

A tank-system design for the hatchery production of sea-urchin larvae

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Gastrula



Prism



Four-arm
Pluteus



Eight-arm
Pluteus

ABSTRACT

In the hatchery production of any species, the larval stage represents the most delicate and critical part of the lifecycle. It is also where some of the greatest gains can be made in streamlining production. Here I propose a tank design for larval culture consisting of a gravity-feed drain and a novel upwelling standpipe that prevents bubbles from contacting the larvae in the culture. This system provides sufficient water movement and slowly replaces the culture water over an adjustable period without loss of larvae. The system can be integrated with either pulse or continuous feeding schedules. A major advantage of the system proposed here is the consistency of water quality throughout the course of the larval phase. This tank system can be extended to smaller laboratory applications and other hatchery species.

1 INTRODUCTION

One of the major challenges that hatchery managers face is maintaining water quality in larval tanks. Like many commercially produced marine invertebrates, green sea urchins have a pelagic larval stage. This stage lasts anywhere from 5 months (Strathmann 1978) to 3 weeks (pers. observ.) – the duration depends on factors such as food levels and temperature. Increasing hatchery production by reducing the larval period invariably involves increasing the temperature of larval cultures. However, the tradeoff is reduced water quality, i.e., faster metabolic rates result in the increased rate of waste accumulation and increased bacterial activity.

Filtering larval-culture water is not practical because the larvae are in suspension and any filtration system is likely to remove or damage the larvae. This issue is resolved in the hatchery production of bivalves by draining the culture water over a sieve (“drain down”), capturing the bivalve larvae (veligers), and washing the sieve into a new tank containing freshly filtered seawater (Fig. 1).



Figure 1. Larval sieving. To maintain water quality in bivalve larval cultures periodic drain down of the tanks is performed. The larvae are caught on a sieve and then washed into a clean tank filled with filtered seawater. Illustrated here is the last stage of the drain down process (washing the larvae into a fresh tank) in the hatchery production of hard clams (*Mercenaria mercenaria*) at the Biosphere Clam Hatchery in Tuckerton, New Jersey.

The drain down process and periodic tank changes result in variable water quality in the culture over time. Water quality deteriorates (at a rate dependant on factors such as larval density, temperature, and feeding rates) until it is replaced (Fig. 2). Food levels in larval cultures also show the same pattern as water quality when large quantities are introduced periodically (pulse feeding). Food density is high when it is first introduced and declines as the larvae feed – again the rate is a function of temperature and larval density.

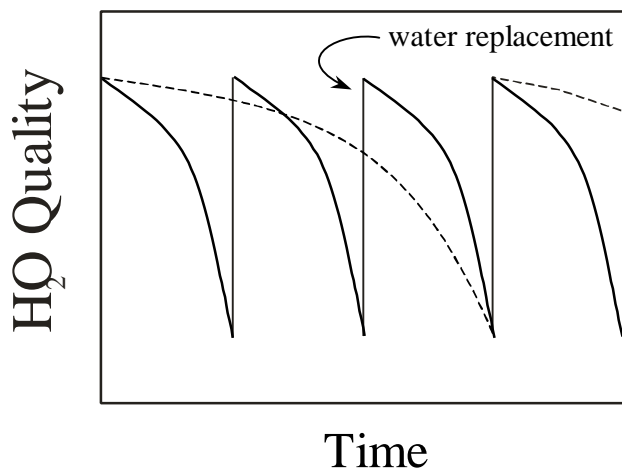


Figure 2. Water quality. The drain down process results in variable water quality through time. The higher the larval density the greater potential for fluctuation in extreme levels of water quality and/or the more cycles of water changes for any culture. The illustration contrasts two hypothetical larval cultures. The dashed line represents a low-density culture and the solid line represents a high-density culture. To maintain the water quality within the same range the high-density culture requires three drain downs to replace the water for every one in the low density-culture. Drain down is a process that can damage sea urchin larvae (plutei lack the protective external shell that bivalve larvae possess) and thus can significantly reduce hatchery productivity. Hatchery managers using the drain down method must balance the tradeoff between low-density cultures (fewer individuals produced) and loss due to damaged larvae. The patterns illustrated for fluctuation in water quality can also be applied to food levels using the pulse feeding method.

The drain down method has been successfully used in the production of green sea urchin larvae (Robinson pers. comm.). However, sea urchin larvae (see illustrations of representative stages above) lack the protective external shell possessed by bivalve larvae. Sieving can damage plutei, and A. Alabi (pers. comm., Island Scallops Ltd., Vancouver Island, BC) has observed damaged and broken rods in the arms after sieving indicating that the process is stressful.

In addition to the difficulties presented by drain down are the problems of gas exchange and water movement in larval sea urchin cultures. Gas exchange and water movement are usually accomplished using an airstone. However, bubbling air through plutei cultures is not recommended (Strathmann 1978) and

elaborate paddle systems for water movement have been designed for the laboratory culture of echinoderm larvae (Fig. 3). The tank design presented here integrates different features of existing larval rearing systems with a unique upwelling standpipe. This system circulates and aerates the culture water without exposing the larvae to bubbles. It also provides a method for continuously replacing culture water over an adjustable period of time. An agricultural drip system can be incorporated into the design to slowly release larval food for continuous feeding.

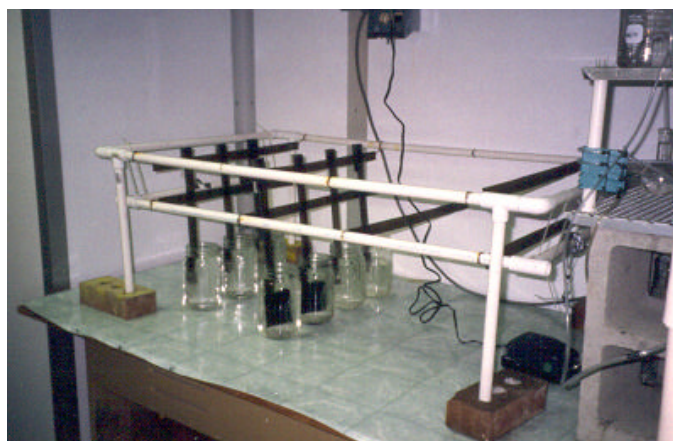


Figure 3. Paddle system. Strathmann (1987) recommends using a paddle system like the one illustrated here to “gently, quietly, and reliably stir large cultures . . .” Although this system has stood the test of time for use in laboratory studies (first reported use was Strathmann, 1971) it would not be practical for hatchery levels of production. The cultures in this illustration are 1 liter containers.

2 TANK DESIGN

2.1 Gravity-feed drain

Robinson (pers. comm.) has successfully adapted halibut larval tanks (Harboe et al. 1998) to culture green sea urchin larvae. The gravity-feed drain in these tanks allows for the regulation of water level and periodic flushing to remove accumulated debris (Fig. 4). Regulating the flow of filtered seawater into the tank controls the turnover period of culture water. The water entering the tank through the inlet is balanced by the release of water through the drain (D_1) in the external vertical pipe. Inserting a screened standpipe in the drain inside the tank (Fig. 4) prevents the loss of larvae from the culture (see below).

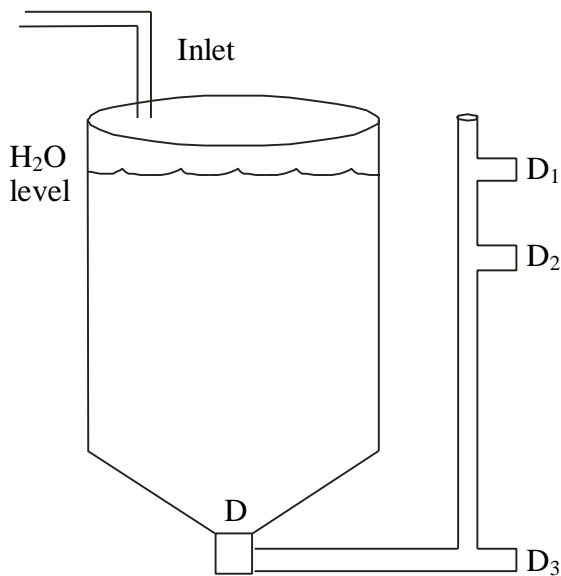


Figure 4. Gravity-feed drain (modified from Harboe, 1998, p. 920). The “Inlet” represents both a filtered seawater source and a drip system for supplying algal food (see text). In the larval halibut tanks a screen mesh covers the internal drain (D) to prevent the loss of larvae. A removable screened standpipe is used in this design (see text). The drains (D₁ – D₃) are fitted with valves. The vertical position of D₁ determines the water level in the tank and is left open. D₂ is closed and used for periodic flushing; D₃ is closed and used to drain the tank. During non-flushing periods the water level in the tank is constant; as filtered seawater enters the tank via the inlet, culture water is released (gravity-feed) at the same rate through D₁.

2.2 Continuous and pulse feeding

Most hatcheries that rear filter-feeding organisms use some form of pulse feeding to provide food (Ramírez et al. 1999). The tank design illustrated in Figure 4 allows for both continuous and pulse-feeding regimes. If pulse feeding turns out to be the preferred method for rearing sea urchin larvae (see Discussion) then the filtered seawater source can be turned off during feeding bouts. If continuous feeding is more efficient then concentrated algae can be added to the larval culture along with filtered seawater to provide a continuous supply of food (at a lower concentration).

2.3 Upwelling standpipe

Figure 5 illustrates the upwelling standpipe in the larval tank and shows photographs of a model used to test the circulation pattern. The standpipe is placed over the drain inside the tank; the lower portion of the standpipe is drilled with holes that are covered by mesh to prevent larvae from entering. The greater the surface area of the lower mesh portion of the standpipe the lower the flow rate of water from the larval tank into the standpipe at any point on the surface of the mesh. A low flow rate insures that larvae will not

be trapped on the mesh. An airstone inside the standpipe acts as an airlift to generate the current. The water inside the standpipe is aerated and simultaneously forced out of the small upwardly angled plastic spouts near the surface (Fig. 5).

If the standpipe were made with slip fittings then it could be removed during the culturing process and would make (minor) flushing of the tank possible (to remove accumulated debris on the bottom of the tank). To replace the standpipe one would simply insert it back into the drain and close the valve at D₁ (Fig. 5). The upwelling generated inside the standpipe would eventually force any larvae inside the standpipe out of the small plastic spouts before opening D₁.

3 DISCUSSION

The partial flow-through system described here could be regulated to turn the culture water over in the tank at a predetermined rate, e.g., 48 hours. The flow rate into the standpipe (Fig. 5 – small horizontal arrows) should be slow enough so as not to trap larvae on the mesh. For example, a cylindrical tank with a volume ~ 3,000 liters (1 m diameter and 2 m deep) could have a standpipe with approximately 0.25 m² of surface area covered with mesh (~1.0 m length of 3-inch PVC pipe). Assuming the flow is evenly distributed across the surface of the mesh then the velocity of the water at any point would only be ~ 4 mm/minute to turn the culture over in a 48 hour period. This calculation also assumes that the filtered seawater added to the culture at the top of the standpipe compensates for the flow out of the small plastic spouts. If more water is pushed out of the spouts via the airlift then the rate of flow into the standpipe would be higher, e.g., 8 – 10 mm/minute – still an acceptably low velocity because sea urchin larvae swim at a speed of 1 – 3 mm/second (M. Hart pers. comm.).

This standpipe could be adapted to work with static cultures as well to provide aeration and water movement. In addition, a modified version of the standpipe could be constructed to work in smaller laboratory settings (1 to 3 liter volumes) and eliminate the need for large bench-top areas for the paddle system. Water changes could be accomplished in these situations (static and small larval cultures) by siphoning water out through the top of the standpipe and then replacing it with filtered seawater. Pouring the filtered seawater through the standpipe would “backwash” any larvae temporarily caught on the mesh back into the culture.

Recently, Ramírez et al. (1999) demonstrated the efficiency of using a continuous, low-food concentration, agricultural drip system in the aquaculture production of shrimp (*Penaeus vannamei*) and the condi-

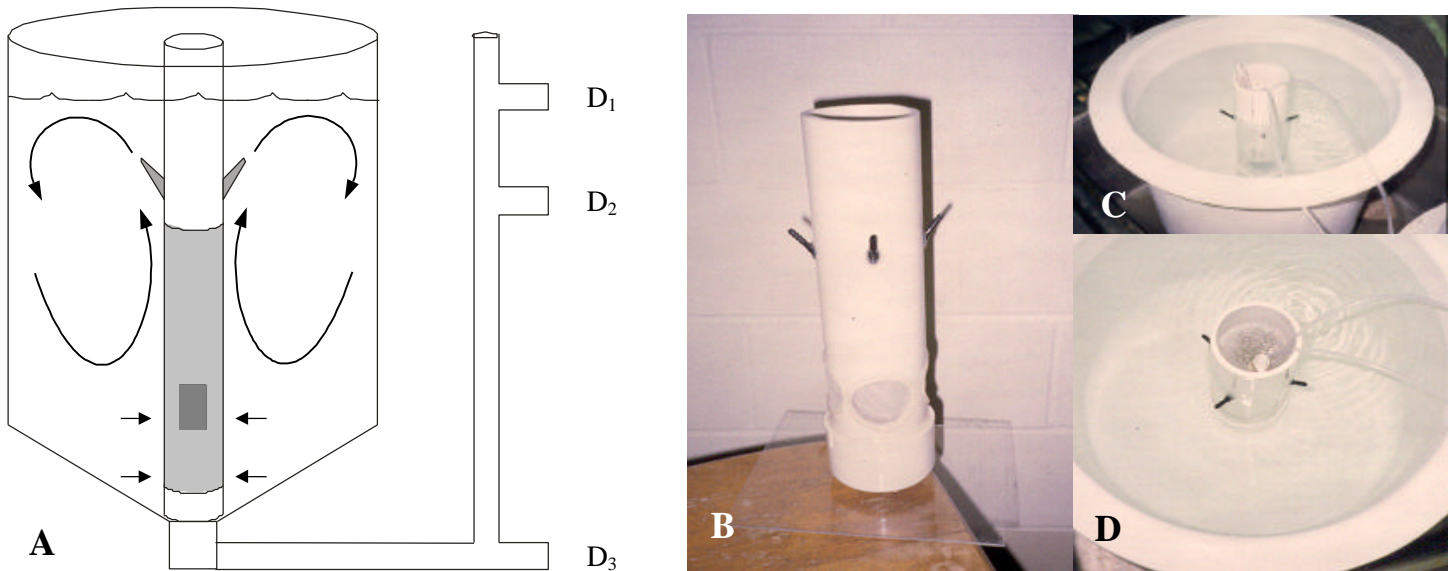


Figure 5. **A.** Schematic of standpipe in larval tank. The bottom of the standpipe fits inside the inner drain of the tank and the top of the standpipe is above the water level. Small plastic spouts (just below the surface of the water) are inserted in the standpipe at an angle to direct the current towards the surface. An airstone inside the standpipe acts as an airlift and separates the aerating bubbles from the larvae. The lower portion of the standpipe has holes covered with mesh screen to prevent larvae from entering the standpipe. Because the surface area of the lower mesh portion of the standpipe (where the water enters) is much larger (~100x) than the cross-sectional area of the spouts (where the water exits) the speed of the current into the standpipe is much less than the speed exiting. If the inlet for filtered seawater is positioned above the standpipe then this water would exit the standpipe via the spouts thus reducing the flow into the standpipe to the water draining out of D₁. The arrows indicate general water circulation in the tank. **B – D.** Photographs of standpipe. A working model of the standpipe was constructed out of 3-inch PVC pipe and tested using food dye in a small 30 liter tank. The vigorous aeration of the water is visible inside the standpipe as well as the gentle upwelling current generated by the spouts in the larval tank.

tioning of scallop (*Argopecten ventricosus*) brood-stock. They observed that pulse feeding methods “do not take into consideration the energetic costs and losses incurred by filter-feeding organisms when they need to adjust to the intake, digestion, and assimilation of irregular and sometimes high food concentrations supplied by [these] methods.” (Ramírez et al. 1999, p. 176). Furthermore, Southgate and Ito (1998) found that partial flow-through culture systems (like the one presented here – Figs. 4 – 5) are more efficient than “conventional static culture methods” in rearing pearl oyster larvae (*Pinctada margaritifera*). I plan to integrate the agricultural drip system of feeding with the larval tank design presented here to run sea-urchin aquaculture trials next year. I will compare larval survival, growth, and settlement rates between traditional static cultures (drain-down and pulse feeding methods) and the partial flow-through system (Fig. 5) integrated with a continuous, low-food concentration, agricultural drip system.

4 REFERENCES

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