North Central Regional Aquaculture Center

Sunfish Culture Guide

In cooperation with USDA\CSREES

NCRAC Culture Series

Sunfish Culture Guide

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North Central Regional Aquaculture Center

The North Central Regional Aquaculture Center (NCRAC) was formed in February 1988. It is one of five regional aquaculture centers administered by the U.S. Department of Agriculture (USDA).

The mission of these centers is to support aquaculture research, development, demonstration, and extension education to enhance viable and profitable U.S. aquaculture production that will benefit consumers, producers, service industries, and the American economy.

They work together within the broader, integrated aquaculture program of the USDA to promote a well-developed sustainable aquaculture industry in the USA. NCRAC programs are jointly administered by Michigan State University (MSU) and Iowa State University (ISU). The Director's office is located at MSU; the Associate Director is at ISU.

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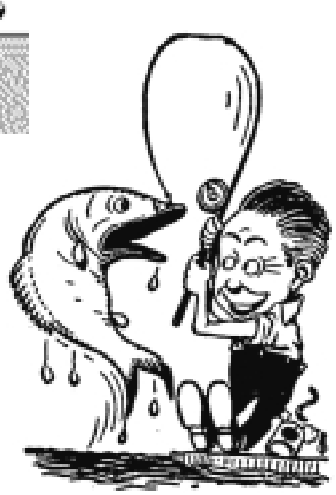
Preface



With the growing interest in aquaculture over the past three to four decades, there is an increasing need to evaluate and develop different fish species for aquaculture. Because of the interest in species other than trout or catfish, coupled with the limited availability of compiled information on other species, the North Central Regional Aquaculture Center (NCRAC) decided to develop culture manuals on potential aquaculture species.

The purpose of the NCRAC culture series is to provide an assemblage of current information regarding the culture of a specific fish species. The guides are written in a style useful to all levels of expertise, from the novice to the expert culturist.

Editor R.C. Summerfelt completed the *Walleye Culture Manual*, Culture Series 101, which is available from NCRAC. The *Sunfish Culture Guide*, Culture Series 102, is the second in the series. The *Yellow Perch Culture Guide* will be the third guide in the series.

The Sunfish Culture Guide, which focuses on *Lepomis* spp., is organized so that each chapter can stand alone. Culturists with different interests and levels of expertise can use all or any part of the guide. Consequently, some repetition of information was unavoidable; however, we believe that repetition will not cause readers undue distraction.

Measurements are given in both metric and English units. The metric system greatly simplifies calculations, but many culturists are more comfortable using the English measurement system. For readers not familiar with scientific abbreviations, Appendix C contains a chart of common abbreviations.

References cited within the text are listed at the end of the manual. Readers with a strong interest in certain topics are encouraged to refer to these references for more in-depth information.

Your editors,

Joseph E. Morris, Charles C. Mischke, and Donald L. Garling

Acknowledgements

The Sunfish Culture Guide is the result of collaborative efforts of the North Central Regional Aquaculture Center (NCRAC) extension members and work groups. Work on the NCRAC sunfish project was made possible through grants from the U.S. Department of Agriculture-Cooperative State Research Education and Extension Service (USDA-CSREES).

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We are grateful to those reviewers and organizations who took time out of their busy schedules to give us constructive input on this publication.

Disclaimer

Photo Credits

The editors thank Sterling K. Johnson (Texas A&M University) for the use of his photographs to depict various fish diseases. We also acknowledge and thank the following individuals who contributed photos for this publication:

- Gary Atchison, Iowa State University
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- Clay Pierce, Iowa State University
- LaDon Swann, Mississippi-Alabama Sea Grant Consortium

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1

Historical Background

Sunfish are important elements to the environmental health of ponds and streams. Certain species have been popular research subjects, key to pond stocking, and important forage bases. Sunfish are now under consideration for their potential suitability in commercial scale aquaculture production.

This chapter explains:

- The term sunfish
- Early research
- Sunfish importance
- Aquaculture potential

1

Historical Background

Authors: C. C. Mischke and J. E. Morris, Iowa State University

The Term Sunfish

The term "sunfish" refers to any of thirty sunfish species in the Centrarchidae family (Pflieger 1975). Centrarchids are strictly a North American fish family that includes bass (*Micropterus* spp.), bream (*Lepomis* spp.), and crappie (*Pomoxis* spp.).

Characteristics

Members of the Centrarchidae family have deep bodies, laterally compressed. The anterior portion of their dorsal fin consists of spiny rays; the posterior portion consists of soft rays. The pelvic fins of sunfish are located directly beneath the pectoral fins.

Manual focus

This guide focuses on the *Lepomis* species of sunfish. Most available literature to date deals with the bluegill and redear sunfish, but the information could apply to most of the *Lepomis* species. Basic production practices are the same for pure sunfish species as for their hybrids (Dupree and Huner 1984).

Early Research

The first studies dealing with sunfish, as with most fish, were observational studies that noted their basic biological activities. Breder (1936) was one of the first researchers to extensively observe the habits of the North American sunfish.

Spawning nests of sunfish.

Sunfish Importance

Sunfish and sunfish hybrids have several characteristics that make them suitable candidates for aquaculture production. They may be well suited to such temperate regions as the North Central Region (NCR), where there is a shorter growing season and cooler water temperatures. However, further research is needed to establish the culture of these fish as a viable aquaculture enterprise.

As with any aquaculture venture, sunfish aquaculture must be approached cautiously, with considerable planning, and careful research.

Assessment of market opportunities should be part of the evaluation, as it is with all potential aquaculture enterprises.

Bluegill sunfish

The bluegill is the most abundant sunfish, and widespread introduction has increased its range in North America, Europe, and South Africa (Pflieger 1975; Carlander 1977). They are abundant in ponds, lakes, and slow-moving streams. Bluegill are intolerant of chronic high turbidity and siltation; they thrive in warm, clear waters where aquatic vegetation or other cover is present (Pflieger 1975).

Redear sunfish

Redear sunfish, frequently referred to as the “shellcrackers,” typically live near the bottom of lakes and ponds, mainly eating snails. They have a rapid growth rate and are usually larger than the bluegill, commonly reaching 267 mm (10.5 in.) (Pflieger 1975; Tomelleri and Eberle 1990).

Green sunfish

Green sunfish are widely distributed and adaptable to a wide range of conditions, for example: high turbidity, low dissolved oxygen, and high alkalinity (Childers 1967; Tomelleri and Eberle 1990).

This wide tolerance usually results in their overpopulation and the suppression of other sunfish populations.

Key pond and research components

Specific species of sunfish have been key components in farm ponds throughout the U.S. They have been stocked extensively in ponds as forage fish for largemouth bass and as sport fish (Swingle 1946; Dupree and Huner 1984; McLarney 1987).

Bluegill are popular research subjects. They are especially used for toxicology studies (Eaton 1970, 1974; Benoit 1975; Sandheinrich and Atchison 1989; Coyle et al. 1993; Little et al. 1993), but also for ecology studies on foraging behavior (Li et al. 1985; Butler 1988; Ehlinger 1989; Gotceitas and Colgan 1989, 1990). Because bluegill are an important forage base and are so wide spread, fishery biologists frequently sample them to determine a pond’s balance and its structure.

Bluegill is the most abundant sunfish, and thrives in warm, clear waters with vegetation.

Redear grow rapidly and are larger than bluegill.

Green sunfish are adaptable to a wide range of conditions.

There is continuing interest in and intensive culturing and marketing of sunfish and their hybrids for pond stocking and food fish production.

Aquaculture Potential

For a fish species to be suitable for aquaculture production on a true commercial scale, it needs to meet both marketing and biological criteria (Webber and Riordan 1976).

Sunfish, especially bluegill and their hybrids, seem to have the potential to become a very commercially viable and marketable aquaculture product.

Marketing criteria

Marketing criteria include appearance, texture, and consumer recognition. Bluegill have a good to excellent flavor and slightly soft texture, which makes them acceptable to a large number of consumers (McLarney 1987). Their flesh is firm, white, and flaky; since the flesh has little fat, most sunfish meat may be kept frozen for long periods (Becker 1983).

Sunfish are highly respected as food fish and bluegill are referred to as “bread and butter fish” (Becker 1983). Bluegill and other sunfish are among the most highly recognized fish species. This improves their marketability.

Biological criteria

Biological criteria include acceptance of artificial feeds (Ehlinger 1989), temperature tolerance (Heidinger 1975), good growth rates (Krumholz 1946; Breck 1993), and the ability to spawn repeatedly (Banner and Hyatt 1975; Stickney 1985).

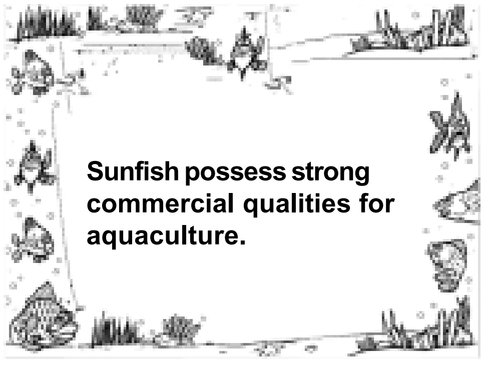
Because sunfish and sunfish hybrids possess many desirable characteristics for aquaculture production, there is increased interest in developing culture techniques.

Research is currently conducted on all aspects of sunfish culture, from out-of-season spawning to grow-out, and finally to marketing.

Summary

Interest in sunfish has progressed from merely observational studies of basic biological information to an interest in intensive and extensive culture practices.

Because sunfish possess desirable production characteristics, research is now conducted in all areas of sunfish aquaculture.



Notes

2

Spawning Behavior & Early Life History

Sunfish species have a number of similar breeding habits. Different morphological characteristics provide cues for species recognition. Similarities in spawning habits allow a wide variety of hybrid development. Research into these habits gives breeders a better understanding of what aspects of the spawning and development process help ensure reproduction success.

This chapter explains:

- Nest building
- Behavioral studies
- Spawning habits
- Hatching and growth
- Larval development

2

Spawning Behavior and Early Life History

Authors: G. D. Dvorak, J. E. Morris, and C. C. Mischke, Iowa State University.

Behavioral Studies

Many observational studies of sunfish spawning behavior have been conducted (Breder 1936; Hunter 1963; Erickson 1967; Keenleyside 1967; Avila 1975; Smith 1975; Clarke et al. 1984). Sunfish were found to be more aggressive when crowded into small areas, regardless of sex or season (Erickson 1967; Avila 1975).

Adequate spacing is necessary to induce bluegill reproductive behavior under intense environmental conditions, according to Bryan et al. (1994).

Species recognition between bluegill and pumpkinseed is based on morphological characteristics and could act as a behavioral isolating mechanism (Keenleyside 1967). Childers (1967) found it necessary to remove the opercula tabs of male redear sunfish when crossing them with female bluegill.

Clarke et al. (1984) investigated the courtship sequences in pure and hybrid crosses of bluegill and pumpkinseed. They found that male behavior was influenced by the behavior and species of the female present, and that females of different species signaled males differently. They concluded that species discrimination is based on differences in courtship behavior.

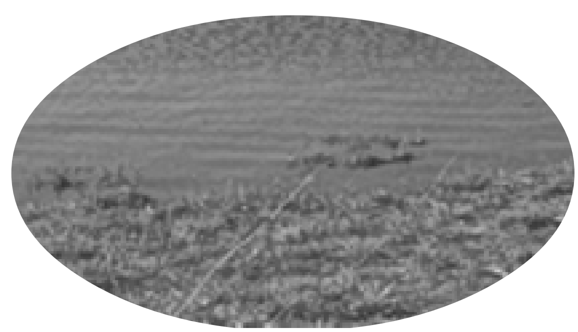
Signal value of certain acts varies with the partner species present. Avila (1975) compared the nesting behavior of male bluegill in surrounded nests [three adjacent occupied nests < 60 cm (24 in.)] and peripheral nests [nests that were separated from adjacent nests by >60 cm (24 in.)]. Peripheral males displayed more reproductive behavior (92%) than surrounded males (76%).

Nest Building

In the wild, sunfish are nest builders in shallow waters and their reproductive habits are remarkably uniform. All sunfish spawn in the late spring or early summer when water temperatures approach 20–29°C (68–84°F) (Breder 1936).

Male sunfish generally appear first at the spawning sites and construct nests along the shoreline in unshaded areas (see below) that have maximum exposure to sunlight (Hunter 1963; Avila 1975). Using his tail, the male sweeps a nest using tail undulations (caudal peduncle), making a shallow circular depression in the soft mud or gravel bottom of quiet shallow water.

Sunfish nests consist of depressions 5–15 cm (2–6 in.) deep and approximately 30 cm (12 in.) in diameter, constructed in water 0.3–1.5 m



Nesting site. Sunfish prefer shallow water to build their nests. These waters are warmer than are the deeper waters.

(1–5 ft) deep (Becker 1983). Most sunfish are colonial nesters, which means they construct nests in densely packed aggregations (Gross and MacMillan 1981).

Often different species can be found nesting together in the same colony (Childers 1967). A bluegill colony is usually comprised of 40–50 nests within a radius of 18–21 m (20–23 yd) (Becker 1983).

Spawning

During the spawning season, males become brilliantly colored, which helps attract females. Additionally, tail sweeping and rim circling behaviors have value both to attract females and threaten other males (Avila 1975). After construction, the male circles the nest and produces courtship calls consisting of a series of grunts (Avila 1975; Gross and MacMillan 1981; Becker 1983).

The courtship behavior of male sunfish closely resembles their aggressive behavior (Breder 1936). A female ready to spawn (gravid female) does not reciprocate this aggressive behavior and remains stationary, which may serve as a signal to the male.

Once a female is attracted to the nest, however, the male circles and guides the female into the nest where the eggs are then deposited and fertilized.

During spawning, the female reclines to one side and the pair swims side by side in a tight circle over the nest. As the female releases her eggs, the male fertilizes them. When sunfish eggs are fertilized, they become water hardened and adhere to the material on the nest bottom.

Female sunfish can produce from 2,000–25,000 eggs per spawn. More than one female may deposit eggs in a single nest (Avila 1975; Becker 1983).

After eggs are deposited, the male chases the female out of the nest. The males defend the nests from predators and fan the nest to keep the eggs aerated and clean of debris, guarding them until the fry begin to swim.

Hatching and Growth of Fry

Eggs usually hatch from 2–6 d after fertilization, depending on species, photoperiod (number of light hours), and temperature.

Bluegill eggs hatch in 2–3 d, (Smith 1975; Beard 1982) but redear sunfish eggs hatch in 6–10 d (Childers 1967). As photoperiod and temperature are increased, hatching time is decreased (Toetz 1966).

Larval sunfish remain in the bottom of the nest, protected by the male and receiving nutrients from their yolk sacs until the swim-up stage.

The critical stage in their development is when larvae switch from endogenous to exogenous feeding—when they no longer rely on their yolk sacs and must obtain food from the environment for energy.

During this critical stage, nutrients from the yolk sac are depleted, and the larvae's mouth becomes more fully developed. The larval fish will starve if they do not begin feeding during this critical stage.

Swim-Up Times

Swim-up times for sunfish vary with the species and the temperature. Times range from 2 d post-hatch for green sunfish (Meyer 1970; Smith 1975) to 3–7 d post-hatch for bluegill (Meyer 1970; Toetz 1966; Smith 1975; Mischke 1995; Mischke and Morris 1997) and 3 d post-hatch for redear sunfish (Meyer 1970).

Meyer (1970) reported swim-up age for bluegill as 3 d post-hatch at a constant temperature of 21°C (70°F). Toetz (1966) and Bryan et al. (1994) reported bluegill larvae swim-up at 4 d post-hatch at 23.5°C (75°F) and 26°C (79°F), respectively.

Childers and Bennett (1961) reported that bluegill larvae reach swim-up at 5–6 d post-hatch at 21°C (70°F). Mischke (1995) and Mischke and Morris (1997) reported bluegill swim-up to be 7 d post-hatch in the 21–25°C (70–77°F) temperature range.

Redear larvae swim-up in 3 d post-hatch at 21°C (70°F) (Meyer 1970), whereas green sunfish have been reported to swim-up in two d post-hatch at 21°C (70°F) and at 25°C (77°F). Additionally, green sunfish fry typically have a greater total length at hatching and swim-up than other sunfish (Taubert 1977).

The first prey items for larval sunfish must be small enough to fit into their mouth. Toetz (1966) reported the mouth gape of larval bluegill at the onset of exogenous feeding to be 230–270 µm (0.009–0.011 in); hence the first prey items must be smaller than this for sunfish larvae to consume it.

If sunfish survive the critical stage in the wild and begin feeding, their first food consists of small zooplankton rotifers, early instar stages of cladocerans and copepod nauplii.

Sunfish growth is rapid, and as mouth size increases, they begin selecting larger plankton prey, such as cladocerans (Siefert 1972). As the fish grow larger, they feed mainly on aquatic insects, small crayfish, and other small fish (Carlander 1977; Stickney 1985).

Larval Development

Larvae form identification is essential to studies of food habits, age, and growth of larvae (Meyer 1970). However, studies of larval sunfish development have been limited to specific species and mainly address such meristic features as: myomere counts (muscle filaments), pigmentation, and ray development (Meyer 1970; Taubert 1977).

Morgan (1951) provides a detailed description (with diagrams) of larval bluegill development. Duwe (1952, 1955) describes the development of the swim bladder in green sunfish and bluegill.

Childers (1967) described some larvae features for all crosses between bluegill, green sunfish, and redear sunfish, but these are limited to pre-hatching events (i.e., hatching percentages, length at hatch). Smitherman and Hester (1962) compared meristic characters of hybrid sunfish to their parentals.

Dvorak (1997) made detailed observations of larval hybrid sunfish development. In Table 2.1, he describes the early development of the female green sunfish x male bluegill (GxB) hybrid larvae reared at a mean temperature of 21.5°C (70.7°F).

Table 2.2 (end of this chapter) shows a comparison of larval development for other sunfish reported by additional researchers.[1]

Prior to hatching, the transparent embryo encircles the yolk sac (Figure 2.1). A single oil globule can be seen in the yolk sac. Additionally, the optic capsule is developed but not pigmented.

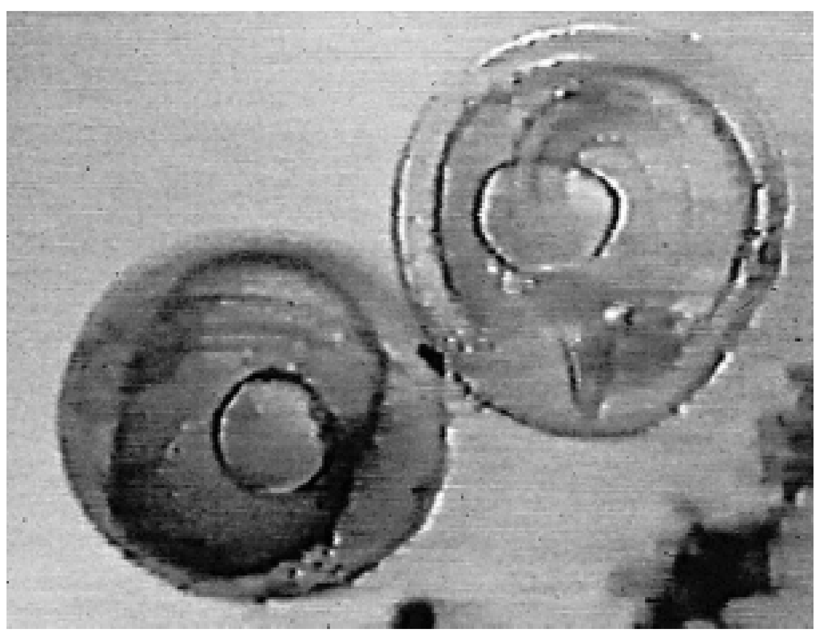


Figure 2.1. Fertilized sunfish eggs just prior to hatching contain the fish embryo, which encircles the yolk sac.

The blood is clear and can be seen circulating around the yolk and into the heart; the heart beats rapidly. The embryo moves vigorously in a shivering-like manner until the egg membrane suddenly ruptures and the larvae are set free.

Post-hatch Times

Within 24 h of hatching, the eyes become pigmented (Figure 2.2). A single, circular oil globule is located posterior in the round yolk sac.

The intestine can be seen from the yolk sac to the anus. Within 24 h, the notochord, myomeres, and median finfold develop.

The G x B hybrid larvae are 4.2 mm (0.016 in.) mean total length (TL) and very inactive at this stage. The heart is clearly seen located in front of the yolk sac.

Blood pigmentation is light pink and can be seen moving throughout the body and around the tail and anus.

2–3 days

Larvae are 4.5 mm (0.18 in.) TL by 2 d post-hatch. The eyes and blood become more darkly pigmented. The otolith is clearly seen, the median finfold is still present, and pectoral fins are visible. The larvae intestine has also widened.

By 3 d, larvae are 4.9 mm (0.19 in) TL (Figure 2.3) and the medial finfold and yolk sac becomes reduced. The yolk sac is more oblong and the larval body is long and slender.

Up to this point, there is no pigmentation on the larvae itself although there is blood pigmentation.

4–5 days

At 4 d post-hatch, the lower jaw begins to form and the mouth of the larvae becomes indented; however, it is not opened to the gut. Total length at this stage is 5.3 mm (0.21 in.).

The pectoral fins get longer and start beating. However, movement of the larvae is minimal due to the weight of the remaining yolk sac.

Table 2.1. Developmental observations of GxB larvae reared to swim-up at 22°C (71°F) listed by days post-hatch (dph).

dph	Length mm (in)	Observations
1	4.2(0.16)	Blood and eyes pigmenting; inactive; intestine seen from yolk sac to anus; median finfold present; heart visible
2	4.5 (0.18)	Eyes and blood more darkly pigmented; pectorals develop; intestine widens; larvae are more active
3	4.9 (0.19)	Pectorals continue developing; median finfold and yolk sac begin reducing; yolk sac is more oblong
4	5.3 (0.21)	Lower jaw forming; mouths begin indenting; pectorals developing and beating
5	5.6 (0.22)	Mouths begin opening weakly; pigmentation above yolk sac; tail becoming more heterocercal and starting to beat
6	5.9 (0.23)	Pigmentation over yolk sac increasing; pectorals beating rapidly; yolk sac almost gone but single oil globule remains; tail more heterocercal; anal fin begins forming; some larvae have full undulatory movements
7	6.1 (0.24)	Gas bladder inflated; free swimming; mouth gape was 0.30 mm (0.01 in)

Larvae measure 5.6 mm (0.22 in.) by 5 d. Pigmentation above the yolk sac begins developing. The pectorals continue getting longer and the larvae beat them rapidly.

Their mouths begin to open weakly and their tail starts to become heterocercal (i.e., develop two unequal lobes) and to beat.

6–7 days

At 6 d post-hatch, larvae pigmentation above the yolk sac increases. The pharyngeal arches are observable and the pectorals are long and beat rapidly. The larvae begins to move about.

The mean TL for the larvae at this stage is 5.9 mm (0.23 in.). The yolk sac almost disappears, but the single oil globule remains (Figure 2.4). The tail now becomes more heterocercal and the anal fin begins forming. Some larvae have full undulatory body movements.

Larvae inflate their gas bladders and swim by 7 d. They have a mean TL of 6.1 mm (0.24 in.).

Their mouth gape is 0.30 mm (0.012 in.), which is larger than the mouth gape of 0.23–0.27 mm (0.009–0.011 in.) reported by Toetz (1966) for larval bluegill at the free swimming stage.

Etnier (1971) reported that G x B hybrid juveniles have larger mouths than parentals.

Figure 2.3. Sunfish fry 3 d post-hatch have a reduced yolk sac and begin to acquire a long and slender body shape.

14 days

Figure 2.5 shows larvae at 14 d post-hatch. The swim bladder is prominent, dark, and circular. Notice that the median fin fold is still present but greatly reduced.

Food can be seen in the stomach and intestine. Additionally, pigmentation begins to develop on the head and body.

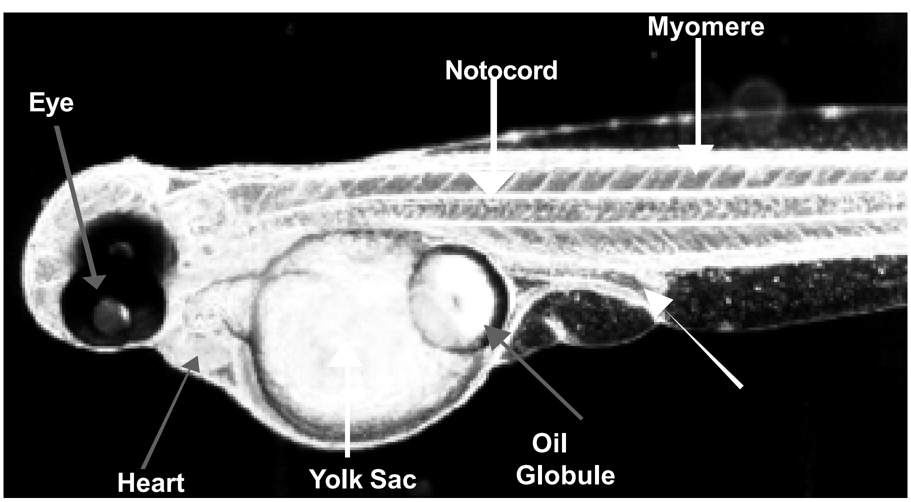


Figure 2.2. Within 24 hours after hatching, the eyes of hybrid sunfish larvae become pigmented and various other structures are clearly observable.

28 days

By 28 d post-hatch, the median fin fold is gone and rays are developed in the caudal fin of the GxB larvae (Figure 2.6).

The larvae are still quite transparent at this stage, but rows of pigmentation can be seen along the body and in patches on the head. Notice that the swim bladder is still dark and prominent at this

Figure 2.4. Hybrid sunfish fry by 6 d post-hatch have almost entirely lost their yolk sac and begun to swim about.

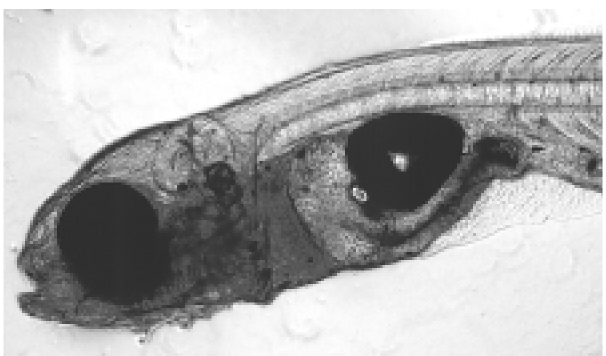


Figure 2.5. At 14 d post-hatch sunfish fry actively feed, have a prominent swim bladder and are developing pigmentation.

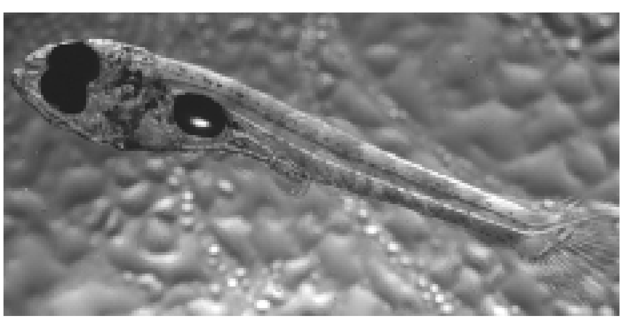


Figure 2.6. Sunfish fry 28 d post-hatch are still transparent, but actively feed and swim.

stage.

Summary

Even though the various sunfish species have similar spawning habits, there are different morphological characteristics that act as cues for species recognition.

Because sunfish have similar spawning behavior, they are relatively easy to cross with one another, thereby producing a wide variety of hybrids. However, because of the morphological differences of some species, such steps as ear flap removal may need to be taken to facilitate spawning.

Sunfish have a rapid early development from spawning to swim-up relative to other species. Most sunfish species hatch 2–6 d after spawning and swim-up 2–7 d after hatching.

The mouth gape of sunfish is very small; therefore, larvae require proportionately small food particles at the critical stage when they switch from endogenous to exogenous feeding. If they are unable to take in the food, they will not survive.

Notes

Table 2.2. Developmental observations made by other researchers on pure strain bluegill, green sunfish and GxB hybrids listed by days post-hatch (dph).

Bluegill Larvae			
dph	Length mm (in)	Observations	Author
0	3 (0.1)		Morgan 1951
0	2-4(0.1-0.2)		Taubert 1977
0	3-4 (0.1-0.2)	Alimentary canal; continuous finfold, pectoral fins, optic vesicles and lens of eye present; heart beating; blood red; single oil globule within yolk sac	Toetz 1966
0	4-5 (0.2)		Childers 1967
1	3-4 (0.1-0.2)	Anus clearly open	Toetz 1966
2	4-6 (0.2)	Alimentary canal open from above yolk sac to anus; mouth not open but jaws present and move weakly	Toetz 1966
3	5-6 (0.2)	Mouth open; mouth gape 0.13 mm (0.005 in); jaw moves weakly; swim bladder present; pectoral fins beating	Toetz 1966
3	5-6 (0.2)	Swim-up	Meyer 1970
3	6 (0.2)	No chromatophores on top of head	Meyer 1970
4	4-5 (0.2)	Alimentary canal open from pharynx to mouth; mouth gape 0.20 mm (0.008 in); continuous finfold reducing, except tail	Toetz 1966
~4	5 (0.2)	Became free swimming	Meyer 1970
5	5-6 (0.2)	Larvae free swimming; oil globule in anterior of yolk; mouth gape 0.23 mm (0.009 in)	Toetz 1966
6	5-6 (0.2)		Taubert 1977
6	5-6 (0.2)	Yolk almost gone; "pin heads" large head and narrow trunk; pigment cells; mouth gape 0.24 mm (0.009 in)	Toetz 1966
7	5-6 (0.2)	Yolk gone except for a tiny oil globule; pelvic fin present; swim bladder black; mouth gape 0.27 mm (0.011 in)	Toetz 1966
8	5 (0.2)		Morgan 1951
8	5-6 (0.2)	Tiny oil globule still present; swim bladder dark; lens of eye orange; mouth gape 0.29 mm (0.011 in)	Toetz 1966
9	5-6 (0.2)	Oil globule gone; swim bladder pigmented; mouth gape 0.33 mm (0.013 in)	Toetz 1966

Table 2.2 cont'd

Green Sunfish Larvae			
0	4 (0.02)	Eyes pigmented; pectoral fin buds	Taubert 1977
0	5 (0.02)		Childers 1967
6	6 (0.02)	Free swimming	Taubert 1977
9	5 (0.02)	Caudal rays developing; profuse spotting with small concise chromatophores on top of head	Meyer 1970
	7 (0.03)	Anal and dorsal rays started to develop	Meyer 1970
	8 (0.03)	All fin rays developed	Meyer 1970
Female Greenfish x Male Bluegill Hybrid Larvae			
0	3 (0.01)	Weighed 0.15 mg (0.000005 oz)	Mischke and Morris 1998
0	5 (0.02)		Childers 1967

Notes

3

Culture Methods

Sunfish have not been cultured on a large scale, so there is limited information regarding the best culture methods. Out of several different culture methods evaluated, pond culture is the most popular method. However, research continues to explore the best type of sunfish and most effective culture method for aquaculture.

This chapter explains:

- Pond culture
- Cage culture
- Indoor larvel fish production
- Fry care and feeding

3

Culture Methods

Authors: C. C. Mischke, J. E. Morris, and R. L. Lane, Iowa State University.

Pond Culture

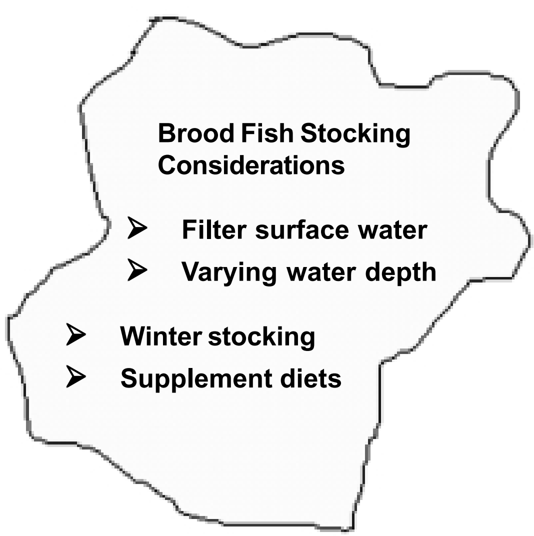
Most sunfish aquaculture production to date has been extensive production of bluegill in small ponds and lakes. Adult sunfish are stocked and allowed to spawn naturally in ponds. Generally, young are raised in the same ponds as the adults (Stickney 1985).

Ponds for specific species

Ponds used for production of a specific sunfish species must be carefully maintained to avoid interbreeding with other sunfish, a common trait in this family.

Water from surface sources must be filtered through a sock-type filter consisting of Saran cloth to prevent the introduction of undesirable fish (ca. 300–µm opening).

Some aquaculturists prefer to use ponds with a maximum depth of 0.9–1.5 m (3–5 ft), however, keeping some shallow areas at least 0.3 m (1 ft) in depth (Higgenbotham et al. 1983).



Water temperature

Brood fish are stocked before water temperatures reach 21°C (69.8°F). According to McLarney (1987), adult sunfish should be stocked in the winter at a rate of 250/ha (100/acre). A stocking rate of 100 pairs/ha (40 pairs/acre) has also been suggested (Dupree and Huner 1984; Engelhardt 1985). Stocking 2 yr old fish at a ratio of one male to one female has been successful (Dupree and Huner 1984; Stickney 1985).

Food requirements

When culturists use good pond culture techniques, sunfish fry are able to obtain their food requirements from the natural production in the pond (e.g., insects and zooplankton).

Brood fish, on the other hand, should be fed with a supplemental pellet feed from 3.2 to 6.2 mm (1/8 to 1/4 in.) in diameter (Dupree and Huner 1984).

A definite protein requirement has not been set for sunfish; however, it is assumed that protein estimates of sunfish brood stock fall between rainbow trout and channel catfish.

Often in pond sunfish grow-out, producers depend on such natural organisms as zooplankton and benthic organisms, for the main food source (Figure 3.1).

However, these natural food sources can become quickly depleted at high fish densities. Producers should consider using supplemental diets to enhance growth in any high-density sunfish pond production.

Using higher protein feeds ($\geq$35%) may improve growth and production potential of hybrid sunfish (green sunfish female x bluegill male)Tidwell et al. (1992). The NCRAC (1998) is in the process of determining dietary requirements for grow-out of bluegill and hybrid sunfish. The dietary phosphorus requirement of hybrid sunfish (GxB) is $<$ 0.5% of the dry diet.

Also, both bluegill and GxB hybrids grow best when fed diets containing at least 10% dietary lipid in the form of fish oil.

Work with hybrids in recirculating systems and in ponds suggests that when the formulated diet supplies virtually all the nutrition, optimum crude protein levels are $>$40%.

However, when fish are grown in ponds and natural food is available, a dietary crude protein level of 36% is considered adequate for maximum mean harvest weight.

Food conversion ratio

Specific growth rates (SGR) and food conversion ratios (FCR) are commonly used in aquaculture to express growth and feed conversion, respectively. When fish growth rates are exponential, as typically occurs over short intervals of time, instantaneous rates should be used to express growth (Busacker et al. 1990).

Figure 3.1. The typical method of sunfish production has been by use of small ponds.

$$\text{SGR (\%/d)} = (\ln W_t - \ln W_i)/T) \times 100.$$

Where: W_f = mean weight at the end of the period

W_i = mean weight at the beginning of the period

T = time in days of the period

The working formula is expressed as:

FCR = [weight of feed fed (g)/live weight gain (g)].

The inverse function of this equation is referred to as feed efficiency.

SGR and FCR measurement studies

To date, the majority of sunfish studies involving measurements of SGR and FCR have dealt exclusively with hybrids.[1] Tidwell et al. (1992) and Webster et al. (1992) reported SGR values of 1.98 and 2.6, respectively. Both studies used small fish (3–5 g).

Webster et al. (1997) reported lower SGR values for larger fish ($>$20 g). An SGR value of 0.37 was reported for large fish stocked in ponds for the summer growing season (Tidwell et al. 1994). The SGR values for sunfish should increase with genetic modification and enhanced culture methods.

Webster et al. (1992) reported FCR values of 3.72 and 3.87 at 32 and 38% crude protein, respectively, using smaller fish (3–4 g). Webster et al. (1997) reported slightly higher FCR values for larger fish ($\geq$20 g). High FCR values were reported for large fish stocked in ponds for the summer growing season (Tidwell et al. 1994).

Stocking rate

NCRAC (1998) suggested the stocking rate for grow-out of hybrid sunfish is 12,355–17,279 fish/ha (5,000–7,000 fish/acre). The accepted food size for sunfish is 227–340 g ($\frac{1}{2}$ – $\frac{3}{4}$ lb). Sunfish generally require more than 2 years to reach this size.

Only one stocking rate is given for pond grow-out. The best results occur when the first year stocking density is relatively high and then reduced to a much lower second year density for final grow-out.

Cage Culture[2]

In areas where regular pond culture is not practical, cage culture (Figure 3.2) may be a viable option. Irregularly shaped ponds, quarry pits, or other bodies of water that cannot be seined easily are all possible areas that may be conducive to cage culture.

According to Morris and Edwards (1991), sunfish must meet these desired species characteristics for cage culture:

- Fast growth
- Tolerance of crowded conditions
- Good growth in regional environments
- Native to region
- Market value

Cage culture advantages

There are certain advantages to cage culture. One of the most important is that many types of water resources can be used that would not otherwise be practical for fish production.

Fish harvesting and management are also greatly simplified. Cage culture requires a relatively low initial investment and allows the continued use of the pond for sport fishing or culture of other species (Masser 1988; Morris and Edwards 1991).

Cage culture also eliminates unwanted fish reproduction. Sunfish cannot spawn in cages that are suspended off the bottom. Therefore, more uniform fish size and more accurate inventories are possible.

Figure 3.2 Cage culture is used well in a variety of ponds.

When fish are grown in cages, they are able to use some of the pond's natural productivity, but not as much as free-roaming fish. Therefore, the protein requirement for cage cultured sunfish is between that of pond cultured fish and fish cultured in recirculating systems.

Stocking rates

Stock fingerling sunfish at 10 cm (4 in.) or larger and grade for uniformity in size.

- 500 sunfish fingerlings can be stocked in a 0.12 X 0.12 X 0.12 m (4 X 4 X 4 ft) square cage.
- 2000 sunfish can be stocked in a 0.24 X 0.24 X 0.12 m (8 X 8 X 4 ft) cage (Masser 1988).
- 200 fish/m^3 (6 fish/ ft^3) can be cage stocked (NCRAC 1998).

The best results usually occur when first year stocking densities are reduced for the second year final grow-out.

Indoor Larval Fish Culture

Although previous sunfish production has been mostly extensive in ponds, there has been success in obtaining fry through some intensive laboratory culture methods. Regardless of where sunfish brood stocks are held (ponds, cages, or tanks), their gametes can be stripped and fertilized in the hatchery.

Childers and Bennett (1961) hand spawned mature gametes from fish into petri dishes. Eggs were stripped from one or more mature females into damp petri dishes followed by stripping milt from one or more males onto the eggs (Figure 3.3).

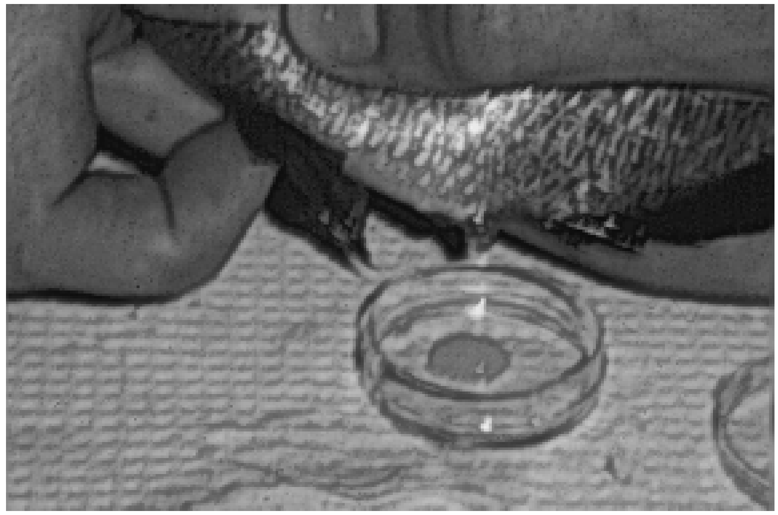
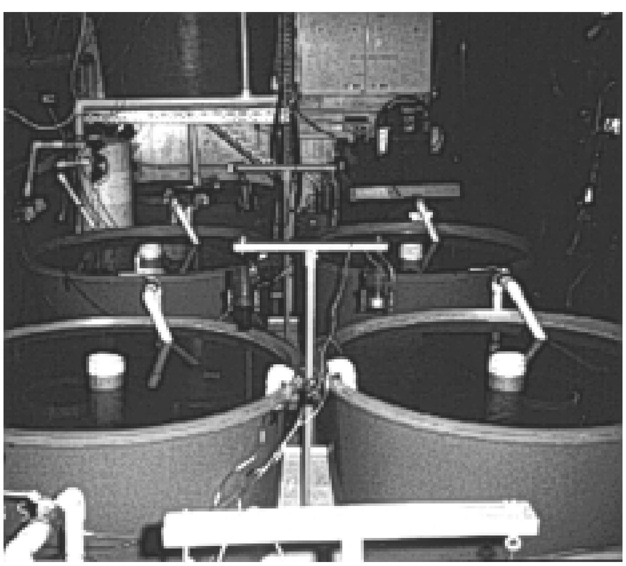


Figure 3.3. One method of reproducing sunfish in the laboratory is to hand-spawn mature gametes from the sunfish into petri dishes.

After mixing milt and eggs, water was added, and a 2 min interval allowed for fertilization to take place.

Fertilized eggs were then carefully placed into clean petri dishes containing aged tap water and allowed to water harden. Next, the petri dishes of fertilized eggs were rinsed with water and placed in aerated aquaria.



Recirculating systems are gaining popularity for fish production in areas where water resources are limited.

Childers and Bennett reported that fertilization occurred with several thousand eggs from various intergeneric crosses (i.e., crosses of different species of sunfish; hybrids). No hatching rates were given.

Toetz (1966) conducted intensive studies on the larval rearing of bluegill. [3] To acquire gametes, he captured wild bluegill and hand spawned the eggs into petri dishes. The testes of the males were extracted and cut up in a watch glass. Water was added, and the suspension then added to the eggs.

Toetz (1966) reported as high as 79% hatch when mature eggs were taken from the posterior-medial portion of the ovaries.

Temperature and photoperiod

Childers and Bennett (1961) successfully induced bluegill and their hybrids to produce gametes through manipulation of temperature and photoperiod. Banner and Hyatt (1975) induced bluegill to spawn by manipulating the temperature and photoperiod and by presenting conspecifics (males and females of the same species).

Bluegill exposed to a 16 h light: 8 h dark photoperiod at 25°C (77°F) released gametes when stripped.

The presence of male bluegill greatly increased female ovarian development. Also, nest digging activities increased when a sharp fluctuation in temperature interrupted ambient conditions appropriate for spawning.

Other experiments have been successful in artificially reproducing sunfish using similar methods (Smitherman and Hester 1962; Merriner 1971; Smith 1975).

Bryan et al. (1994) manipulated temperature and photoperiod and introduced artificial spawning nests to induce courtship and spawning of bluegill in the laboratory. After spawning, males were allowed to defend the nests for 12 h. Nests were then removed and placed into 38-L (10 gal) flow-through aquaria. At 26°C (79°F), eggs hatched in 36 h.

Larval sunfish were transferred to rearing chambers at 3 d post-hatch and fed Artificial Plankton Microcapsules® (Argent Chemical Laboratories, Redmond, Washington). At 9 d post-hatch, brine shrimp was fed 3 times daily. However, no survival rates were given.

Mischke and Morris (1997) developed a protocol for handling brood stock and out-of-season spawning for intensive culture of sunfish. They induced natural spawning of adult bluegill in the laboratory through the manipulation of temperature and photoperiod (Figure 3.4).

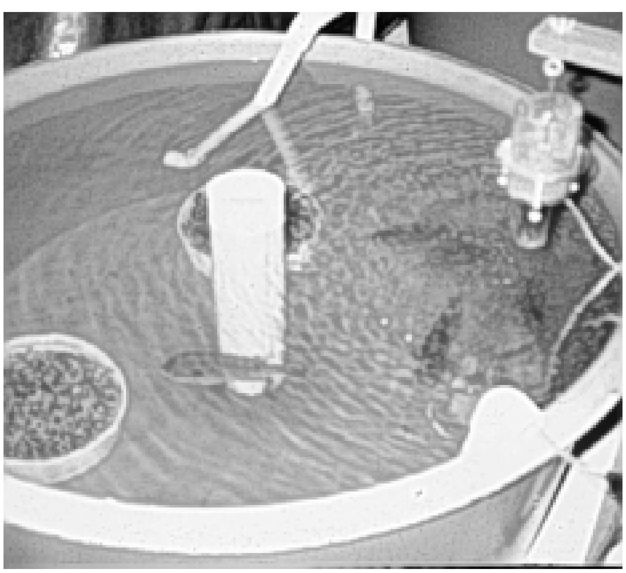


Figure 3.4. Out-of-season spawning of bluegill brood stock can be successful in indoor tanks.

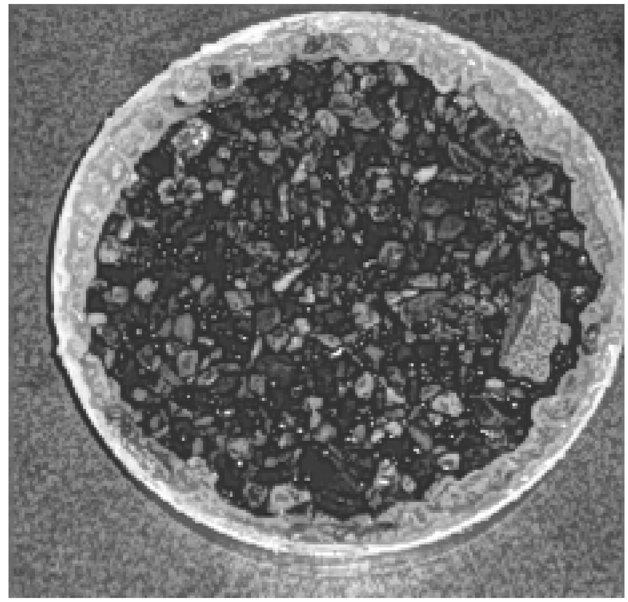


Figure 3.5. Artificial nests are used for out-of-season spawning of sunfish in indoor tanks.

Brood fish spawned multiple times on artificial nests (Figure 3.5). After spawning, the nests were transferred to a hatching tank and the eggs were allowed to hatch. Once hatched, larval fish were removed from the nests and transferred to aquaria for swim-up (Figure 3.6).

Larval sunfish require brine shrimp for an initial period before offering commercial diets. Increased growth rates and fry survival occurred when brine shrimp were fed for longer periods.

Fry Care and Feeding

If sunfish fry are held in aquaria while they absorb their yolk sacs, they should be siphoned from the aquaria before they reach swim-up. Bluegill larvae reach swim-up at 7 d post-hatch, so they should be siphoned from the aquaria just before this time (Mischke 1995).

Larvae may then be transferred to rearing chambers or tanks held at 25°C (77°F), the preferred growth temperature of larval bluegill (Beitinger and Magnuson 1979; Bryan et al. 1994), and offered feed.

The critical stage in larvae sunfish development is from 4–9 d after hatching, since they switch from endogenous (energy derived internally from the yolk sac) to exogenous (external) feeding (Toetz 1966; Smith 1976).

During this critical stage, nutrients from the yolk sac are utilized and the sunfish's mouth opens. The larval fish will starve if they do not begin feeding at this stage (Figure 3.6).

Food size

Proper feed for larval sunfish must be small enough that the fish can physically handle it. Toetz (1966) reported the mouth gape of larval bluegill at the onset of exogenous feeding to be 230–270 µm (0.009–0.011 in.). First food items must be smaller than this for sunfish to consume it.

The mouth gapes of redear sunfish and pumpkinseeds are very close to that of the bluegill. The mouth gapes of warmouth, green sunfish, and longear are larger.

Sunfish that survive the critical stage in the wild will begin feeding. Their first food consists of small plankton, including rotifers and copepod nauplii (Figure 3.7). Growth is rapid and as the mouth size increases they begin selecting larger prey items (Siefert 1972).

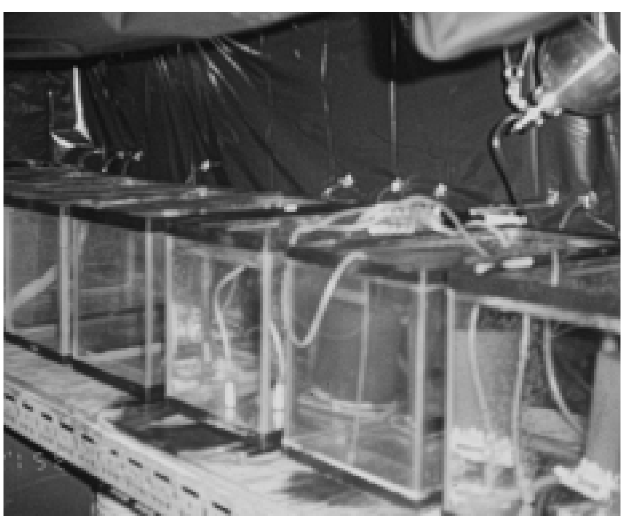


Figure 3.6. Aquaria like these are used to hold the sunfish fry until they absorb their yolk sacs and are able to accept such feeds as brine shrimp.

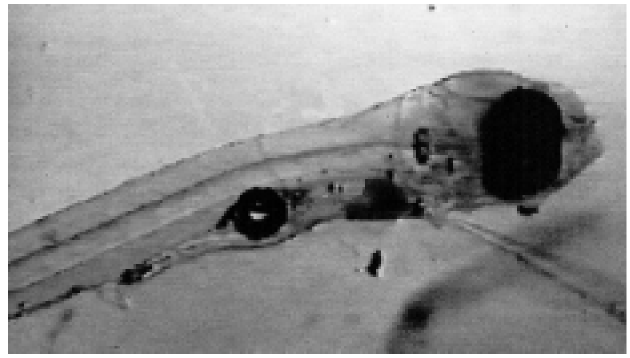


Figure 3.7. Larval bluegill begin feeding at 7 d post-hatch and must have adequate food supplies of the right size at this stage or the fish will starve.

Commercial feed

When intensively culturing sunfish fry, a commercial feed <250 µm (0.01 in.) should be used as the first feed, and progressively larger feeds offered as the sunfish grow. When changing to larger feed sizes, mix the larger feed with the smaller sized feed for a couple of feedings to allow the fish to adapt to the larger feed size.

Bryan et al. (1994) transferred larval sunfish to rearing chambers at 3 d post-hatch and fed Artificial Plankton Microcapsules® (Argent Chemical Laboratories, Redmond, Washington). At 9 d post-hatch, brine shrimp were fed to the fish 3 times daily. No survival rates were given.

Mischke and Morris (1998) transferred larval bluegill to rearing chambers at 7d post-hatch and conducted several feeding studies. They found that larval bluegill would not digest commercial feeds at the onset of exogenous feeding. However, feeding brine shrimp nauplii (newly hatched brine shrimp) (Figure 3.8) for 14 d and then weaning larvae to Fry Feed Kyowa® B-250 (Biokyowa, Incorporated, Tokyo, Japan) increased survival rates to approximately 43%.

Comparisons

If sufficient land or water resources are not available, a recirculating system may be a viable means of sunfish grow-out. Recirculating systems usually use tanks for production; therefore, much less land is required than for pond culture.

Also, recirculating systems use a fraction of the water that would be needed for pond culture. Through reuse and treatment of water, recirculating systems use $<$10% of the water required by ponds to produce similar yields (Losordo et al. 1992).

There are, however, higher fixed costs associated with recirculating systems than with pond production systems (e.g., pumping costs and oxygenation). Recirculating systems require a higher level of management than pond production systems.

Unlike pond production, a truly complete diet is required for sunfish in recirculating systems. Crude protein levels required for recirculating are estimated to be $\geq$40% (NCRAC 1998).

Stocking densities for sunfish grow-out in recirculating systems have not been adequately determined at this time.

Summary

Even though pond culture is the most common culture method for sunfish (both for spawning and grow-out), there are many culture methods available. Sunfish may be suitable for grow-out in cages and recirculating systems. There are also encouraging results of intensive, out-of-season spawning techniques for sunfish in the laboratory.

A combination of these methods may be desirable; for example, use out-of-season spawning in a recirculation system and then grow-out in cages or ponds.

Individuals should choose culture methods most suited to production needs and geographic area. Land, labor, and capital restraints must also be considered.

Very specific requirements for feeding and stocking for first and second year grow-out are not yet available, but the general values given here can be used as a starting point and refined for individual requirements.

Figure 3.8. Culturing brine shrimp to feed to first-feeding sunfish can increase survival rates.

Notes

4

Brood Stock Management

Careful sunfish brood stock management is essential to ensure quality of stock and the highest level of reproduction. The purity of a particular brood stock species begins with careful selection and identification of the fish, depth and filtration of ponds, and amount and size of feed provided. Research into hormone use may reveal ways to maintain desired production levels.

This chapter explains:

- Field collection methods
- Species and sex determination
- Pond management
- Hormone research

4

Brood Stock Management

Authors: C. C. Mischke and J. E. Morris, Iowa State University

Field Collection Methods

The type of field method used to collect species depends on the individual and the collection purpose. Each type, however, requires the proper equipment and diligence. There are three major collection types:

- Electro fishing
- Trap nets
- Seine nets

Nets

Sunfish brood stock collected using electro fishing equipment, trap nets, seine nets (Figure 4.1) or other types of nets, require proper permits that are acquired from natural resource agency personnel.

It is important to make certain the proper steps are taken and any permits necessary are acquired.

Anesthetize net-captured fish immediately in 19 L (5 gal) of water with 1.5 g (0.05 oz; 80 ppm) of Finquel® (Argent Chemical Laboratories, Redmond, Washington).

Electro fishing

Place fish captured by electro fishing in fresh water and allow them to recover from the stress of electro fishing for approximately 1/2 h before anesthetizing.

After capture, brood stock can be stripped of gametes (eggs and milt) and either released or kept for future brood stock. Keep in mind that if Finquel® is used, there is a 21-d withdrawal time for bluegill >7.6cm (3 in.) in length.

Egg and sperm collection

When fish are stripped of gametes on site, remove the fish from the anesthetic and dry the urogenital pore with a soft cloth to prevent the anesthetic from interfering with fertilization.

Then strip the eggs from ripe females into a damp glass petri dish by applying gentle pressure on the abdomen. Immediately fertilize the eggs by stripping the milt from one or more males directly onto the eggs.

After stripping the eggs and milt:

- Return the fish to the pond.
- Swirl petri dish vigorously to mix gametes.

Figure 4.1. Seining is one possible method of sunfish brood stock acquisition if proper permits are obtained.

- Wait 2 min to allow fertilization to take place (Childers and Bennett 1961).
- Pour the fertilized eggs (in lots of 200–300) into clean glass petri dishes filled with aged tap water.
- Allow approximately 15 min for the eggs to water harden.

Once the eggs water harden:

- Wash the fertilized eggs by lowering and raising the petri dishes 4–5 times in a large beaker of dechlorinated tap water.
- Place petri dishes in aquaria with gentle aeration after washing, or
- Add enough water to each petri dish to cover the fertilized eggs, and
- Replace the water approximately 6 times each day during incubation.

Transportation

If the adult sunfish are kept for future brood stock, take care in their transport and acclimation to their new environment.

Fish should be transported in water with plenty of oxygen and 1% NaCl (uniodized salt) to reduce osmoregulatory (physiological regulation of internal water/solute concentration) stress.

Once fish arrive at the holding facilities, they must be acclimated to the different water chemistry and temperature. Do this by slowly exchanging hauling water with system water.

Species and Sex Determinations

It is very important to identify different sunfish species, especially when trying to produce hybrids. Table 4.1 contains characteristics of the most common sunfish.

Identification of hybrid sunfish can be difficult because hybridization results in fish with multiple species characteristics. Pflieger (1975) and Tomelleri and Eberle (1990) provide keys, description, and illustrations for more specific identification of sunfish species.[1]

Physical identification

Identifying the brood stock sex is critical when producing any hybrid to ensure stocking proper male to female ratios.

The ease of sexing brood fish is a function of the time of year. As with most fish, the sunfish are more easily sexed during the breeding season.

As the spawning period nears, the males take on distinctive, brilliant spawning colors, and milt is usually easy to express from the vent (Dupree and Huner 1984).

Females have a much fuller and rounder abdomen than males during the breeding season (Figure 4.2).

A mature male bluegill's urogenital opening usually terminates in a small, funnel-shaped pore (McComish 1968). The area around the opening tends to be darkly pigmented. These features help to identify male from female.

Figure 4.2. Observations of the urogenital openings of adult sunfish can help to determine the sex of the fish.

According to Brauhn (1972), male bluegill characteristics include:

- Square, heavily pigmented opercula lobe
- Black pigmented gular area
- General dark cast to the body
- Definitive spot at the posterior base of dorsal fin

The female's urogenital opening resembles a small, swollen doughnut-like ring, probably the result of a slight eversion of the urogenital tract (McComish 1968).

According to Brauhn (1972), other female characteristics include:

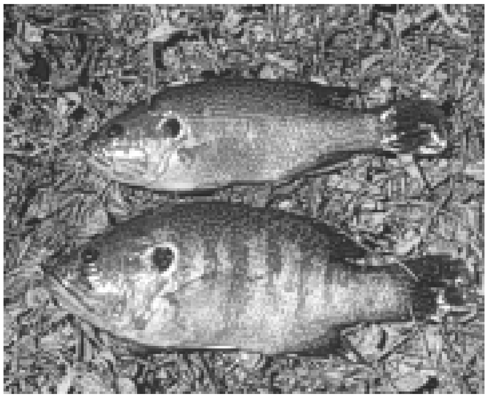
- Rounded, less pigmented opercula lobe
- Yellow pigmented gular area
- General light appearance to the body
- Reduced spot at the posterior base of dorsal fin.

Extraction identification

Another method of sexing brood fish is probing for eggs (extraction identification). First, use

Table 4.1. Key to sunfish identification. (Adapted from Pflieger 1975)

	Warmouth	Green Sunfish	Redear Sunfish	Pumpkin-seed	Longear Sunfish	Bluegill
Species	*Lepomis gulosus*	*Lepomis cyanellus*	*Lepomis microlophus*	*Lepomis gibbosus*	*Lepomis megalotis*	*Lepomis macrochirus*
Relative Mouth Size	Large	Large	Small	Small	Moderate	Small
Pectoral Fin		Short, round	Long, pointed	Long, pointed	Short, round	Long, pointed
Gill Rakers		Long, slender	Short, stout	Short, stout	Short, thick	Long, slender
Rear Margin of Bony Gill Cover		Hard, inflexible	Thin, flexible	Thick, inflexible	Thin, inflexible	Thin, flexible
Ear Flap	Tipped with bright red in breeding males	Black with whitish yellow margin	Black with whitish border and prominent orange/red spot	Never prolonged, light-colored border	Considerably elongated	Entirely black
Back and Size Coloration	Rich olive-brown with numerous dark brown mottlings	Bluish-green with emerald and yellow reflections	Golden or light-olive green	Golden or light-olive green	Blue-green speckled with yellow and emerald	Dark olive-green with emerald and brassy reflections
Belly Coloration	Light yellow	Pale yellow or white	Yellow to orange-yellow	Yellow to orange-yellow	Yellow to orange	Reddish orange
Other Characteristics	Red iris in eye, patch of teeth on tongue			Wavy blue lines on cheek		Distinct black blotch in base of soft dorsal fin



Male and female green sunfish. The male often appears smaller. The female shows the typical swollen, doughnut like ring.

gentle pressure on the abdomen, palpating from the middle of the abdomen back to the vent.

The fish is a male if milt (white liquid) is expelled from the urogenital opening. If no milt appears, the fish is probably a female; check with a capillary tube to be certain.

A 1.1–1.2 mm (0.043–0.047 in.) wide capillary tube 5–10 cm (2–4 in.) in length should be used (Figure 4.3). Hold the fish upside down and gently insert the capillary tube through the urogenital opening.

Once the capillary tube is inserted:

- Angle it back towards the tail and slightly to one side.
- Gently insert the tube through the oviduct and into the ovary.
- Use gentle force and slightly twirl the tube back and forth to help insertion.
- When the tube is inserted, place a finger over the end of the tube and remove it.

If eggs are seen in the tube, the fish is a female. If no eggs are present and no milt was seen from palpation of the abdomen, then a certain sex determination cannot be made, and the fish should not be used.

Pond Management

Because interbreeding is common in this fish family, it is important in sunfish brood stock management to *not* hold brood fish in ponds that are contaminated with other sunfish. Therefore, ground water should be used for filling sunfish ponds; or, if surface water is used, it should be filtered to prevent introduction of undesirable fish.

Filtration and depth

A common and simple method of filtration of surface waters is the use of a Saran® or nylon sock filter. The filter material should have approximately the same size mesh as mosquito netting to prevent admission of larval fish into the pond. Due to the small mesh size, daily to weekly cleaning is required (McLarney 1987). Also, before filling and stocking brood fish ponds, ponds should be dried thoroughly and all depressions treated with an appropriate fish toxicant.

The preferred depth of brood fish ponds is from 0.9–1.5 m (3–5 ft) with some shallow areas 0.3

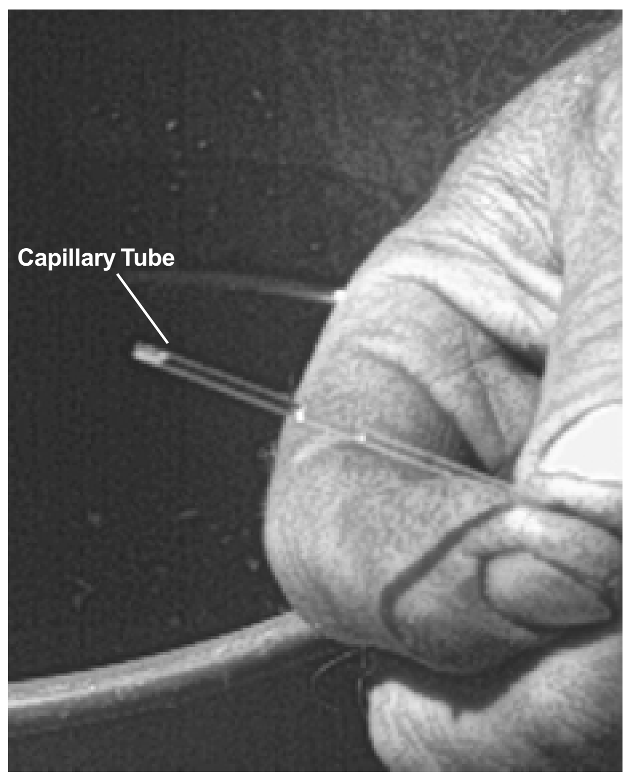


Figure 4.3. Insertion of a capillary tube into the sunfish urogenital opening is the most reliable method for sexing adult fish.

m (1 ft) in depth (Higgenbotham et al. 1983). Fish should be stocked at a rate of 100 pairs/ ha (40 pairs/ acre) and a ratio of one male to one female (Dupree and Huner 1984; Engelhardt 1985; Stickney 1985).

Floating feed allows the producer to observe fish on a regular basis.

Food size and amount

It is important for brood stock to be in good physical condition for maximum spawning success. High quality dry feeds should be fed several times daily.

When >225 kg/ha (200 lbs/acre) standing crop is maintained, brood fish should be offered a pellet feed 3.2–6.2 mm (1/8–1/4 in.) in diameter to supplement available natural feed, according to Dupree and Huner (1984). A floating feed is preferred to a sinking feed because floating feed allows the producer to observe fish on a regular basis.

The amount of feed depends on the water temperature. When temperatures are above 21°C (70°F), feed brood stock 5–7 times per week at a rate of 3% of the standing crop.

At temperatures from 13–21°C (55–70°F), feed the brood stock on alternate days at a rate of 1–2% of the standing crop; and when water temperatures are <13°C (55°F) do not feed the brood stock (Dupree and Huner 1984).

Hormonal Injections

The use of hormones , pending INAD approval, could also be useful for induction of sunfish spawning. A small amount of research has been done in this area; however, its priority may increase in the future.

The United States Food and Drug Administration (USFDA) has recently approved Chorulon®, an HCG product, as a new animal drug for male and female brood fish under the direction of a licensed veterinarian (NCRAC 1999).

Neal (1961) injected bluegill with combinations of human chorionic gonadotropin (HCG) and mammalian follicle stimulating hormone (FSH). Results showed that FSH alone did not increase gonadal weight, but gonadal weight did increase using a FSH-HCG combination.

Additional findings

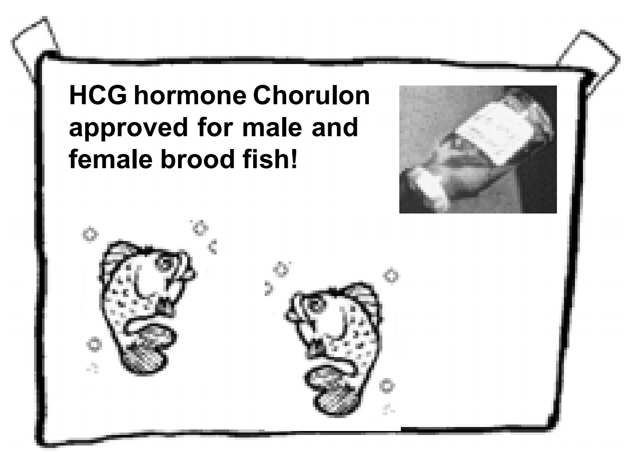
Other research on sunfish should be applicable to bluegill. Sneed and Dupree (1961) injected gravid green sunfish with a thyroid-stimulating hormone (TSH), HCG, and acetone-dried buffalo pituitaries.

Three injections of 225 international units (IU) of HCG combined with 2, 3, and 5 units of TSH per pound of body weight resulted in the ovulation of the fish. However, it was found that HCG or pituitary extract alone would not cause ovulation.

Leutinizing hormone-releasing hormone (LH-RH) in other fish has been used at the rate of 0.1 mg/kg (0.0000015 oz/lb) of fish body weight (Argent Chemical 1975), but a specific amount for bluegill was not given.

The hormone was injected into the intermuscular tissue, but the hormone alone did not cause the fish to reproduce. The careful combination of photoperiod, water, temperature, and proper nutrition was also required.[2]

Mischke et al. (2001) noted success in spawning both bluegill and hybrid sunfish using



both a combination of water temperature and photoperiod.

Summary

Because sunfish species are so similar, the species and sex of the fish is extremely important when managing brood stock of sunfish. Sunfish species can be determined using various keys.

Sex of sunfish is best determined during the spawning season by probing with capillary tubes. In general, sunfish do spawn quite readily; however, there has been research on hormonal injection to facilitate gamete production.

Notes

5

Hybridization

Hybridization is increasingly recognized as a viable part of fish management and aquaculture production. Fertility considerations, growth rates, and vigor give hybrids a potential advantage as a food fish and use in pond management.

This chapter explains:

- Hybrid importance
- Historical hybrid research
- Hybrid production methods

5

Hybridization

Authors: C. C. Mischke, G. D. Dvorak, and J. E. Morris, Iowa State University

Hybrid Importance

Hybrids are increasingly recognized as important to overall fish management. Centrarchid sunfish hybrids (Figure 5.1) show potential for aquaculture and fishery management because:

- Sunfish are extremely fecund, each female producing an average of 80,000 eggs/year in several successive spawns (Carlander 1977).
- Sunfish communities can become overpopulated and stunted quickly.

Many hybrid sunfish are used to control population through:

- Their reduced reproductive potential.
  - The result of skewed sex ratios
  - Predominately males
- Their abnormal reproductive behavior.
  - (Krumholz 1949; Childers and Bennett 1961; Heidinger and Lewis 1972; Lewis and Heidinger 1971).

Hybrid sunfish also exhibit:

- Hybrid vigor with improved growth.
  - (Childers 1967; Kurzawski and Heidinger 1982; Brunson and Robinette 1983; Engelhardt 1985)
- High acceptance of artificial feeds.
  - Lewis and Heidinger 1971; Brunson and Robinette 1983; Tidwell et al. 1992)
- Greater tolerance to cooler water and poor environmental conditions.
  - Heidinger 1975; Brunson and Robinette 1983)
- High vulnerability to angling.
  - (Kurzawski and Heidinger 1982; Engelhardt 1985; Brunson and Robinette 1986).

These characteristics make hybrid sunfish good candidates for aquaculture, especially in the Midwest.

Figure 5.1. Common sunfish hybrids include the various crosses between bluegill (top) and green sunfish (bottom).

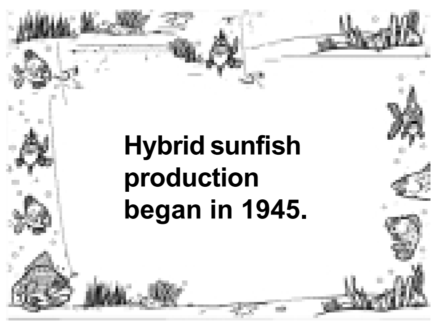
Historical Hybrid Research

The earliest research on stocking hybrid sunfish for population control was done by Ricker (1945). He found female redear sunfish x male bluegill (RxB) resulted in only 2% females, good growth rates, and excellent stocking in small ponds.

Krumholz (1949) also observed that in small ponds stocked with hybrid sunfish, RxB hybrids exhibited faster growth and were relatively heavier for their length when compared to parental stocks.

Childers and Bennett (1961) completed crosses between bluegill, redear sunfish, and green sunfish. They found:

- Female bluegill x male green sunfish (BxG) produced significant numbers of F_1 offspring naturally.
- Female green sunfish x male redear sunfish (GxR) will produce significant numbers of F_1 offspring naturally.
- Only female redear sunfish x male green sunfish (RxG) F_1 hybrids exhibited a 50:50 sex ratio.
- All others had >70% males.



They found female green sunfish x male bluegill (GxB), RxB, and the female bluegill x male redear sunfish (BxR) F_1 hybrids did not reproduce.

Heidinger (1975) investigated the growth of hybrid sunfish (GxB) at low temperatures [<15°C (59°F)] when stocked with channel catfish. He found that, in all cases, hybrid sunfish gained weight while channel catfish lost weight. Heidinger concluded, on the basis of temperature requirements, hybrid sunfish are better adapted for colder climates than channel catfish.

In Mississippi, Brunson and Robinette (1983) looked at hybrid sunfish growth at low temperatures for 112 d with an average temperature of 10.4°C (50.7°F). They compared winter growth of young-of-the-year bluegill to that of the GxB hybrid. They found that hybrids had increased weight and length compared to bluegill; they also outgrew bluegill by a ratio of approximately 2:1.

Intermediate characteristics

The intermediate nature of hybrids has been reported in several studies. Etnier (1971) collected female green sunfish x male pumpkinseed hybrids and GxB hybrids and their parental species from three lakes in north-central Minnesota. He found that both hybrids had larger mouths and larger food organisms in their stomach than either the bluegill or pumpkinseed, but not green sunfish.

Green sunfish have larger mouths than either bluegill or pumpkinseed. The female pumpkinseed x male bluegill hybrid was found to be intermediate in many meristic measurements, including their pharyngeal arches and teeth (Heckman 1969). Smitherman and Hester (1962) found intermediate characteristics of hybrids between all crosses of bluegill, redear sunfish, and the redbreast sunfish.

Reproduction findings

Increased growth rate is primarily attributed to the reduced fertility of hybrids, which also helps to eliminate the overpopulation frequently associated with sunfish populations (Krumholz 1949; Childers 1967). With less energy diverted to reproduction, more energy can then be used for growth.

Some F_1 hybrids were fertile (Ricker 1945; Laarman 1979), but they typically showed low fecundity and highly skewed sex ratios, which limits their reproductive potential. The fecundity of bluegill females was reported to be 280 times greater than GxB F_1 hybrid females (Laarman 1979).

Some hybrids were reproductively isolated from their parental species. Brunson and Robinette (1987) attempted to backcross F_1 GxB hybrids with females of each parental species in both ponds and the laboratory but were unsuccessful.

In the lab experiment, fertilization was accomplished, but the embryos failed to survive. They concluded that gametes of the hybrids might not have been compatible with parental gametes.

Hybrid Production Methods

There are two methods used to produce hybrid sunfish: stripping and fertilizing eggs in the hatchery (Figure 5.2) or stocking parent species and allowing them to spawn, usually in ponds.

Method 1

Childers and Bennett (1961) and Smitherman and Hester (1962) described this laboratory method for stripping and fertilizing eggs:

- Select females of the desired sunfish species containing mature gametes.
- Hand spawn them into damp, glass petri dishes. Note:
  - Petri dishes must be glass in order for this technique to work properly.
  - Fish are considered to contain mature gametes if their eggs or milt are easily extruded using gentle pressure on the abdomen.
- Dip the petri dishes into clean, de-chlorinated water.
- Shake vigorously to remove excess water.
- Hold females with their vents over the petri dishes and strip by gentle stroking motions with the fingers on either side of the abdomen toward the vent.
- After eggs are stripped into the petri dishes, strip milt from one or more males onto the eggs using the same stroking motions used for stripping the eggs.

When milt and eggs are stripped:

- Mix by vigorously swirling the petri dish.
- After mixing, add de-chlorinated water.
- Allow 2 min for fertilization to occur.
- Place the fertilized eggs into clean glass petri dishes containing aged tap water, and
- Allow the eggs to become water hardened (ca. 15 min).
- Wash the eggs by raising and lowering the petri dish several times in a container of aged tap water. Note:
  - Fertilized eggs adhere to the petri dishes.
  - Non-fertilized eggs will not.
- Place the petri dishes of fertilized eggs into aerated aquaria.

Hatch time

Eggs should hatch from 41–46 h after fertilization, depending on the photoperiod and temperature. As the amount of light and temperature are increased, hatching time is decreased (Toetz 1966).

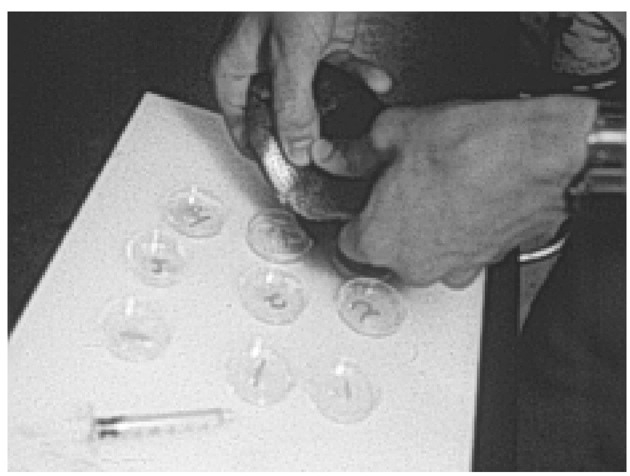


Figure 5.2. Stripping and fertilizing eggs in the laboratory is a common method of hybrid sunfish production.

Method 2

The second method of producing hybrid sunfish is to stock parent species in empty ponds. To prevent contamination with undesirable sunfish:

- Dry the spawning pond thoroughly.
- Treat all depressions with approved fish toxicants before filling and stocking with brood fish.

Filtration process

Water from surface sources must be filtered to prevent introduction of undesirable fish. As discussed in Chapter 4, a common and simple filtration method of surface waters is use of a Saran® or nylon "sock" filter.

The filter material should have approximately the same size mesh as mosquito netting to prevent admission of larval fish because of the small mesh size, daily to weekly cleaning is required (McLarney 1987).

Area required

Dupree and Huner (1984) found that ponds <0.4/ha (1/acre) in area are preferable for hybrid production. After the pond has been properly prepared:

- Select mature male and female sunfish.
- Stock them into the ponds.

Stock parent fish at a 1:1 ratio of males to females suggest Dupree and Huner (1984) and Engelhardt (1985).

They also suggest stocking rates of:

- 100 pairs/ha (40 pairs/acre) for small brood fish.
- 75 pairs/ha (30 pairs/acre) for large brood fish.

Spawning temperatures

Spawning activity begins when the water temperature reaches:

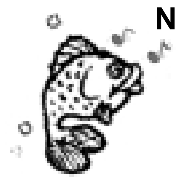
- 21°C (70°F) for green sunfish.
- 24°C (75°F) for redear sunfish.
- 27°C (80°F) for bluegill.

Spawning continues as long as the temperature remains above these levels (Dupree and Huner 1984).

Summary

There is interest in production of hybrid sunfish because of the potential reduction in fecundity, generally due to skewed sex ratios of some sunfish crosses. Hybrids can also show hybrid vigor and grow faster than either parent species.

Many sunfish crosses have been considered; however, the most common crosses are between bluegill and green sunfish, and bluegill and redear sunfish.



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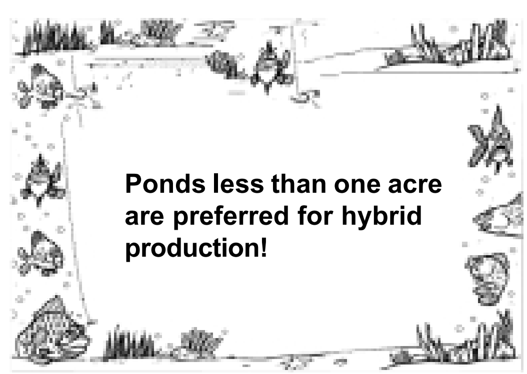


6

Water Temperature Influences on Survival & Growth

Various experiments show that external environments profoundly affect fish body temperature. In turn, this affects all growth components for fish considered for aquaculture.

This chapter explains:

- Environmental effects
- Growth and survival rates

6

Water Temperature Influences on Survival and Growth

Authors: C. C. Mischke and J. E. Morris, Iowa State University

Environmental Effects

Fish are poikilothermic animals, which means that their body temperature fluctuates with the external environment; therefore, temperature profoundly affects their physiology.

All components of growth are influenced by temperature (Kitchell and Windell 1968; Beitinger and Magnuson 1979), including:

- Food consumption
- Digestion rate
- Maintenance
- Metabolism
- Specific dynamic action
- Effective food conversion

Consequently, most fish species have an optimum temperature for growth and survival.

Sunfish prefer to build nests in the warmer temperatures of shallow waters. Optimal water temperature for sunfish fertality and growth is 20–30°C (68–86°F).

Temperature ranges[1]

Research shows that temperature affects fish both in fertility and growth rates. Researchers report the optimal temperature range for sunfish is 20–30°C (68–86°F) (Breder 1936; Banner and Hyatt 1975; Carlander 1977). Most sunfish spawn when temperatures are within this range (Carlander 1977).

Growth rates for sunfish generally increase as temperatures increase up to approximately 30°C (86°F). They decrease as temperatures increase above 30°C (86°F) (Carlander 1977; Lemke 1977; Beitinger and Magnuson 1979).

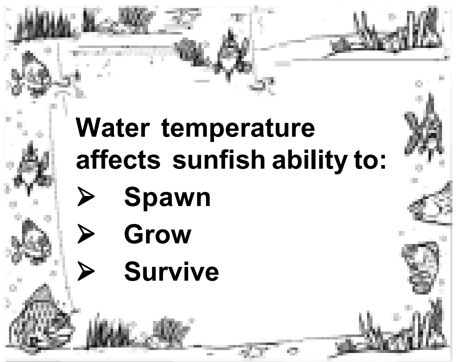
Growth and Survival Rates

Temperature affects fish capacity to survive and grow. The highest mean specific growth rate (2.35%/d) occurred at 30°C (86°F) from bluegill grown for 30 d at 2°C (4°F) temperature increments from 20–36°C (68–97°F) reported Lemke (1977).

Growth of bluegill is the greatest at 31°C (88°F), but not significantly different from fish at 25°, 28°, and 31°C (77°, 82°, and 88°F) according to Beitinger and Magnuson (1979).

Temperature not only affects sunfish growth, but also impacts survival. Bluegill eggs hatch at temperatures ranging from 23–34°C (73–93°F).

Banner and Van Arman (1973) successfully hatched bluegill eggs at 34°C (93°F), but 50% mortality occurred. They also reported eggs



Notes

at different temperatures had a maximum percent hatch at 22°C (72°F) in one experiment and a maximum percent hatch at 24°C (75°F) in a second experiment.

Fry have a greater thermal tolerance range than eggs, and juveniles have a greater thermal tolerance than fry or eggs (Banner and Van Arman 1973). Wrenn et al. (1979) found a hatching success mean of 95% (range 76–99%) for bluegill eggs within the 23–34°C (73–93°F) temperature range.

Summary

Water temperature has a profound influence on fish ability to spawn, grow, and survive. Optimal growth of sunfish is reported to be at water temperatures of 18–23ºC (64–73ºF). These temperatures have the most positive influence on all components of fish physiology.

7

Industry Status

Currently, a limited number of aquaculture producers exist. The industry has plenty of room for new producers. However, more information needs to be available on the most successful aquaculture production techniques.

This chapter explains:

- Industry status
- Locations
- Primary markets

7

Industry Status

Authors: C. C. Mischke and J. E. Morris, Iowa State University

Industry Status

Sunfish culture, on a commercial scale, is a very small industry at this time. Because the number of sunfish producers is small, the industry is wide open for new producer entry.

As the global world shrinks and the population expands, new food sources and production methods are needed.

Most common sunfish species

Bluegill is the most commonly raised sunfish species for a number of reasons talked about in-depth elsewhere in this guide. These prolific fish are also adaptable to environmental conditions and lend themselves well to certain hybrid varieties.

Most common culture methods

Pond culture is currently the most commonly used culture method. However, cages, raceways, and recirculating systems are also used.

According to a 1999 NCRAC survey, a variety of sunfish species and hybrids are cultured, but bluegill remains the most commonly cultured sunfish species. Most sunfish culture occurs from the Midwest east and south.

Locations

Very few producers are located in western United States. Table 7.1 summarizes the current number of sunfish producers reported by the Aquaculture Buyer's Guide (Aquaculture Magazine 1999).[1]

Of the listed producers of sunfish, most were located in Texas.

The North Central Regional Aquaculture Center (NCRAC) conducted a survey of state aquaculture contacts to determine the number of sunfish producers in each state, the specific species produced, the culture methods used, the most commonly used markets, and the prices received. Table 7.2 gives the number of producers in each state.

As with the Aquaculture Buyer's Guide listing, most of the commercial sunfish producers are located in Texas and Wisconsin. Many of the surveys were not returned, but it is clear that the sunfish industry is small and very dispersed throughout the U.S.

Primary Markets

The primary market for sunfish is sport-fish stocking and fee-fishing operations. Some states did report a market for food fish, bait, and scientific research use.

Summary

There are a small number of sunfish producers in the U.S., with the majority located in Wisconsin and Texas. Because of the relatively small number of sunfish producers, there is ample opportunity for entry into the industry by new aquaculture producers.

However, the limited number of producers means there is limited information on proven production techniques on a commercial scale.

Notes

Shrinking world + expanding population = need for new food sources that are:

- **Plentiful**
- **Practical**
- **Profitable**

Table 7.1. The number of states in the U. S. with sunfish producers and the total number of sunfish producers as listed in the 1999 Aquaculture Buyer's Guide, Aquaculture Magazine.

Species Cultured	Number of States with Producers	Total Number of Producers
Bluegill	31	65
Green Sunfish	7	8
Redear	16	25
Pumpkinseed	4	4
Longear	4	5
Coppernose	5	11
Hybrid Bluegill*	8	
Fingerlings	4	13
Market Size	4	5
Hybrid Sunfish*	11	13

*The specific species cross was not given.

Table 7.2. The number of commercial sunfish producers, reported by state aquaculture extension specialists and aquaculture coordinators, 1999.

State	Number of Commercial Producers	State	Number of Commercial Producers
Alabama	4	Montana	NR
Alaska	0	Nebraska	NR
Arizona	1	Nevada	NR
Arkansas	NR	New Hampshire	0
California	NR	New Jersey	1
Colorado	0	New Mexico	0
Connecticut	NR	New York	0
Delaware	0	North Carolina	7
Florida	0	North Dakota	NR
Georgia	NR	Ohio	43
Hawaii	0	Oklahoma	NR
Idaho	20	Oregon	NR
Illinois	NR	Pennsylvania	NR
Indiana	0	Rhode Island	0
Iowa	2	South Carolina	0
Kansas	8	South Dakota	0
Kentucky	3	Tennessee	6
Louisiana	8	Texas	75
Maine	0	Utah	2
Maryland	20	Vermont	0
Massachusetts	NR	Virginia	1
Michigan	27	Washington	0
Minnesota	NR	West Virginia	3
Mississippi	NR	Wisconsin	80
Missouri	20	Wyoming	NR

NR = state aquaculture specialists did not respond to survey.

8

Production of Polyploid Sunfish

Polyploid sunfish are those with three or more full sets of chromosomes rather than the usual two. This type of sunfish has the potential to overcome current barriers limiting sunfish use in aquaculture.

This chapter explains:

- Sunfish aquaculture positives and negatives
- Overcoming obstacles
- Polyploidy, triploid, and tetraploidy induction techniques

8

Production of Polyploid Sunfish

Authors: D. L. Garling, P.D. Wilbert, A.R. Westmaas, and S.M. Miller, Michigan State University; R. Sheehan, P. S. Wills, and J.M. Paret, Southern Illinois University—Carbondale

Sunfish Aquaculture

Many factors make sunfish strong aquaculture candidates for commercial production in the Midwest. The bluegill sunfish is the most frequently pursued game fish in the United States, particularly by first time anglers and youth. Sunfish are also desirable as a food fish in Midwest markets.

Positive characteristics

Sunfish brood stock are easy for culturists to acquire in high numbers, in contrast to such species as the walleye and striped bass. Less experienced culturists find that sunfish readily spawn in ponds, which is in contrast with other types of fish. Sunfish are also not inclined to be cannibalistic, like the striped bass and walleye.

Polyploid sunfish could overcome barriers limiting sunfish in aquaculture.

A number of sunfish species and hybrids are known to readily accept pellet feeds, which is important for survival. Sunfish also exhibit fast initial growth and steady growth at lower temperatures than channel catfish, which is an important characteristic for production in cooler regions. Sunfish are raised in the Midwest for pond stocking and in limited numbers for food fish.

Negative characteristics

Despite these important positive attributes of sunfish, several factors have slowed progress towards widespread commercialization of these fish for the food fish market.

The development of a food fish production industry has been impeded because sexual maturation occurs prior to the attainment of marketable sized fish. Sexual maturation leads to slow, inefficient growth and uncontrolled reproduction. These same problems affect their value as a sport fish in recreational ponds.

Sunfish hybrids

To limit reproduction in bluegill sunfish, fish culturists developed sunfish hybrids. The most commonly available sunfish hybrid is a cross between the green sunfish female and the bluegill male.

The first generation (F_1) of this cross, although sexually fertile, is predominantly male. In some populations, males comprise greater than 95% of the population; but this number can vary greatly (Table 8.1). However, production of second-generation (F_2) progeny can occur despite skewed sex ratios.

This hybrid can also back cross with parental species. A considerable amount of energy that could go into growth is expended in such

Table 8.1. Literature reported sex ratios of GxB hybrids.

Cross	Broodstock Source	% Males	Reference
	Michigan	81	Hubbs and Hubbs (1933)
	Michigan	87	Laarman (1973)
	Central Illinois	97	Childers and Bennett (1961)
G x B	Southern Illinois	80	Ellison and Heidinger (1978)
	Texas	66-78	Crandall and Durocher (1980)
	Mississippi	95	Brunson and Robinette (1987)
B x G	Central Illinois	64-70	Childers (1967)
	Southern Illinois	71	Lewis and Heidinger (1971)

prespawning behaviors as nest building and aggressive territoriality among males. These behaviors may affect the density at which sunfish can be reared.

Overcoming Obstacles

Induced polyploidy in sunfish could potentially overcome many current obstacles to the development of food fish aquaculture in the Midwest. Polyploidy refers to the condition of having three or more full sets of chromosomes instead of the two found in normal diploids (Table 8.2). Triploid fish produce few, if any, sperm or eggs because they have three instead of the normal two sets of chromosomes.

Induced triploidy in other species resulted in fish with reduced testes and ovaries development and delayed sexual maturation. If triploid sunfish exhibit similar characteristics, problems associated with the onset of sexual maturation before attaining market size, such as the loss of dietary energy to gonadal development and spawning behaviors, will diminish.

Efficient growth

Efficient growth through sexual maturation is especially important for sunfish because the market value greatly decreases for fish less than 0.51 kg (0.33 lb). Induced triploidy could promote good growth and feed conversion through market size.

Induced triploidy could overcome problems of uncontrolled reproduction since few, if any, triploid individuals produce sperm or eggs. Any sperm or eggs produced usually fail to create viable young. Ponds that contain only triploid bluegill sunfish would, consequently, not stunt as a result of overpopulation.

Tetraploidy was induced in a variety of fish, including the bluegill sunfish, rainbow trout,

Table 8.2. Ploidy terminology where n is the number of complete sets of chromosomes and x is the number of chromosomes for the bluegill sunfish.

Ploidy Term	Cell Type	n	x
haploid	egg or sperm	1	24
diploid	body cells	2	48
triploid	body cells	3	72
tetraploid	body cells	4	96

and tilapia. Tetraploid trout crossed with diploids produce viable offspring at lower survival levels than crosses between normal diploids. However, triploid trout produced by tetraploid x diploid crossings out performed triploid trout produced by heat shocks.

Under ideal production conditions, tetraploid bluegill sunfish can be used to produce triploids through tetraploid x diploid crosses, eliminating the need to treat eggs separately.

Polyploidy Induction Techniques

Michigan State University (MSU) and Southern Illinois University-Carbondale (SIUC) worked jointly (funded by the NCRAC) to develop methods to induce and evaluate polyploidy in bluegill sunfish and its hybrid with green sunfish.

MSU researchers were the first to produce triploid and tetraploid bluegill using cold shocks. SIUC researchers were the first to produce triploid hybrid sunfish.

They subsequently produced triploid bluegill using hydrostatic pressure shocks and refined flow cytometry methods that facilitate ploidy determinations in larval sunfish.

SIUC researchers evaluated several shock types, magnitudes, and durations and found that hydrostatic pressure shocks were superior to temperature shocks: Pressure shocks produced high survival (>90%), 100% triploidy, and no deformed fish.

Methods recently developed at Iowa State University (ISU) reliably reproduce bluegill sunfish out-of-season. They are then reared from hatch to juvenile size under laboratory conditions (Mischke and Morris 1997, 1998). These advances significantly enhanced the commercial potential for sunfish production.

Three Chromosomes Sets

Triploids possess three rather than two sets of chromosomes and are created by interrupting the process of meiosis. Meiosis is the type of first cell division that results in production of either

Table 8.3. Polyploidy induction techniques for sunfish.

Species	Ploidy	Method, Initiation, t/p[1], Duration	% Induction	% Survival rtc[2], Stage	Reference
Largemouth Bass	3N	pressure, 5 min, 55,158 kPa (8000 psi), 1 min	100	55, prehatch	Garrett et al. (1992)
Bluegill	3N	pressure,1-1.5 min, 55,158 kPa (8000 psi), 5 min	100	72, hatch	Westmaas (1992)
Bluegill	4N	cold, 40 min, 7.5ºC (45.5ºF),15 min	40	<1, 17-12 d	Miller (1995)
Hybrid Sunfish (B x G)	3N	pressure, 2 min, 48,264 kPa (7000 psi), 4 min	100	90, hatch	Wills et al. (1994)
White Crappie	3N	cold, 5 min 5ºC (41ºF), 60 min	72-92	33-40, hatch	Baldwin et al. (1990)

[1] t/p = temperature or pressure used in the stock treatment
[2] rtc = % survival relative to control = (treatment survival/control survival) X 100

eggs or sperm.

The second meiotic division of the egg is completed shortly after a sperm enters an egg. Since fertilization is external in sunfish, application of a treatment to inhibit the second meiotic division is possible.

The second meiotic division may prevent any process that causes depolymerization of microtubules essential to formation of the spindle apparatus. The spindle apparatus separates the chromosomes during the second meiotic division.

Various treatments have been used to inhibit the second meiotic division, including cold shocks, chemical treatments, hydrostatic pressure, and heat shocks.

The resulting egg has two identical sets of maternal chromosomes. Subsequent fertilization of the egg creates a 3N triploid individual with three or a triploid set of chromosomes.

Triploid Induction Techniques

The most effective treatments used to produce triploid sunfish are pressure shocks (Table 8.1). The pressure shock units used by most researchers are based on a design originally used by Dasgupta (1962).

A hydraulic press with a pressure gauge applies the desired pressure to a treatment vessel. The pressure unit treatment vessels consist of a cylinder and piston constructed by a machine shop.

The vessel used at MSU was:

- A stainless steel cylinder 14 cm (5.5 in.) in length, with an outer diameter of 5 cm (2 in.) and a 3.8 cm (1.5 in.) inner diameter.
- A 10.8 cm x 1.27 cm (4.2 x 0.5 in.) stainless steel base welded to the bottom of the cylinder for stability (Figure 8.1).
- A solid brass piston 17.8 cm (7 in.) in length.
- Compression maintained from two rubber ‘O’ rings around the base of the piston.

A hole drilled up through the center of the piston had a release valve at the top that could be opened to allow air to escape. This also released pressure on the apparatus when a

Figure 8.1. Applying pressure in the production of triploids.

treatment was complete. Decompression speed of the press was nearly instantaneous.

Materials required

Sunfish must be hand spawned prior to shock treatments. Sunfish collected from ponds during the spawning season or induced to spawn in tanks should be checked for ripeness just before spawning.

Fish are ripe if they release eggs or milt when gentle pressure is applied to their sides just in front of the vent using a thumb and forefinger.

The following materials are needed :

- A small glass dish.
- Pasteur pipette and pipette bulb (available from a scientific supply house).
- Soft cloths (e.g., undershirts).
- Stopwatch.

- Equipment to incubate the treated eggs.
- Press and treatment vessel.

Hand spawning technique

Begin hand spawning by gently drying the genital areas of both the male and female fish.

Apply gentle hand pressure to:

- The sides of the female to expel eggs onto a dry glass container.
- Obtain milt in the same way from two males for each female spawned.
- Collect the milt with a Pasteur pipette and pipette bulb.

As soon as the milt is added to the eggs in the glass container:

- Gently stir.
  - Fertilization time is arbitrarily noted as when eggs and sperm are mixed.
  - This time determines when to apply pressure shocks.
- Start the stopwatch at this time.

The process

After milt is applied to the eggs, slowly add water to the eggs to activate the sperm and wash out excess milt.

As soon as the eggs are washed or just prior to the pressure treatment:

- Pour the fertilized eggs (Figure 8.2), with approximately 15 ml of water, into the pressure cylinder.
- Place the piston into the cylinder and:
  - Make certain the valve is open.
  - Press it down into the cylinder until water comes out of the valve opening.
  - Close the valve.
- Place the treatment vessel under the hydraulic press.
- Use initiation time, pressure, and treatment duration (recommended in Table 8.3).
- Release the press pressure on the piston.
- Open the cylinder valve after the shock treatment time is complete.

The eggs can then be incubated as for out-of-season spawning or in incubation chambers (Mischke and Morris 1998).

Cold Shock Treatments

Cold shock treatments (tetraploid induction) are the only treatments used successfully to induce tetraploidy in the bluegill sunfish. Cold shock treatments are applied using a water bath maintained at the treatment temperature.

Bluegill are first:

- Spawned
- Eggs fertilized
- Eggs washed (described above)

Fertilized eggs are poured into treatment/ incubator chambers prior to starting the cold shock treatment.

Materials required:

Two-piece chambers can be cut from sections of PVC pipe. Each chamber should be approximately 5 cm (2 in.) in length by either 7.62 (3 in.) or 6.35 cm (2.5 in.) in width.

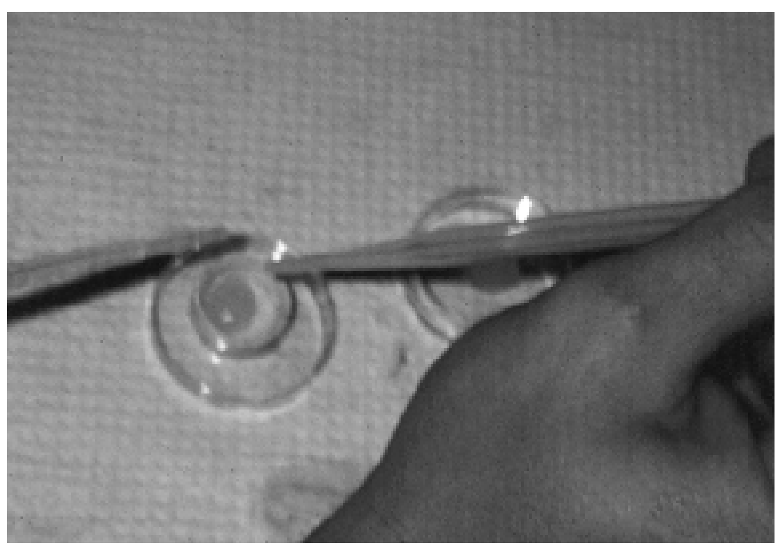


Figure 8.2. Fertilized sunfish eggs being prepared for pressure treatment.

Using silicone:

- Attach fine mesh screen (420 mm (0.016 in.) to one end of each 5 cm (2 in.) long pipe piece.
- Leave the other end open.
- Place the larger diameter cup over the smaller cup containing eggs to create a closed chamber.

These materials allow water to pass through the chamber, but retain the eggs and the newly hatched fry.

The process

The most effective cold shock treatment used to produce tetraploid bluegill was a 15-min application at a temperature of 7.5°C (59°F) 40 min after fertilization.

After completing the shock treatment interval:

- Remove the incubation chambers from the cold water bath.
- Place in an incubation unit (Figure 8.3).

The chambers can be placed in a commercial Heath-type drip incubator or an up-welling bucket incubator. An up-welling bucket incubator can be made from:

- Two buckets the same size (19-liter (5-gal) buckets are easiest to use).
- Cut out the bottom of one bucket, leaving a 1.3 centimeter (1/2 in.) lip around the edge.
- Attach a round piece of fiberglass replacement screening to both sides of the lip using silicone cement.
- Drill a hole in the side of the second bucket, just above the bottom.
- Glue a male hose adapter into the hole using silicone cement.
- Place the screen-bottomed bucket tightly into the hose adapted bucket.
- Connect a water line to the male hose adapter to pass water through the screen and incubation chambers.
- During incubation, supply the unit with 26°C (79°F) water.
- Keep the flow at approximately 1.5 liters/min (ca. 0.4 gpm).

Fry culture

Transfer the fry to ponds for grow-out to market size after they are 14–28 d post-hatch. For best grow-out results, process intensive fry (larval) culture as described by Mischke and Morris (1998).

Ploidy Testing

Bluegill fry can be tested for ploidy at 7–12 d post-hatch (shortly after yolk absorption) using a flow cytometry ploidy analysis technique developed at MSU. At least five fry from each shock treatment should be tested for ploidy. Unfortunately, ploidy level determination by flow cytometry is expensive.

Figure 8.3. Triploid eggs being placed in an incubator for hatching.

Ortho Cytofluorograph Analysis for Cellular DNA Content of Fixed Cells with DNA Doublet Discrimination program to compile and analyze sunfish ploidy data.

MSU provides sample preparation and ploidy determination protocols to commercial laboratories, along with the equipment necessary to run ploidy analysis, and may, on a limited basis, run ploidy analysis of fry at cost.

Blood samples of larger bluegill can be used to determine ploidy by flow cytometry, karyotyping, or the use of a Coulter-counter with channelizer (Ihssen et al. 1990).

Summary

The production of 100% triploids is very important for food fish producers and recreational fisheries managers, since even a few normal diploid fish would result in unwanted reproduction.

The laboratory techniques using pressure shocks to induce triploidy in sunfish can be used to eliminate uncontrolled reproduction and make sunfish more suitable for commercial food fish production. It may also make them more valuable for sale in the recreational fisheries market.

The development of procedures for tetraploid induction may decrease labor costs for triploid *Lepomis* production, while ensuring 100% triploidy. The determination of whether induced triploidy and tetraploidy can be done commercially to confer more rapid growth, more efficient growth, or both, are key factors.

These keys still require research to determine the cost to benefit ratio for induced triploidy.

Notes

9

Fee–fishing

Fee-fishing ponds and lakes provide multiple benefits to the community as a whole, to fish culturists, and to the owners/operators. A successful fee-fishing business must be carefully planned and operated to be truly rewarding to the operator and the angler.

This chapter explains:

- Fee-fishing ponds
- Successful fee-fishing business characteristics
- How to start a fee-fishing business
- Follow up information

9

Fee-Fishing

Author: D. L. Garling, Michigan State University

Fee-Fishing Ponds

Fee-fishing ponds, also called catch-out ponds or pay lakes, are usually small, heavily stocked bodies of water containing one or more kinds of fish. The type of fish stocked in these ponds depends on pond conditions.

Cold-water ponds are normally stocked with trout; warm-water ponds are usually stocked with channel catfish or channel catfish with hybrid sunfish.

Ponds stocked with largemouth bass, or largemouth bass and bluegill, do not usually supply the harvest rate necessary for a successful fee-fishing business.

Ponds should also be stocked with catchable size catfish or be located in areas where fishing opportunities are limited. A warm-water pond can be stocked with a hardy fish, such as carp, and managed on a “fish-for-fun” basis.

Figure 9.1. Fee fishing operations get the majority of their business from families with young children.

Operators of fee-fishing ponds may charge:

- A basic fee for 1/2 or 1 d of fishing.
- A basic fee for each pound or inch of fish caught (high quality fish).
- Both for the fishing privilege and the fish caught.

Facilities may include:

- Snack bar
- Picnic facilities
- Bait and tackle shop
- Boat rentals
- Public rest rooms
- Parking

The Fee Angler

A 1975 survey of Michigan fee-fishing operators indicated that the majority of customers (75%) were composed of families with young children (Figure 9.1). Other customers were usually individual adult males or special groups (e.g., the Rotary Club or Boy Scout Troops).

When asked what the most important characteristics of a fee-fishing operation was for them, Virginia fee anglers ranked their preferences for return and enjoyment as:

- Manager's attitude (ability to deal effectively with people)
- Water quality
- Natural area beauty
- Companionship
- Fish size
- Facilities
- Ease of access
- Number of fish caught
- Weather
- Privacy

Fee anglers do not rank catching fish high on their priority list; probably because they expect to catch fish while at a catch-out pond.

They may rank other factors higher than catch rate or size of fish caught, but no fee-fishing pond will remain in business long if its customers fail to catch fish!

Successful Business Characteristics

The most common characteristic of a *successful* fee-fishing business is providing a desirable family outing.

Parents are satisfied if their children catch quality fish in a reasonable length of time, usually without regard to cost.

Scenic

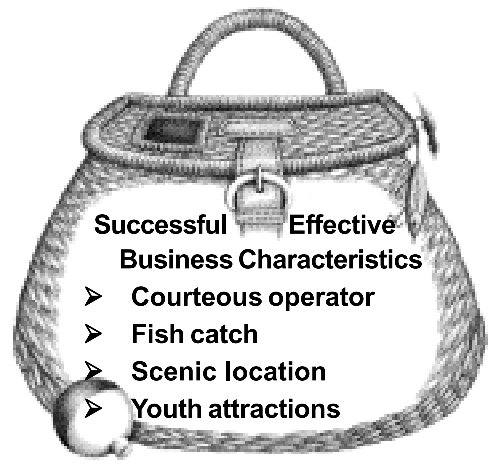
Ponds should be located in a "natural" area screened from urban distractions. Trees are effective screens when placed between roads, parking areas, and other evidences of man and the actual fishing area.

Fee-fishing areas should include picnic facilities (e.g., tables, benches, trash receptacles) and public rest rooms.

Youth attraction

Fee-fishing facilities can also succeed when planned as an amusement park with such youth attractions as playground equipment.

Most successful fee-fishing areas are located within 80.5 km (50 mi) of population centers with



50,000 or more people or near other types of public attractions.

People skills

Most importantly, the fee-fishing operator must be willing and able to work with people. It is essential to be polite and courteous even under the most difficult situations (for example, being awaken at 5:00 AM for bait).

The best advertisement for a successful business is word of mouth from satisfied, repeat customers. Other forms of advertising, such as road signs and brochures at tourist stops, can help attract a first time fee-fishing angler and help launch a business.

The friendly, helpful operator will bring customers back!

How to Start a Fee-Fishing Business

Starting a fee-fishing business requires solid planning and objective thinking. It requires research into time, pond location, type of fish, and legal considerations.

Also consider:

- Site selection and beautification
- Pond construction
- People management
- Pond management
- Advertisement and information
- Liability insurance and legal assistance
- Economic analysis & average return

Site selection[1]

The best site for profitable fee-fishing is a pond within 80.5 km (50 mi) of a population center of >50,000. The site should be in a peaceful, natural setting with easy access. It must have an adequate supply of high quality water to facilitate holding large numbers of fish.

Springs and wells are the most desirable sources of water. Rivers and streams are less desirable because they can be a source of unwanted fish, fish diseases, silt from runoff, and may flood the pond.

Ground water may be an acceptable source of water if a well or spring to maintain water quality supplements it.

Surface runoff is usually not a desirable water source for fee-fishing ponds since water availability and pond level then becomes seasonal, fluctuating with the rainfall.

Runoff water can also carry sediment and nutrients into a pond, which encourages excessive and unwanted aquatic plant growth.

Pond construction

Before construction, check with your state's Conservation or Natural Resources Agency to determine what permits are required.

- Permits may be required to buy and sell game fish.
- Pond-building permits may be required, depending on the:
  - Location of the pond.
  - Water source.
  - Character of the waterway receiving the pond effluent.

The Natural Resource Conservation Service provides information and technical assistance in pond construction and design.

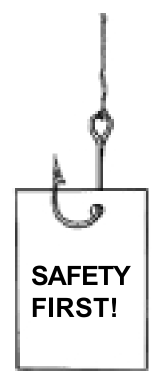
Follow the guidelines established by the U.S. Department of Agriculture, Natural Resource Conservation Service to construct ponds.

Site beautification

It is important to locate the fee-fishing pond in a pastoral, natural setting.

After pond site selection:

- Develop a plan to show the location of the pond(s) and other facilities (e.g., snack bar, rest rooms, parking lot, and picnic tables).
- Include landscape features in the plan.
- Ensure that all trees, shrubs, and power lines are at least 6–9 m (20–30 ft) away from the pond bank to avoid snagged fishing tackle.
- Avoid natural screens of deciduous trees, like poplars, to prevent leaf litter in ponds, unless the trees are on the leeward side of ponds or coniferous trees screen them.
  - Place deciduous trees no closer than their maximum mature height.
  - Plant these behind a windbreak of conifers to avoid unwanted fall leaf litter in the pond.
- Use fast growing conifers for an effective natural screen between the ponds, parking lots, roadways, and buildings.



Safety

To promote pond safety, maintain one or more safety stations around the pond. A safety station consists of:

- A pole with a connected rope and floatation device that can be thrown to someone who inadvertently falls into the pond.
- A clear site line should also be maintained between the pond and the pond operator's location.

Pond management

Fee-fishing pond management differs from recreational pond management and lake management. The goal of fee-fishing is to provide adequate numbers of catchable fish to clientele, rather than sustained fishing recreation over time.

Stock the ponds with catchable size fish when needed rather than stocking just once or twice each year.

Stocking considerations

Rainbow trout (*Oncorhynchus mykiss*) are usually stocked in cold-water ponds [summer temperatures do not exceed 21°C (70°F)] because they are easier to catch than brown trout (*Salmo trutta*) and more hardy than brook trout (*Salvelinus fontinalis*).

Supplies of catchable sized rainbow trout that are ≥23 cm (9 in.) are also easier to locate than supplies of catchable brook or browns. Warm water ponds are generally stocked with catchable sized channel catfish ≥30 cm (12 in.) or hybrid sunfish ≥15 cm (6 in.).

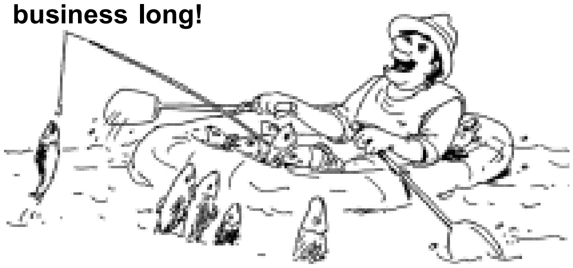
Although bluegill, crappie, perch, bullhead, northern pike, and walleye can be stocked in larger recreational fee-fishing lakes, they should not be stocked in small fee-fishing ponds. Bluegill, crappie, perch, and bullhead often overpopulate and stunt in small ponds and lakes.

Northern pike and walleye eat large amounts of fish and only produce reliably fishable populations in lakes >16 ha (40 acre).

Wild fish should never be stocked in fee-fishing ponds or lakes because they can introduce parasites and diseases.

Stock fee-fishing ponds with enough fish to ensure a good catch rate. However, stocking very high densities of fish may result in crowded conditions, which leads to disease outbreaks. Sick or dying fish are not good advertisements for a fee-fishing pond.

Stock ponds as needed—or you won't stay in business long!



Actual stocking rates of catchable size fish depend on how quickly they are caught. It also depends on the operator's ability to maintain water quality through aeration and the addition of water from springs or wells.

Feeding considerations[2]

It may be necessary to feed the fish; however, feeding fish should be avoided unless you have the ability to run water through the pond to remove excess nutrients added by uneaten feed and fish wastes.

Even if the fish did eat all of the feed, a significant amount of food can be converted into fish wastes.

Excess nutrients from feed and fish waste leads to excessive plant growth. This makes fishing difficult and the area unattractive. If feeding is necessary to maintain the weight and health of fish, it is better to feed:

- No more than once a day or every other day.
- Only at rates the fish can completely eat in a few minutes.
- Use floating feed to visually determine if the fish are actively eating the feed.

Use available resources

Use every resource available for greater chance of success. The Fisheries and Wildlife specialist for a State's Cooperative Extension Program, for example, can provide additional information on cold water and warm water pond management for fishing and weed control. They can also provide a list of commercial fish sources, and assistance in controlling fish predators.

People management

People management is as important as fish management when operating a fee-fishing business. Remember that fee anglers rank the manager's attitude and ability to work with people as their highest preference in selecting a fee-fishing pond.

Advertisement and information[3]

Active forms of advertisement (TV, radio, newspapers) may be necessary to initially attract customers to a fee-fishing pond. Later, passive advertisements, such as road signs (where allowed) and word of mouth, are generally sufficient to attract local customers.

Pamphlets distributed at tourist stops (highway rest stops, restaurants, and motels) or through your state's Travel Bureau can help in attracting vacationers.

Some state Travel Bureaus or regional Chambers of Commerce print travel guides with local attraction advertisements.

Advertisements should always include:

- Directions to the area
- Facilities available
- Activities available
- Operation schedule (season, days, and hours open)
- Species available
- Fees

Rules and Regulations

Prominently post:

- **Prices**
- **Fishing regulations**
- **Activity rules**

Once fee anglers arrive, use signs to clearly direct customers to parking areas and points of information.

Prominently post prices for fishing and other services (fish cleaning and bagging) to avoid confusion and later misunderstandings. Clearly post all rules.

Generally, fee-fishing businesses prohibit swimming for liability reasons. Businesses that stock catchable size fish usually require that all fish caught are kept by the person to avoid hooking mortality.

Additional suggestions

A snack bar and bait and tackle shop can serve as a focal point to provide:

- Information
- Additional services

Snack bars and/or bait and tackle shops are significant sources of additional income to the fee-fishing operator and promote goodwill.

Distributing a free pamphlet (such as How To Fish) at the snack bar with the fee-fishing business name, location, and schedule of operation acts as an advertisement as well as demonstrating goodwill.

The pamphlet can explain how to have an enjoyable day at the pond, provide fishing tips, and encourage repeat customers.

Posting photos of customers with impressive catches tends to bring them back with friends to show off the pictures and then to point out where they landed "the big one!"

Photos can also boost the expectations for a good catch and a good time for the incoming anglers.

Property care

Place numerous trash receptacles around the snack bar, picnic area, and ponds to reduce litter problems. Make certain that the cans are covered and emptied often to limit attracting flying pests like bees and flies, which customers will certainly notice.

Since a fee-fishing business is often operated close to the owner's home, it is important to make private areas off limits to customers.

Clearly identify areas open to fee-fishing and associated activities and those off-limit in order to protect privacy and keep goodwill.

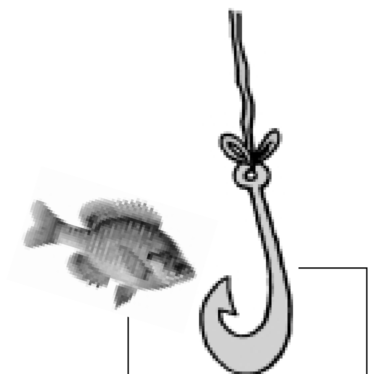
Liability insurance and legal assistance[4]

Liability insurance is an absolute necessity to protect a fee-fishing business against unforeseen accidents. Some states have laws that limit a landowner's liability. For example, Michigan Act. No. 110 (House Bill 4202) may limit liability for individual landowners operating a fee-fishing business.

Sections 3 and 4 of the law limit the liability of a landowner, tenant, or leasee. A person who paid a fee to fish on their land and were injured can sue them, but only if **all** of the following situations occurred:

- The injuries were caused by an unreasonably risky, harmful condition;
- The owner, etc. knew or should have known of the condition;
- The owner, etc. failed to make the condition safe or to warn the person of the condition;
- The injured person did not know or had no reason to know of the condition.

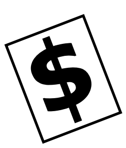
Insurance companies that are experienced in covering outdoor recreation businesses should be contacted to determine the cost and coverage available before going into business.



Consider cost and return before investing!

Incorporating the business

Additional liability protection and certain tax advantages may be afforded by incorporating. Including only the pond area and associated facilities as part of the corporation assets may be a benefit.



Economic Business Analysis

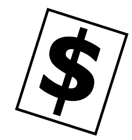
Before beginning any business venture, it is wise to research the possibility of success. Michigan can serve as an example of the economics of a fee-fishing business in the north central region.

In 1975, 50 Michigan fee-fishing businesses were surveyed. The 34 respondents were located throughout the state. Almost all of the operations were family-run, provided a supplemental source of income, and required little or no part-time help.

For analysis, the operations were then divided into small or large operations based on their annual gross incomes.

Only large operations provided the primary source of income for their owners.

Average Dollar Return



Average returns to labor, management, and investment were low for both sizes of operations. The average annual return of a small operation was not sufficient to cover annual operating costs when labor and management costs (fair wages to owner or owner's family) were included in the calculations.

Although large operators had greater returns overall, they still provided low returns to labor and to management.

It was found that the average return of investment for both small and large businesses were negative. However, while all the businesses provided income to the owner, not all of the individual businesses had negative returns.

Location, facilities, management, and other factors affected the returns to fee-fishing operators.

Unfortunately, insufficient data were collected to estimate the optimum size of a fee-fishing operation. But in 1975, an initial investment of at least $30,000 for fee-fishing firms was estimated to "break even" under average circumstances.

This estimate should be increased to current dollar equivalents for planning purposes.

Follow Up Survey

A 1991 follow-up survey indicated that slightly <40% of licensed Michigan fish growers had fee-fishing operations. Approximately 9% of all licensed fish growers surveyed were involved in fee-fishing only and 11% were primarily involved in fee-fishing operations.

Rainbow trout were the most commonly used fish species, although some growers provided brook or brown trout, catfish, bluegill, or largemouth bass.

Most fee-fishing operators did not charge an entrance fee, but did require that all fish caught be purchased, priced either by the inch or pound. Most operators provided cleaning, bagging, and icing of fish caught; but few provided additional services such as farm tours, refreshments, tackle, or souvenirs.

In the south, additional services have been found to generate significant income for the fee-fishing operator.

Only a few Michigan operators aggressively advertised their business. Most relied on word-of-mouth advertising or repeat customers.

The primary competition for fee-fishing operations was from public access recreational fisheries (lakes and rivers) and other types of family-oriented recreation.

Summary

Fee-fishing lakes are becoming a popular means of income, especially around heavily populated urban areas where there are limited opportunities to fish.

Fee-fishing lakes can provide the fish culturist with a different channel to market fish.

Obviously, the costs and estimated returns for a fee-fishing business should be carefully considered before any investments are made.

With the proper location, management, and facilities, a fee-fishing business can be rewarding and profitable to the operator.

Notes

10

Calculating Chemical Treatments

Disease recognition and treatment for fish is essential for maintaining healthy stock and maximizing investment. Treatment requires the ability to accurately calculate rates for approved chemicals in easy-to-use formulas.

This chapter explains:

- Disease and mortality causes
- Drug and chemical treatment overview
- Pond/tank treatment calculations
- Measurement facts
- Conversion factors

10

Calculating Chemical Treatments

Authors: M. Riche and D. L. Garling, Michigan State University

Disease and Mortality Causes

Diseases are a common problem in fish culture. The best method for controlling disease problems is prevention. Disease prevention includes:

- Providing good water quality.
- Minimizing handling stress.
- Using only clean and disinfected equipment.
- Using a good quality, correctly stored feed.

Despite careful and conscientious prevention efforts, however, disease problems can still occur. When a problem occurs, it is important to quickly identify and treat the problem.

Four RECOUP steps used in treating a disease can help prevent its reoccurrence.

Die-off rates

When mortalities do occur, the rate at which fish die can be used for an initial disease diagnosis. Generally, fish death rates follow one of three patterns:

- Rapid <1–2 d
- Slow for a few days then a rapid die-off
- Slow rate over a long period of time

Die-off causes

If fish die-off occurs within 1–2 d, they are most likely dying as a result of an environmental disaster (e.g., lack of oxygen or lightening) or a toxic chemical.

Death due to lack of oxygen in a pond occurs most often just before sun up when the levels of oxygen are lowest in the pond. The largest fish die first.

If a large number of all sizes of fish die, especially after a thunderstorm, your pond may have been struck by lightening. Fillet a few of the dead fish. If there are areas of bleeding along the backbone, the fish may have died from electrocution.

If fish die as a result of a toxic chemical, the smallest fish die first because their metabolism is faster than that of larger fish. The faster metabolism of small fish results in quicker uptake of a toxic chemical.

Fish dying slowly for a few days, followed by a rapid die-off within 1 or 2 d, are most likely the victims of a virulent virus or bacteria. A fish health specialist is required for a specific identification of the cause and treatment recommendation.

Contact the aquaculture extension specialist, State Department of Agriculture aquaculture

R E C O U P

Reduce stressors (e.g., low DO, high ammonia, high or low temperatures)

Correct pathogen identification (e.g., parasite, bacteria, and virus)

Use safe and appropriate treatments

Prevention

coordinator, or state aquaculture association to find a fish health specialist.

Bacterial infections generally result from stress. If the stress causing the virulent bacterial infection is identified and corrected (e.g., low oxygen, silty water, rapid temperature changes, or heavy parasite load), the severity of die-off may be reduced.

If fish die at a slow rate over a long period of time (weeks or months), they are most likely dying as a result of:

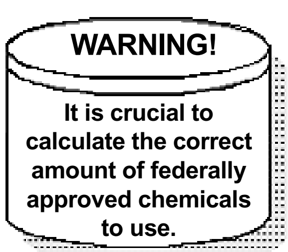
- Prolonged stress from poor water quality.
- Too low or high a temperature.
- Large daily temperature changes.
- A low virulent bacteria or virus.
- A heavy parasite load.

This type of slow die-off pattern may correct itself if the fish are dying due to water quality problems caused by too many fish in the system. The causes of a slow die-off, however, are most often the stressors types that lead to more rapid die-offs from virulent bacteria. Consequently, it is important to quickly identify and correct the cause of the die-off.

Drug/Chemical Treatments Overview

Drug and chemical treatments should only be given after stress has been reduced and the disease causing pathogen identified.

Safe and appropriate treatments kill the pathogen without harming fish. There is often a small difference in the drug amount that kills the pathogen and the amount harmful to fish.



Feed treatments[1]

Medicated fish feeds are available to treat bacterial diseases for specific fish species. The U.S. Food and Drug Administration is currently reviewing reports on permitting the use of these feeds to treat bacterial diseases in other species of fish under the supervision of an attending veterinarian.

Until rules are developed for the extra-label use of medicated fish feeds, they can only be used to treat fish listed on the feed label.

Pond/Tank Treatment Calculations

To calculate the correct amount of chemical to use, find out what pathogen you are dealing with and the chemical dose that will kill it. "Dose" refers to the strength of the treatment.

To calculate a safe and appropriate dose, measure the area of the pond or tank and the volume of water the fish are in. **Always** check calculations twice, since small errors can result in dead fish. It is particularly important to be careful where decimals are used. Calculate:

- Area
- Water volume
- Correct amount

Calculate the area

The area of most ponds and tanks is determined by using equations for the area of a circle, rectangle, or right triangle. The area of irregular shaped ponds is determined by dividing the pond into smaller areas that resemble these shapes.

The smaller areas are calculated and added together to get the total area. Figure 10.1 shows these equations.

Calculate the water volume

A pond or tank without water coming in or going out is called static. Water volume in a

Facts to Remember

Area of rectangle = length x width

Area of circle = radius^2 x 3.14 (π)

radius = $\frac{\text{diameter}}{2}$

1 acre = 43,560 square feet

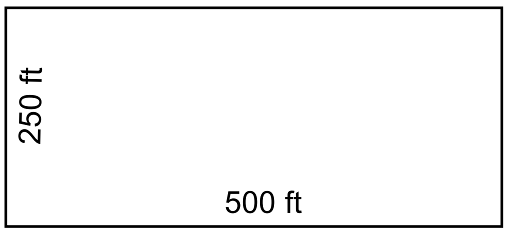
1 acre-foot = 1 acre of water 1 foot deep

Rectangular Pond

Area = 500 ft x 250 ft = 125,000 ft^2

Expressed as:

$$\frac{125{,}000}{43{,}560} = 2.9 \text{ acres}$$

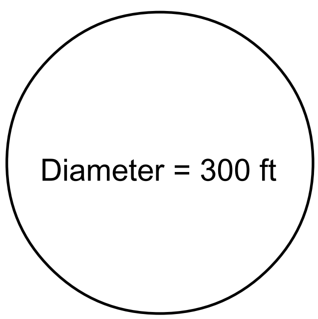


Circular Pond

Area = 150^2 x 3.14 = 70,650 ft^2

Expressed as:

$$\text{Acreage} = \frac{70{,}650}{43{,}560} = 1.6 \text{ acres}$$



Irregular Shaped Pond

Area 1 = 80 ft x 70 ft = 5,600 ft^2

Area 2 = 200 ft x 90 ft = 18,000 ft^2

Pond Area = 5,600 + 18,000 = 23,600 ft^2

Expressed as:

$$\text{Acreage} = \frac{23{,}600}{43{,}560} = 0.5 \text{ acres}$$

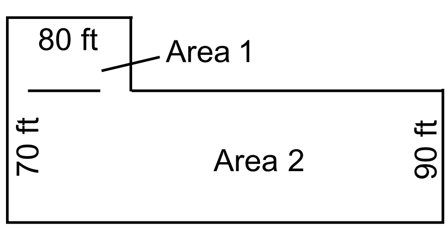


Figure 10.1. Areas for rectangle, circle, and right triangle.

static pond or tank is the amount of water under the area calculated. It is determined by multiplying the area by the water depth. In a pond, use average water depth (Figure 10.2).

Calculating the volume of water in a flow-through system is more complicated. The water leaving the tank must also be included in the volume for calculating the dose. The water flowing out removes some of the chemical previously added.

To keep the same dose during the entire treatment, the amount of chemical leaving with the water must be replaced. In a flow-through system, you must know the flow rate. Flow rate is the amount of water leaving a tank or pond in 1 min.

The flow rate is determined by measuring the amount of time it takes to fill a 5 gal bucket with the incoming or outgoing water. It is expressed as:

5 gal/min to fill = gal per min

Calculate the correct amount

The chemical dose to use is usually given as the weight or amount of chemical to apply in a certain amount of water. There are a number of ways the weight and volume are given.

To reduce the chance of using too much or too little chemical, it is best to change weights and volumes into familiar units with conversion factors.

For example, if you are more familiar with pounds than kilograms, convert the kilograms to pounds (conversion factors and conversion tables are given at the end of this chapter).

Percent Active Ingredient

Before measuring any chemical, read the label carefully. It is important to note the percent active ingredient.

The percent active ingredient is the **amount** of chemical actually doing the work.

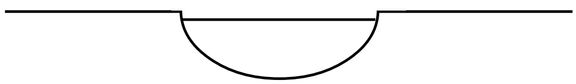
Example: The chemical formalin is a mixture of water and formaldehyde.

Calculating Depth and Water Volume

Pond with uniform slop on bottom

Maximum pond depth = 15 ft

Depth = 1/2 (maximum depth) = $\frac{15}{2}$ = 7.5 ft



If the surface area on pond is 3 acres, the volume = 3 acres x 7.5 ft = 22.5 acre-feet

Pond with irregular bottom

1. Measure the depth at several uniformly spaced points.
2. Add these values together and divide by the number of depth readings.

Example: Depth in feet:

A = 6
B = 10
C = 8
D = 5
E = 3
F = 6
38

Average depth = $\frac{38}{6}$ ft = 6.3 ft

Pond with irregular bottom

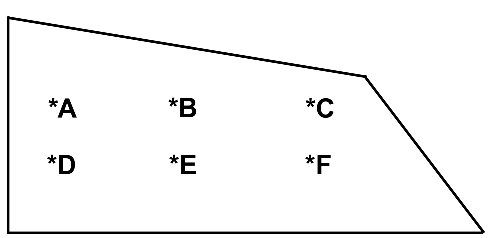


Figure 10.2. Area and volume for a regular shaped pond.

Formalin can be 37% formaldehyde with 63% water or 20% formaldehyde with 80% water. The percent of formaldehyde is the percent active ingredient.

Since only the formaldehyde does the work, it is necessary to use a correction factor so the right amount of active ingredient is used.

The easiest way to calculate the dose is with the following equation:

$$D = V \times K \times CF \times AI$$

Where: D = Dose
V = water volume
K = amount needed to kill pathogens
CF = conversion factors
AI = (100%)/(% active ingredient)

Some sample calculations are given below. For ease of discussion, English units will be used for many of the samples.

Scenario 1: Pond Treatment.

After 4 d of early morning dissolved oxygen readings you are happy to see the dissolved oxygen is back to saturation.

Although you didn't lose any fish when the dissolved oxygen was low, the fish are acting like the dissolved oxygen is still low.

A gill parasite is present in some of the sicker fish examined. A positive identification indicates the parasite is *Trichodina*.

You know formalin is effective against *Trichodina* but you also know it will decrease the dissolved oxygen if there is high level of organic matter in the pond. However, you need to take action.

Steps to take:

1. Checking with the state extension agent to ensure formalin is safe and legal, you decide to treat your 4 acre pond.

   Treatment: Formalin is 37% active ingredient (stock solution)

   Treatment Rate: 15 ppm (using Formalin as 100% dosage)

2. You measure the pond and determine the following dimensions (Figure 10.2):

   Pond area is measured as Length x width, or:

   600 ft x 300 ft = 180,000 ft²

3. You are more comfortable using acre instead of thousands of sq ft, so you use the correct conversion factor to change the area to acres.
   The conversion factor indicates there is 43,500 ft² in every acre.

   Pond area is now measured as:

   $$\frac{180,000}{43,500} = 4.13 \text{ acres}$$

4. You now calculate the volume of water under the area. The measurements indicate the average depth is 7 ft.
   Volume of water is measured as area of pond x average depth, or:

   4.13 acres x 7 ft
   = 28.9 acre-feet (A-ft)

5. You calculate the dose or amount of formalin to use. The dose is the amount of formalin to get 15 ppm of active ingredient.
   Using the equation:

   D = V x K x CF x AI

   You get: 28.9 A-ft x 15 ppm x (0.33 gal / 1.0 A-ft) x 1 = 143 gal
   Treatment: The dose is 143 gal of formalin (37% active ingredient) in 28.9 ac ft of water.

Scenario 2: Irregular Shaped Pond

A buyer is interested in purchasing bluegill stocked in your 0.20 ha (0.5 acre) pond last fall. However, you notice that the bluegill have white patches on their skin.

It looks like "ich," so you collect a few for diagnosis. The diagnosis indicates you were right; your fish have "ich", (*Ichthyopthirius*). The buyer said he doesn't want diseased fish.

You know there is a way to correct the situation. Steps to take:

1. You determine chemical A is effective in treating *Ich*. Checking with the state extension agent to ensure the chemical is safe and legal, you treat your pond.

   Treatment: Chemical A is 20% active ingredient.
   Treatment Concentration: 25 ppm; repeat every 5 d as needed.

2. You measure the pond and determine the following dimensions (Figure 10.3):

The pond is an irregular shape. The best way calculate the area is to:

- Divide the pond into two regular areas
- Calculate each area
- Add the two areas together to get the total pond area
  Area A is the area of a right triangle, measured as (Length x width)/2, or:

  (100 ft x 200 ft)/2 = 20,000 ft²

  Area B is the area of a rectangle and

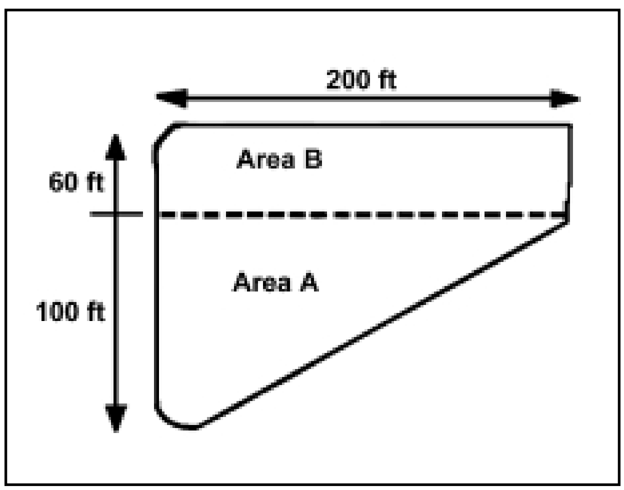


Figure 10.3. Area and volume of an irregular-shaped pond.

measured as (Length x width), or:

60 ft * 200 ft = 12,000 ft^2

Total area of the pond is measured as Area A + Area B, or:

20,000 ft^2 + 12,000 ft^2 = 32,000 ft^2

3. You now calculate the volume of water under the area. The measurements indicate the average depth is 6.5 ft.

   Volume of water is measured as Area of the pond * average depth, or:

   32,000 ft^2 * 6.5 ft = 208,000 ft^3

4. After volume is determined, calculate the dose or amount of chemical A to use.

   The dose is the amount of chemical A to get 25 ppm of active ingredient.

   Using the equation:

   D = V x K x CF x AI

   You get: 208,000 ft^3 x 25 ppm x (1.04 fl oz/1,000 ft^3) x (100%/20% active ingredient) = 27,040 fl oz

5. It is easier to measure gallons than 27,040 fl oz, so you convert to gallons.

   The conversion factor indicates there is 128.0 fl oz in each gal.

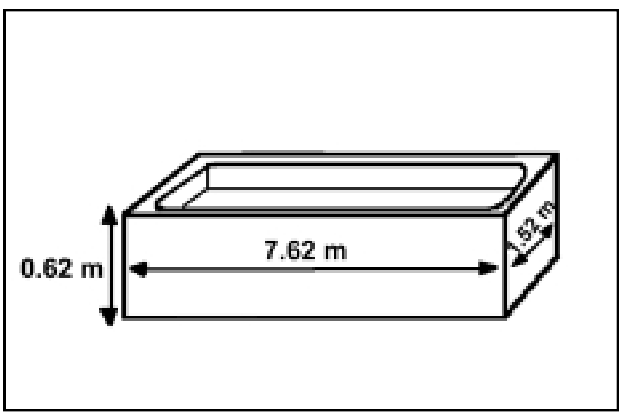


Figure 10.4. Area and volume of a rectangular tank.

27,040 fl oz x (1.0 gal / 128.0 fl oz) = 211.25 gal

Treatment: The dose to use is 211.25 gal of chemical A in 208,000 ft^3 of water.

Scenario 3: Static bath in a tank.

After coming back from a fish culture workshop, your nephew tells you he doesn't think the fish like the new food you bought before you left. Even though they didn't eat, he kept feeding them because he thought they would get hungry.

The tank water looks brown, so you turn up the water flow and the water clears. When the water clears, you notice the fish have white to pinkish patches of slime on their skin.

A diagnosis identifies the parasite *Epistylis* on their skin and gills. You are afraid to use formalin because the gills are already damaged. You find out salt (sodium chloride) is effective against *Epistylis*.

1. Checking with the state extension agent, you learn salt is a low priority regulatory drug and may be used for treatment.

   Treatment: Salt (sodium chloride)

   Treatment Concentration: Static bath of 1% salt for 10–30 min.

2. A static bath requires turning off the water. After providing aeration to the tank, you take the measurements. The tank has the following dimensions (Figure 10.4).

   Area of the tank is measured as length x width, or:

   7.62 m x 1.52 m = 11.58 m^2

3. You calculate the water volume under the area. The water depth is 0.62 m.

   Volume of water is measured as area of the tank x depth of water, or:

   11.58 m^2 x 0.62 m = 7.18 m^3

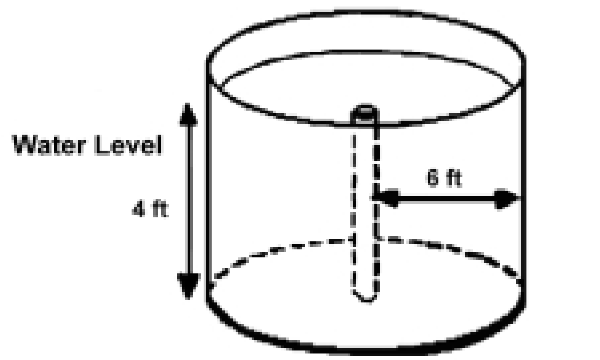


Figure 10.5. Volume of a circular tank.

4. After determining the volume, calculate the salt dose to use. The dose is the amount of salt to get a 1% solution, which is the same as 1 lb of salt for every 100 lb of water.
5. To determine the amount of salt to use, convert the volume to pounds of water. The conversion factor indicates there are 2,201.5 lb of water in each m^3.

   Using the equation:

   D = V x K x CF x AI

   You get: 7.18 m^3 x (1 lb of salt / 100 lb of water) x (2,201.5 lb/m^3) x (100%/ 100% active ingredient) = 158 lbs

   Treatment: The dose to use is 158 lb of salt or 7.18 m^3 of water.

Scenario 4: Flow through bath in a tank.

The bluegills in your circular tanks are swimming slowly near the top of the tank. For a week you have been trying to reduce the ammonia level in your recirculating system. Some of the fish are swallowing air and the gill covers won't close normally.

Your fish have been diagnosed as having bacterial gill disease with tissue swelling. After flushing the tank with freshwater, you contact the state extension agent. The agent informs you salt is a low priority regulatory drug.

1. You decide to treat the fish with salt to kill the bacteria and soothe the gills. The fish are having difficulty getting enough oxygen, however, so you don't want to turn the water off. Therefore, you choose to treat them with a flow-through bath.

   Treatment: Salt (sodium chloride)

   Treatment Concentration: 1% solution for 1 h.

   To maintain the 1% solution for the entire hour, you need to keep adding salt to replace the salt flowing out with the water.
2. Calculate the volume of the tank (Figure 10.5) to find the flow rate of water leaving the tank.

   Area is measured as 3.14 x (distance from center to edge) ^{2}, or:

   3.14 x 6 ft x 6 ft = 113 ft^2

   Volume is measured as area * depth of the water, or:

   113 ft^2 x 4 ft = 452 ft^3

   Treatment application: 1.0% solution is the same as 1 lb of salt in every 100 lb of water.
3. Salt is most easily measured in pounds. Convert the volume of water into pounds of water. A cubic foot of water weighs 62.43 lb (refer to the Conversion Chart at the end of this chapter).

   Using the equation:

   D = V x K x CF x AI

   You get: 452 ft^3 x (1 lb of salt/100 lb of water) x (62.42 lb of water/1.0 ft^3) X (100%/% active ingredient) =282 lb of salt

The initial dose is 282 lb of salt in 452 ft^3 of water. Then:

- Drop the water level about 0.5 ft.
- Add 282 lb of salt.
- Allow the water to come up to the original level.
  - This allows the salt to dissolve before it begins to leave the tank.
- Allow additional salt when the water begins to flow out of the tank.
  - This replaces the salt leaving with the water.

4. First calculate the flow rate to determine the amount of additional salt needed.

   You determine it takes 16 sec to fill a 5 gal bucket with the incoming water.

5. Calculate the flow rate (gallons per seconds), which is:

   (5 gal/16 s) x (60 s/1 min)
   = 18.8 gal/min

Knowing the flow rate allows you to calculate how much salt will leave the tank during the treatment and how much salt to replace.

Since the tank contains 1.0 lb of salt for every 100 lbs of water, convert the gallons of water leaving the tank to pounds of water leaving the tank. Each gallon of water weighs 8.32 lb (see Conversion Chart). With these factors, calculate the amounts needed.

6. Use the following equation to calculate the salt leaving the tank:
   (18.8 gal/1.0 min) x (8.32 lb of water/ 1.0 gal) x (1.0 lb of salt/100.0 lb of water) = 1.6 lb of salt/1.0 min

   Treatment: For a 1 hr treatment, you need an additional (1.6 lb of salt/1.0 min) x (60 min/1.0 h) or:

   96 lb of salt/1.0 h of treatment

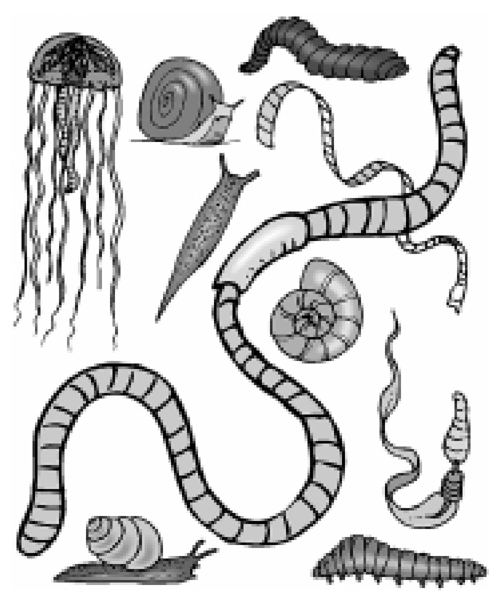
   Use an automatic delivery device to deliver:

   96 lb of salt at 1.6 lb of salt/min

Summary

Disease is a common problem in fish culture. When a problem occurs it is vital to recognize it immediately, seek professional advice, and quickly treat the problem. Implementing the RECOUP steps can resolve the problem, reduce stress, and limit loss.

Calculating a safe and appropriate dose will kill the pathogen without harming the fish. Calculations should always be checked twice for accuracy since small errors can result in dead fish.



Useful Conversions

1 liter (l) of water
- = 1 kilogram (k)
- = 2.205 pounds (lb)

1 milliliter (ml) or
cubic centimeter (cm^3) of water
- = 1 gram (g)
- = 0.0353 ounce (oz)

1 gallon (gal) of water
- = 8.34 pounds (lb)
- = 3.785 kilograms (kg)

1 cubic foot (ft^3) of water
- = 62.36 pounds (lb)
- = 28.29 kilograms (kg)

1 fluid ounce (fl oz)
- = 1.043 ounces (oz)
- = 29.57 grams (g)

1 parts per million (ppm)
- = 1.226 grams per acre/foot (g/acre/ft)
- = 0.0283 grams per cubic foot (g/ft^3)
- = 2.718 pounds per acre/foot (lb/acre/ft)
- = 0.0038 grams per gallon (g/gal)
- = 0.0000623 pounds per cubic foot (lb/ft^3)

1 cubic foot per second (ft^3/sec)
- = 448.83 gallons per minute (gal/min)
- = 26,930 gallons per hour (gal/h)
- = 646,320 gallons per day (gal/d)
- = 1.699 cubic meters per minute (m^3/min)
- = 101.93 cubic meters per hour (m^3/h)
- = 2446 cubic meters per day (m^3/d)

1 percent solution
- = 38 grams per gallon (g/gal)
- = 1.3 ounces per gallon (oz/gal)
- = 0.622 pounds per cubic foot of water ($lb/ft^3/H_20$)

1 acre
- = 43,560 square feet (sq ft)
- = 208.7 x 208.7 feet (ft)
- = circle diameter 235.5 feet (ft)

1 acre per foot
- = 1 surface area 1 foot deep
- = 43,560 cubic feet (ft^3)
- = 2,718,144 pounds (lb)
- = 1,233.49 cubic meters (m^3)
- = 325,851 gallons (gal)

Rotenone applications = 3 ppm

General dilution formula:

Desired concentrations x desired volume/concentration of stock solution = volume of stock solution to dilute to the desired volume.

Conversion of Length Units

Unit	Inch	Foot	Yard	Millimeter	Centimeter	Meter
Inch	1	0.0833	0.0278	25.40	2.540	0.0254
Foot	12	1	0.3333	304.8	30.48	0.3048
Yard	36	3	1	914.4	91.44	0.9144
Millimeter	0.0394	0.0033	0.0011	1	0.1	0.001
Centimeter	0.3987	0.0328	0.0109	10	1	0.01
Meter	39.37	3.281	1.0936	1,000	100	1

Conversion of Weight Units

Unit	Grain	Ounce	Pound	Milligram	Gram	Kilogram
Grain	1	0.0023	0.000143	64.8	0.0648	0.000065
Ounce	437.5	1	0.0625	28,350	28.35	0.0284
Pound	7,000	16	1	453,590	453.6	0.4536
Milligram	0.0154	0.00036	0.000002	1	0.001	0.000001
Gram	15.43	0.0353	0.0022	1,000	1	0.001
Kilogram	15,430	35.27	2.205	1,000,000	1,000	1

Conversion of Volume Units

Unit	Gallon	Quart	Pint	Fluid Ounce	Cubic Foot	Cubic Inch	Milliliter	Liter	Cubic Meter
Gallon	1	4	8	128	0.1337	231.0	3,785.4	3.785	0.00378
Quart	0.25	1	2	32	0.0334	57.75	946.36	0.946	0.00095
Pint	0.125	0.5	1	16	0.0167	28.88	473.18	0.473	0.00047
Fluid Ounce	0.0078	0.0313	0.0625	1	0.00104	1.805	29,573	0.0296	0.00003
Cubic Foot	7.481	29.92	59.84	957.5	1	1,728	28,317	28.32	0.02832
Cubic Inch	0.0043	0.0173	0.0346	0.5541	0.00058	1	16.39	0.0164	0.000016
Milliliter	0.00026	0.00106	0.0021	0.0338	0.000035	0.060	1	0.001	0.000001
Liter	0.2642	1.057	2.1134	33.81	0.0353	61.02	1,000	1	0.001
Cubic Meter	364.2	1,057	2,113	33,810	35.3	61,000	1,000,000	1,000	1

11

Common Sunfish Parasites

Parasites, which are commonly found in fish, can become a real problem in aquaculture production—affecting fish health, pond stability, and marketability. Knowledge and prevention remain the most effective tools. The Federal Food and Drug Administration (FDA) regulates all drugs used for fish diseases.

This chapter explains:

- Parasites and hosts
- Chemical treatments
- Prevention
- Parasite groups
- Parasite characteristics and effects

11

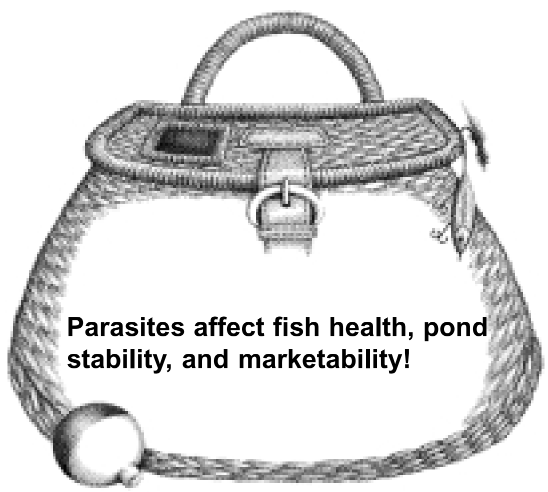
Common Sunfish Parasites

Authors: T. Sampson, Michigan Department of Natural Resources, Fish Division and D. L. Garling, Michigan State University

Parasites and Hosts

Parasites depend on other organisms, called hosts, for survival. They may be found inside the host (endoparasites) or around the host (ectoparasites). Parasites affect their hosts in various degrees, ranging from mild irritation to mortality.

Some parasites require several different hosts to complete their life cycle. The particular host associated with the adult parasite is commonly referred to as the final host or definitive host. Other hosts that occur in the life cycle are called intermediate hosts; these hosts may include other fish, snails, and birds.



Parasitic indications

Although careful pond maintenance reduces the risk of parasites, they may still occur from a number of sources. General signs of parasitic infection include:

- Gulping at the surface
- Flared gills
- Flashing
  - In an effort to remove an irritation on the skin, fish will attempt to rub the irritated area against the side or bottom of a raceway.
  - This exposes the shiny underbelly, which gives the appearance of a flash of light.
- Lethargic behavior, off feed, or laying on the bottom
- Excess mucus production, fin erosion, or skin lesions
- Tremors

These symptoms may be associated with other diseases, so it is best to get the advice of an expert for the cause.

Chemical Treatments for Fish Infected with Parasites

In the past, chemical therapeutics were used to eliminate fish parasites. Over the years, however, laws changed and some of the chemicals used in aquaculture were found to be either harmful to humans or harmful to the environment. To insure human safety, the U.S.Food and Drug Administration (FDA) must approve all chemicals used as treatments for parasites and diseases, either added to water for direct treatment or added to feeds.

Drugs must be specifically labeled for aquaculture use. To insure safety and drug efficacy, culturists should follow all label directions and warnings. Currently, few chemical therapeutics are approved for use in aquaculture. Of the approved drugs, only formalin is effective against fish parasites.

Use of low priority drugs[1]

The FDA also permits the use of low regulatory priority aquaculture drugs. This means the FDA reviewed these types of compounds and determined them to be new animal drugs of low regulatory priority. As a result, the FDA determined that several of these “low priority” drugs can be used to combat parasites.

This designation does not mean that the FDA has officially approved these substances for use, even though the expectation of detrimental effects is low. It does mean that these compounds must be used under specific guidelines and tolerances established by the FDA. Among these compounds are:

- Acetic acid
- Garlic (whole form)
- Magnesium sulfate
- Sodium chloride

These were used successfully to treat some parasitic infestations.

Under certain conditions authorized by FDA, unapproved new animal drugs may be used in conformance with the terms of an Investigational New Animal Drug (INAD) application. Additional information on approved, low regulatory priority and INADs can be obtained from the FDA Center for Veterinary Medicine.

Chemically treating large scale culture ponds is not practical. Focus instead on maintaining good water quality and following good cultural practices.

Prevention

The best “treatment” is proper cultural practices that prevent parasitic infestations from occurring. Among these practices are:

1. Eliminate predators and intermediate hosts such as other fish, snails, herons and kingfishers.
2. Use a water source with high quality water that is free of fish.
3. Quarantine any new fish for a period sufficient to determine if they are carrying any parasites.
4. Maintain low rearing densities to reduce stress in the fish. High rearing densities facilitate rapid transmission of diseases when they do occur.
5. Maintain good water quality by monitoring dissolved oxygen content. The dissolved oxygen content of rearing water should be between 90 and 100% saturation.

Reduce the amount of organic matter in ponds that removes oxygen as it decomposes.

Parasite Groups

A variety of fish parasites exist, but many of these can be listed in ten major types.

These include:

1. Protozoa—single celled animals.
2. Monogenetic Trematodes—flukes with posterior organs of attachment.
    - Simple life cycles involving only one host.
    - Primarily external parasites found on the gills, skin, and fins.
3. Digenetic Trematodes—flukes with a mouth and ventral suckers.
    - Complex life cycles involving intermediate hosts.
    - Adults are generally internal parasites found in the intestine, throat, stomach, gall bladder, and urinary bladder.
    - Young stages, called metacercariae, are found in the skin, muscle, eyes, and internal organs.

Best Practice

- **Proper culture treatments**
- **Good water quality**
- **Limit fish stress**
- **Reduce bird & snail populations**

4. Cestodes (Tapeworms)—worms with flattened, segmented bodies.
    - The head usually has suckers or hooks.
    - Adults are generally found in the intestine.
    - Juvenile forms are found in other internal organs as well as the intestine.
5. Nematodes (Roundworms)—thin, elongated worms with a cylindrical shaped body.
    - Adults are usually found in the intestinal tract.
    - Some are external found outside the rectum, skin and muscles.
    - Larval stages are found in various internal organs.
6. Acanthocephala (Spiny Headed Worms)—worms with flattened or cylindrical bodies.
    - Possess a hook bearing appendage (proboscis) with which it attaches to its host.
    - Adults are generally found in the intestine with larval forms found in the liver.
7. Copepods— external parasites.
    - Found on the gills, skin, and fins.
    - They may be louse like, worm like, or grub like.
8. Leeches— external parasites.
    - Attaches to the host with suckers.
    - Suckers may be larger or smaller than their body diameter.
9. Glochidia—larval clams encapsulated in the fins and gills of the host.
10. Fungi—plant-like organisms that lack chlorophyll.

Parasites affect pond stability

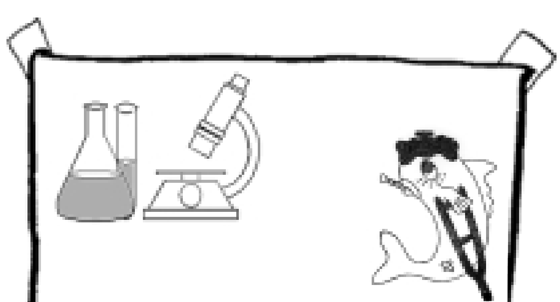
Parasite Characteristics and Effects

Parasites left unrecognized and untreated create a serious problem that may seriously affect any investment return for the aquaculture operator.

Following are tables listing the most common parasites, their:

- Causative agents
- Characteristics
- Effects
- Treatments
- Prevention
- Effects on humans

The family these parasites belong to heads each set of tables and Figures 11.1–11.4 shows specific types of these parasites.



There are 10 major types of parasitic groups.

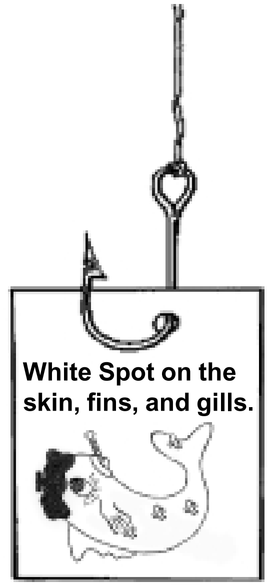
Family = Protozoa—Single Cell Animals

Chilodon	
Causative Agent	A single celled animal of the genus and species Chilodonella cyprini.
Characteristics	An oval shaped body 50-70 µm (0.002-0.003 in.) in length. Individuals can only be seen with magnification although colonies may be seen on the fish as blotchy gray areas.They are capable of movement due to fine hairs called cilia that cover their body. Chilodon are capable of movement due to fine hairs called cilia that cover their body. Chilodonella reproduces on the fish by dividing in half. It produces a resistant cyst that can survive through periods of unfavorable environmental conditions. It is most frequently observed when fish move into shallow water to spawn.
Effects	Large infestations of Chilodonella cause fish to become lethargic, lie on their sides or rise to the surface, and may lead to death. Infestations on the gills cause the fish to secrete large amounts of mucous, which impairs respiration.
Treatments	A 1 h bath in a 250 mg/l (0.033 oz/gal) formalin solution, where possible. 1% salt solution may also be used.
Prevention	Avoid high concentrations of organic material in rearing ponds. Reduce crowding and maintain good water quality by monitoring dissolved oxygen content.
Effects on Humans	Not harmful to humans.

Epistylis	
Causative Agent	A single celled animal of the genus Epistylis.
Characteristics	A microscopic protozoan with cilia for movement. This parasite is generally found on the skin, fins, and gills of fish. It is most common during summer in water with high organic content. Lesions appear as short, slimy, fungus-like growths.
Effects	Hemorrhaging may occur at the site of attachment. The sites of hemorrhages are susceptible to secondary bacterial infections. Heavy infestations can cause large mortalities.
Treatments	A 1 h bath in a 200 mg/l (0.027 oz/gal) formalin solution, where possible. A 15 min bath in a 2% salt solution is effective in controlling this parasite but not practical in large scale pond culture. In ponds, treatments with potassium permanganate have been shown to provide some relief.
Prevention	Avoid high concentrations of organic material in rearing ponds. Reduce crowding and maintain good water quality by monitoring dissolved oxygen content.
Effects on Humans	Not harmful to humans.

Ichtyobodoiasis (Costia)	
Causative Agent	A single celled animal of the genus Epistylis. The parasite ranges in size from 10-20 µm (0.0004-0.0008 in.) in length by 5-10 µm (0.0002-0.0004 in.) wide.
Characteristics	Ichtyobodo lives on the skin and gills of fish utilizing sloughed epithelial cells from the fish for food. Generally, the fish and the parasites live in a state of symbiosis where neither adversely affects the other. Under situations of environmental stress or poor health, the fish's immune system may become compromised, allowing the parasites to reproduce at an immense rate to the point where they begin attacking live cells. Ichtyobodo is most active when water temperature is between 10-25° C (50-77° F) and cannot survive in temperatures >30° C (86° F). Ichtyobodo is transmitted from fish to fish either in its adult form or as an encysted form. Fish in poor health or exposed to such environmental stressors as low dissolved oxygen, overcrowding, or some other pathogen are more susceptible to epizootics than those living in a more favorable environment.
Effects	Fish may become lethargic and go off feed. They may be seen "flashing" or rubbing against the side or bottom of a pond or tank in an attempt to alleviate the irritation. A thickening of mucus may appear on areas of the body most heavily affected.
Treatments	Several chemical therapeutics are available for small rearing units where fish are easily accessible. Among these are: 1-min dip in a 2-5% solution of acetic acid and a 1-2 min dip in a 3-5% solution of sodium chloride.
Prevention	When possible, quarantine any new fish prior to placing them in with existing fish when possible. Avoid any water source with wild fish population, maintain optimum water quality, and reduce crowding.
Effects on Humans	Not harmful to humans.

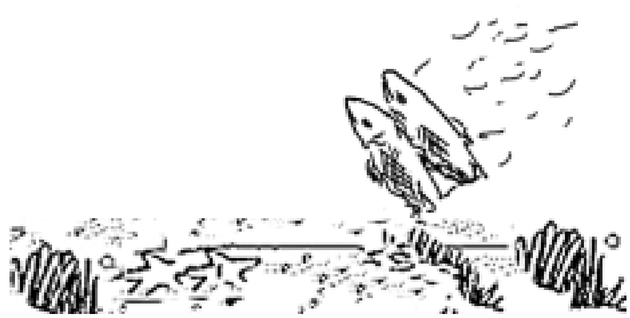
Figure 11.1. White Spot (Ich) is the largest protozoan found on fish.



White Spot (Ich)	
Causative Agent	An external protozoan of the genus and species Ichthyophthirius multifiliis (Figure 11.1). It is the largest protozoan found on fish.
Characteristics	Adults appear as white spots up to 1 mm (0.04 in.) in diameter within the epithelial layers of the skin, fins, and gills of fish. Upon maturity, the adult leaves the fish host and becomes enclosed in a cyst. Within the cyst it divides and multiplies to produce 400-2,000 young. When water conditions are favorable, the young (metacercariae) leave the cyst and become free swimming until they find a fish host. If they do not find a fish host within a few days they die. Depending on the water temperature, the entire life cycle takes from 4 d to 3 wk. Ich is most active when water temperature is between 15-24^{o}C (60 - 75^{o}F).
Effects	Fish may become lethargic and lie on the bottom of the pond. They may be seen "flashing" or rubbing against the side or bottom of a pond or tank.
Treatments	Heavy infestations affecting all fish may not respond to chemical therapeutics and may result in severe mortality. Treatment with formalin at 15-25 mg/l (0.002-0.003 oz/gal) in ponds for 5-7 d may work if started when Ich is first seen. If possible, raise water temperature to 29^{o}C (85^{o}F) for 3 wk. **Caution**: Formalin may also kill algae, which could lead to lowered dissolved oxygen levels in ponds, especially at elevated temperatures.
Prevention	Quarantine any new fish prior to placing them in with existing fish when possible. Avoid any water sources with wild fish populations, maintain optimum water quality, and reduce crowding.
Effects on Humans	Not harmful to humans.

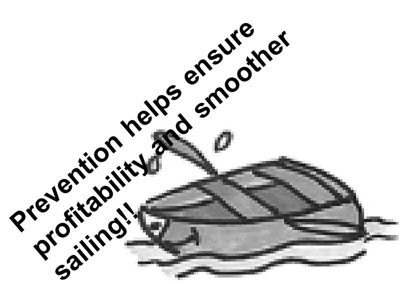
Trichodina	
Causative Agent	A single celled animal of the genus Trichodina. There are several species.
Characteristics	A microscopic saucer shaped protozoan with cilia at each end of the cell. It attaches to fish by means of an adhesive sucking disk on its underside. Trichodina reproduces both sexually and through binary fission. The presence of this parasite is usually an indication of poor water quality and/or too high a rearing density.
Effects	Large infestations of Trichodina cause fish to become lethargic, lie on their sides or rise to the surface, and may lead to death. Infestations on the gills cause the fish to secrete large amounts of mucous, which impairs respiration and often leads to a secondary bacterial infection.
Treatments	A 1 h bath in a 250 mg/l (0.033 oz/gal) formalin solution, where possible.
Prevention	Avoid high concentrations of organic material in rearing ponds, reduce crowding, and maintain good water quality by monitoring dissolved oxygen content.
Effects on Humans	Not harmful to humans.

Notes



Family = Trematodes—Flukes

Black Spot (Black Grub)	
Causative Agent	Encysted larval flukes of the genus Uvulifer spp.
Characteristics	Black pigmented cysts approximately 1 mm (0.04 in.) in diameter slightly raised from the skin, fins, mouth, or flesh of fish. The life cycle begins when the adult fluke living inside the intestine of a Kingfisher produces eggs that are released in the feces. The eggs hatch producing a free-swimming larva (miracidia) that must find a snail host within several hours or die. In the snail, the miracidia continues to develop and multiply. They leave the snail as free-swimming larvae (cercariae) that must find a fish host within several hours or die. Once they enter the fish a cyst is formed around the parasite, which remains with the fish until a Kingfisher eats it, where it then continues to develop into an adult fluke.
Effects	In general, even heavy infestations of these parasites do relatively little damage to the fish. There is some evidence that heavily infested juvenile fish may experience excessive blood loss, physiological stress, and even death. Also, fish with heavy infestations on the eyes may become blind.
Treatments	No chemical treatment.
Prevention	Removal of intermediate hosts (snails and Kingfishers) will disrupt the life cycle of this parasite. Copper Sulfate ($CuSO_4$) applied to a pond at a rate of 1.0 ppm, kills algae and reduces snail populations. Removal of roosting places around a pond discourages Kingfisher predation.
Effects on Humans	Not harmful to humans.



Yellow Grub and White Grub	
Causative Agent	Encysted larval digenetic trematodes of the genera Clinostomum spp. (Yellow Grub) (Figure 11.2) and Posthodiplostomum spp. (White Grub) (Figure 11.3).
Characteristics	Whitish or yellow pigmented cysts about 1-2 mm (0.04-0.08 in.) in diameter slightly raised from the skin, fins, mouth, or flesh of fish (may also occur in visceral organs). **Yellow grub life cycle** The Yellow Grub life cycle begins when the adult fluke (grub), living inside the throat of such wading birds as egrets or herons, produces eggs that wash out of the bird as it eats. The white grub life cycle begins when the adult fluke (grub), living inside the intestine of such wading birds as egrets or herons, produces eggs that are released from the bird through its feces. **Yellow and white grub eggs** The eggs hatch, producing free-swimming larvae (miracidia) that must find a snail host within several hours or die. In the snail, the miracidia continue to develop and multiply. They leave the snail as another free-swimming larvae (cercariae) that must find a fish host within several hours or die. Once they enter the fish, a cyst is formed around the larval parasite (metacercariae). The parasite remains with the fish until eaten by a heron or egret where it continues to develop into an adult.
Effects	Generally, even heavy infestations of these parasites do relatively little damage to the fish. Infestations of these flukes do affect the marketability by making the fish aesthetically unpleasing and difficult to sell as food.
Treatments	Several chemical treatments reduce infestations in laboratory research, although these chemicals are expensive and currently not approved for use by the US FDA.
Prevention	Removal of intermediate hosts (snails and wading birds) disrupts the life cycle of these parasites. Copper Sulfate ($CuSO_4$) applied to a pond at a rate of 1.0 ppm, for algae control, reduces snail populations and removal of roosting places around ponds discourages heron and egret predation.
Effects on Humans	Not harmful to humans.

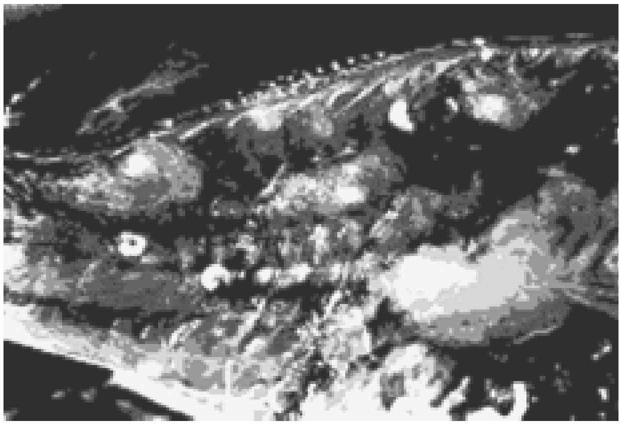


Figure 11.2. The yellow cysts identified the Yellow Grub parasite that begins life inside the throat of wading birds.

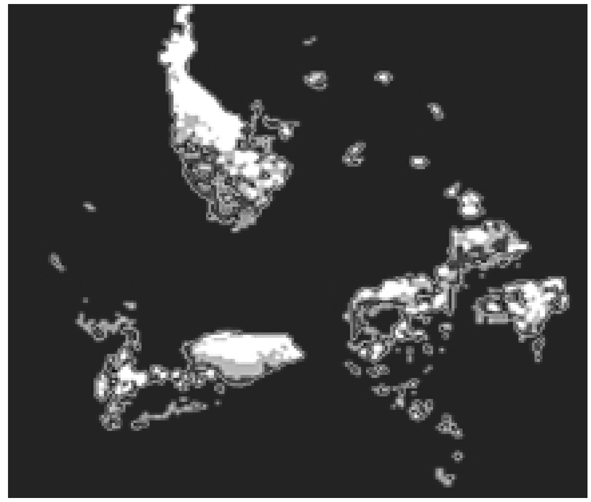


Figure 11.3. The white cyst identifies the White Grub parasite that begins life in the intestine of wading birds.

Bass Tapeworm	
Causative Agent	A cestode of the genus and species Proteocephalus ambloplitis.
Characteristics	The adult tapeworm may grow to a length of 300-400 mm (12-16 in.). It fixes itself to the intestinal wall of the final host (commonly bass and sunfish) by means of muscular suckers on its head. The body of the tapeworm consists of numerous segments called proglottids. Each proglottid has its own reproductive system. When a proglottid is filled with eggs, it breaks away from the body of the tapeworm and is passed from the fish into the water. The water dissolves the casing of the proglottid and the eggs are released. In order for the life cycle to continue, certain copepod crustaceans (the first intermediate host) must eat the eggs. In the body of the crustacean, the eggs hatch and develop into a larva called a procercoid. Once the crustacean is eaten by another fish (the second intermediate host), the procercoid larva bores through the wall of the digestive tract and invades the visceral organs of the fish such as the liver, spleen, or reproductive organs. The life cycle comes full circle when a bass or sunfish (the final host) eats the fish containing the procercoid larva, which then attaches to the intestine and develops into adult tapeworms.
Effects	Large infestations of Proteocephalus ambloplitis can cause nutritional deficiencies leading to fish sterility and death.
Treatments	No approved chemical therapeutic.
Prevention	Use parasite free brood stock. Avoid using a water source with wild fish that may be infected.
Effects on Humans	Not harmful to humans.

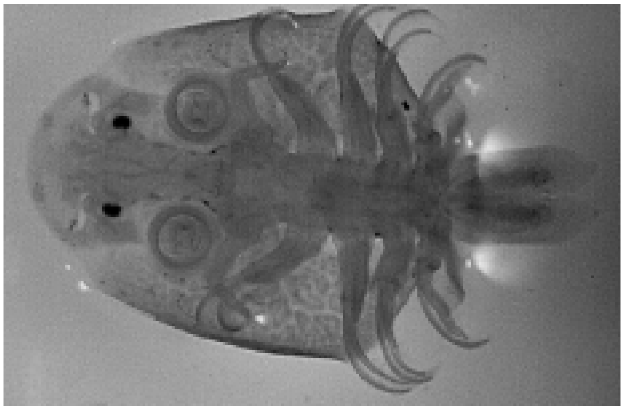


Figure 11.4. Fish lice have a clear, gelatinous disc shaped body with eight legs and a small tail and are visible to the naked eye.

Summary

There are several common parasites of sunfish that may occur in culture situations. However, because there are so few approved chemicals for treatment of parasites in aquaculture, the best "treatment" of parasites is through prevention.

Maintaining good water quality, reducing bird and snail populations, and limiting fish stress, greatly reduce parasite outbreaks.

Argulus (Fish Lice)	
Causative Agent	A monogenetic fluke of the genus Argulus (Figure 11.4).
Characteristics	A clear, gelatinous disc shaped body with eight legs and a small tail. Visible to the naked eye they generally grow to between 7-10 mm (0.3-0.4 in.). It has two hooks for attaching itself to a fish and a proboscis between its eyes, which it forces under the scales and into the skin. It feeds by sucking the fish's blood. Mating takes place in late spring and summer. The female deposits her eggs along the sides of a pond in long, flatish tubes. Females die after egg deposition and the eggs hatch in about a month. After hatching, the young will mate and attach themselves to a fish host.
Effects	Heavy infestations can cause large mortalities. This parasite is also suspected as a carrier of viral infections.
Treatments	A 1 h bath in a 200 mg/l (0.027 oz/gal) formalin solution where possible. A 15-min bath in a 2% salt solution is effective in controlling this parasite, but not practical in large scale pond culture.
Prevention	Avoid high concentrations of organic material in rearing ponds, reduce crowding, and maintain good water quality by monitoring dissolved oxygen content.
Effects on Humans	Not harmful to humans.

12

Collecting Fish Samples for Disease Diagnosis

Because disease can be common among fish, it is essential to recognize potential diseases and know what steps to take. Knowledge of how to correctly collect and ship samples of fish suspected of disease is vital for an accurate and timely diagnosis.

This chapter explains:

- Disease management
- Locating a fish health specialist
- How to collect samples
- How to ship samples
- Forms to include

12

Collecting Fish Samples for Disease Diagnosis

Authors: M. Riche and D. L. Garling, Michigan State University

Disease Management

Disease is a common problem in fish culture. It can reduce hatchery efficiency and production, which increases costs and reduces profits. Disease occurs as a result of interactions between the fish, pathogens, and the environment. Disease outbreaks occur when a fish's resistance is lowered due to stress caused by environmental fluctuations and poor management practices (Figure 12.1).

Prevention requires a minimizing of environmental fluctuations and practicing good farm management. Daily observation of fish behavior and feeding activity is necessary to allow early problem detection so that an accurate diagnosis and treatment can be performed early.

It is essential to:

- **Quickly recognize the beginning of a problem.**
- **Correctly collect samples.**
- **Correctly ship samples.**
- **Provide timely and accurate information on the problem.**

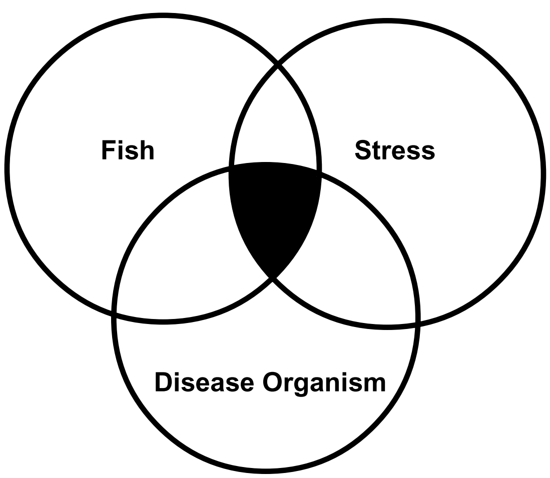


Figure 12.1. Diseases occur in fish populations when three ingredients are present: fish, a stressor, and a disease organism. The shaded area in the figure indicates disease when these three ingredients overlap.

Despite careful and conscientious prevention, disease problems can still occur. When a problem does occur, it is important to catch and correct the problem quickly. A fish culturist must have an existing plan to handle the problem.

Sound strategy is knowing:

- The nearest fish health diagnostic laboratory.
- How to collect diseased specimens.
- How to properly get the specimens to the laboratory.

It is essential that a fish culturist and fish health specialist work closely together. It is the fish health specialist's responsibility to make a timely and accurate diagnosis. It is the fish culturist's responsibility to provide the specialist with the tools to do the job. Working closely together increases the likelihood of a quick, accurate diagnosis followed by appropriate treatment.

For the fish health specialist to be effective in treating the disease, it is essential for the culturist to act quickly. A case history form should accompany the diseased fish to the laboratory. The form provides information necessary for the fish health specialist to provide a timely and accurate diagnosis.

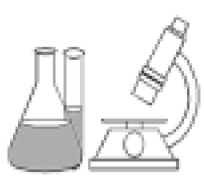
Locating a Fish Health Specialist

Specific identification of the cause of fish disease and treatment recommendations usually require examination by a fish health specialist. To find the nearest fish health specialist, contact:

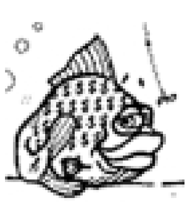
- An aquaculture extension specialist.
- State Department of Agriculture aquaculture coordinator.
- State aquaculture association.

The fish culturist should always contact a fish health specialist prior to bringing in or shipping specimens. This ensures the laboratory can handle the case in a timely and efficient manner. The fish health specialist can often suggest the most appropriate method for shipping fish when they are contacted prior to capturing the fish. This enhances the likelihood of successful shipping and a more accurate diagnosis.

How to Collect Samples



Collecting samples requires more than just capturing sick fish. It requires collecting and recording some important observations and information to help the fish health specialists make a diagnosis.



Prior to collecting fish specimens:

- Perform a water quality analysis.
- Use a pond side water quality kit to ensure an adequate analysis.

Record mortality rates

Recording any daily mortality rates is an important step in finding and understanding a disease problem.

These records are valuable tools that provide the fish health specialist important clues.

A pattern similar to curve one, where mortality is rapid and approaches 100% within 1–2 d, suggests mortality is due to a severe environmental problem.

Problems associated with this pattern include:

- Low dissolved oxygen
- Temperature and pH
- Carbon dioxide
- High ammonia and nitrite levels
- Toxic chemical(s)

To help the fish health specialist determine the cause of death, test the water for:

- Dissolved oxygen
- Carbon dioxide
- Conductivity and pH
- Temperature
- Ammonia and nitrite levels
- Alkalinity
- Hardness
- Biological oxygen demand (BOD)
- Total suspended solids (TSS), where practical

Record the results on a case history form.

Use live fish

To enhance the chance of a quick and accurate disease diagnosis, use live fish. Submitting live fish for diagnosis allows the fish health specialist to observe behavior and clinical symptoms first hand.

Live fish can either be transported directly to the disease diagnostic laboratory by the fish culturist, or shipped. Direct transport is the preferred method, since the fish generally arrive at the diagnostic laboratory more quickly and in better condition than when shipped. This method also allows the fish health specialist to ask the culturist any relevant questions regarding the disease incident.

Collecting live fish at the surface with a dip net or cast net is the best method for capturing diseased fish. Sick fish rarely eat, so using a hook and line for collecting fish is usually ineffective.

Similarly, fish caught in a seine are often healthy and can lead to a misdiagnosis.

Dead fish collection

If live fish are not available, use dead fish, but only if they have been dead <6 h. Fish dead >6 h exhibit an increased tissue decomposition, reduced parasitic loads, or can contain bacteria that confuse the results.

Dead fish that do not have red gills, normal coloring, or lack mucous are generally unacceptable for diagnosis.

Collect water sample

When sending specimens to a disease diagnostic laboratory, collect a water sample in a clean, sealed jar or bottle at the same time the fish are collected.

Before collecting the water sample, rinse the container two to three times with the water to be collected. Seal the container under water being careful to eliminate any air bubbles.

Water samples are often helpful to a specialist in making a diagnosis. Not all disease diagnostic laboratories are equipped to analyze water quality samples, however.

The fish health specialist or local extension specialist can help locate a suitable water quality analysis laboratory.

Collecting samples also requires collecting and recording important observations and information to help the fish health specialist make a diagnosis.

These include:

- Gathering representative specimens exhibiting clinical signs of disease.
- Collecting water samples.
- Recording any observations on a case history form.

Shipping Samples

Fish can be sent to the fish health specialist by a variety of methods. Contact the nearest available fish health specialist for the best shipment method.

Contact the nearest available fish health specialist for the best shipment method.

These include:

- Transporting or shipping live fish.
- Shipping fish on ice.
- Shipping frozen fish.
- Shipping fish fixed and preserved.

Remember to make certain the case history form accompanies any fish to the diagnostics laboratory to help the specialist.

Choosing specimens

When transporting or shipping sick fish, select smaller individuals. Choose representative fish that clearly exhibit clinical signs of the disease.

Some clinical signs to look for include:

- Lethargy (lack of energy)
- Refusal to eat
- Erratic swimming
- Red or white spots
- Fungus
- Discoloration of skin, gills, or eyes
- Lesions or ulcers
- Fin erosion

- Cysts
- Internal or external parasites
- Hemorrhaging.

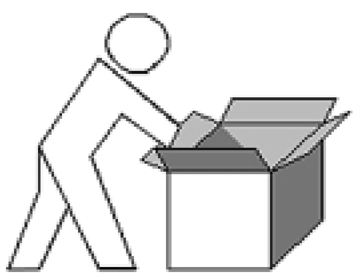
If the fish health specialist recommends shipping freshly killed fish, the fish should be euthanized prior to preparation for shipping. The most common methods for euthanasia are over anesthetization with tricaine methanesulfonate (Finquel®), benzocaine, or carbon dioxide.

To assist a fish health specialist make a diagnosis, include 2–3 healthy fish for comparison. Keep healthy fish separate from diseased fish and clearly marked.

It is important to properly prepare fish for shipping to maintain tissue integrity for an accurate diagnosis.

Plastic bag shipments

When fish cannot be transported to the fish health specialist directly, the next most desirable method is to ship them live. It is important to select specimens that will survive since disease organisms quickly leave dead fish. This can contaminate other fish and lead to a misdiagnosis.



The specimens should be shipped in heavy duty plastic bags. Double bagging is best to reduce the risk of puncture or leakage. Then:

- Fill the bags with clean water approximately 1/3 full to leave sufficient room for oxygen addition.
- Place 3–7 fish in the bag.
- Add pure oxygen to fill the bag.
- Ship the bags in a cooler or box.
  - Cardboard boxes can be used, but Styrofoam is preferred to reduce changes in temperature.
  - Pack crushed ice around the bags for long trips.

Iced shipments

If the captured specimens are unlikely to survive the trip to the disease diagnostic laboratory, they can be shipped on ice.

- Place the fish in sealed bag without water but on crushed ice.
  - Shipping fish in water increases tissue decomposition, potentially confusing the diagnosis.
  - No more than one fish should be placed in a bag.
- Ship the fish in an insulated and sealed container to prevent leaking.

Frozen specimen shipments

When live fish can't be transported or shipped immediately, they can be shipped frozen. This method is less desirable than iced specimens because thawing frozen specimens destroys cells, making some diseases hard to diagnose.

Freezing does not destroy bacteria, viruses, and fungus, so their detection is still possible.

To ship the specimens:

- Contact the carrier about their regulations regarding the shipment of packages containing dry ice.
- Place diseased fish in a heavy duty bag with a small amount of water and freeze.
- Cover the frozen fish (in the bag) with dry ice in a well insulated container.
  - A cooler
  - Styrofoam box
- Include the case history sheet.
- Ship the specimens immediately after packing.
  - Samples remain frozen for 24–36 h.

When fish are thawed, coloration is lost. This makes it important to record any unusual coloration on the case history sheet before freezing the sample.

Preserved specimen shipments

This method is the least preferred method of shipping fish to the fish health specialist. Some diseases cannot be detected after preserving the specimens in a fixative.

Bacteria, viruses, and fungus are killed by fixatives and make their detection difficult. If there is no other alternative, this method can be used to determine some parasites and the presence or absence of some bacteria (some commonly used fixatives for preserving specimens are listed in Table 12.1).

The best fixative for soft tissues, such as organs, is the buffered formalin. For whole fish, the preferred fixative is Bouin's solution. When preserving fish, it is best to use freshly euthanized fish. It is important when preserving freshly euthanized fish to:

- Use smaller fish
  - They generally preserve better.
  - They require less fixative.
- Place fish sized <5 cm (2 in.) directly into the fixative.
  - If fish >5 cm (2 in.) are used, cut open the abdominal cavity to expose the internal organs to the fixative.
- Use approximately 10 times more fixative than tissue.
  - A whole fish weighing 50 g requires 500 ml of fixative.
  - A whole fish weighing 100 g requires 1,000 ml of fixative.

After 24–48 h:

- Carefully remove the fish from the fixative and transfer to a 70% solution of alcohol.
  - Ethyl alcohol is preferred.
  - Either ethyl alcohol (ethanol) or isopropyl alcohol can be used.
- Place the specimens in a heavy duty sealed plastic bag containing the alcohol.
  - Double bag to reduce the risk of puncture or leakage.

Contact the carrier about their regulations regarding the shipment of packages containing alcohol.

Summary

Disease outbreaks reduce hatchery efficiency and production, increase costs, and reduce profits. Disease occurs as a result of interactions between fish, pathogens, and the environment. When a problem occurs, it is important to catch and correct the problem as quickly as possible.

The responsibility of the fish culturist is to:

1. Quickly recognize beginning problems.
2. Correctly collect samples.
3. Correctly ship the samples.
4. Provide timely and accurate information regarding the problem.

The fish culturist and fish health specialist who work closely together increase the likelihood of a quick, accurate diagnosis, followed by an appropriate treatment.

Note: A sample of the form to include with your fish specimens is included at the end of this chapter.

Table 12.1. Several different fixatives are commonly used for preserving fish specimens.

Bouin's Solution	
Saturated Pictric Acid	750 ml
Concentrated Formalin	250 ml
Galcial Acetic Acid	50 ml
Buffered Formalin	
Concentrated Formalin	100 ml
Distilled Water	900 ml
Sodium Phosphate Monobasic	4.0 g
Sodium Phosphate Dibasic	6.5 g
Davidson's Fixative	
95% Ethyl Alcohol	300 ml
Concentrated Formalin	200 ml
Glacial Acetic Acid	100 ml
Distilled Water	300 ml

Case History Form

Laboratory Name: ____________________

Case No.: ____________________

Address: ____________________

____

____

Date Rec'd: ____________________

Rec'd By: ____________________

Phone: ____________________

For Laboratory Use Only

Owner Information

Owner's Name: ____________________

Phone: (Home): ____________________

Address: ____________________

____

____

Phone (Farm): ____________________

General Information

Species: ____________________

Density Stocked: ____________________

Type of Culture System: ____________________

Source of Fish: ____________________

Feed Type and Amount (% BW/day): ____________________

General Specimen Information

How many specimens are being submitted? ______________________________

How are fish being submitted (live, iced, frozen)? ______________________________

Were all the fish collected from the same tank or pond? Yes ☐ No ☐

Date the fish were collected: ______________________________

Is a water sample being submitted? Yes ☐ No ☐

Describe how many fish are dying a day and how long they have been dying: ______________

____

____

____

____

Is this a recurring problem? Yes ☐ No ☐ When did it last occur? ______________

When was the pond or tank stocked? ______________________________

When was the last time the tank or pond was treated? ______________________________

What was it treated with? ______________

What was the concentration? ______________ How often was it treated? ______________

______________________________ ______________________________

Water Quality

Water Source: ______________________ Flow (gpm): ______________________

Temperature: ______________________ Dissolved Oxygen (ppm): ______________

pH: ____________ Ammonia (ppm): __________ Nitrate (ppm): ______________

Hardness (mg/L Ca CO_3): ______________ Alkalinity (mg/L $CaCo_3$): ______________

Water Color: ______________________ Total Suspended Solids: ______________

Has the color changed recently? Yes ☐ No ☐ When did it change? ____________

Was there a change in weather? Yes ☐ No ☐

Describe the change in the weather? ____

____

____

____

____

Fish Behavior

Are fish eating? Yes ☐ No ☐

Are fish listless? Yes ☐ No ☐

Are fish flashing? Yes ☐ No ☐

How long have fish been off feed? ____

Are fish swimming erratically? Yes ☐ No ☐

Are fish swimming on their side? Yes ☐ No ☐

Are fish staying near the inlet, outlet, or aeration devices? Yes ☐ No ☐




Disease Signs (Check those that apply)

Is there redness on the following: Belly ☐ Skin ☐ Side ☐ Fin ☐ Mouth ☐ None ☐

Color of Gills: Dark Red ☐ Bright Red ☐ Pale ☐ Grey ☐ Brown ☐

Appearance of Gills: Frayed ☐ Clubbed ☐ Uneven ☐ Yellow Mucous ☐

Appearance of Fins: Frayed ☐ Eroded ☐ Redness ☐ Bloody ☐ Fine ☐

Do the fish appear emaciated? Yes ☐ No ☐ Fine ☐

Are there sores or ulcers? Yes ☐ No ☐ Where are they located?________________

Are cysts present? Yes ☐ No ☐ Where are they located?________________

Are the eyes cloudy? Yes ☐ No ☐

Are the eyes bulging? Yes ☐ No ☐

Are there air bubbles in the skin, eyes, or fins? Yes ☐ No ☐

Does there appear to be an excess of mucous? Yes ☐ No ☐

Are there signs of fluid filled body cavity or bloating? Yes ☐ No ☐

Is the body coloration normal? Yes ☐ No ☐ How is coloration different?

Is there a white cottony growth on skin, gills, or fins? Yes ☐ No ☐

Give a brief description of other clinical signs: ____

Additional Comments: ____

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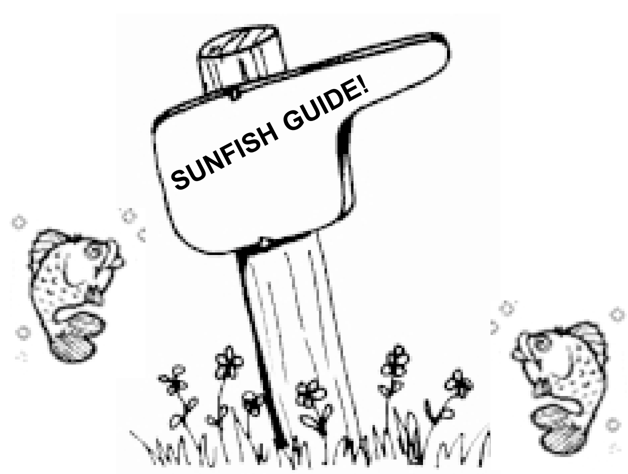


Appendices

Appendix A: Chapter Notes

Chapter 2

[1] A number of researchers have observed and verified the development of green sunfish and bluegill larvae.

Chapter 3

[1] Tidwell et al. and Webster et al. studies are excellent resources for information on the measurements of SGR and FCR in hybrid small fish.

[2] Cage culture works well in ponds with irregular shapes and can be lined up in a row for efficiency of space.

[3] For further information on hatch rates and larval rearing of bluegill, see Toetz (1966).

Chapter 4

[1] For a more in depth look at species characteristics through the last two decades, read both Pflieger (1975) and Tomelleri and Eberle (1990)'s studies.

[2] Hormones have only recently come into use as an animal drug and hormonal use and injections should only be done under close supervision of a qualified veterinarian to avoid serious damage.

Chapter 6

[1] Temperature plays an important role in every aspect of aquaculture and can positively or negatively affect sunfish survival, reproduction, and growth. This can impact the profitabality of an aquaculture venture.

Chapter 7

[1] If you are interested in aquaculture, contact the NCRAC for the resource location nearest to you. There are experts available to help you get started and review with you options, pitfalls to watch out for, and ways to help make your business successful.

Chapter 9

[1]Obtain additional information on site development for parks or campgrounds from your state's Cooperative Extension Service. For example, Michigan Extension Service Bulletin No. E-1252, Site Development Process, provides valuable information on the recreational site development process.

The Southern Regional Aquaculture Center (SRAC) also has several publications on fee-fishing, SRAC Publications 479A-482.

[2]The Fisheries and Wildlife specialist for your State's Cooperative Extension Program can provide additional information on the management of cold water and warm water ponds for fishing, weed control, a list of commercial fish sources, and assistance in controlling fish predators.

[3]Additional information on marketing your fee-fishing business is available from: Michigan State University Cooperative Extension Service Bulletin E-2409, Promoting fee-fishing operations as tourist attractions.

[4]Additional information on the benefits of incorporating may be obtained from the your state's Cooperative Extension Service.

Chapter 10

[1]Lists of approved drugs and chemicals that are of low regulatory priority can be acquired from your state aquaculture extension specialist or on the Internet through the AquaNIC (http://aquanic.org) home page.

Chapter 11

[1]Access the FDA Internet homepage for more information on drugs available for aquaculture and INADs at:

http://www.fda.gov/cvm.

Appendix B: Contacts for Additional Information

This list of names and addresses for organizations and websites offer additional information on aquaculture and fish health. These contacts can offer more in-depth resources for specific topics related to sunfish culture.

Your particular state's Fisheries Cooperative Extension Program or the North Central Regional Aquaculture Center Contact (Table B.1) provides further information on many of the topics covered.

Ted Batterson, Director of the North Central Regional Aquaculture Center at:

Michigan State University
13 Natural Resources Building
East Lansing, MI 48824-1222

The associate director of the North Central Regional Aquaculture Center can be reached at:

Joseph E. Morris
Iowa State University
124 Science II
Ames, Iowa 50011-3221

An excellent source for general aquaculture information is the Aquaculture Network Information Center (AquaNIC). The AquaNIC website contains valuable information and numerous resource links. The AquaNIC web address is: http://aquanic.org.

Table B.1. Contacts for additional or specific information.

Contact Name	Postal & Email Address	Telephone/Fax
Mr. Fred P. Binkowski	Great Lakes WATER Institute University of Wisconsin-Milwaukee 600 E. Greenfield Avenue Milwaukee, WI 53204 sturgeon@csd.uwm.edu	(414) 382-1723 (414) 382-1700 Fax: (414) 382-1705
Mr. Jeffrey L. Gunderson	Minnesota Sea Grant Extension Program University of Minnesota-Duluth 2305 East 5th Street Duluth, MN 55812 jgunderson@extension.umn.edu	(218) 726-8715 Fax: (218) 726-6556
Dr. Ronald E. Kinnunen	Michigan State University - Upper Peninsula 702 Chippewa Square Marquette, MI 49855-4811 kinnunen@msue.msu.edu	(906) 228-4830 Fax: (906) 228-4572

Table B.1 continued

Contact Name	Postal & Email Address	Telephone/Fax
Mr. Charles D. Lee	Department of Animal Science and Industry Kansas State University - Call Hall Manhattan, KS 66506-1600 clee@oz.oznet.ksu.edu	(913) 532-5734 Fax: (913) 532-5681
Mr. Jerry Mills	Brown Co. Extension Agent Agriculture 1019 1st Ave., S.E. Aberdeen, SD 57401-4799 brownco@www.ces.sdstate.edu	(605) 626-7120 Fax: 605) 626-4012
Dr. Joseph E. Morris	Department of Natural Resource Ecology and Management Iowa State University 124 Science II Ames, IA 50011-3221 jemorris@iastate.edu	(515) 294-4622 Fax: 515) 294-5468
Dr. Robert A. Pierce II	School of Natural Resource University of Missouri-Columbia 302 Anheuser-Busch Natural Resources Building Columbia, MO 65211-7240 piercer@missouri.edu	(573) 882-4337 Fax: (573) 882-1977
Dr. Chester Hill	Williston Research and Extension Center North Dakota State University 14120 Hwy 2 Williston, ND 58801 chet.hill@ndsu.nodak.edu	(701) 774-4315
Staff	Department of Animal Science Illinois-Indiana Sea Grant College Program Purdue University 1026 Poultry Building West Lafayette, IN 47907-1026	(765) 494-6264 Fax: (765) 494-9347
Ms. Laura G. Tiu	Piketon Research & Extension Center Ohio State University 1864 Shyville Road Piketon, OH 45661-9749 tiu.2@osu.edu	(740) 289-2071 Fax: (740) 292-1953
Mr. Michael D. Plumer	University of Illinois Extension Carbondale Extension Center Dunn-Richmond Economic Development Center 150 E. Pleasant Hill Rd. Carbondale, IL 62901 plumerm@mail.aces.uiuc.edu	(618) 453-5563

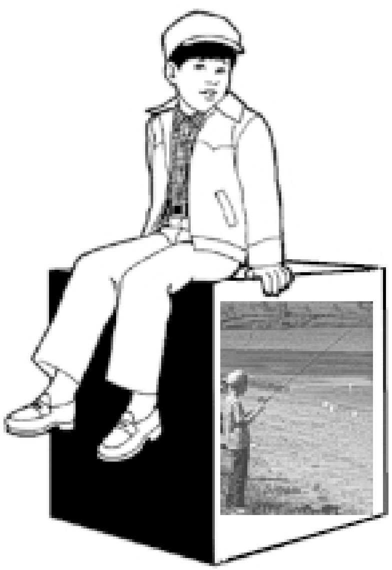
Appendix C: Abbreviations, Acronyms, Common Names

AquaNIC	Aquaculture Network Information Center
BOD	Board of Directors
°C	degree of Celsius
CES	Cooperative Extension Service
cm	centimeter
d	day
°F	degree Fahrenheit
FCR	Food Conversion Rate
FDA	Food and Drug Administration
FFO	Freshwater Farms of Ohio, Inc.
FMA	fish meal analog
ft, ft^2, ft^3	foot, square foot, cubic foot
g	gram(s)
gal	gallon(s)
GxB	hybrid sunfish
h	hour(s)
ha	hectare(s)
HACCP	Hazard Analysis Critical Control Points
IAC	Industry Analysis Council
IAFWA	International Association of Fish and Wildlife Agencies
IGF-1	insulin-like growth factor
in.	inch(es)
INAD	Investigational New Animal Drug
ISU	Illinois State University Iowa State University

JSA	Joint Subcommittee on Aquaculture
kg	kilogram(s)
L	liter(s)
lb	pound(s)
m, m^2, m^3	meter, square meter, cubic meter
µm	micrometer
mg	milligram(s)
min	minute(s)
mL	milliliter(s)
MSU	Michigan State University
N	number
NADA	New Animal Drug Applications
NCR	North Central Region
NCRAC	North Central Regional Aquaculture Center
NDSU	North Dakota State University
OAA	Ohio Aquaculture Association
OSU	Ohio State University
oz	ounce(s)
ppm	parts per million
Purdue	Purdue University
RAC(s)	Regional Aquaculture Center(s)
RAS	recirculating aquaculture systems

SGR	specific growth rate
SIUC	Southern Illinois University-Carbondale
TC	Technical Committee (TC/E = Technical Committee/Extension; TC/R = Technical Committee/Research)
TL	total length
TSS	total suspended solids
UM	University of Missouri
UMn	University of Minnesota
USDA	U. S. Department of Agriculture
USFWS	U.S. Fish and Wildlife Service
yd	yard
YOY	young-of-the-year
yr	year(s)
>	more than
<	less than

Other Abbreviations and Acronyms



Common and Scientific Names of Sunfish

Common Names	Scientific Names
Bluegill	*Lepomis macrochirus*
Redear	*Lepomis microlophus*
Green Sunfish	*Lepomis cyanellus*
Pumpkinseed	*Lepomis gibbosus*
Warmouth	*Lepomis gulosus*
Longear	*Lepomis megalotis*
Red Breast Sunfish	*Lepomis auritus*

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