LOST RIVER SUCKER CULTURE MANUAL

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Introduction

The following procedures have been used by the California-Nevada Fish Health Center for the culture of Lost River Suckers (*Deltistes luxatus*) in 2006 and 2009-2015. These procedures build upon practices used by other catastomid culture facilities and a working knowledge of specific requirements for suckers from Upper Klamath Lake, Oregon. This manual is directed towards fish culturists and biologists that will be involved in the propagation of Lost River Suckers. These culture methods may also be applicable for Shortnose Sucker (*Chasmistes brovirostris)*.

Considerations and Concerns:

1) Chronic mortality and "fall out" of larval fish transitioned from live prey to pelleted feed has been a concern in LRS propagation. Transition from live Artemia to dry pelleted diet (OTOHIME, manufactured by Marubeni Nisshin Feed Co., Ltd, Tokyo, Japan, and purchased directly from Reed Mariculture, Inc. Campbell, CA, 95008, USA) occurs when larval fish are between 10-20mm. Potential factors associated with this mortality event:
    a. Poor feed acceptance due to texture, size, or palatability.
    b. Mismatch in digestive development and feed. Poor absorbance.
    c. Micronutrient imbalance
    d. Lack of proper microbiome to aid in digestion and absorption. Given the benthic / epiphyte diet of sucker fry in Upper Klamath Lake, bacterial flora may be a critical element in larval nutrition.
2) Gill epithelial hyperplasia (figure 2) is another propagation concern. This condition can overlap with fallout of 20 -30mm fry but also occurs in older fish. If affected fry are handled (e.g. netted) they often lose equilibrium and die. Both respiratory and ion imbalance are likely causes.

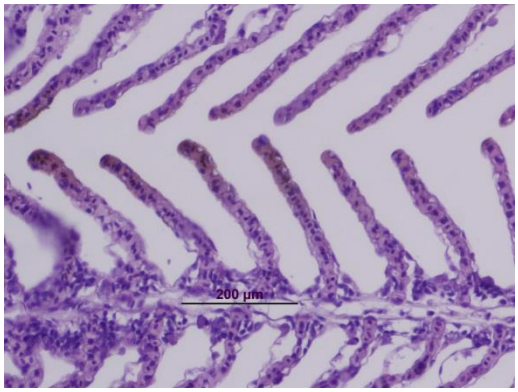


Figure 1. Photomicrograph of a normal LRS gill.

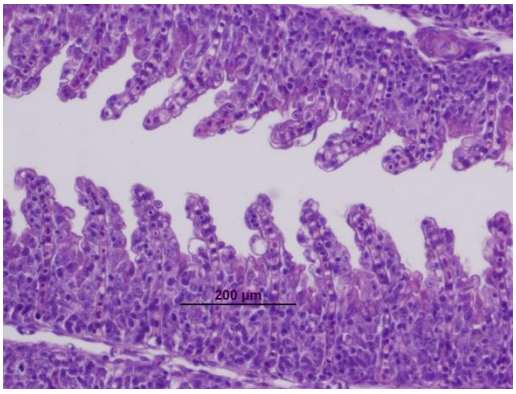


Figure 2. Photomicrograph of a hyperplastic LRS gill. Note that there is little no intestinal space between gill lamellae.

3) Premature hatch (larvae hatching prior to development of fully formed eyespots):. There does not appear to be a difference in survival between these larvae and those that hatch once eyed eggs are observed in a hatchery setting. Initially, it was thought elevated water temperatures could be the primary mechanism for early hatch. Early hatch has occurred in eggs held at 9 - 16°C.
   The primary concern with early hatch larvae in a hatchery setting is;
   - Fish will remain quiescent within their rearing environment for longer periods of time than their fully developed cohorts. The lack of mobility increases mortality due to bacterial or fungal growth entrapping sedentary larvae. Early hatch larvae become extremely labor intensive at this point. To minimize heavy loss, quiescent larvae are hand sorted daily until they are observed actively swimming within the water column.
4) Variable and poor growth of fry fed dry diets. Since 2006 the FHC has tried a wide range of commercially available feeds in an attempt to achieve consistent growth. Diets such as; commercial salmon diets, marine larval diets, algal feed, freeze dried copepods, and others have been tried. OTOHIME, a transitional marine derived feed top coated with vegetable oil has been the most readily accepted diet. Feed consumption and palatability are in constant flux. Monitoring feed consumption is an important task and must be performed daily to maintain clean rearing environments, maximize growth, and minimize the buildup of harmful bacteria or fungus. It is necessary to perform accurate pound counts every two weeks as feed adjustments are made regularly. Tank density and location of feed presentation should be evaluated on weekly basis to enhance feed rate of the tank population.

Spawning Procedures

Broodstock:

1. Egg collection site(s) should be close to natural spawning areas to avoid transportation of adult stock.
2. Feral broodstock are captured near their spawning grounds to target mature fish. Only gravid fish are selected for spawning.
3. All brood fish are check by lightly squeezing the abdomen. If gametes are given freely they are immediately collected and the fish released.

Egg Take Procedures:

NOTE: Female Lost River suckers are known to contain 44,000 – 236,000 eggs (NRC 2004). Female Lost River suckers use in 2014 & 2015 spawning operations by the FHC averaged 181-272 eggs/g.

1. Prior to spawning, establish a fertilization ratio of _x_male(s) to _y_female(s) (1 male can be used to spawn multiple females).
2. Obtain brood fish utilizing trammel nets and select only gravid individuals.
   - **DO NOT** attempt to force eggs from a female; **DO NOT** squeeze hard. If fish is gravid eggs will easily be expelled from the vent.
3. Thoroughly dry fish with soft towel prior to expressing gametes.
   - **DO NOT** allow water from hands, gloves, etc. to be introduced to eggs or milt prior to fertilization. When water is added prematurely to eggs water hardening will close the micropyle and reduce fertilization. **Do Not** allow water or feces into milt sample. If water is added prematurely spermatocytes become activated and will not be motile during fertilization.
4. Collect eggs and sperm of individual fish in separate collection containers. The FHC used disposable Dixie cups. If excessive feces are expelled into the semen sample it should be discarded.
5. Once eggs are collected they are held in separate sealable containers (plastic Tupperware or glass Pyrex containers are recommended).
6. At the time of fertilization, add milt directly to the container containing eggs.
7. The FHC recommends collecting and retaining desired eggs lots first. Obtain milt and add immediately to initiate fertilization. We have had successful fertilization when gamete collection and mixing occur within 10 minutes.

8. Add enough lake water to cover all eggs to activate spermatocytes and allowed to incubate for 15 minutes.
9. After fifteen minute incubation period, water, milt and, ovarian fluid is carefully poured off.
10. Lake water is again added to the container and the fertilized eggs can now be sealed and placed into an ice chest (maintain temperature) for transport.

Transport of Eggs:

1. Transport of eggs to the rearing facility should occur in sealed containers held within an ice chest to maintain temperature. Multiple stops to monitor dissolved oxygen and temperature may be necessary depending of transport

distance. Small doses of oxygen can be bubbled into the transport containers to maintain O_2 levels if necessary.

a. It is useful to obtain dissolved oxygen and temperature readings from the spawning location for reference.

Handling of Eggs:

1. Upon arrival to the hatchery facility fertilized eggs are rinsed 2x for 5 minutes with a 1:100 iodophor solution; use clean tempered non-chlorinated water to make iodophor solution.
2. Sucker eggs are highly adhesive. Eggs will be clumped together upon arrival. Gently break egg clumps by hand and gently stir to ensure adequate contact with iodophor.
3. Discard the initial iodophor rinse for removal of organics and repeat the disinfection process to ensure adequate surface contact with fertilized eggs.
4. Following iodophor treatment eggs are ready to be placed into prepared incubators.

<u>Incubation Procedures</u>

NOTES: Four points to consider during the incubation of sucker eggs, larvae, and fry:

- Hatchery water supply should be free of serious fish pathogens (eg. Filtered UV sterilization or well water).
- Rearing unit(s) need to be kept clean to maintain proper water quality parameters.
- Maintain proper water temperatures and low light conditions (minimize UV exposure to eggs and larvae as much as possible).
- Isolate eggs and fry into many small sized lots to prevent massive losses.

Preparation for Incubation:

1. Insure all upwellers have been properly disinfected with iodine (minimum of 100ppm) and rinsed thoroughly.
2. Each upweller is fed by its own water line with enough water flow/pressure to gently roll the eggs within the upweller.
    - The FHC recommends using PVC gate valves on the water delivery manifold. Use a PVC petcock at the upweller. This will allow for finite water control. Avoid the use of standard PVC ball valves as water flow control is inadequate.

- Good rolling action aids in the prevention of eggs adherence and helps to minimize fungal infection.

3. Upwellers should be placed within a contained tank. As eggs adhere and become clumpy the upweller will overflow or become clogged. Secondary containment of overflow eggs and larvae is crucial.

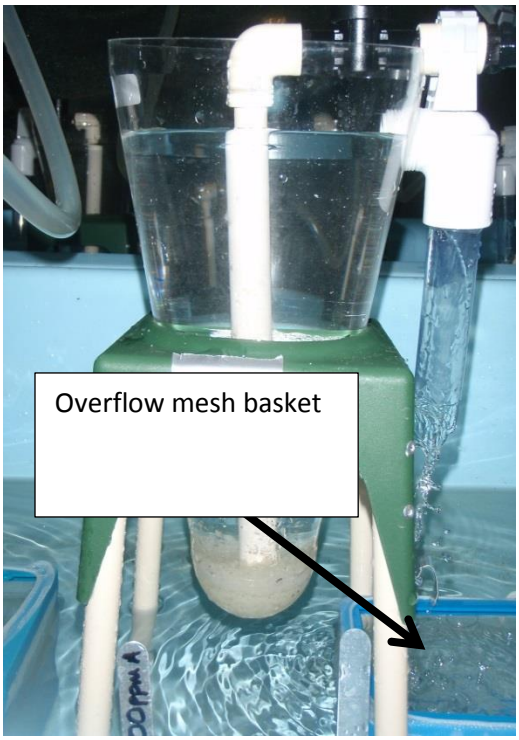


Figure 3. Mini upwelling jar from Pentair Aquatic Ecosystems

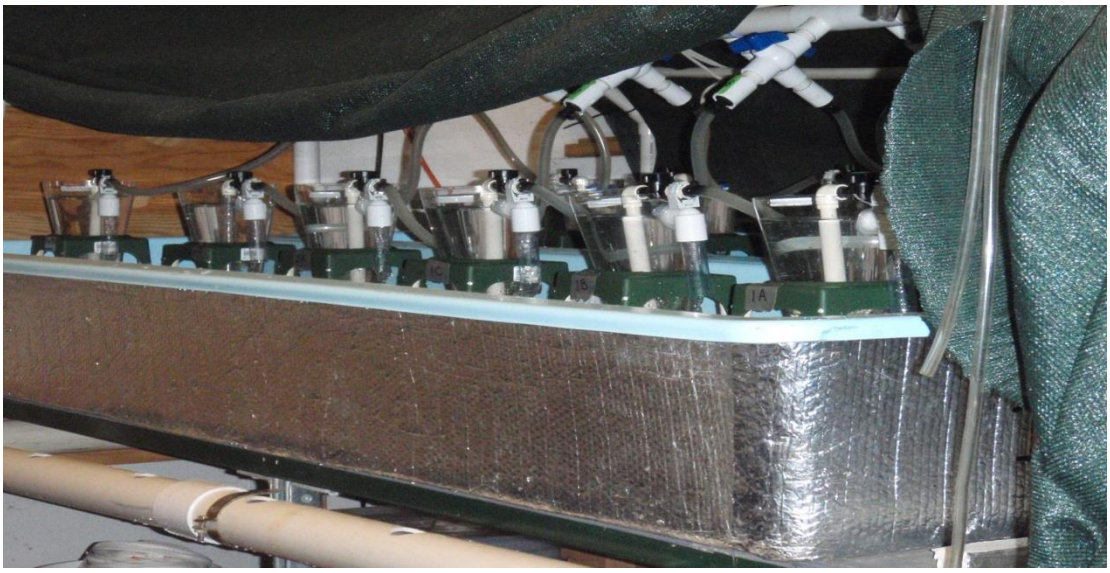


Figure 4. Individual upwellers and water supplies for egg incubation

Figure 5. Covering eggs during incubation aids in minimizing UV light. Constant exposure may cause retinal damage, increase chance of early hatch and, increase bacterial / fungal growth withing the rearing unit.

Incubation of Fertilized Eggs:

1. Load incubators at low densities.
    - The FHC recommends loading no more than 30mL's of water hardened eggs in the ~1.75L mini upweller (Pentair Aquatic Ecosystems part number J32 & J32H for PVC upweller stand).
    - Lower densities equivocate to better rolling action which will minimize fungal growth.
2. Monitor eggs twice daily to ensure upwellers are operating properly (adequate flow to gently roll eggs) and signs of fungal growth.
    a. It is encouraged that a log book be used to record daily observations.
    b. When eggs begin adhering to one another, gently separate them by hand (clumping of eggs encourages the growth of fungus).
3. Loss of viable eggs due to fungus can be reduced if egg density is extremely low (no contact) or formalin treatments are provided from 1 day post-spawn to eyed egg stage (days prior to hatch). FHC procedures are as follows:
    a. Turn off inflow to upweller
    b. Calculate exact volume of water within the upweller to be treated
    c. Calculate the amount of Formalin to be used (the FHC recommends treating between 1000-1300mg/L static for 10min.)

- Calculation example:

  Treating at 1300mg/L

  1300mg/L * 1.75L (volume of upweller) = 2275mg
  2275mg / 1000 = 2.275g
  2.275g / 1.081 (specific gravity of Formalin) = 2.1mL's

  Add 2.1mL's of Formalin to a 1.75L upweller static for 10min.

d. Add Formalin to upwellers and gently mix multiple times over the 10min time period using a feather, small net or, spatula to insure Formalin contact with all eggs.

- Multiple upwellers can be treated at one time.

e. Restore water flow to upweller and insure gentle rolling is achieved.

❖ ***Any individual responsible for administering Formalin treatments were proper personal protection equipment, be educated on Formalin health hazards via Safety Data Sheet and be given safety orientation by supervisor or the facilities Safety Officer.***

NOTE: The decision to treat with Formalin should be made prior to or within 24hrs of incubation. The treatment of eggs with Formalin is a preventative measure used to inhibit or slow the growth of fungus. <u>Should Formalin be used it must start day1 post arrival and continue each day until the first observed hatch at which time treatments must stop to avoid damaging newly hatched larvae.</u> Experimental work utilizing different concentrations of Formalin were conducted at the FHC. Fertilized eggs used in the experiment were held at extremely low densities (300 eggs per 2.8L rearing basket. Baskets measured 16.5cm x 12.7cm x 13.3cm; frames covered by a **500µm mesh bag). Results indicate that there was no difference in egg loss due to fungus when treated with formalin for 10 minutes at concentrations ranging from 0-1300ppm. Observationally however, there was a slight increase in the number of eyed eggs in the 1300ppm treatment group when compared to their untreated cohorts.** ***It appears that if eggs are held at low enough densities (>30mL's of water hardened eggs per 2.8L rearing baskets) and do not contact each other, Formalin may not needed.***

4. When fungal outbreaks occur and the facility want to maximize larval hatch, eggs are sorted by hand. This process is highly labor intensive but critical. FHC sorting procedures are as follows:
   a. Clumps of eggs with observed fungal growth are removed from the upwellers and placed into glass Pyrex sorting dishes containing tempered water.
   b. Eggs and dish are placed onto light table for better background lighting.
   c. Eggs are teased apart by hand, forceps and, disposable clear plastic bulb pipettes.

d. Dead eggs are separated and weighed or counted to calculate approximate loss then placed into an iodophor disinfection container (<100ppm) before being froze then disposed.
e. Viable eggs are retained in a 2.8L isolation rearing basket with 500µm mesh netting and placed into the secondary retention tank with the upwellers. Contaminated eggs no longer come into contact with eggs held in upwellers. Salvaged eggs are given the chance to develop.

NOTE: The retention of any potentially viable egg may be necessary when working with federally list fish species. Retaining salvaged eggs will result in additional hatch.

5. Any hand tools, nets or cleaning brushes are disinfected with iodophor (<100ppm) after use in any incubation unit.
6. Additional screening measures are incorporated into the rearing units at the FHC to ensure no larvae are incidentally passed through the effluent system. All water leaving the rearing unit is passed through are 500micron mesh bag. Bags are checked twice daily. This method is highly efficient at catching eggs, larvae and waste. Mesh bags tend to foul within 1-2 days. The FHC recommends changing bags daily. Used bags are placed into 5 gallon buckets containing 5ppm chlorine for a minimum of 20 minutes prior to being disposed of or rinsed sufficiently for reuse.

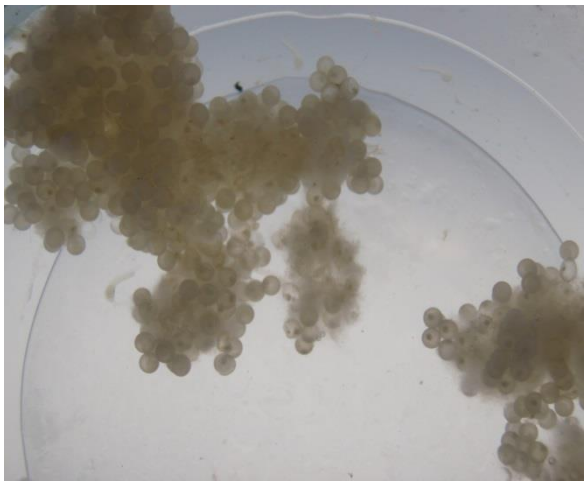


Figure 6. Clumped eggs with fungus.

Incubation of Eyed Eggs:

1. Removal of all dead eggs is critical as they provide a medium for fungal growth.
    - Sorting of live and dead eggs is performed manually as described in the previous section and all tools used are disinfected between units. FHC sorting procedures are reiterated and described below:

        a. Water to incubator is turned off

b. Eggs and water are poured from incubator into a Pyrex glass dish
c. Glass dish is placed onto a light table
d. A pipette is used to sort dead/fungal eggs from “good” eggs
e. “Good” eggs are placed in a new Pyrex dish filled rearing unit
f. Once all eggs are sorted water is turned back on to the upweller and “good” eggs are poured back into upweller.
g. Dead/fungal eggs are disinfected and disposed of.

Figure 7. Manual sorting of eggs using the Pyrex dish, light table and, clear plastic pipette.

2. It is important to disinfect and rinse upwellers while sorting and prior to the reintroduction of viable eggs (<100ppm iodophor).
    a. Fish culture equipment should be disinfected before it is moved between modules.
    b. It is recommended that there be an adequate amount of tools and upwellers to maintain a steady work flow while disinfection is taking place.
3. Heating or chilling of water for egg or larvae incubation may be necessary if ambient water temperatures increase or decrease out of range. The FHC recommends rearing temperatures of 13˚C however temperatures ranging 9-14˚C are acceptable.
4. Incubators should be checked twice daily for any indication of problems or signs of early hatch.

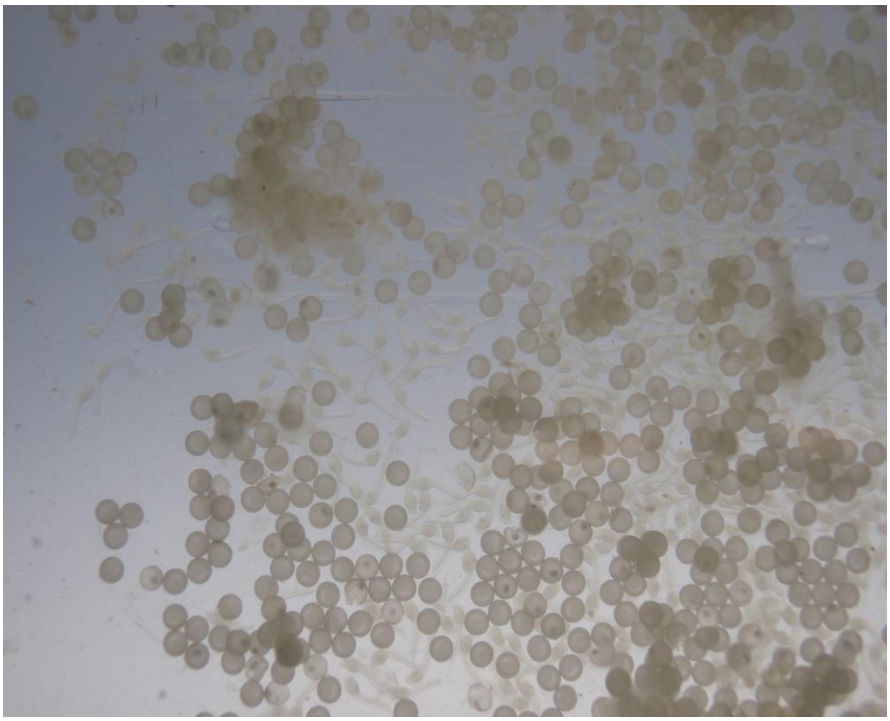


Figure 8. Early hatch; larvae with no visible eye spots

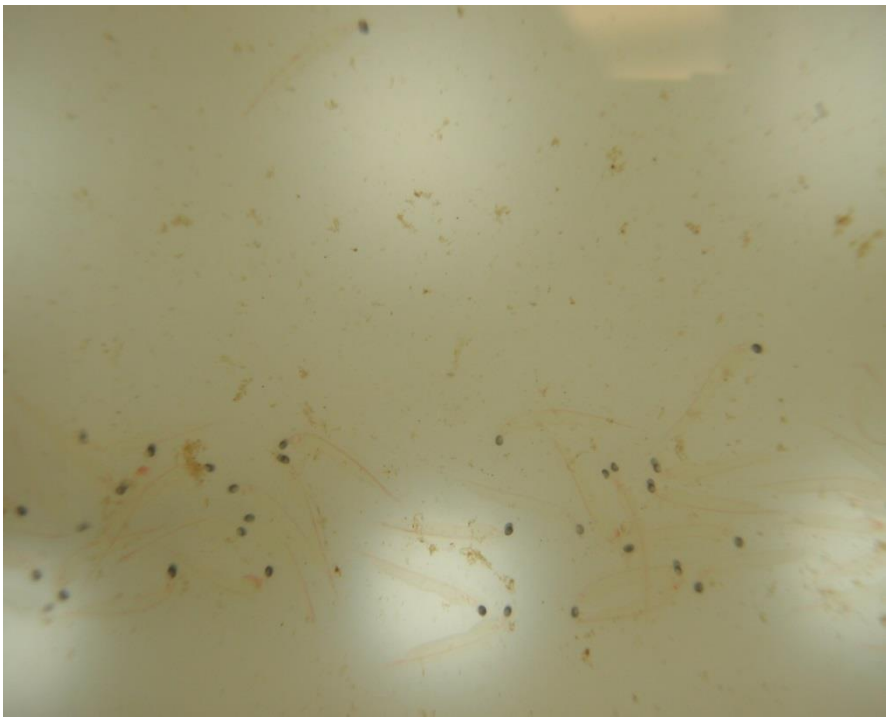


Figure 9. Fully developed larvae withvisible eye spots

Hatching &Tanking of Lost River Suckers

The Fish Health Center has explored a number of ways in which to tank larval LRS, our primary method is described below.

1. As larvae hatch within the upwellers they are sorted, counted and separated from unhatched eggs by hand. Larvae are placed into a 6.5” x 5” x 5.25” hanging basket with a 500 micro mesh (Pentair Aquatic Eco-Systems part number A934) within a 40L aquarium. Unhatched eggs with no signs of fungus are returned to upwellers.
    - Larvae Sorting Steps:
        a. Water to incubator is turned off
        b. Larvae, unhatched eggs, and water are poured from incubator into a Pyrex glass dish
        c. Glass dish is placed onto a light table
        d. A clear plastic disposable pipette with the tip slightly modified by cutting off approximately 0.25” to increase the boar size is used to sort larvae from unhatched eggs
        e. Larvae are counted and placed in a new Pyrex dish with tempered water from the rearing system
        f. Once all larvae are removed and counted water is turned back on to the upweller for unhatched eggs.
        g. Larvae are then poured into the hanging rearing baskets described above.
        h. Dead / fungus eggs are disposed.

NOTE: Hatching can take place over multiple days (1-5) depending on the number of larvae that hatch prematurely or develop fully. Hatch rates can be increased by retaining eggs within a Pyrex dish and left on the light table. Water temperatures will slowly increase and induce larvae into hatching. This method of hatching is used at the FHC rearing facility when fungal outbreaks jeopardize the survival of eyed eggs. Culturists will observe “shock hatching” taking place within minutes of introducing eggs to the light table. The FHC has held eggs in glass dishes containing approximately half volume of water on a light table 00:05min – 17hrs to increase hatch and survival of larvae entrained by fungus.

Figure 10. Rearing baskets containing eggs and larvae. Clear PVC pipe from upwellers can be seen within rearing baskets. This is done to retain any eggs that have been pulled through the PVC pipe due to buoyancy or overflow.

2. Once larvae are actively swimming within the rearing nets they are ready to receive live prey items (refer to **Hatching and Feeding-out of Artemia Nauplii** section on page 17).
    a. LRS will remain in rearing baskets until all larvae are actively hunting live prey items and traveling freely within the water column. Larvae are translucent. The orange pigmentation of Artemia makes the observation of consumed prey items easy.
    b. Not all hatched larvae survive. As fallout begins dead larvae accumulate on the bottom. Fungus will develop quickly. Surviving larvae unable to swim will become trapped and die. Nets must be checked twice daily and all dead larvae removed.
        - Dead larvae are removed from the net via pipette.
            a. When dead larvae become impinged on the net and can't be removed by pipette live larvae are carefully placed in a Pyrex dish with water from the rearing system by untying and removing the mesh net from the basket frame.
            b. The net turned inside out and gently swayed (to dislodge live larvae from the net) back and forth in the Pyrex dish.
            c. A new mesh net is then put on the frame, the basket hung back into the aquarium.
            d. Larvae are poured from the Pyrex dish into the clean net.
            e. The dirty net must then be disinfected and cleaned with iodophor (<100ppm) and rinsed for future use.

Benefits of using individual rearing baskets:

1. Allows for separation and tracking of individual spawning crosses / family groups.
2. Mesh size is adequate for water flow and retains larvae.
3. Enables culturist to remove, handle or treat individual fish populations without risk to all other fish / nets within the aquarium
4. Observationally, larvae are not actively swimming at the time of hatch. Rearing nets allow them to continue development without the constant agitation induced by upwelling.

NOTE: Artemia cysts and dead nauplii will settle on the net bottom as well as on non-swimming larvae. This results in bacterial growth due to the protein rich environment and fouls the rearing net. Fouled nets must be changed, disinfected, and cleaned daily or every other day. This process is very labor intensive as larvae become entrapped under the net's frame, take extreme caution when handling nets containing larvae

3. At approximately 10mm, actively feeding larvae are transferred into larger rearing tanks. The FHC uses 40L acrylic aquaria with adequate water flow, desired water temperature (14-16˚C) and, aeration.
    - Current practices at the FHC are:
    a. While holding rearing baskets within the 40L aquarium, untie and remove 500 micron mesh netting.
    b. Carefully turn netting inside out releasing larvae into the aquarium.
    c. Gently move net left to right freeing any additional larvae still in the net.
    d. Larvae are now free swimming within the 40L aquarium.
    e. Rearing nets are disinfected with iodophor (<100ppm) and rinsed for future use.

NOTE: Larvae can be released directly in the aquarium as described above or a pound count performed at this time. The function of a pound count is to obtain an approximate biomass and estimate the number of fish within a given population. Procedures for a larval fish pound count are as follows:

1. ***Obtain a scale capable of reading 0.01 – 0.001g.***
2. ***Tare a 500-1000mL beaker containing approximately half volume of water.***
3. ***Fill glass Pyrex dish approximately a quarter the way full.***
4. ***Place rearing basket containing actively swimming larvae into Pyrex dish.***
5. ***Untie and free larvae into Pyrex dish as previously described.***
6. ***Using a small nylon net, scoop out subset of larvae, drain off excess water and place larvae into tared beaker and record the weight in grams.***
7. ***Repeat this process until all larvae from the rearing net have been weighted.***

a. ***It will be necessary to pour weighed larvae into the 40L aquarium and tare new beaker before proceeding. This may be performed a number of times during the pound count process.***

4. ***Obtain 20-30 larvae, euthanize using MS-222, weight individual larvae (balance will need to be capable of reading 0.001g for accurate weights), record weights. The FHC also recommends obtaining standard lengths using digital calipers at this time.***
5. ***Calculate the average weight of larvae.***
6. ***Divide the mean weight of individual larvae by the total biomass to obtain estimated number of fish per aquarium / rearing unit.***
7. ***Pound count data will be used to track growth and establish daily feeding rations.***

8. Larvae remain in 40L aquaria until they are approximately 15-20mm. Once larvae reach size they are transferred to 450L (120gal) fiberglass rectangular tanks.
    - Transferring procedures are as follows:
    1. Using 6-8 inch nets gently scoop up larvae and transfer to larger rearing tank.

NOTE: This is an opportune time to perform an additional pound count as described above. Large weighting vessels may be needed. Scales that read 0.01g are acceptable.

9. Fish are still being offered live prey items at this time however; pelleted feed is now being introduced into daily feedings.
    a. Using the data obtained from the pound counts, offer 2-3% body weight per day of dry feed by hand multiple times over 8hr work day.
10. Once larvae are observed feeding on dry pelleted feed they are weaned off Artemia over a 2-3 week period.
11. Once LRS have reached 40-50mm they are moved into a larger 670L (180gla) fiberglass rectangular tank for grow out.
    - Transferring procedures are as follows:
    1. Using 8-12 inch nets gently scoop up larvae and transfer to lager rearing tank.

NOTE: This is an opportune time to perform an additional pound count as described above. Large weighting vessels may be needed. Scales that read 0.01g are acceptable.

NOTE: The time and number of fish that can be housed in any rearing container / tank is density dependent and will fluctuate based on size, weight and feeding response. It is the opinion of the FHC that density considerations are determined by the available surface area of the tank bottom required for foraging. Observationally, LRS begin feeding / foraging on the tank bottom (benthic orientation) between 20-30mm.

Hatching and Feeding-out of Artemia Nauplii

NOTE: Multiple culture vessels of Artemia must be started each day to insure adequate amounts of live prey items are viable for feeding larvae. FHC recommends having 2-3 culture vessels running at one time; 1) vessel containing hydrated cyst; 2) vessel containing Artemia at 24hrs old; 3) vessel containing Artemia at 48hrs old.

Hatching Artemia:

1. Hydrate 8-10 grams of Artemia cysts (Pentair Aquatic Ecosystems part number BS90) in 1L of fresh water within the culture vessel (Pentair Aquatic Ecosystems part number BS6 & HS1 for aluminum stand)for 30-45minutes with gentle aeration (This is optional however rehydrating cysts in freshwater tends to increase hatch rate).
2. Add 6-7L of pre tempered 25ppt saltwater
3. Insure aquarium heaters within each culture vessel are set at 75F (~24˚C) and overhead lights are turned on (use standard household fluorescent bulbs, two aquarium heaters are sufficient unless ambient air temperatures are extremely cold)
4. Artemia will hatch overnight and will be used the following morning (Multiple culture vessels are utilized in an alternating fashion. Cysts started in the morning will be used 24hrs later(the following morning) and 32hrs later (in the afternoon)
5. Should a DHA additive (Selcon™ Concentrate; American Marine Inc., Ridgefield, CT) be incorporated, initially follow manufactures recommended volumes.

Feeding Artemia in the Aquariums:

1. Removed aerator from hatcher and turn overhead light off
2. Allow Artemia to settle to the bottom of the culture vessel for ~5minutes (note that cysts will settle out as well)
3. Open valve and drain 500mL of hatcher contents into a beaker
4. Pour the contents through the 250 micron sieve, gently rinse in fresh water (minimizes saltwater shock on larvae), and add contents to 500mL glass bottle containing fresh water.
5. Shut off water inflow of aquaria/trough to be fed.
6. Once excess water has drained out, allocate the contents of the feeding bottle to **ALL** aquariums containing larvae.

Note: The AM feeding each morning will contain the highest number of Artemia. There are enough Artemia nauplii hatched from 8-10grams of cysts to feed 20,000 – 30,000 larvae twice a day. If feedings seem light, increase the weight of cysts used.

7. Leave water shut off for 10minutes before restoring flow
8. Repeat process for each aquarium containing larvae
9. Check water flow before leaving

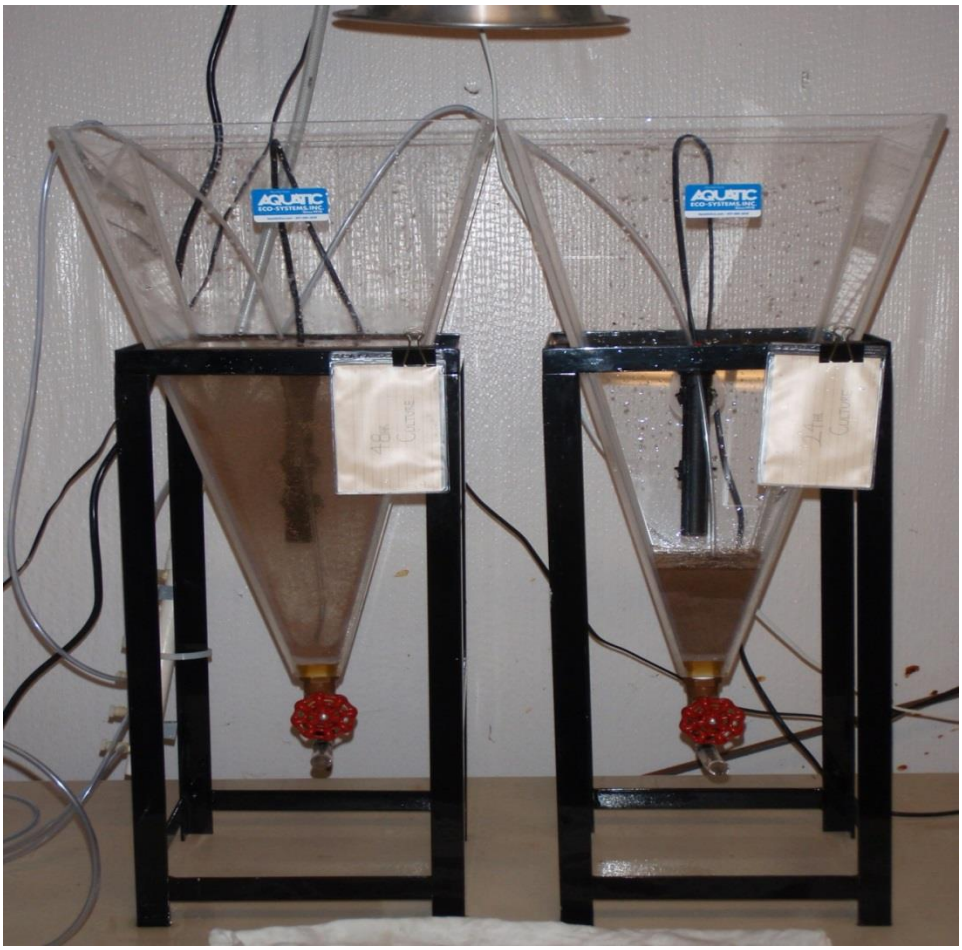


Figure 11. Artemia culture vessels with 24hr containing 1L of freshwater for rehydrating cycts and 48hr drained halfway after am feeding.

Pelleted Feed

NOTES: LRS feeding responses change regularly. The FHC monitors feed consumption daily. Feed amounts are slightly increased daily (1-5g) when no food is observed on the tank bottom. Feed allocations are decreased slightly (1-5g) daily when food is observed accumulating. It is crucial that fish receive adequate feed allotments for growth however; continual monitoring is required so excess feed does not accumulate within the rearing unit increasing bacterial or fungal growth.

1. The FHC uses the marine derived pelleted diet OTOHIME (manufactured by Marubeni Nisshin Feed Co., Ltd, Tokyo, Japan, and purchased directly from Reed Mariculture, Inc. Campbell, CA, 95008, USA) lightly top coated with vegetable oil.
2. During larval transition from live prey items to OTOHIME small amounts of pelleted feed are offered multiple times throughout the day by hand (previously described). Fish are observed for active feeding response. Dry feed is incorporated with Artemia for 2-3 weeks before being weaned from live prey items (described in previous section).
3. Once weaned, larvae are fed 5% body weight per day (BW/d) by hand over multiple daily feedings.
    a. Obtain tank biomass (describe in previous section)
    b. Multiply desired feed percentage by tank biomass to get grams of feed per day.
        - Ex. 1000g of fish per tank
        - Desired feed rate of 5% body weight per day
        - 1000g of fish x 0.05 = 50g of feed per day

4. Feed rations will range from 1-5% BW/d over the rearing period.
5. Feed sizes are increased incrementally every 2-3 weeks. Fish will receive pelleted feed size “A” for two weeks then, a blending of feed size “A” + “B” (ratio of 1:1) for one week before only receiving only “B” for two weeks. Then “B” + “C” for one week before only receiving “C” etc.
        a. Fish may stall on a given pellet size and the feeding regime will need to accommodate.
        b. Do not allow fish to stay on smaller feed sizes for long, growth will slow. The FHC will normally offer smaller feed for no more than 1-2 weeks to accommodate smaller fish.
6. 24hr Belt feeders are used for the remainder of the rearing period.

- When fish are actively feeding there will be little to no food observed on the tank bottom overnight. The FHC infers that active foraging takes place at night. Offering feed 24rhs / day is beneficial.

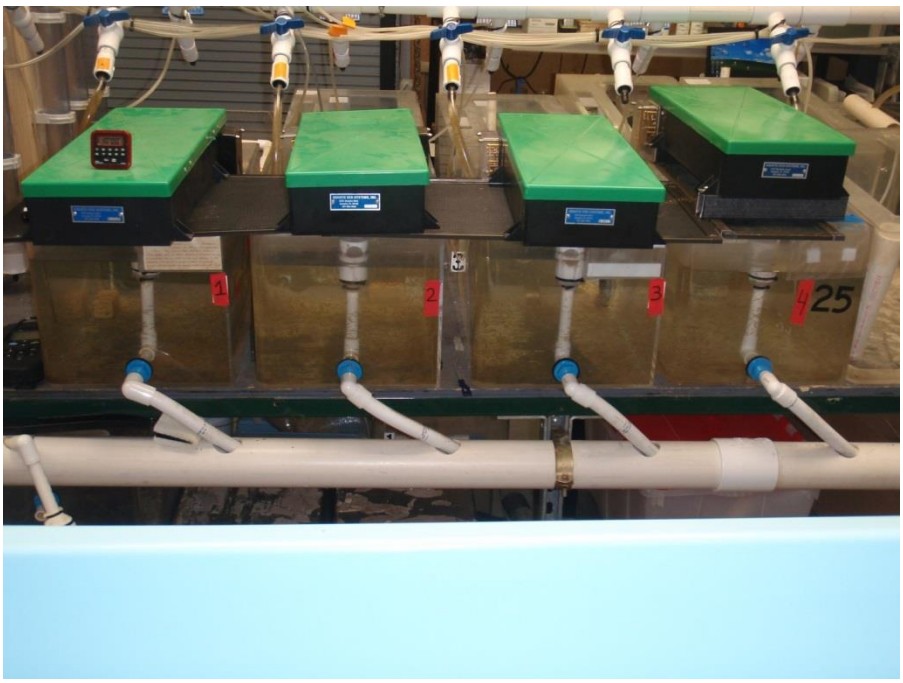


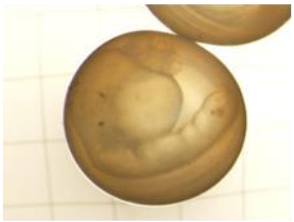
Figure 12. 24hr belt feeds placed on top of 40L aquariums containing actively feed larvae.

Considerations:

1. In order to obtain optimal growth and maintain a clean rearing environment tanks must be checked daily.
2. Observations are made and recorded as to presence or absences of feed.
3. It is critical to maintain current and accurate pound counts in order to calculate feed amounts.
4. Feed amounts will change and should be based on feeding response.
5. Optimal rearing temperatures are still to be determined. FHC rearing temperatures range from 12-20˚C.

Developmental Photos:

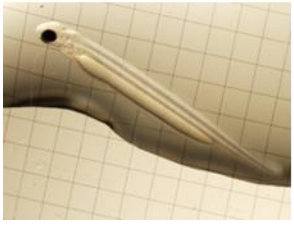
Fertilized Lost River Sucker Egg at Time 0



7 Days Post Fertilization

11 Days Post Fertilization
Eye Spots Are Now Visible

12 Days Post Fertilization
Eye Spots Are Dark and Easily Seen



Lost River Sucker Larvae
1 Day Post Hatch,13 Days Post Fertilization

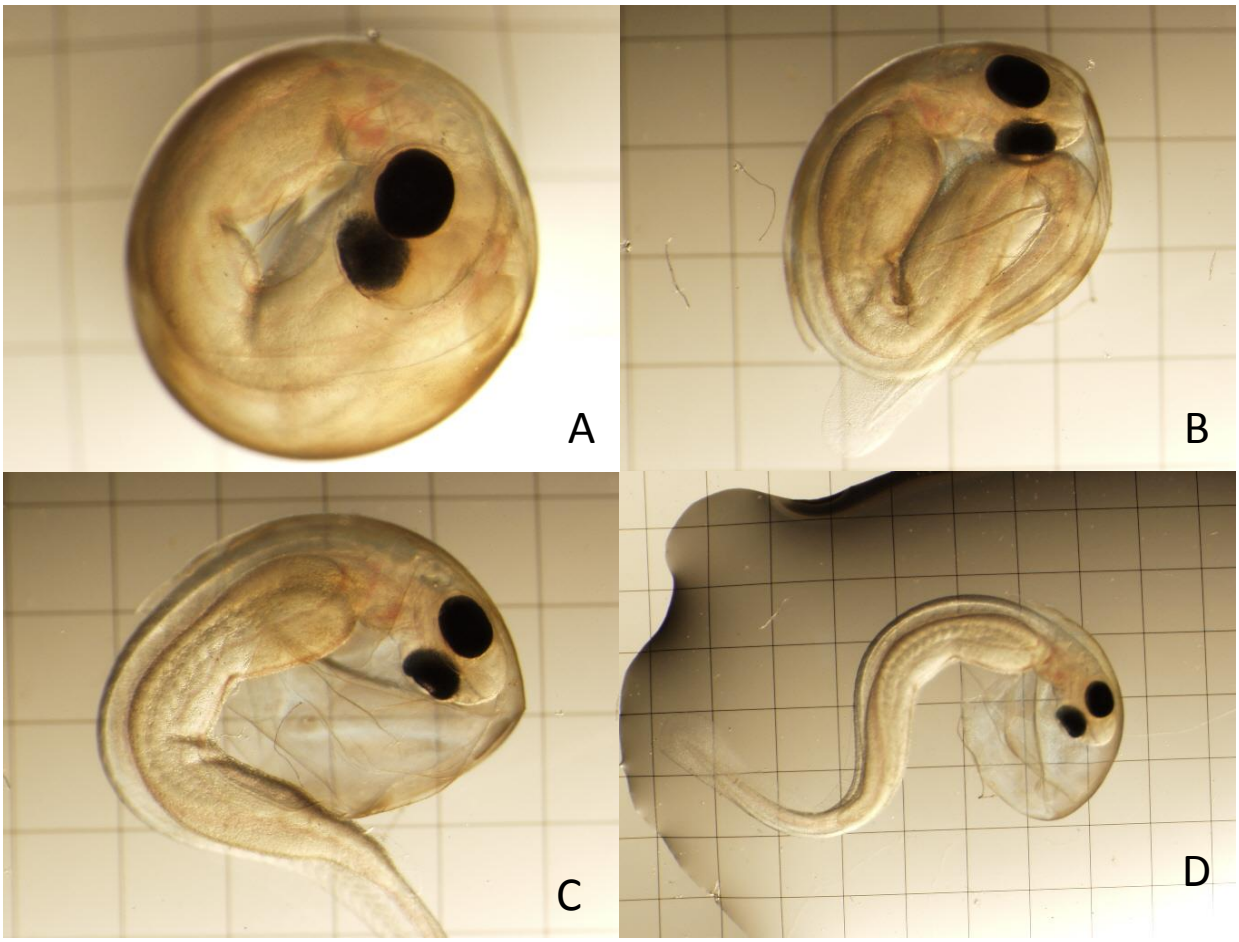
Lost River Suckers That Hatched Prematurely:

Prematurely Hatched Lost River Sucker Larvae
1 Day Post Hatch, 7 Days Post Fertilization

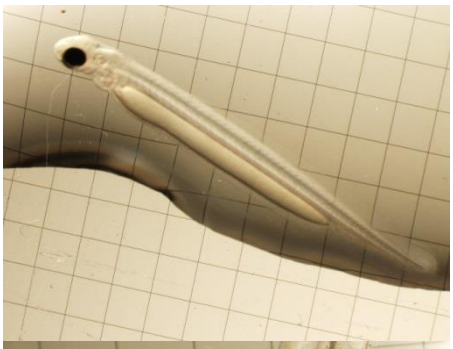
Prematurely Hatched Lost River Sucker Larvae
2 Days Post Hatch, 8 Days Post Fertilization

Close up of Prematurely Hatched Lost River Sucker Larvae
2 Days Post Hatch, 8 Days Post Fertilization

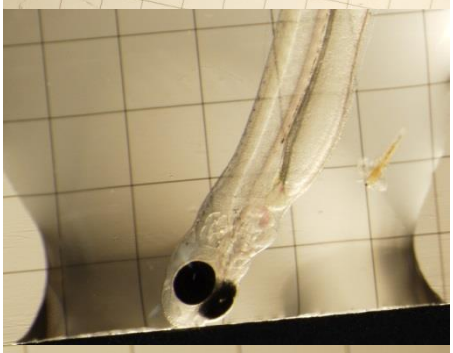
Lost River Sucker Larvae Hatching 14 Days Post Fertilization:



Lost River Sucker Larvae 1 Day Post Hatch to 15 Days Post Hatch:



Lost River Sucker Larvae
1 Day Post Hatch



Lost River Sucker Larvae
3 Days Post Hatch
Artemia Nauplii on the Right

Lost River Sucker Larvae
9 Days Post Hatch
Artemia Nauplii Anterior Left
Artemia Cyst Anterior Right

Lost River Sucker
15 Days Post Hatch

Ca-Nv Fish Health Center Publications Concerning Lost River Suckers

Foott, J. Scott, R. Stone, and R. Fogerty. 2009. Effects of simulated algal bloom pH on juvenile Lost River Sucker energetics and growth

Foott J. Scott, R. Stone, A. Wilkens, and J. Rasmussen. 2012. Juvenile Lost River Sucker survival and energetics in Upper Klamath Lake mesocosm cages: July - December 2011

Foott, J. Scott, R. Stone, and A. Wilkens. 2013. Juvenile Lost River Sucker survival in Upper Klamath Lake mesocosm cages July 2012 - March 2013

Foott, J. Scott, R. Stone, J. Jacobs, and A. Wilkens. 2014. Juvenile Lost River Sucker survival in Upper Klamath Lake mesocosm net pens (July 2013 - October 2013) and labratory hypoxic challenge

Jacobs, J., R. Stone and J. Foott, 2015. Juvenile Lost River Sucker Sentinel Survival in Upper Klamath Lake Mesocosm Netpens (June-September 2014)