. Identify when the parameter will be measured/monitored.
- iv. Identify who will do the work.

Column 6. Describe what corrective action will be taken if critical limits are exceeded.

Column 7. Describe what will be done to verify that the process is working.

Column 8. List specific records that will be kept to facilitate decision-making.

APPENDIX 2.

Aquaculture Production Hazard Analysis Forms

Form 1. Business and Product Description

INSTRUCTIONS: Complete this form with information about the business operator and the final cultured product.

BUSINESS INFORMATION:

Business Name: ______________________ Contact Person: ______________________

Business Address: ______________________

City ______________________ State ______________ Zip: ______________

Phone ______________________ Email: ______________________

PRODUCT INFORMATION:

Market/scientific name of product (e.g. eastern oyster, *Crassostrea virginica*): ______________________

Describe Finished Product: (e.g. live, whole, unshucked) ______________________

Source, Type and Size of Seedstock/Fry/Fingerlings: (e.g. Joe's Oyster Hatchery, 8mm seed) ______________________

Production Method: (e.g. nursery and growout culture in ADPI bags) ______________________

Intended Use and Consumer: e.g. (human consumption, direct sales to consumer at farm markets) ______________________

Form 2. Production Flow Chart

INSTRUCTIONS: Create a schematic of the operation with the steps listed in sequential order. Briefly describe the individual steps. Always begin with source water management and broodstock (or seedstock) source management. See examples at the end of this appendix.

Form 3. Hazard Analysis Worksheet

INSTRUCTIONS: Complete this form. Make additional copies if necessary (e.g. if there are 9 business steps, you will need 3 sheets

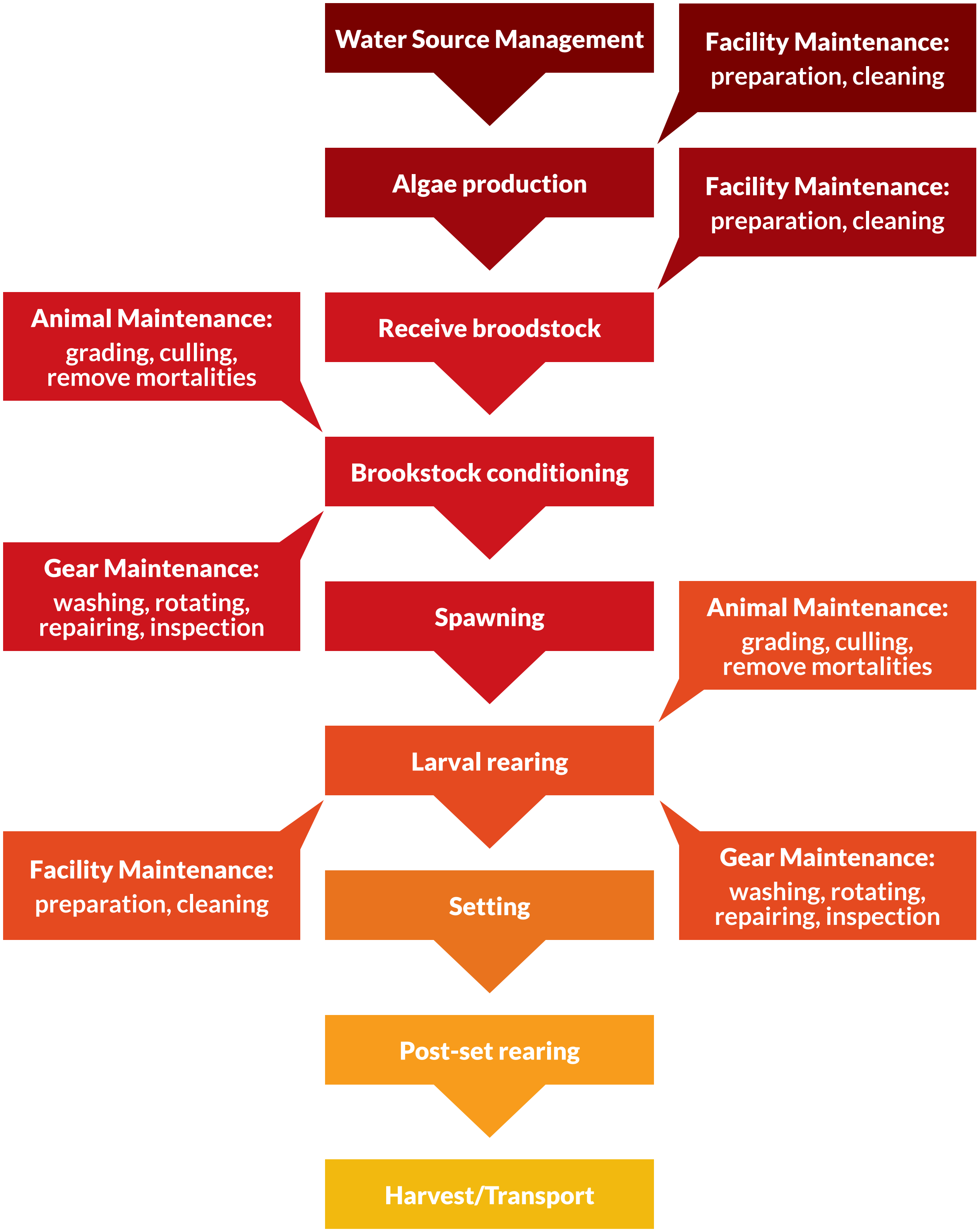
(1) List production steps individually from the Production Flow Chart:	**(2) Select the potential hazards at this step**						**(3) Explain how these hazards affect production (see MANUAL)**	**(4) List appropriate hazard management strategies (see MANUAL)**
	Operational	**Invasives**	**Diseases**	**Predators**	**Biofouling**	**Environmental**		
Step:								
Step:								
Step:								

Form 4. Production Risk Management Plan

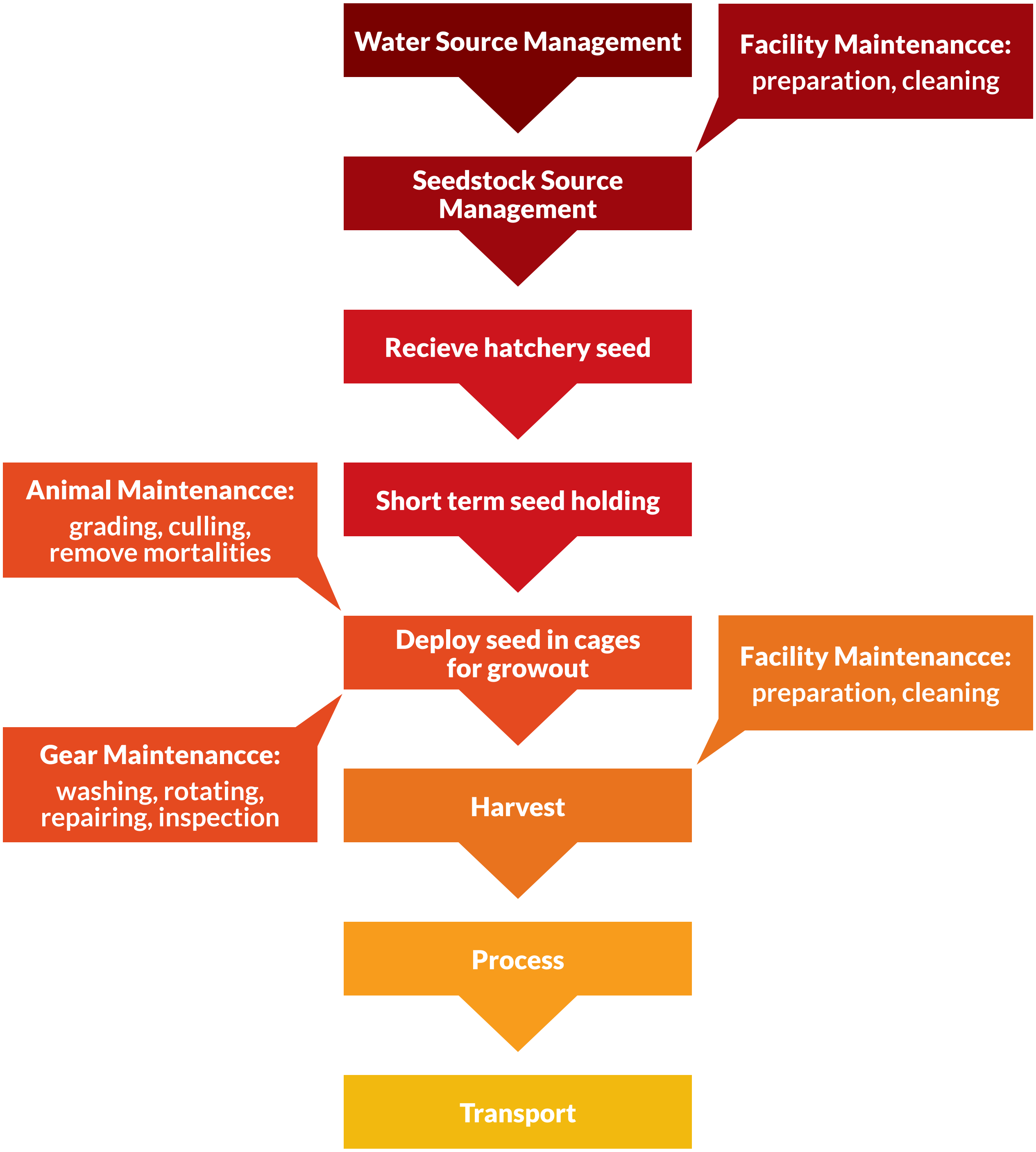
INSTRUCTIONS: Complete this form. Make additional copies if necessary (e.g. if there are 9 business steps, you will need 3 sheets).

(5) Monitoring					**(6) Corrective Action** **(What is done when item monitored is not as expected; e.g. adjust temperature, call aquatic animal health professional)**	**(7) Verification** **(What is done to ensure that the corrective action has resulted in return to normal)**	**(8) Record Type** **(e.g. temperature logbook)**
List production Step	**What**	**How**	**Frequency**	**Who**			
Step:							
Step:							
Step:							

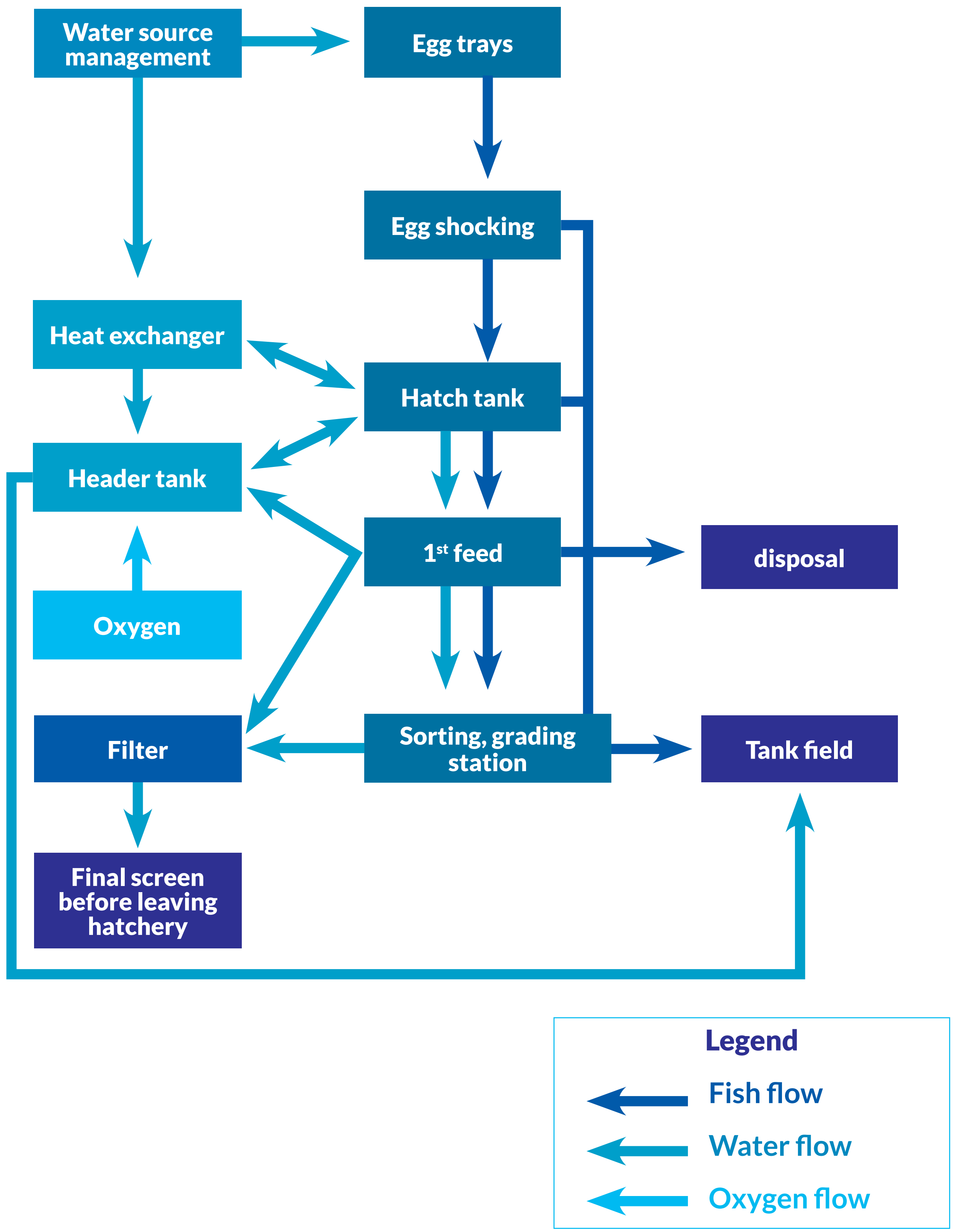
Example 1. Shellfish Hatchery Flow Chart



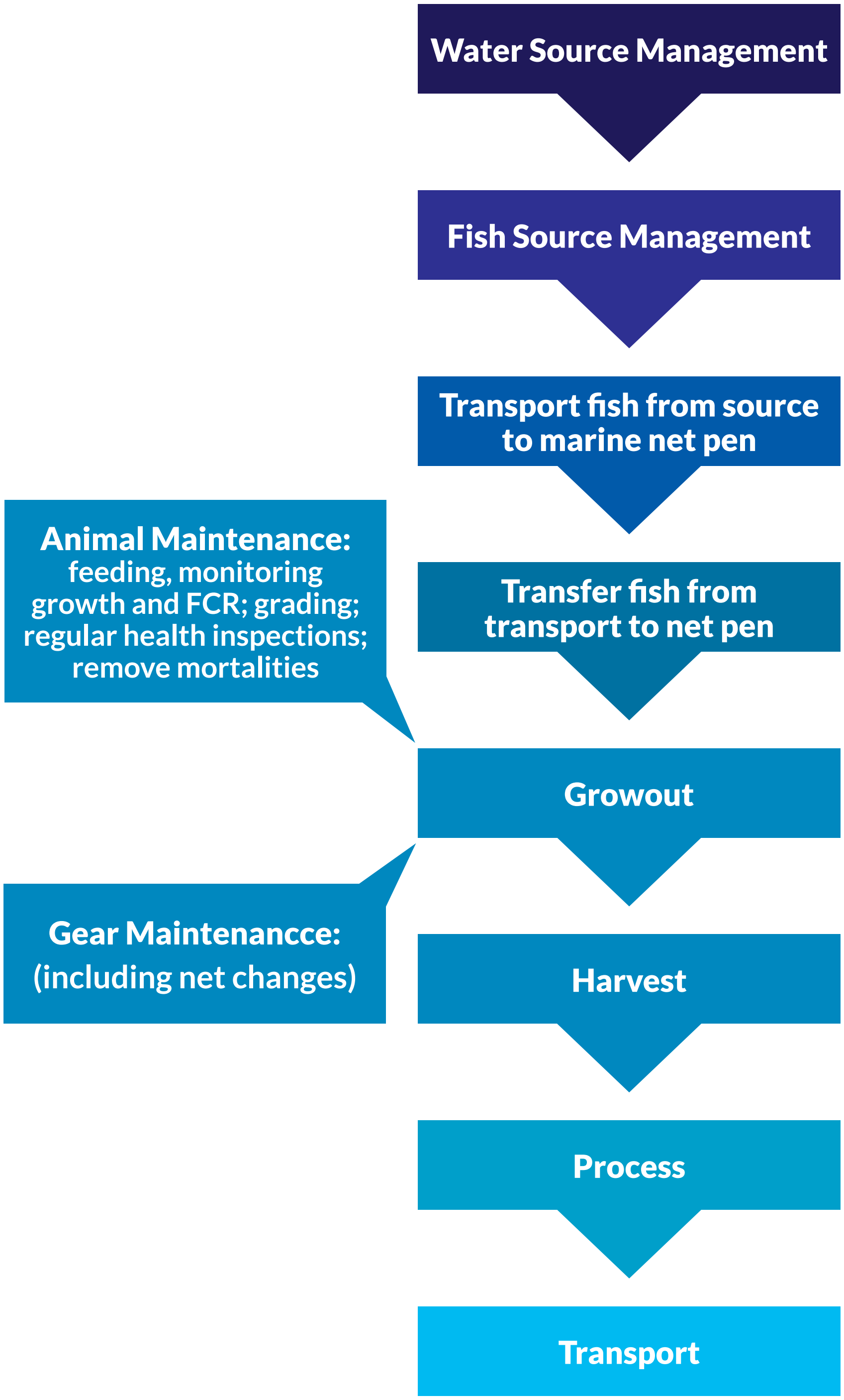
Example 2. Shellfish Growout Flow Chart



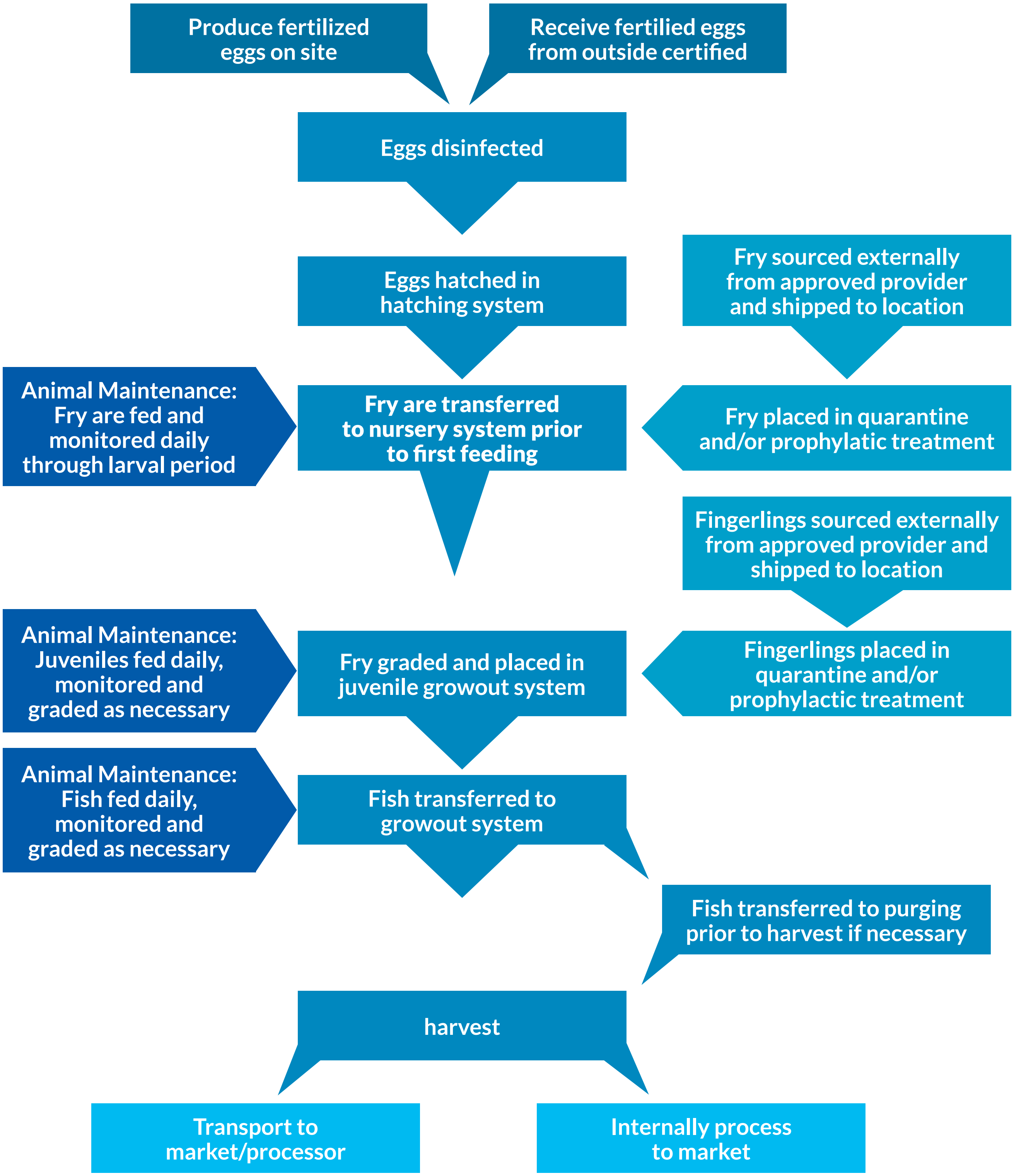
Example 3. Finfish Hatchery Flow Chart



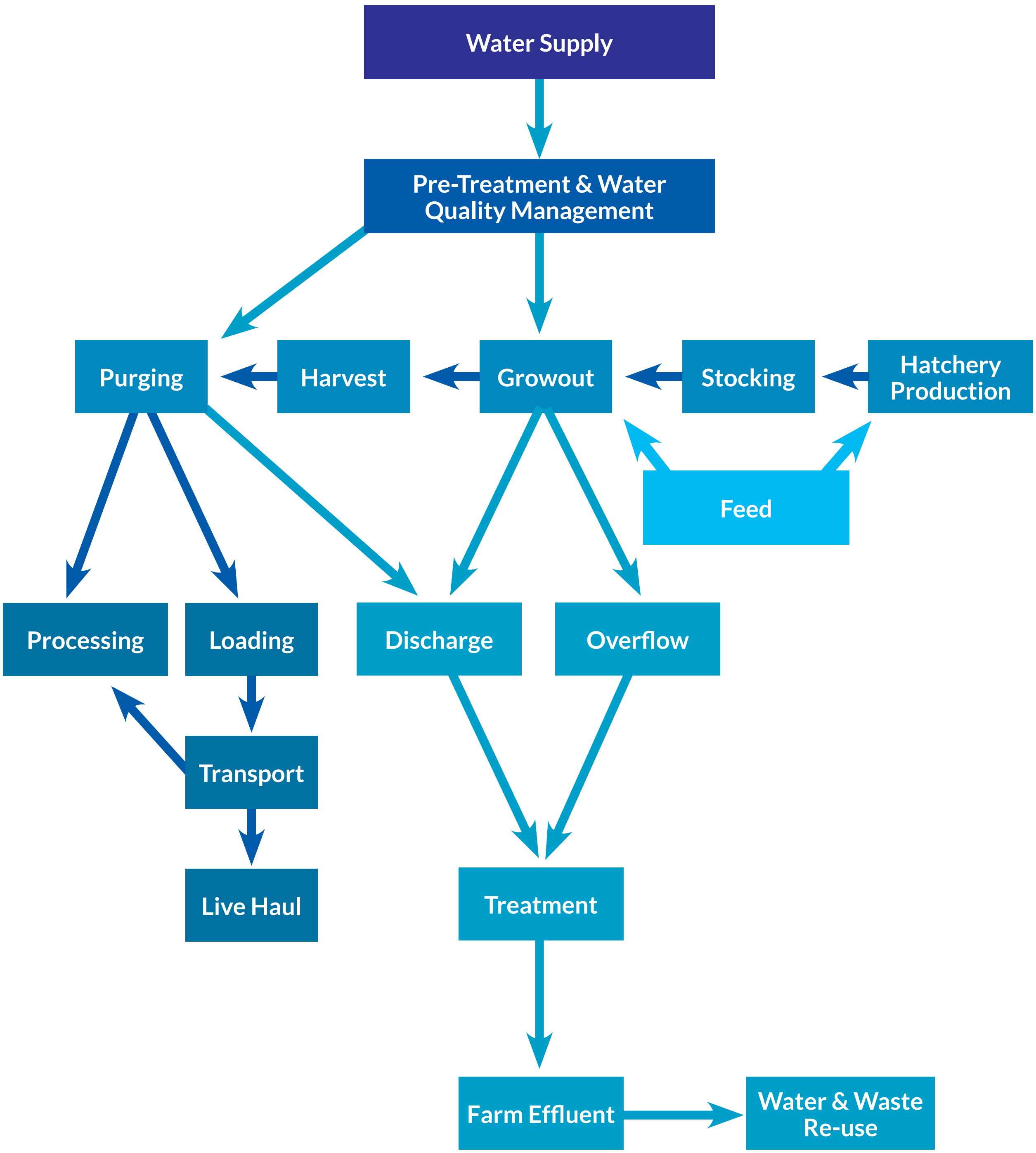
Example 4. Marine Finfish Net Pen Flow Chart



Example 5. Recirculating Aquaculture System Flow Chart



Example 5. Finfish Pond Culture Flow Chart



APPENDIX 3.

Aquatic Animal Health Professional/ State Aquaculture Coordinator Contact List

Connecticut

State Shellfish Pathologist (SHELLFISH)
Connecticut Department of Agriculture, Bureau of Aquaculture
P.O. Box 97, Milford, CT 06460
Contact: Inke Sunila
(203) 874-0696
Email: dept.agric@snet.net

Connecticut Veterinary Diagnostic Laboratory (FINFISH)
University of Connecticut, 61 North Eagleville Rd., U-3089, Storrs, CT 06269
Contact: Sal Frasca
(860) 486-3738
Email: CVMDL@uconn.edu

Fish Health Laboratory (FINFISH)
Connecticut Department of Energy and Environmental Protection
Contact: Richard Van Nostrand
(860) 673-3695
Email: richard.vannostrand@ct.gov

Delaware

(none known at this time)

Maine

Fish Vet Group (FINFISH)
350 Commercial Street, Portland, ME 04101-5597
Contact: Jason Collins
(207) 699-5902
Email: jason.collins@fishvetgroup.com

Kennebec River Biosciences (SHELLFISH, FINFISH)
41 Main Street, Richmond, ME 04357
Contact: Cem Giray
(207) 737-2637 x207
Email: cgiray@kennebecbio.com

University of Maine Animal Diagnostic Laboratory (FINFISH)
5735 Hitchner Hall
Orono, Maine 04469-5735
Contact: Deborah A. Bouchard
(207) 581-2767
Email: deborah.bouchard@maine.edu

Maine Department of Inland Fisheries and Wildlife
Maine Department of Marine Resources
Burns Road, Augusta, Maine 04330
Contact: Colby Wells, DVM, Fish Pathologist
(207) 287-2813
Email: colby.wells@maine.gov

Maryland

(none known at this time)

Massachusetts

(none known at this time)

New Hampshire

New Hampshire Veterinary Diagnostic Laboratory (FINFISH)
University of New Hampshire
129 Main Street, Durham, NH 03824
Contact: Inga F. Sidor
(603) 862-2726 or (603) 862-2743
Email: inga.sidor@unh.edu

New Jersey

Haskin Shellfish Research Laboratory (SHELLFISH)
6959 Miller Avenue, Port Norris, NJ 08349
Contact: Lisa Calvo
(856) 785-0074 or cell: (609) 440-4560
Email: calvo@hsrl.rutgers.edu

New Jersey Public Health, Environmental and Agriculture Laboratory (FINFISH)
3 Schwartzkopf Drive
Ewing, NJ 08628
Contact: Amar Patil
(609) 671-6400 or (609) 406-6999
Email: amar.patil@ag.state.nj.us

New York

School of Marine and Atmospheric Sciences (SHELLFISH)
Stony Brook University
Stony Brook, NY 11794-5000
Contact: Bassem Allam
(631) 632 8745
Email: bassem.allam@stonybrook.edu

Fish Disease Diagnostic Laboratory (FINFISH)
Cornell University
Ithaca, NY 14853
Contact: Paul R. Bowser
(607) 253-4029 or lab: (607) 253-4028
Email: prb4@cornell.edu

Pennsylvania

Animal Diagnostic Laboratory (FINFISH)
Penn State University
University Park, PA 16802
Contact: not available at this time
(814) 863-0837
Email: adlhelp@psu.edu

Rhode Island

Aquatic Diagnostic Laboratory (SHELLFISH, FINFISH)
Roger Williams University
One Old Ferry Road, Bristol, RI 02809
Contact: Roxanna Smolowitz
(401) 254-3299 or cell: (508) 566-0379
Email: rsmolowitz@rwu.edu

Vermont

(none known at this time)

Virginia

Shellfish Pathology Laboratory (SHELLFISH)
Virginia Institute of Marine Science
1375 Greate Road, P.O. Box 1346, Gloucester Point, VA 23062
Contact: Ryan Carnegie
(804) 684-7713
Email: carnegie@vims.edu

Washington, D.C.

(none known at this time)

West Virginia

(none known at this time)

USDA Agriculture Research Service

National Cold Water Marine Aquaculture Center
120 Flagg Road, Kingston, RI 02881
Contact: Dina Proestou, Research Geneticist
(207) 422-2700
Email: dina.proestou@ars.usda.gov

Directory of Aquatic Veterinarians and Disease Diagnostic Laboratories

http://www.aquavetmed.info/

Directory of State Aquaculture Coordinators

http://www.nasac.net

APPENDIX 4.

Aquaculture Extension Professional Contact List

Connecticut

Anoushka Concepcion
Connecticut Sea Grant/UCONN Extension
University of Connecticut
1080 Shennecossett Road
Groton, CT 06340-6048
Tel. (860) 405-9105
Fax (860) 405-9109
Email: anoushka.concepcion@uconn.edu

Tessa Getchis
Connecticut Sea Grant/UCONN Extension
University of Connecticut
1080 Shennecossett Road
Groton, CT 06340-6048
Tel. (860) 405-9104
Fax (860) 405-9109
Email: tessa.getchis@uconn.edu

Dr. Robert Pomeroy
Connecticut Sea Grant
University of Connecticut
1080 Shennecossett Road
Groton, CT 06340-6048
Tel. (860) 405-9215
Fax (860) 405-9109
Email: robert.pomeroy@uconn.edu

Delaware

John W. Ewart
Aquaculture & Fisheries Specialist
Delaware Aquaculture Resource Center/Delaware Sea Grant Marine Advisory Service
College of Earth, Ocean, and Environment (CEOE)
University of Delaware
700 Pilottown Road
Lewes, DE 19958
Tel. (302) 645-4060
Fax (302) 645-4213
Email: ewart@udel.edu

Doris T. Hicks
Seafood Technology Specialist
Delaware Sea Grant Marine Advisory Service
College of Earth, Ocean, and Environment (CEOE)
University of Delaware
700 Pilottown Road
Lewes, DE 19958
Tel. (302) 645-4297
Fax (302) 645-4213
Email: dhicks@udel.edu

Dr. Dennis McIntosh
Assistant Research Professor and Extension Specialist - Aquaculture
Department of Agriculture and Natural Resources
Ag Annex Room 126
Delaware State University
1200 N. DuPont Highway
Dover, DE 19938
Tel. (302) 857-6456
Fax (302) 857-6402
Email: dmcintosh@desu.edu

Maine

Chris Bartlett
Marine Extension Associate
Maine Sea Grant Program
Marine Technology Center, City of Eastport
16 Deep Cove Road
Eastport, ME 04631
Tel. (207) 853-2518
Fax (207) 853-0940
Email: cbartlett@maine.edu

Deborah Bouchard
Laboratory Manager – Research Coordinator
University of Maine Animal Health Laboratory
Aquaculture Research Institute
348 Hitchner Hall
Orono, ME 04469
Tel. (207) 581-2767
Fax (207) 581-4430
Email: deborah.bouchard@maine.edu

Dana Morse
Marine Extension Associate
Maine Sea Grant Program/University of Maine Cooperative Extension
Darling Marine Center
193 Clark's Cove Road
Walpole, ME 04573
Tel. (207) 563-8186
Fax (207) 563-3119
Email: dana.morse@maine.edu

Mike Pietrak
Aquaculture Research Institute
179 Hitchner Hall
Orono, ME 04469
Tel. (207) 581-4344
Email: michael.pietrak@umit.maine.edu

Sarah Redmond
Marine Extension Associate
Maine Sea Grant College Program and UMaine Cooperative Extension
Center for Cooperative Aquaculture Research
33 Salmon Farm Road
Franklin, ME 04634
Tel. (207) 422-6289
Cell (207) 841-3221
Email: sarah.redmond@maine.edu

Maryland

Dr. Reginal M. Harrell
Professor of Fisheries and Wildlife Science and Extension Specialist
1449 AnSc/Ag Eng Bldg (Bldg 142)
University of Maryland
College Park, MD 20742
Tel. (301) 405-4708
and
Wye Research & Education Center
P.O. Box 169
124 Wye Narrows Drive
Queenstown, MD 21658
Tel. (410) 827-8056 x140
Fax (410) 827-9039
Email: rharrell@umd.edu

Dr. Andrew M. Lazur, Director
Maryland Sea Grant Extension
1212 Symons Hall
University of Maryland
College Park, MD 20742
Tel. (301) 405-7992
Fax (301) 314-9091
Email: lazur@umd.edu

Dr. Donald Meritt
Shellfish Aquaculture Specialist
UMCES Horn Point Environmental Laboratory
P.O. Box 775
Cambridge, MD 21613
Tel. (410) 221-8475
Fax (410) 221-8490
Email: meritt@umces.edu

Matt Parker
Aquaculture Business Specialist
University of Maryland Sea Grant Extension
Anne Arundel County Office
97 Dairy Lane
Gambrills, MD 21054
Tel. (410) 222-6759
Fax (410) 222-6747
Email: mparke11@umd.edu

Tom Rippen
Sea Grant Seafood Technology Specialist
University of Maryland Eastern Shore
30921 Martin Court
Princess Anne, MD 21853
Tel. (410) 651-6636
Fax (410) 651-7656
Email: terippen@mail.umes.edu

Dr. Daniel E. Terlizzi
Extension Specialist, Water Quality
Institute of Marine and Environmental Technology
701 E. Pratt Street
Baltimore, MD 21202
Tel. (410) 234-8837
Fax (410) 234-8896
Email: dterlizz@umd.edu

Donald W. Webster
Eastern Shore Agent
University of Maryland
Wye Research & Education Center
P.O. Box 169
124 Wye Narrows Drive
Queenstown, MD 21658
Tel. (410) 827-5377 x127
Fax (410) 827-9039
Email: dwebster@umd.edu

Massachusetts

Dr. Joseph K. Buttner
Assistant Professor
Department of Biology
Salem State College
352 Lafayette Street
Salem, MA 01970
Tel. (978) 542-6703
Fax (978) 542-6863
Email: joseph.buttner@salemstate.edu

Dr. Craig Hollingsworth
University of Massachusetts Extension Aquaculture Program
Department of Plant, Soil & Insect Sciences
Agricultural Engineering Building
University of Massachusetts
Amherst, MA 01003
Tel. (413) 545-1055
Fax (413) 545-5858
Email: chollingsworth@umext.umass.edu

Diane C. Murphy
Fisheries & Aquaculture Specialist
Cape Cod Cooperative/Woods Hole Oceanographic Institution Sea Grant
Barnstable County Deeds & Probate
PO Box 367
3195 Main Street
Barnstable, MA 02630-0367
Tel. (508) 375-6953 (office)
Tel. (508) 274-7065 (mobile)
Fax (508) 362-4923
Email: dmurphy@barnstablecounty.org

Josh Reitsma
Marine Program Specialist
Cape Cod Cooperative Extension/Woods Hole Oceanographic Institution Sea Grant
Barnstable County Deeds & Probate
PO Box 367
3195 Main Street
Barnstable, MA 02630-0367
Tel. (508) 375-6950
Email: jreitsma@barnstablecounty.org

New Hampshire

Dr. Ken La Valley
Associate Director/Program Leader
Commercial Fishing/Aquaculture Technology Specialist
NH Sea Grant Extension/UNH Cooperative Extension
201 Taylor Hall
University of New Hampshire
Durham, NH 03824
Tel. (603) 862-4343
Fax (603) 862-0107
Email: ken.lavalley@unh.edu

Michael Chambers
Marine Aquaculture Specialist
NH Sea Grant Extension/UNH Cooperative Extension
Jere A. Chase Ocean Engineering Lab
24 Colovos Road
Durham, NH 03824
Tel. (603) 862-3394
Email: michael.chambers@unh.edu

New Jersey

Lisa M. Calvo
Aquaculture Program Coordinator
New Jersey Sea Grant Consortium
Haskin Shellfish Research Laboratory
Rutgers University
6959 Miller Avenue
Port Norris, NJ 08349
Tel. (856) 785-0074
Cell (609) 440-4560
Email: calvo@hsrl.rutgers.edu

George (Gef) E. Flimlin
Professor/Marine Extension Agent
Rutgers Cooperative Extension of Ocean County
1623 Whitesville Road
Toms River, NJ 08755
Tel. (732) 349-1152
Fax (732) 505-8941
Email: flimlin@aesop.rutgers.edu

New York

Antoinette Clemetson
Marine Fisheries Specialist
New York Sea Grant
Cornell University Research & Extension Center
3059 Sound Avenue
Riverhead, NY 11901-1098
Phone: (631) 727-3910
Fax: (631) 369-5944
Email: aoc5@cornell.edu

Mark Malchoff
Extension Program Leader, and Aquatic Resource Specialist
Lake Champlain Sea Grant Program
101 Hudson Hall
Plattsburgh State University of New York
101 Broad Street
Plattsburgh, NY 12901-2681
Tel. (518) 564-3037
Email: mark.malchoff@plattsburgh.edu

Gregg Rivara
Aquaculture Specialist
Cornell Cooperative Extension
3690 Cedar Beach Road
Southold, NY 11971
Tel. (631) 852-8660
Fax (631) 852-8662
Email: gjr3@cornell.edu

Kim Tetrault
Community Aquaculture Specialist
Cornell Cooperative Extension
3690 Cedar Beach Road
Southold, NY 11971
Tel. (631) 852-8660
Fax (631) 852-8662
Email: kwt4@cornell.edu

Dr. Michael Timmons
Agricultural & Biological Engineering
Cornell University
302 Riley-Robb Hall
Ithaca, NY 14853
Tel. (607) 255-1630
Fax (607) 255-4080
Email: mbt3@cornell.edu

David White
Program Coordinator and Recreation/Tourism Specialist
New York Sea Grant
SUNY College at Oswego
Oswego, NY 13126-3599
Tel. (315) 312-3042
Email: dgw9@cornell.edu

Pennsylvania

Ann Faulds
Associate Director
Pennsylvania Sea Grant
1350 Edgmont Avenue
Suite 2570
Chester, PA 19013
Tel. (215) 806-0894
Email: afaulds@psu.edu

Dr. Steven Hughes
Aquaculture Research & Education Center
Box 200, 1837 University Circle
Cheyney University
Cheyney, PA 19319
Tel. (610) 399-2400
Email: shughes@cheyney.edu

Rhode Island

Azure Dee Cygler
Fisheries and Aquaculture Extension Specialist
Rhode Island Sea Grant/Coastal Resources Center
Graduate School of Oceanography
University of Rhode Island
220 South Ferry Road
Narragansett, RI 02882
Tel. (401) 874-6197
Email: azure@crc.uri.edu

Dr. Dale F. Leavitt
Aquaculture Extension Specialist
220 M&NS
Roger Williams University
One Old Ferry Road
Bristol, RI 02809-2921
Tel. (401) 254-3047 (office)
Cell (401) 450-2581
Fax (401) 254-3310
Email: dleavitt@rwu.edu

Jennifer McCann
Director of Extension
Rhode Island Sea Grant
Graduate School of Oceanography
University of Rhode Island
South Ferry Road
Narragansett, RI 02882
Tel. (401) 874-6127
Fax: (401) 874-6920
E-mail: mccann@crc.uri.edu

Dr. Michael Rice
Department of Fisheries, Animal & Veterinary Sciences
University of Rhode Island Fisheries Center
Woodward Hall, Room 19
Kingston, RI 02881
Tel. (401) 874-2943
Fax (401) 874-7575
Email: rice@uri.edu

Vermont

Dr. Jurij Homziak
Extension Assistant Professor/ Director Lake Champlain Sea Grant
Lake Champlain Sea Grant Program
Forest Service Northern Research Station
University of Vermont
705 Spear Street
South Burlington, VT 05403
Tel. (802) 856-0682
Email: jurij.homziak@uvm.edu

West Virginia

Dr. Kenneth Semmens
West Virginia University
1052 Agriculture Science Building
P.O. Box 6108
Morgantown, WV 26505-6108
Tel. (304) 293-2657
Fax (304) 293-6954
Email: Ken.Semmens@mail.wvu.edu

Regional Leadership

Dr. Reginal Harrell, Director
Northeastern Regional Aquaculture Center (NRAC)
University of Maryland
2113 Animal Sciences Building
College Park, MD 20742-2317
Tel. (301) 405-6511
Fax. (301) 314-9412
Email: rharrell@umd.edu

David Alves
Northeast Region Aquaculture Coordinator
NOAA Aquaculture Office
National Marine Fisheries Service
Northeast Regional Office
55 Great Republic Drive
Gloucester, MA 01930
Tel. (978) 281-9210
Fax (978) 281-9117
Email: david.alves@noaa.gov

National Leadership

Max Mayeaux
Program Specialist
Division of Animal Systems
USDA National Institute of Food and Agriculture
1400 Independence Avenue SW, Stop 2201
Washington, DC 20250-2201
Tel. (202) 401-3352
Fax (202) 401-6156
Email: mmayeaux@nifa.usda.gov

Dr. Michael Rubino, Director
NOAA National Aquaculture Program
1315 East West Highway
Silver Spring, MD 20910
Tel. (301) 427-8325
Email: michael.rubino@noaa.gov

APPENDIX 5.
Educational Resources

General Business and Risk Management (Northeast U.S.)

The following resources may be helpful to the prospective or new aquaculture farmer. Please note that this list is not exhaustive and only includes some of the most common publications. Many of these resources can be borrowed from state or university libraries, or by contacting the local extension office (see Appendix 4 for contact information).

Buttner, J., Flimlin, G., Webster, D., 2008. Freshwater aquaculture species for the northeast. Northeastern Regional Aquaculture Center, NRAC Publication No. 102-2008

Buttner, J., Flimlin, G., Webster, D., 2008. Marine aquaculture species for the northeast. Northeastern Regional Aquaculture Center, NRAC Publication No. 103-2008.

Flimlin, G., Buttner, J., Webster, D., 2008. Aquaculture systems for the northeast. Northeastern Regional Aquaculture Center, NRAC Publication No. 104-2008.

McIntosh, D., 2008. Aquaculture risk management. Northeastern Regional Aquaculture Center, NRAC Publication No. 107-2008.

Pomeroy, R., 2003. Aquaculture record keeping. Connecticut Sea Grant Publication No. CTSG-03-15.

Secretan, P.A.D., Nash, C.E., 1989. Aquaculture and Risk Management. Aquaculture Development and Coordination Programme. United Nations Development Programme, Food and Agriculture Organization of the United Nations. Rome.

Shellfish Aquaculture (Northeast U.S.)

The following resources may be helpful to the prospective or new aquaculture farmer. Please note that this list is not exhaustive and only includes some of the most common publications. Many of these resources can be borrowed from state or university libraries, or by contacting the local extension office (see Appendix 4 for contact information).

General Bivalve Shellfish Culture

Baptist, G., Merritt, D.W., Webster, D.W., 1993. Growing microalgae to feed bivalve larvae. Northeastern Regional Aquaculture Center, NRAC Publication No. 160.

Flimlin, G., 2000. Nursery growout methods for aquacultured shellfish. Northeastern Regional Aquaculture Center, NRAC Publication No. 00-002.

Flimlin, G., Leavitt, D.F., Smolowitz, R., 2014. Dead and Dying Shellfish: What to Do? Northeastern Regional Aquaculture Center, NRAC Publication No. 220-2013.

Gosling, E. 2003. Bivalve Molluscs: Biology, Ecology and Culture. Blackwell Publishing, Oxford, United Kingdom.

Helm, M.M., Bourne N., Lovatelli, A., 2004. Hatchery culture of bivalves. A practical Guide. FAO Fisheries Technical Paper. No. 471. Rome, FAO.

Loosanoff, V.L., Davis, H.C., 1963. Rearing of bivalve mollusks. Advances in Marine Biology. 1:2-136.

Menzel, W., 1990. Estuarine and Marine Bivalve Mollusk Culture. CRC Press, Inc., Boston, Massachusetts.

Shumway, S.E., (Ed.), 2011. Shellfish Aquaculture and the Environment. Wiley.

Spencer, B.E., 2002. Molluscan Shellfish Farming. Blackwell. Fishing News Books. Malden, Massachusetts.

Walton, W.C., Murphy, D., 2005. Shellfish aquaculture: tools, tips, and techniques. Woods Hole Sea Grant Video Publication No. WHOI-V-05-004.

Eastern oyster (*Crassostrea virginica*)

Bohn, R.E., Webster, D.W., Merritt, D.W., 1995. Producing oyster seed by remote setting. Northeastern Regional Aquaculture Center, NRAC Publication No. 220.

Kennedy, V.S., Newell, R.I.E., Eble, A.F., (Eds.), 1996. The Eastern Oyster *Crassostrea virginica*. Maryland Sea Grant College, University of Maryland System, College Park, Maryland.

Brooks, W.K., 1891. The Oyster. The Johns Hopkins Press, Baltimore, Maryland.

Galtsoff, P.S., 1964. The American oyster *Crassostrea virginica* (Gmelin). U.S. Fish and Wildlife Service Fishery Bulletin. 64, 1-480.

Matthiessen, G.C., 1989. Small-scale Oyster Farming: A Manual. National Coastal Resources Research and Development Institute. Newport, Oregon.

Matthiessen, G.C., 2001. Oyster Culture. Fishing News Books, Blackwell Science, Maldon, Massachusetts.

Wallace, R.K., Waters, P., Rickard, F.S., 2008. Oyster hatchery techniques. USDA Southern Regional Aquaculture Center, Publication No. 4302.

Northern quahog (*Mercenaria mercenaria*)

Belding, D.L., 1910. A report upon the quahaug and oyster fisheries of Massachusetts including the life history, growth and cultivation of the quahaug (*Venus mercenaria*), and observations on the set of oyster spat in Wellfleet Bay. Commonwealth of Massachusetts Department of Conservation, Division of Fish and Game.

Castagna, M., Kraeuter, J.N., 1981. Manual for growing the hard clam *Mercenaria*. Special Report No. 249, Virginia Institute of Marine Science, Gloucester Point, Virginia.

Castagna, M., 1984. Methods of growing *Mercenaria mercenaria* from post-larval to preferred-size speed for field planting. Aquaculture. 39, 355-359.

Hadley, N.H., Whetstone, J.M., 2007. Hard clam hatchery and nursery production. USDA Southern Regional Aquaculture Center, Publication 4301.

Kraeuter, J.N., Castagna, M., (Eds.), 2001. Biology of the Hard Clam. Developments in Aquaculture and Fisheries Science – 31, Elsevier, New York.

Malinowski, S., 1986. Small-scale Farming of the Hard Clam on Long Island, New York. New York State Urban Development Corporation, New York

Manzi, J.J., Castagna, M.. 1989. Clam Mariculture in North America. Elsevier Press. New York.

Rice, M.A., 1992. The Northern Quahog: The Biology of Mercenaria mercenaria. Rhode Island Sea Grant, Narragansett, Rhode Island.

Whetstone, J.M., Sturmer, L.N., Oesterling, M.J., 2005. Biology and culture of the hard clam (Mercenaria mercenaria). USDA Southeastern Regional Aquaculture Center, Publication No. 433.

Bay Scallop (*Argopecten irradians*)

Belding, D.L., 1910. The scallop fishery of Massachusetts including an account of the natural history of the common scallop. Commonwealth of Massachusetts Department of Conservation, Division of Fish and Game, Marine Fisheries Service No. 3.

Castagna, M., Duggan, W.P., 1971. Culture of the bay scallop, Aequipecten irradians. Proceedings of the National Shellfisheries Association. 61, 80-85.

Karney, R.C., 1991. Ten years of scallop culture on Martha's Vineyard, in: Shumway, S.E., Sandifer, P.A., (Eds.), Scallop Biology and Culture. World Aquaculture Workshops, No. 1. World Aquaculture Society, Baton Rouge, Louisiana, pp. 308-312.

Shumway, S.E., Parsons, G.J., (Eds.), 2006. Scallops: Biology, Ecology and Aquaculture, 2nd Edition. Developments in Aquaculture and Fisheries Science Volume 35. Elsevier, San Diego, California.

Surier, A., Karney, R., Leavitt, D., 2010. Hatchery culture of the bay scallop. USDA Northeast Regional Aquaculture Center, Publication No. 214-2010.

Widman, J.C. Jr., Choromanski, J., Robohm, R.A., Stiles, S., Wikfors, G.H., Calabrese, A., 2001. Manual for hatchery culture of the bay scallop, Argopecten irradians irradians. Connecticut Sea Grant College Program, CTSG-01-03.

Softshell clam (*Mya arenaria*)

Belding, D.L., 1909. A report on the soft-shell clam fishery of Massachusetts including the life history, growth and cultivation of the soft-shell clam (*Mya arenaria*). Commonwealth of Massachusetts Department of Conservation, Division of Fish and Game, Marine Fisheries Service No. 1. (republished in 1916 and 1930).

Buttner. J.K., Weston, S., Beal, B.F., 2010. Softshell clam culture: hatchery phase, broodstock care through seed production. USDA Northeastern Regional Aquaculture Center, Publication No. 202-2010.

Weston, S., Buttner, J.K., 2010. Softshell clam culture: basic biology and general culture considerations. USDA Northeastern Regional Aquaculture Center, NRAC Publication No. 201.

Blue mussel (*Mytilus edulis*)

Bayne, B.L. (Ed.), 1976. Marine mussels: their ecology and physiology. Cambridge University Press, New York.

Gosling, E.M., (Ed.), 1992. The Mussel *Mytilus*: Ecology, Physiology, Genetics and Culture. Elsevier, Amsterdam.

Jamieson, G., (Ed.), 1989. Special section on mussel culture. World Aquaculture. 20(3), 8-105.

Lutz, R.A., (Ed.), 1979. Mussel Culture and Harvest: A North American Perspective. Elsevier, Amsterdam.

The Island Institute, 1999. The Maine Guide to Mussel Raft Culture. Produced for the Maine Aquaculture Association.

Finfish Aquaculture (Northeast U.S.)

The following resources may be helpful to the prospective or new aquaculture farmer. Please note that this list is not exhaustive and only includes some of the most common publications. Many of these resources can be borrowed from state or university libraries, or by contacting the local extension office (see Appendix 4 for contact information).

AFS-FHS (American Fisheries Society-Fish Health Section). 2010. FHS (Fish Health Section) blue book: suggested Procedures for the Detection and Identification of Certain Finfish and Shellfish Pathogens, 2010 Edition. American Fisheries Society-Fish Health Section, Bethesda, Maryland.

Bowser, P., 2012. General Fish Health Management. Northeastern Regional Aquaculture Center, NRAC Publication No. 111.

Huguenin, J.M., 1993. Fish Counting and Measurements in Situ: A technology assessment. Northeastern Regional Aquaculture Center, NRAC Publication No. 221.

Killian, H.S., Heikes, D., Van Wyk, P., Masser, M., Engle, C.R., 1998. Inventory Assessment Methods for Aquaculture Ponds. Southern Regional Aquaculture Center, SRAC Publication No. 395.

McIntosh, D., 2010. An overview of baitfish culture in the northeast. USDA Northeastern Regional Aquaculture Center, Publication No. 218.

Regenstein, J.M., 1992. Processing and Marketing Aquacultured Fish. Northeastern Regional Aquaculture Center, NRAC Publication No. 140.

Wooster, G.A., Hsu, H., Bowser, P.R., 1993. A Manual for Nonlethal Surgical Procedures to Obtain Tissue Samples for use in Fish Health Inspection. Northeastern Regional Aquaculture Center, NRAC Publication No. 112.

Seaweed Aquaculture (Northeast U.S.)

The following resources may be helpful to the prospective or new aquaculture farmer. Please note that this list is not exhaustive and only includes some of the most common publications. Many of these resources can be borrowed from state or university libraries, or by contacting the local extension office (see Appendix 4 for contact information).

Edwards, M., Watson, L., 2011. Cultivating *Laminaria digitata*. Aquaculture Explained No. 26. Irish Sea Fisheries Board.

Flavin, K., Flavin, N., Flahive, B., 2013. Kelp Farming Manual: A Guide to the Processes, Techniques, and Equipment for Farming Kelp in New England Waters. Ocean Approved, Portland, ME. 123pp.

FAO (Food and Agricultural Organization of the United Nations), 1990. Training manual on *Gracilaria* culture and seaweed processing in China. Training Manual 6. Online at http://www.fao.org/docrep/field/003/ab730e/AB730E00.htm#TOC.

Redmond, S., Green, L., Yarish, C., Kim, J., Neefus, C., 2014. New England Seaweed Culture Handbook Nursery Systems. Connecticut Sea Grant Publication No. CTSG-14-01. 92 pp. http://seagrant.uconn.edu/publications/aquaculture/handbook.pdf.

Yarish, C., Redmond, S., Kim, J., 2012. *Gracilaria* Culture Handbook for New England. Wrack Lines (report). Connecticut Sea Grant Paper No. 72. http://digitalcommons.uconn.edu/wracklines/72.

Yarish, C., Redmond, S., Kim, J., 2012. *Gracilaria* Culture Handbook for New England. Wrack Lines (video). Connecticut Sea Grant Paper No. 70. http://digitalcommons.uconn.edu/wracklines/70.