TECHNICAL REPORT

Advances in purple-hinge rock scallop culture on the US West Coast

WESTERN REGIONAL AQUACULTURE CENTER

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Table of Contents

Figures

Cover photo: Joth Davis

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Background

In the late 1970s, the rock scallop, *Crassadoma gigantea* (also known as purple-hinge rock scallop and giant rock scallop), was identified as a promising candidate species or marine aquaculture. Since that time, rock scallops have intermittently been the focus of aquaculture-related research along much of the North American West Coast. Growers have also been experimenting with rock scallop culture, either in collaboration with researchers or on their own in hatcheries and at growout lease sites when natural sets of seed have been available, illustrating a strong and abiding interest in culture of this species.

The rock scallop is considered an excellent candidate for sea farming, having several advantages over other shellfish: It reaches a size of more than 100–200 mm (4–8 in), depending on the habitat, and is broadly distributed, occurring in many subtidal areas, from Baja California to southeast Alaska, in a wide range of ocean environments. Thus, it is a candidate for culture coast-wide in existing and prospective west coast mariculture facilities, ranging from onshore seawater tanks to nearshore and offshore open-ocean operations. In addition, a strong market potential makes this a favorable aquaculture species.

Rock scallops are highly prized among west coast sport divers for the large adductor muscle, which is 1.5-to-2 times larger than the adductor muscle of similarly sized weathervane scallops (*Patinopecten caurinus*), a species that also reaches a large size—greater than 200 mm (8 in) (RaLonde et al. 2012). Rock scallops, however, are virtually unknown to the general public because commercial harvest is prohibited throughout their range, primarily due to their patchy distribution and consequent high susceptibility to overharvesting and local depletion. One notable exception of harvesting, permitted in California under a special arrangement, illustrates the high value and marketability of this product: live large scallops sold for $5.00–$8.00 per scallop at farmers markets (Richards et al. 2009).

Rock scallops are also believed to have high market potential in sushi markets, including those with smaller-sized adductor muscles. Notably, rock scallops were ranked highly in a flavor profile analyses that included Atlantic sea scallops (*Placopectin magellanicus*) and Atlantic bay scallops (*Argopectin irradians*). The adductor muscle is high in protein and omega three fatty acids and low in carbohydrates (Leighton & Phleger 1981; RaLonde et al. 2012).

Despite the recognition that rock scallop is a promising species for aquaculture and considerable efforts that have been directed toward resolving culture bottlenecks, work on rock scallop aquaculture has occurred in "fits and starts" (see reviews Bourne 1989; Leighton 1991; Shumway & Parsons 2006) and has fallen short of developing techniques needed for commercial production. A few groups have achieved production of small (100s to 1000s) batches of scallops, suggesting commercial production of *C. gigantea* is within reach, but bottlenecks in routine production continue to hinder the culture of this species on larger scales.

Photo: Courtesy of Brandon Stevens

The intent of this WRAC research project was to advance marine aquaculture along the west coast of the United States by developing production techniques for the purple-hinge rock scallop. To help move rock scallop culture closer to commercialization, we addressed two key bottlenecks: lack of commercial quantities of seed and the need to develop cost effective growout techniques. Specifically, we focused our research on spawning induction, production of sterile (triploid, tetraploid) seed, and manipulation of the cementing behavior during the growout production phase.

Rock Scallop Life Cycle

Key characteristics of the rock scallop life cycle (Figure 1) to consider for aquaculture production include:

Suspension feeders. Rock scallops extract phytoplankton (microscopic algae) and other associated food—including decomposed matter—from seawater throughout their life, as is typical for many other suspension-feeding bivalves.

Separate sexes. A dioecious bivalve, individual scallops are typically male or female and distinguished by gonad color, orange for females and white for males. Hermaphrodites occur, but are relatively rare.

Broadcast spawners. Rock scallops release eggs and sperm directly into the water, where fertilization occurs. Once fertilized, embryos develop in less than 48 hours into veliger larvae.

Planktonic larval stages. Larval rock scallops swim in the water column and undergo three distinct pelagic stages: trochophore, early veliger (dissoconch I), and late veliger (dissoconch II). This scallop, like most bivalves, begins feeding on plankton once past the trochophore stage.

Prolonged metamorphosis. Rock scallops transition to a more benthic existence via settlement onto a suitable substrate at the pediveliger stage where they subsequently metamorphose to the juvenile stage.

Metamorphosis is prolonged in this species, as compared to many other bivalves, taking three to six weeks to fully develop the structure (ctenidia) needed to feed on suspended plankton. During this period, scallops are believed to rely on pedal feeding (using the foot) for nutrition and are characterized by smooth pale valves. The colorful striation associated with the juvenile shell occurs once feeding on suspended plankton begins.

Temporary (byssally) attached juvenile stage. Unlike most other scallop species that live unattached on the sea bottom, juvenile rock scallops attach themselves to vertical or horizontal substrates such as filamentous algae and hard substrates, including rocks, shells, and man-made structures (e.g., pier pilings, docks, oil and gas platforms)

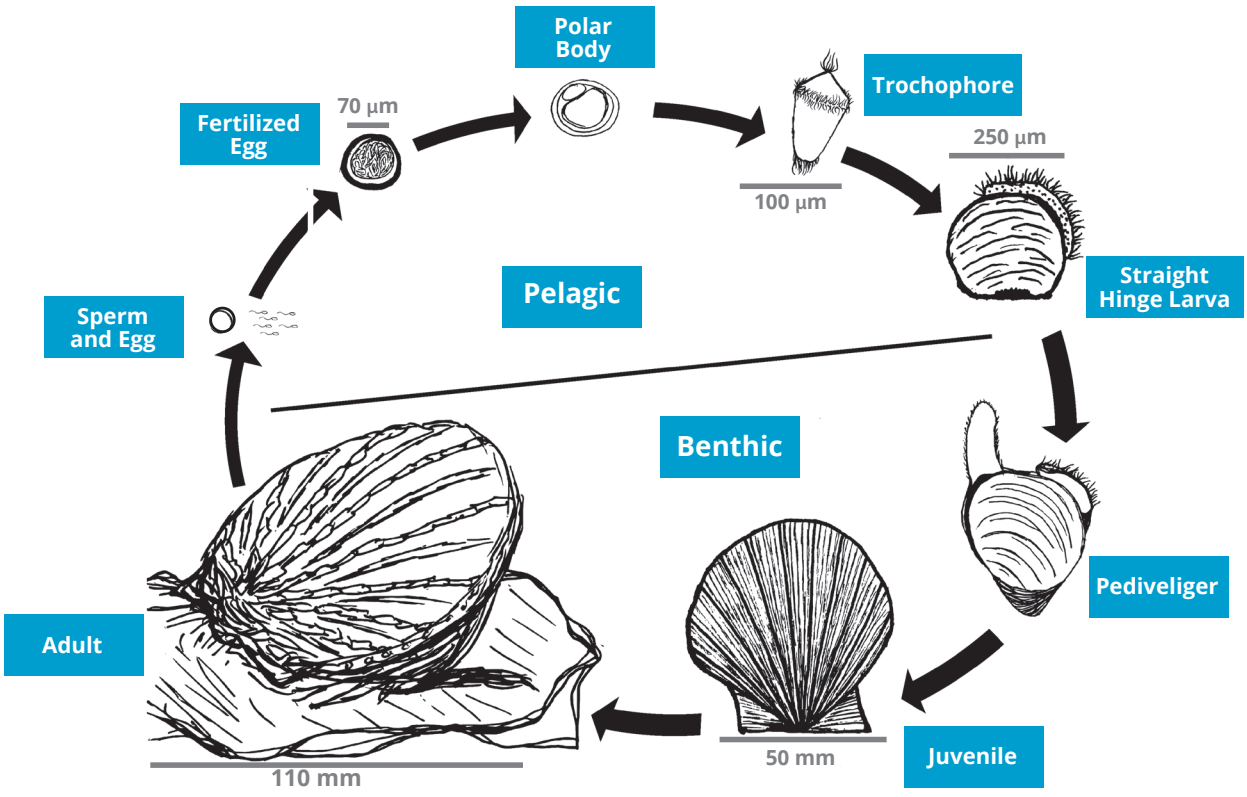
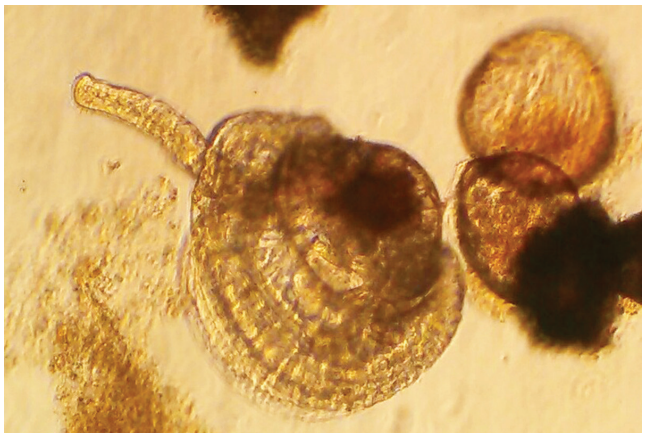


Figure 1. Life cycle of the purple-hinged rock scallop, *Crassadoma gigantea*.

Credit: Cassiel Nortier-Tilly, California Sea Grant, University of California Santa Barbara



Photos, top to bottom: Paul Olin, Joth Davis

using byssal threads, similar to mussel attachments. Attachment is not permanent, allowing rock scallops to release their byssal threads and move to another location.

Permanent (cemented) attached stage. Typically, as rock scallops grow and mature, they permanently affix themselves by cementing their right (bottom) valve to a hard substrate, which can include aquaculture growout gear. This behavior is usually initiated within a specific size range (25–50 mm / 1–2 in shell length), but commencement and duration of the cementation phase varies (see section "Manipulation of the Cementing Behavior").

Subtidal ocean inhabitants. A subtidal marine species, rock scallops have a low tolerance for extended exposures to high temperatures (>22°C (71.6°F)) and low salinity (<25 ppt). They commonly are found in high flow areas, presumably due to the strong attachment achieved through cementation, but also grow in calmer marine waters.

Spawning Induction

As with many bivalves, a variety of cues have been used to induce reproductive scallops to spawn in the laboratory and hatchery. These include temperature shock, air exposure, exposure to UV sterilized seawater, chemical induction (hydrogen peroxide, serotonin), combined excess algae and raised temperatures (simulating natural summer spawning conditions), and addition of a suspension of male gametes. These methods were tested to various degrees during this project, along with strip spawning (excising the gonad with a scalpel to expose and collect the gametes). The spawning method that was most reliable across locations was chemical induction via injection of serotonin into either the gonad or adductor muscle.

Chemical Induction

We evaluated the influence of injection site (gonad versus adductor muscle) on the efficacy of serotonin as a chemical inducing agent. We collected scallops off Santa Barbara, California, and visually determined sex and gonad index (ripeness). To view the gonad we inserted a thin flexible instrument (i.e., paint brush, broom bristle, or small cable tie) through the byssal notch (small opening near the hinge where the foot extends out) and "tickled" the scallop by slowly moving the instrument along the mantle and interior of the scallop. Once the scallop gaped, we inserted an object (rubber cork, clothes pin) to keep the two valves open until we completed our examination. Sex was determined by the gonad color (orange for female, white for male), and gonad index was based on the color pattern (transparent, blotchy, opaque), length (distance wrapped around adductor muscle), and fullness (plumpness). Scallops with the highest gonad index were chosen for the spawning trial.

Serotonin was injected into a cohort of 12 females and 6 males, with half of the scallops injected in the gonad and the other half in the adductor muscle using methods adopted from Bourne et al. 1989 (0.2 ml of $2x10^{-4}$ solution of serotonine creatine sulphate (8 mg dissolved in 10 ml filtered seawater). Twice as many females were used because, similar to other bivalves, males spawn more readily than females. Once injected, we placed individual scallops into a container filled with seawater, recorded the time when the scallops commenced spawning and calculated the percentage of scallops that spawned.

All but one male and one female spawned, 83% and 92% respectively. Both scallops that did not spawn had

been injected in the gonad; 78% and 100% spawning success when injected in the gonad and adductor muscle, respectively. These non-responsive scallops also were the last in their groups to receive injections. Overall, male scallops spawned sooner, within 10 to 30 minutes following chemical injection, whereas females took a bit longer, with spawning occurring 30 to 90 minutes after injection. One exception was a male scallop that took nearly two hours to release sperm. For the spawning trials in Washington, injections were made into the adductor muscle (not gonad). Success was highly variable and seemed to be dependent on the time since collection. For animals that did spawn, it typically occurred 20-to-60 minutes post-injection for males and 45-to-90 minutes for females.

Conclusions

Injection of serotonin into the adductor muscle is a reliable method for inducing spawning in ripe rock scallops. Coordinating the timing of gamete release from males and females is required as males typically spawn more quickly.

Broodstock Conditioning

While serotonin injections often were reliable for inducing spawning in scallops that had just been collected from the wild, it often did not work for scallops that had been held in the laboratory for extended time periods (weeks or months). Further, on numerous occasions during the project, it was difficult to obtain "ripe" spawnable scallops that had been in natural habitats in Washington. Because of these problems, we evaluated methods for reproductive conditioning.

Reproduction Conditioning: To explore methods for facilitating reproductive readiness of rock scallops for spawning, we conducted conditioning trials at two locations in Washington: the Taylor Shellfish Farms hatchery on Dabob Bay and the Baywater SeaLab (now Pacific Hybreed) located at the NOAA NW Fisheries Science Center located in Manchester.

Dabob Bay: Adult scallops were collected from Hood Canal and either held in polyethylene fish totes with dedicated air supply and a flow rate of 60 liters per hour at the hatchery (Trial 1) or within lantern nets suspended in the bay (Trial 2). Trial 1 began in April 2017 and consisted of 30 scallops held in the hatchery for a 4–5 week conditioning period. Scallops were continually fed a mixed microalgae diet, and the temperature was gradually increased from ambient (~10°C / 50°F) to 14°C / 57°F). Spawn attempts and/or visual assessments of gonad index commenced in May and were ongoing through September. For Trial 2, 15 broodstock remained suspended in lantern nets in Dabob Bay to "naturally" condition on wild phytoplankton until August. They were then brought into the hatchery and held under the same conditions as Trial 1 for 10–15 days prior to spawn attempts. None of the spawn attempts in Trial 1 or Trial 2 were successful, and visual inspection revealed low gonad indexes for scallops in both groups.

Baywater SeaLab: In December 2017, rock scallops (N=17) were collected from Agate Pass, Washington and maintained at the Baywater SeaLab for a 6-week conditioning period in a similar holding system to that used at Dabob Bay. Scallops were feed a mixed microalgal diet, and the temperature was gradually increased by 2°C / 3.6°F, from ambient (12°C / 54°F) to 14°C / 57°F. Scallops were fed a combination of live algae and REED Mariculture Shellfish Diet 1800™. The live algae consisted of the flagellate, T-Iso (*Tisochrysis lutea*, CCMP463) and a diatom (*Chaetoceros gracilis*) (e.g., CHAGRA) mixed at a ratio of 50/50 by cell number and fed to broodstock at a total concentration of 50,000 cells per ml. Microalgae was added to a head tank and pumped into the scallop broodstock tote via peristaltic pump (Masterflex Corp.). REED Mariculture Shellfish Diet was added daily to the head tank. A total cell density of approximately 100,000 cells per ml was fed to scallops continuously over a 24-hour period. This feeding regime was maintained over the conditioning period with T-Iso and CHAGRA substituted at times with other algal strains reared in the PSRF algal facility.

Prior to initiating the conditioning trial, the gonad index was visually determined for a subsample of scallops using the previously described gonad assessment methods. The indexes were low and it was assumed that scallops were not ripe for spawning. After approximately six weeks of conditioning, we again evaluated gonad index. No significant increase in the index was noted for either male or female scallops. A second group of rock scallops (N=30), also maintained at the Pacific Hybreed hatchery facility, were introduced to a second conditioning trial. Prior to the start of this trial, a subsample of scallops was examined and gonad index assessed. The sex of each scallop in the sample was easily determined, but the gonad indexes were again low. In an attempt to increase gonad development, scallops were held under similar conditions (temperature,

salinity, water flow rate) as before and fed a combination of live algae and REED Mariculture Shellfish Diet 1800™ for an additional four weeks (ten weeks total). For a subsample of scallops, the gonad index was visually assessed in mid-May, late June, and mid-July with little or no increase noted. With the exception of 2 individuals (females) that had a moderate index in mid-August, the other scallops (20 examined) exhibited reduced indexes.

Conclusions

After conducting dedicated and similar conditioning trials in Washington with older scallops coming from a wild population and younger scallops (≥ 3 years) that had been reared in the hatchery, our research team was unable to induce significant gonadal development in either group. Scallops in both groups were actively feeding and producing copious quantities of waste (feces and pseudofeces). Mortality of scallops was low with less than 5% loss over the conditioning period for either group. Coincidently, a similar biomass of Pacific oysters was readily conditioned and spawned at the Baywater SeaLab during the same time frame using the same methods. It is largely unknown why rock scallops failed to undergo initial or renewed gametogenesis in the hatchery environment. It could be that the quantity and/or quality of food was inadequate. In past years and prior to WRAC funding, routine scallop spawns were attained by bringing in wild scallops that were either in or close to full reproductive condition. Unfortunately, access to wild scallops in Washington has been largely curtailed due to regulatory concerns over relative abundance and other considerations, making it imperative to develop broodstock conditioning protocols for hatchery maintained populations.

Photo: Courtesy of Brandon Stevens

Photo: Courtesy of Bodega Marine Laboratory

Production of Sterile Seed

A primary emphasis of our WRAC-funded research was the development of methods for producing triploid rock scallops. Triploid seed have long been used in Pacific oyster (*Crassostrea gigas*) culture to address a marketing problem: during the summer months, Pacific oysters become less palatable, their taste often described as chalky and bitter due to the presence of male and female reproductive tissue. Triploid seed offered a solution to these problems by limiting production of gonad tissue such that the oyster became effectively sterile and energy usually used for reproduction was redirected into growth. A change in product quality, as experienced by Pacific oysters, is not expected to be a problem with rock scallops because their large, adductor muscle is the primary product, not the whole organism as with oysters. Nonetheless, sterile rock scallop seed is of interest to many because it 1) would reduce genetic interaction between farmed and wild populations, a concern for aquaculture areas that have wild populations of rock scallops in close proximity, and 2) may also increase adductor muscle size, the body part prized for human consumption.

Triploids

To explore the potential for producing triploid rock scallops, we developed methods based on those used to produce triploid oysters and other species of shellfish. One such method requires manipulating oocytes soon after fertilization in order to retain two duplicate sets of chromosomes (2N) instead of the normal single set (N). When sperm, which normally also has one set of chromosomes (N), is

combined with these 2N eggs, a triploid (3N) animal is produced. This phenomenon is achieved by temporarily interrupting normal development by applying a "shock," using temperature, pressure, or chemicals, to the oocyte after it comes in contact with sperm (Figure 2). Success of triploid production requires determining the timing, duration, and intensity of the shock such that triploid production is maximized and mortality is minimized.

Our first triploid trials evaluated the use of 6-dimethylaminopurine (6-DMAP) as the "shock." Wild brood animals were collected from Port Gamble Bay, WA and transported to the nearby Taylor Shellfish Farms hatchery on Dabob Bay. Scallops were held in seawater tanks where they underwent broodstock conditioning and were continually fed to enhance gonad development prior to spawning. Conditioning involved maintaining animals at 14°C / 57°F for at least six weeks and feeding them daily with a high ration microalgae diet (Reed Mariculture Shellfish Diet 1800™ mixed with multiple species grown on site).

To determine the optimal dose of 6-DMAP for producing triploid scallops, we fertilized scallop eggs, held them for 60 minutes, exposed them to one of eight dosages (treatments) for 20 minutes, and then maintained them in beakers and assessed survival and ploidy of the resulting larvae after three days. We found that 50% and greater

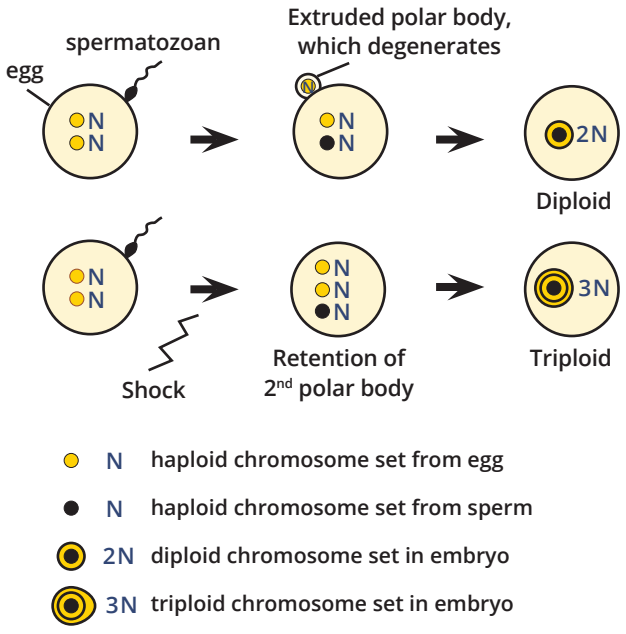


Figure 2. The process of triploidy induction following application of a shock.

Adapted from design by: FAO (https://www.fao.org/3/y5720e/y5720e0c.htm)

triploid induction occurred at concentrations of 400 µM and above; the percentage of triploids essentially plateaued at 500 µM and above; and, as expected, survival declined with increased dose (Figure 3A).

A second (unreplicated) large-scale trial was run when a mass spawn occurred unexpectedly. Three groups of about 15 million fertilized eggs were placed in high-density conical culture tanks and exposed, approximately 60 minutes post-fertilization, for 20 minutes to one of three shock treatments: 300 µM 6-DMAP, 500 µM 6-DMAP, and no 6-DMAP. As with the earlier trial, the proportion of triploids produced increased and survival decreased with increasing dosage (Figure 3B). Based on these results and using response surface method analyses, we determined that a dose of 425 µM 6-DMAP would be optimal for producing a high proportion of triploids with adequate survival. Reducing the duration of the treatment from 20 minutes to 15 minutes may increase survival and is suggested for future trials.

Tetraploids

Another way to produce triploids is to mate a regular diploid (2N) animal with an animal that has four sets of chromosomes—a tetraploid (4N). In this case, tetraploid animals are produced and maintained as broodstock. Tetraploid males have sperm that are 2N (diploid) instead of the usual 1N (haploid); thus, when tetraploid males are mated with normal females, the resulting embryos have three chromosome sets. This method has proved to be very effective for oyster culture. Not only does it reliably produce cohorts that are 100% triploid, but because no "shock" is required, there is no added mortality.

The tetraploid method for production of triploid rock scallop seed seems promising. However, the general method to produce tetraploid scallops requires using adult female triploids—which were not available. We explored another possible method where haploid sperm with no functional DNA is used to "fertilize"—more correctly "activate"—the oocytes obtained from a normal diploid female. This method would then involve using a "shock" to block extrusion of both the first and second polar bodies, yielding embryos with four sets of chromosomes, all from the female scallop. As a first step, we evaluated methods for producing sperm with no functional DNA. In the ultraviolet spectrum, ultraviolet -C (UVC) is known to damage DNA, which is why it is used in sanitizing (germicidal) lamps. We exposed sperm to UVC irradiation to determine 1) optimal exposure

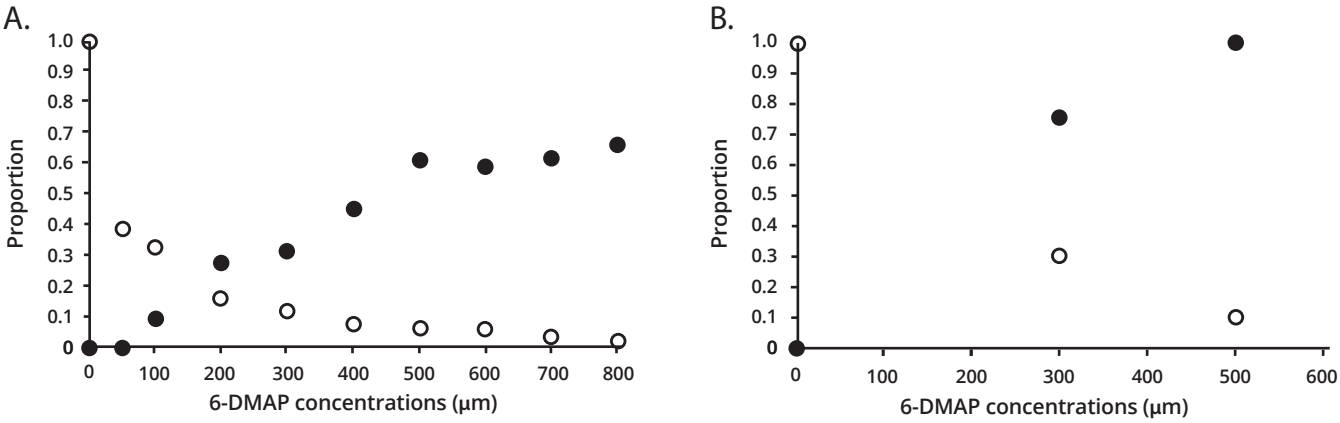


Figure 3. Effect of exposure of fertilized rock scallop eggs to varying concentrations of 6-DMAP on proportion and survival of triploid (3N) scallops during two trials: A) eight concentrations of µ6-DMAP; B) two concentrations of µM 6-DMAP. Closed circles, proportion produced; Open circles, survival.

time for retaining sufficient sperm motility needed for successful fertilization, and 2) satisfactory yield of haploid (1N) embryos indicating that the sperm lacked functional DNA.

We distributed 240,000 oocytes into three replicate 1 liter (L) beakers filled with 800 milliliters of water and set the beakers into a 17°C / 63°F water bath. Unfertilized and fertilized controls were set aside. Sperm were put in petri dishes and then exposed for 60, 70, 80, 90, 100, 110, 120, and 130 seconds to UVC (800 microwatts per square centimeter). We compared the motility of sperm exposed to UVC with normal unexposed sperm, categorizing motility on a qualitative scale from 0–5, with 0 being no motility and 5 being motility equal to that of the unexposed sperm. We found, as expected, that increased exposure time dramatically decreased sperm motility and thus the chance for successful fertilization (Figure 4). This information enabled us to identify exposure times that would support oocyte activation. The next step was to test the ability of the UVC-exposed sperm to successfully fertilize oocytes without contributing any DNA to the resulting embryos. To do this, we placed each group of UVC-treated sperm in 1L beakers containing unfertilized oocytes. To reduce bacterial contamination that commonly occurs in these types of small cultures, we added two antibiotics (Penicillin G and Streptomycin). Approximately 12 hours post fertilization, we sub-sampled larvae (trochophore stage) for ploidy analysis. Samples unfortunately could not be evaluated for all treatments and replicates due to a laboratory mishap, but ploidy was determined for four samples: a single replicate for the 80- and 100-second groups, and two replicates for the 90-second group (Figure 4). Ploidy was determined utilizing a Partec flow cytometer following protocols routinely used at Taylor Shellfish Farms for evaluating ploidy in bivalve larvae. In each of these samples, only 1N peaks were observed, indicating that exposure to UVC for 80, 90, and 100 seconds was effective at producing sperm without functional DNA. A similar experiment is needed to ascertain whether reducing the duration of UVC exposure likewise would be effective.

Conclusions

Our results, while limited, are promising and lay the foundation for further work on the development of tetraploid rock scallops using a shock to cause retention of both polar bodies in oocytes activated by sperm with

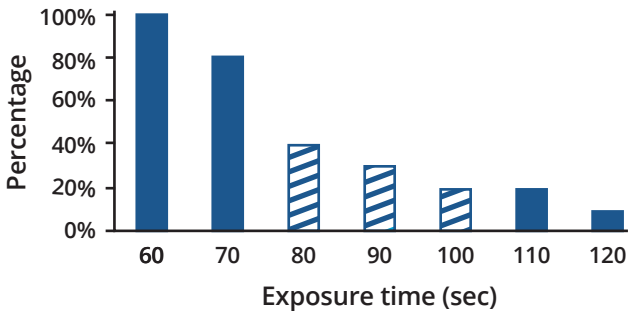


Figure 4. Effect of UVC irradiation exposure times on rock scallop sperm motility (relative to controls) and haploid embryo (dashed bar) production. Solid blue bars denote unknown ploidy status due to missing data.

nonfunctional DNA. This alternate method would be advantageous compared to the practice used to produce Pacific oyster tetraploids, because it avoids having to first produce triploid scallops. If this new approach works, the timeline for developing tetraploid scallops (and hopefully 100% triploid seed production) will be greatly reduced compared to current approaches.

Maturation

With concerns about genetic risks to wild stocks in mind, we also investigated the maturation process to determine what size and age rock scallops mature. This information is needed to evaluate the potential for interbreeding between farmed and wild populations. That is, if rock scallops mature at a large size with a prolonged juvenile stage, growers may be able to market the scallops before they contribute reproductively, thereby posing little risk of genetic mixing with wild populations.

We characterized the maturation cycle of rock scallops, specifically identifying size and age at first maturation and the variation and temporal progression of the process. Rock scallops for this analysis were produced from a single spawn at the Taylor Shellfish Farms hatchery in April 2015 and deployed five months later (September) in Dabob Bay. From January 2016 to December 2017, ten scallops were randomly sampled biweekly from the group. For each scallop, the shell height was recorded and a gonad sample taken and preserved for histology. Tissue sections were later examined via light microscopy, with sex and reproductive phase (based on six categories; Figure 5) recorded for each individual.

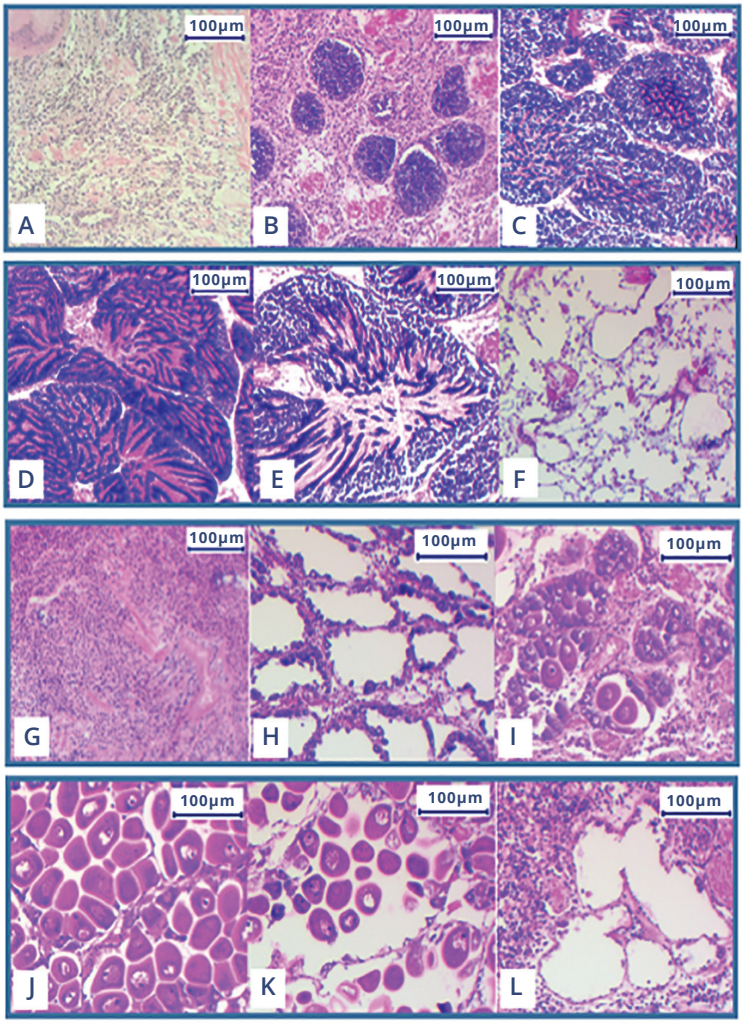


Figure 5. Gonadal maturation stages for male (A–F) and female (G–L) *Crassadoma gigantea*. Males stages: **A** inactive, **B** early active, **C** late active, **D** ripe, **E** partially spawned, **F** spent; Female stages: **G** inactive, **H** early active, **I** late active, **J** ripe, **K** partially spawned, **L** spent.

A total of 428 individuals were sampled over a 32-month period. The sex of the scallops was first histologically observed at about 12 months. We found significantly more males (50.1%) than females (3.5%), a few hermaphrodites (0.93%), and many individuals without visible germ cells that were classified as "unknown" (44.9%). Rock scallops gradually progressed from immature to mature. The first mature (ripe) scallop was detected at 20 months, with the first spent (spawned out) individual detected at 23 months. The majority of individuals matured within 26 months and were either partially spawned or fully spent within 29 months.

Logistic regression models were used to estimate age and size at first maturation from the subsampled scallops. First maturation—the point at which 50% of individuals reached sexual maturity (Ropes 1968)—occurred at a shell height of 55.25 mm (2.2 in) and an age of 25.1 months (Figure 6).

Rock scallops mature at a relatively small size (~55 mm / 2.2 in SH) and young age (2 years), smaller than the size that would typically be marketed and/or taken by recreational divers. This indicates that farmed scallops grown near wild scallop populations may pose a risk for genetic mixing with wild populations, depending on factors such as proximity, sex ratios, and relative fecundity. Such a risk may be reduced if a market for a smaller adductor muscle is developed and scallops are removed from the environment prior to fully maturing, or if sterile (triploid) scallops are raised.

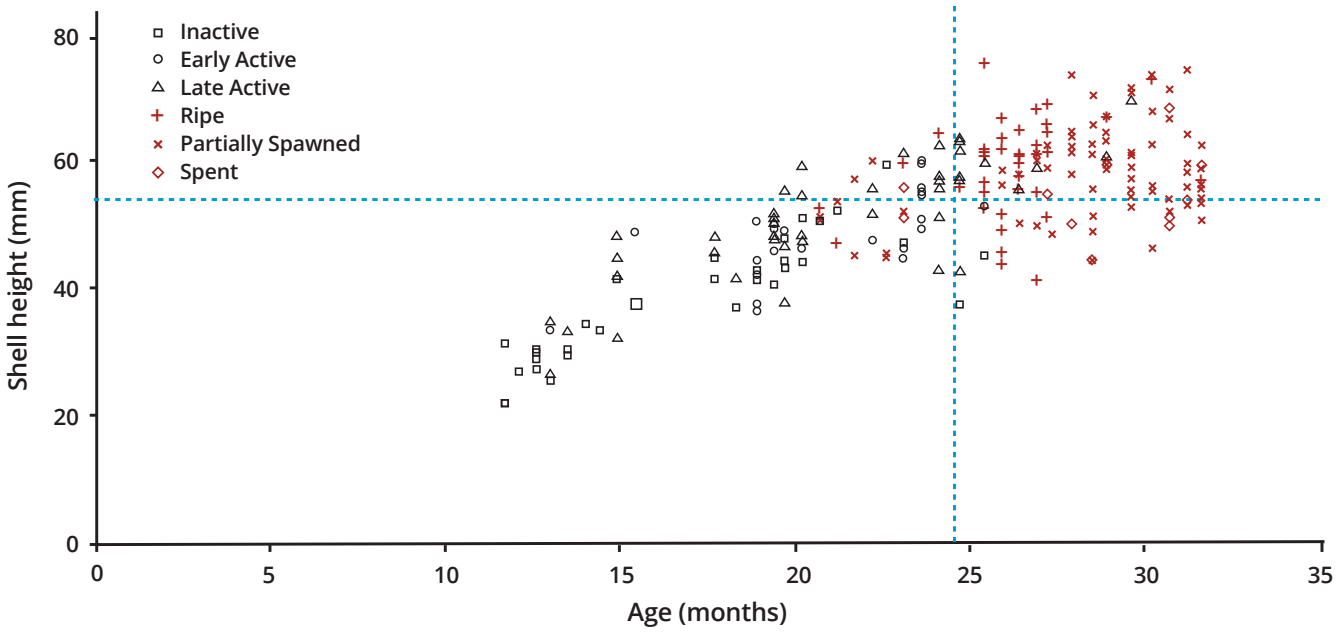


Figure 6. Size and age of *C. gigantea* assigned to six maturation stages: Inactive (square), early active (circle), late active (triangle), ripe (plus), partially spawned (x), and spent (diamond). Black symbols represent immature individuals (inactive, early active, late active), red symbols are mature individuals (ripe, partially spawned, spent). Blue lines indicate age (25.1 months post-spawn) and size (55.25 mm) at first maturation, determined by single predictor logistic regressions.

Growout: Manipulation of Cementing Behavior

Unlike growout methods for other commonly cultured bivalves, such as oysters, clams, and mussels, growout methods for the purple-hinge rock scallop, *Crassadoma gigantea*, require manipulation of a *delayed* cementing stage—in which scallops transition from temporary attachment with byssal threads to permanent attachment through cementation to a hard substrate.

During this stage, the rock scallop not only cements itself to the substrate, but it also conforms to the shape of the substrate as it deposits new shell during growth. As a result, both the product and culturing gear can be damaged during harvest, reducing yield and increasing production cost, respectively. Notably, unlike other cementing organisms, such as some oysters, rock scallops have the ability to continue cementing throughout their life even if detached at some point (Culver et al. 2006), although the occurrence of recementing is influenced by many factors, including size and environmental conditions.

We conducted field studies to better understand the role of rock scallop cementation and potential ways to manipulate it for growout. Based on our earlier research (Culver et al. 2006), along with observations in nature, we hypothesized that rock scallops need to be in a fixed stable state for good growth and high survivorship, which is attained through different means, depending on scallop size and environmental conditions. Specifically, the fixed state for juvenile rock scallops is achieved through byssal attachment to substrates. Then upon reaching a certain size, often around 20–35 mm (0.8–1.4 in) shell height (SH), byssal attachment is no longer adequate to maintain a stable fixed state and cementation is initiated (alongside maturation). As scallops continue to grow, they reach a size where they are heavy enough to be fixed in place and permanent attachment (cementation) is no longer needed in most environments.

Based on this attachment hypothesis, we designed experiments to address three questions: 1) Can the cementing behavior be inhibited through periodic disturbance with resulting adequate growth?, 2) Does the cementing behavior decrease as scallops grow?, and 3) Is continuous cementing needed for optimal growth? To address the first two questions, we deployed replicated bags with 50 scallops (~30 mm / 1.2 in shell height (SH)) at seven bay sites in Washington and one bay site in California. For Washington-based scallops, the cages were emptied at approximately 100-day intervals, and the scallops assessed for growth, survivorship, and cementation. In the case of cemented scallops, these individuals were easily dislodged and returned to the cage for additional growth. We found that the California scallops, which typically grow more quickly in the southern warmer water environments as compared to Washington, rapidly cemented to the mesh and could not be dislodged without damage to the shell and/or gear on a practical time frame; thus, the California experiment was discontinued. In Washington, however, after 25 months, average survivorship was high (73%), and shell growth and adductor muscle growth was good (78.9 mm / 3.1 in SH, 10.1 g / 0.36 ounces), albeit significantly different among sites (Figure 7). The disturbance method proved successful at minimizing cementation to the gear, especially as the scallops reached a size of 50–55 mm

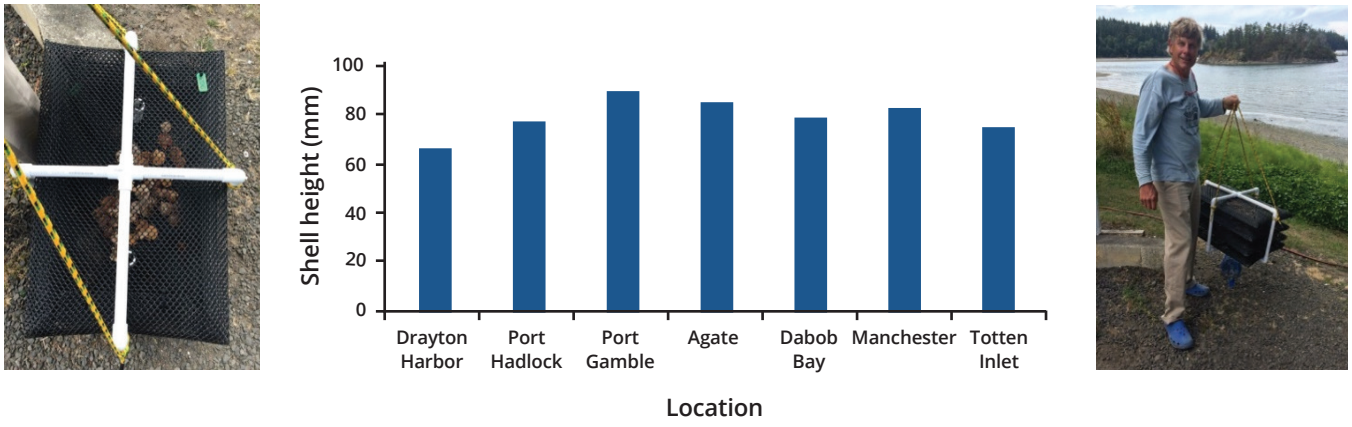


Figure 7. Growth of *C. gigantea* deployed in stacked mesh bags (shown in photos) at different sites in Washington.

Photos: Carrie Culver

(2–2.2 in) SH, the size that they become mature. At this size range, less than 1% of scallops continued to exhibit cementing behavior.

Second, we assessed the influence of continual attachment on scallop growth in California. Scallops (~30 mm / 1.2 in SH) were artificially attached to either short (70 mm / 2.75 in) or tall (140 mm / 5.5 in) flat panels and grown in replicated shellfish culture trays (Figure 8). After nearly 25 months, scallop survivorship was high (89%). Shell growth was significantly greater for scallops that continually cemented to panels (tall panel treatment) as compared to those that grew past the panels' edge and continued to grow without cementing to the substrate (short panel treatment); 102.3 mm vs 96.4 mm/4.0 in vs 3.8 in. However, adductor muscle weight was not significantly greater: 21.2 g vs 19.7 g/0.75 oz vs 0.69 oz. These findings indicated that partial attachment, as compared to continuous attachment, was adequate for good adductor muscle growth.

In addition to these experiments, at one site in Washington, we compared growth between scallops grown in the mesh bags and disturbed periodically to scallops artificially attached to substrates. After 36 months, survivorship was high and growth was adequate for both methods. Similar to the California results, the attached scallops had greater shell growth (92.9 mm vs 85.2 mm/3.7 in vs 3.4 in), but substantially larger adductor muscles (14.5 g vs 9.4 g/0.51 oz vs 0.33 oz). Given the growth rate of the unattached scallops, and assuming linear growth, it would take nearly 20 additional months for the unattached scallops to produce an adductor muscle equal to that produced by the attached scallops. Additional studies are needed to clarify whether similar growth would occur elsewhere. If so, attachment of seed could potentially reduce the time to harvest significantly—estimated at two years for an adductor muscle weighing 20 g (0.71 oz).

Our results support our attachment hypothesis and indicate that cementing may be primarily a juvenile rock scallop behavior that is exhibited during a small window of time, and may not be required at all depending on the growout conditions and desired time to market. However, promoting attachment—even for a small amount of time—may positively affect growth. For farming rock scallops, manipulating attachment—be it promoting or inhibiting attachment—even for a small amount of time will have costs, with pros and cons associated with each method. Specifically, promoting attachment has more upfront gear

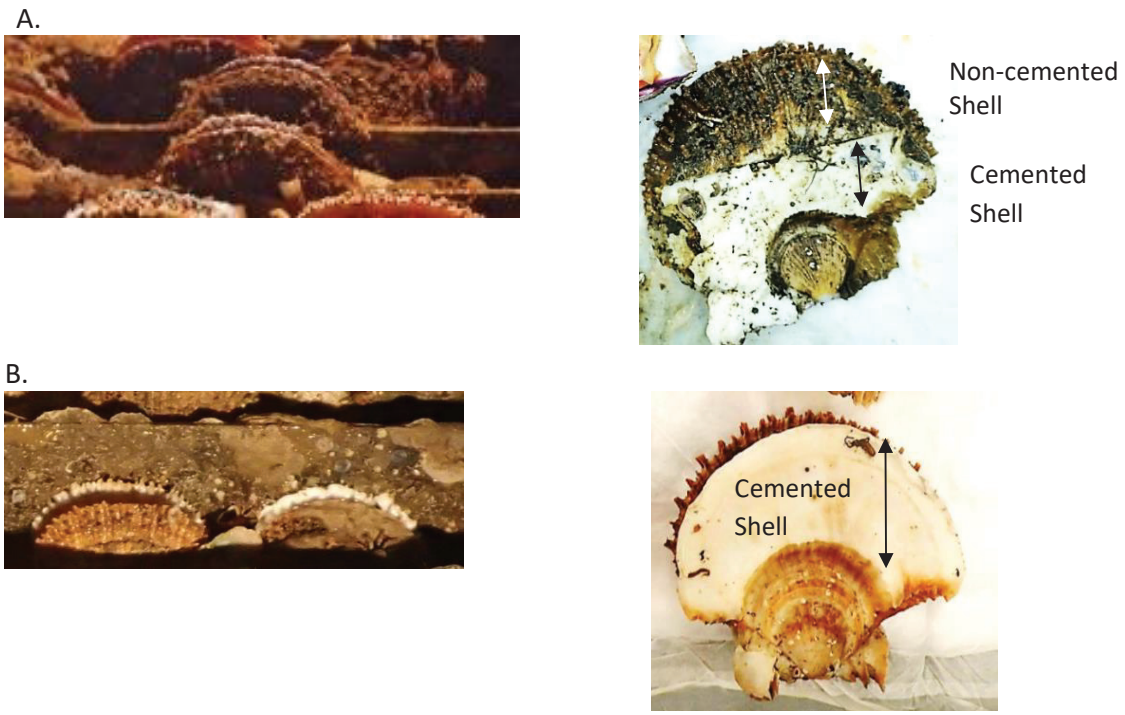


Figure 8. Growth of *Crassadoma gigantea* on short (A) and tall (B) flat panels in California. Attached valve of removed scallops is shown and illustrates non-cemented (brown shell) growth of scallop once past edge of short panel vs continued cemented growth (white shell) when grown on tall panel.

Photos: Carrie Culver

and labor costs, but it may be beneficial in certain locations and conditions (such as in California and high current areas), where the behavior is difficult to inhibit. Further, attachment can increase growth—thereby reducing time to market—and provides a means for branding the animals (see Culver et al. 2006). Inhibiting attachment through continued disturbance may be beneficial where local conditions enable scallops to maintain a fixed state such that cementation can be inhibited. While inhibiting attachment requires manually detaching scallops periodically during a certain time frame (about 12–14 months depending on the location) and results in decreased growth and increased time to market, it does not require special gear or setup. Growers will need to assess the feasibility of manipulating the cementing stage based on their systems and environmental conditions and consider time to market in order to identify the most optimal growout method for their circumstances.

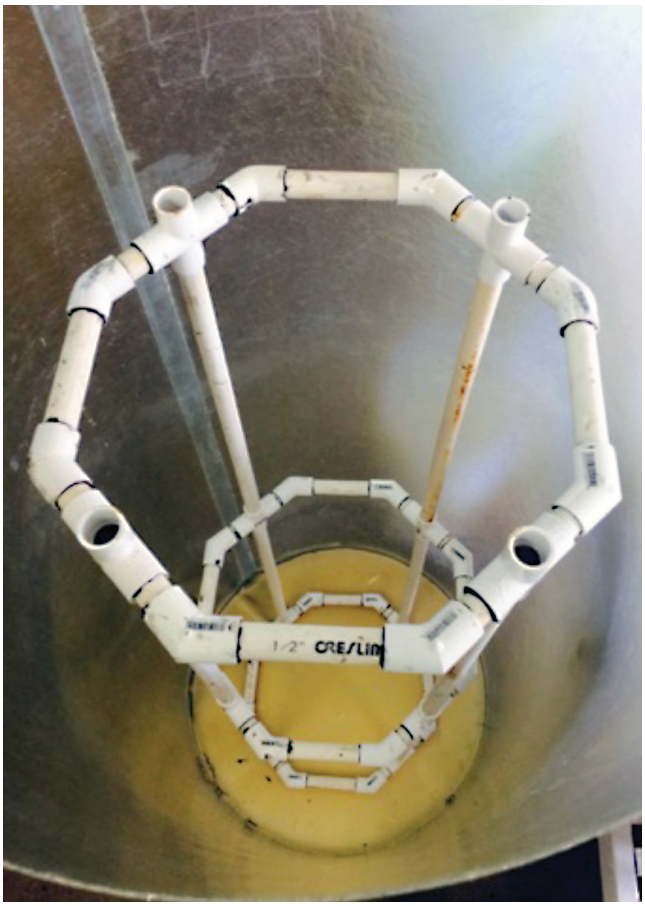


Photo: Courtesy of WRAC Rock Scallop Work Group

Photo: Karl Menard

Summary

The purple-hinge rock scallop remains a very promising aquaculture candidate for the US West Coast. To move rock scallop culture into commercialization, however, requires more research. Specifically, careful experimentation is needed to identify conditions for broodstock conditioning and larval rearing through metamorphosis. While we were not able to produce diploid seed through controlled spawning, we believe the natural spawns that occurred were associated with elevated water temperatures and lunar phase. We also noticed higher larval metamorphosis and survival in a tank with good aeration (an air diffuser was present) suggesting aeration and water circulation/flow warrant further investigation.

Additional research on production of triploid and tetraploid rock scallops, informed by our work, may also be required in some states to address regulatory concerns over genetic mixing.

Although not addressed here, research regarding up-take and retention of saxitoxin—a natural toxin produced during some harmful algal blooms and responsible for paralytic shellfish poison in humans—is also needed to inform potential harvest restrictions and market development for cultured rock scallops. Farmed bivalves are subject to shellfish sanitation assessments prior to sale, which protect the public from saxitoxin and other types of naturally occurring toxins and contaminants, and rock scallops would also require such testing. Despite these hurdles, by working together to address them, commercial rock scallop culture will undoubtedly become a reality.

References

Bourne N, Hodgson CA, Whyte JNC. 1989. A manual for scallop culture in British Columbia. Canadian Technical Report of Fisheries and Aquatic Sciences. 1694. 215 p.

Culver CS, Richards JB, Page HM. 2006. Plasticity of attachment of the purple-hinge rock scallop, *Crassadoma gigantea*: implications for commercial culture. Aquaculture. 254:361–369.

Helm MM, Bourne N, Lovatelli A. (comp./ed.) 2004. Hatchery culture of bivalves. A practical manual. FAO Fisheries Technical Paper. No. 471. Rome, FAO. 177p.

Leighton DL. 1991. Culture of Hinnites and related scallops on the American Pacific coast. In Winston Menzel, Ph.D., ed. Estuarine and Marine Bivalve Mollusk Culture. Boca Raton, Florida: CRC Press, Inc. pp. 100–110.

Leighton DL, Phleger CF. 1981. The suitability of the purple-hinge rock scallop to marine aquaculture. Rep. No. T-SCSGP 001. California Sea Grant College Program Publ., Center for Marine Studies, San Diego University, San Diego.

RaLonde R, Brenner K, Oliveira ACM. 2012. Alaska's purple-hinge rock scallops considered for aquaculture development. Global Aquaculture Advocate. May/June 2012:46–48.

Richards JB, Culver CS, Fusaro C. 2009. Shellfish harvesting as a biofouling control strategy on oil and gas platforms: Development of a profitable, symbiotic marine business in southern California. Ronald C. Baird Sea Grant Science Symposium: The ecology of marine wind farms. Nov 2–4, 2009. Newport, Rhode Island.

Ropes JW. 1968. Reproductive Cycle of the Surf Clam, *Spisula solidissima*, in offshore New Jersey. Biol Bull 135:349–365.

Shumway S, Parsons GJ. 2006. Scallops: Biology, Ecology and Aquaculture. 2nd Edition. Elsevier: Amsterdam, the Netherlands.

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