The Giant Clam: an Ocean Culture Manual

Edited by Hilconida P. Calumpong

ACIAR

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* Floating ocean nurseries

† Galvanised wire cages and wire-reinforced cement substrates

Preface

The giant clam represents a traditional food source for the people of the Indo-Pacific region. The demand for the meat or shells, coupled with the over-exploitation by Taiwanese boats have decimated and locally exterminated populations of some species. Thus, in the 1970s and 1980s, research into the reproduction and larval culture of giant clams became important. The Micronesian Mariculture Demonstration Center (MMDC) in Palau played a key role in moving mariculture of giant clams from the laboratory to mass culture. The University of Papua New Guinea (Motupore Island Research Centre) was likewise involved in giant clam larviculture, but on a smaller scale than the MMDC. In the early to mid 1980s, Australia became involved with giant clam mariculture, first in a small government-funded project through the University of New South Wales and then in a separate large project funded by the Australian Centre for International Agricultural Research (ACIAR) and administered by James Cook University of North Queensland. The ACIAR-funded giant clam project began with Australia, Fiji, Philippines and Papua New Guinea but by Phase II in the late 1980s it included Tonga, Cook Islands, Kiribati and Tuvalu, while losing Papua New Guinea. Also, in the late 1980s, the International Centre for Living Aquatic Resources Management (ICLARM) set up a coastal aquaculture centre near Honiara, Solomon Islands in which giant clams are the main organism being cultured. Other hatcheries and ocean nurseries have been started, notably in Micronesia and most recently in Tonga and Cook Islands.

The markets for giant clams are at present almost exclusively in Asia and the Pacific islands. The demand is for adductor muscle, mantle (fresh, dried, frozen) and shells. Although more work is required on marketing aspects, the general indication for future development and expansion of giant clam cultivation is positive.

This manual provides a guide to the practicalities of giant clam aquaculture. It deals with ocean nursery culture operations (see Figure 1.2, page 14). A companion manual, also published by ACIAR, details the hatchery and land nursery culture of the giant clams.

Acknowledgments

This manual is the product of research in eight countries: Australia, Cook Islands, Fiji, Kiribati, Philippines, Solomon Islands, Tonga and Tuvalu. The work was largely funded by the Australian Centre for International Agricultural Research through projects 8322 and 8733. The research was also supported in part by the Australian International Development Assistance Bureau (AIDAB).

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Chapter 1

Introduction

Giant clams (Tridacnidae: Bivalvia) are the largest bivalves in the world. They inhabit shallow clear waters of coral reefs around countries in the Indo-Pacific region like Papua New Guinea, Indonesia, Malaysia, Palau, northern Australia and the Philippines (Rosewater 1965; Lucas 1988; Lucas et al. 1991).

Aside from achieving great proportions, giant clams are unique bivalves in that they harbour symbiotic algae, called zooxanthellae, which supply food to the clams through photosynthesis. Therefore, giant clams have their own built-in 'food factories' which need only sunlight, water and carbon dioxide as basic raw materials. They also filter microscopic particles from water flowing over their gills, like other bivalve molluscs (oysters, scallops, etc.).

Giant clams have two main parts, the **shell** (which can be used for identification of the different species) and the soft **flesh** which is covered by the **mantle** (the coloured flesh of the clam). The mantle has an elongate incurrent aperture and a round excurrent aperture for seawater to pass over the clam's gills. Beneath the mantle are the different organs, including the heart, gills, kidney, adductor and retractor muscles, digestive tract, gonad, foot and byssus. Figure 1.1 illustrates the external and internal parts of a giant clam.

1.1 Identification of giant clams

There are eight known species of living giant clams. These species may be identified by their shape, shell characteristics (such as presence and absence of scales), relative sizes and the growth habit of adult clams on reefs.

Identification is facilitated by knowing the differences between the two genera of giant clams, *Tridacna* and *Hippopus*. Differentiation of these two genera is based on the external surface of the

byssal region and by the colour of the mantle. In *Hippopus* species, the area of the byssal orifice possesses tightly fitting teeth while in *Tridacna* species, teeth are absent. Additionally, the mantle of *Tridacna* species generally has bright colours and projects laterally beyond the lip margin of the shell, while the mantle of *Hippopus* species is usually dull and limited within the shell margin. A rare species from Tonga and Fiji, *T. tevoroa*, is somewhat transitional in these characteristics. The characteristics of each species of giant clam are described and illustrated in Boxes 1.1–1.8. A key to the identification of the eight giant clam species is shown in Box 1.9.

1.2 Importance of giant clams

Giant clams have long been part of the diet of island and coastal peoples of the Indo-Pacific region. The entire flesh of these clams is edible except for the kidneys, which are bitter. The adductor muscle, which comprises about 10-15 % of the entire flesh, is most sought after. This is the large muscle which draws the shells together.

The shells of giant clams are used locally for a variety of purposes (sinks, ashtrays, soap cases, house decors, pots). They are also important items as souvenirs in the tourist trade, for shell and stone craft and for tile-making.

1.3 Culture of giant clams

Natural stocks of most species of giant clams have been severely reduced in many areas by over-harvesting, almost to the point of extinction. Clam culture provides (1) food to coastal inhabitants; (2) income to farmers from raw materials for new industries; (3) a means to restock coral reefs depleted of clams and to promote reef conservation.

Because of their mariculture potential, giant clams have been the subject of intense studies at institutions in Australia, Palau, the Philippines, Solomon Islands and elsewhere in the Pacific.

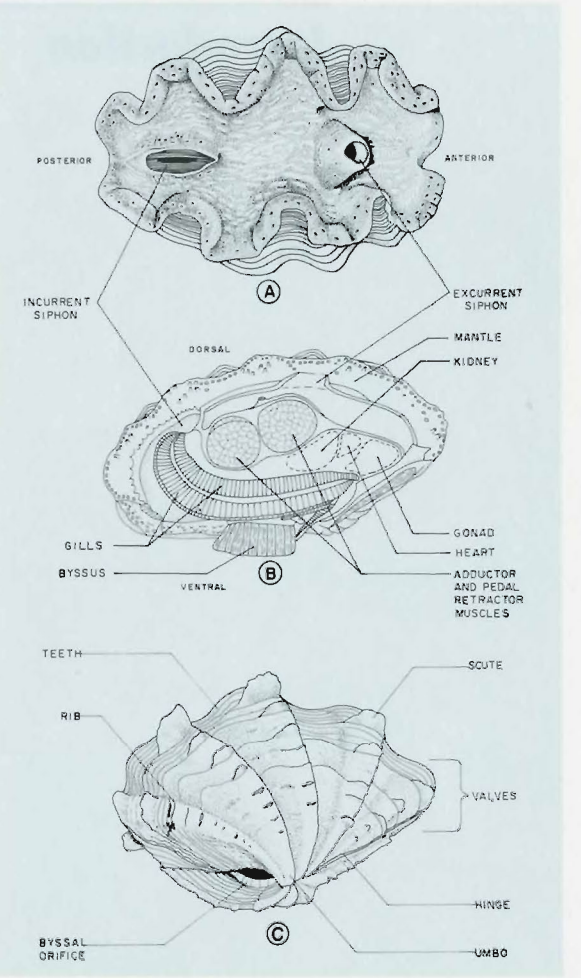
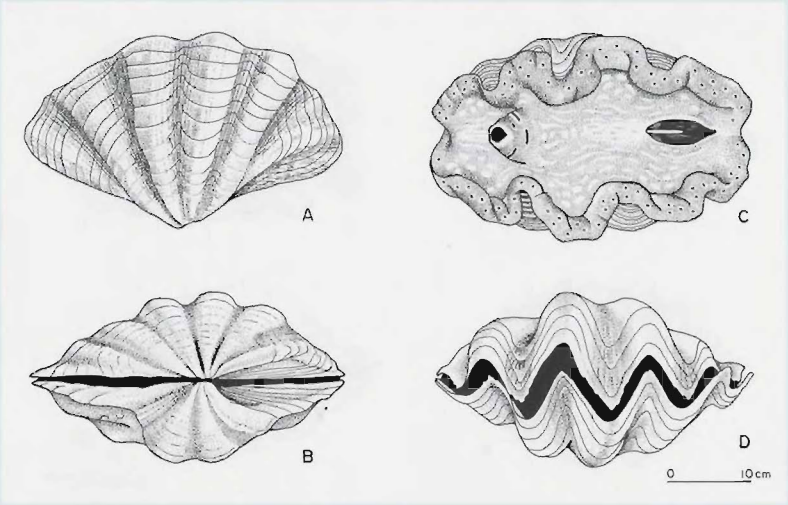


Figure 1.1 Parts of a giant clam. A. Dorsal view. B. Lateral view showing the internal organs. C. Lateral view of shell.

Box 1.1 *Tridacna gigas*



A. Lateral view. B. Ventral view. C. Mantle view. D. Dorsal view.

Scientific name: *Tridacna gigas*

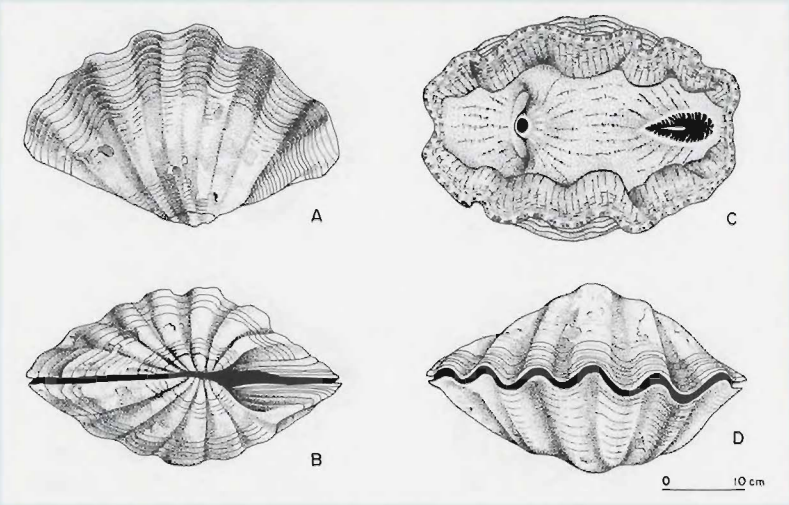
Common name: True giant clam

Description: Largest species, attaining lengths of over 100 cm and weights of 200–500 kg; shell white in colour, fan-shaped (side view) with deep grooves; edge of the shells bear elongate, triangular projections. Large individuals are unable to close their shells completely because of the well-developed mantle.

Habitat: Generally found on sand and among corals on shallow reefs but may be found at depths of 20 m; some individuals may be exposed during low tide.

Distribution: Known to be previously distributed more widely in the Indo-Pacific region but now restricted through overfishing to Australia (Great Barrier Reef), Indonesia, Papua New Guinea, Philippines (Palawan and Sulu Sea), Solomon Islands and ports of Micronesia.

Box 1.2 *Tridacna derasa*



A. Laterol view. B. Ventral view. C. Mantle view. D. Dorsal view.

Scientific name: *Tridacna derasa*

Common name: Smooth giant clam

Description: Second largest species, reaching 50 cm or more in length; shell white and very smooth, the teeth at the lip margin rounded; shell thick and heavy.

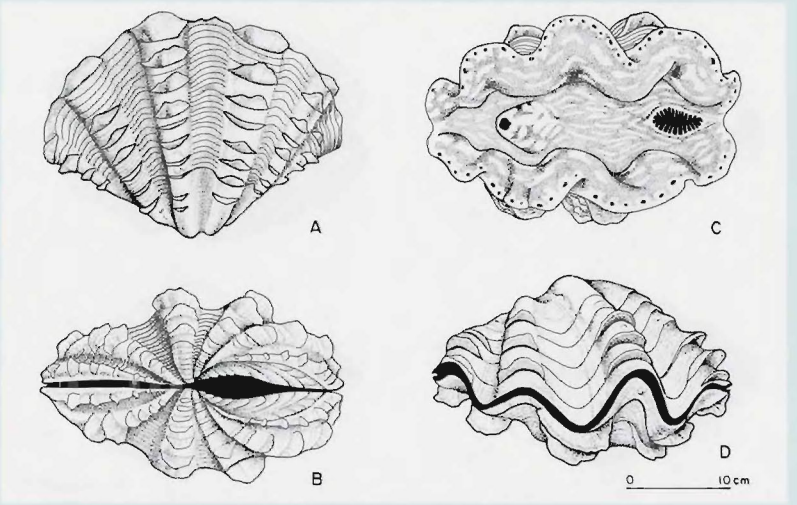
Tridacna derasa is often confused with another species, *H. porcellanus*, which also has a white, very smooth shell and rounded lip margin. However, the byssal region is heart-shaped in *H. porcellanus*.

Juveniles of *T. derasa* and *T. gigas* are also similar in appearance. To differentiate, *T. gigas* possesses deep grooves on its shell surface. Also, the mantle of *T. derasa* is almost always brightly coloured (blue and green) while that of *T. gigas* is usually drab (yellowish brown to tan).

Habitat: Occurs on outer edges of reefs at about 4–20 m and appears to favour oceanic environments more than fringing reefs adjacent to large island masses.

Distribution: Australia (Great Barrier Reef), Pacific Islands to Tonga, Philippines (Palawan; Sulu Sea; Bato, Leyte; Guiuan, Samar; Scarborough Shoals, South China Sea; Lubang Is., Mindoro), Indonesia.

Box 1.3 *Tridacna squamosa*



A. Lateral view. B. Ventral view. C. Mantle view. D. Dorsal view.

Scientific name: *Tridacna squamosa*

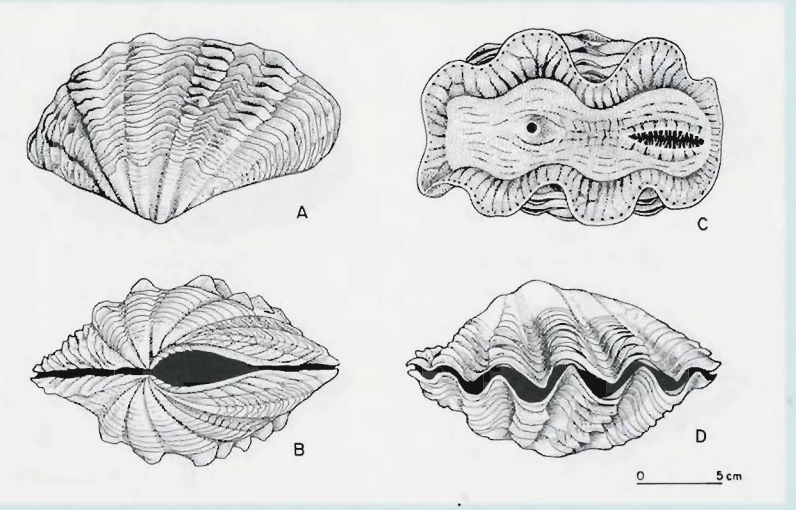
Common name: Scaly or fluted clam

Description: May reach 35–40 cm long; equilateral shell with widely spaced fluted scales which become larger towards the edge of the valve; shell colour is variable, some may be white, tinged with lemon yellow towards the margin, or may be yellow with deep orange in between grooves, or just plain yellow or orange. This species is differentiated from other scaly species (*T. maxima* and *T. crocea*) by the presence of well spaced scales from the umbo towards the lip margin of the shell.

Habitat: Attached by a weak byssus to living coral or coral rubble at depths up to 18 m on reefs usually dominated by *Acropora*; found in both oceanic and nearshore reefs.

Distribution: Found from East Africa and the Red Sea to French Polynesia.

Box 1.4 *Tridacna maxima*



A. Lateral view. B. Ventral view. C. Mantle view. D. Dorsal view.

Scientific name: *Tridacna maxima*

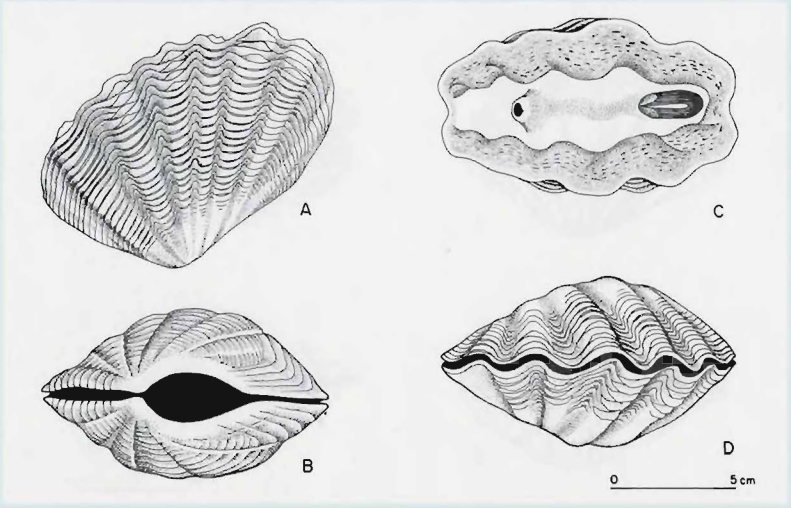
Common name: Elongated clam

Description: Can reach 35–40 cm in length but is usually much smaller; bright mantle colouring; shell is elongated at one side with closely spaced scales near the margin; shell colour varies from plain white to yellow or white tinged with orange.

Habitat: Partly embedded in coral or firmly attached to coral heads.

Distribution: Found from East Africa and the Red Sea to French Polynesia.

Box 1.5 *Tridacna scrocea*



A. Lateral view. B. Ventral view. C. Mantle view. D. Dorsal view.

Scientific name: *Tridacna crocea*

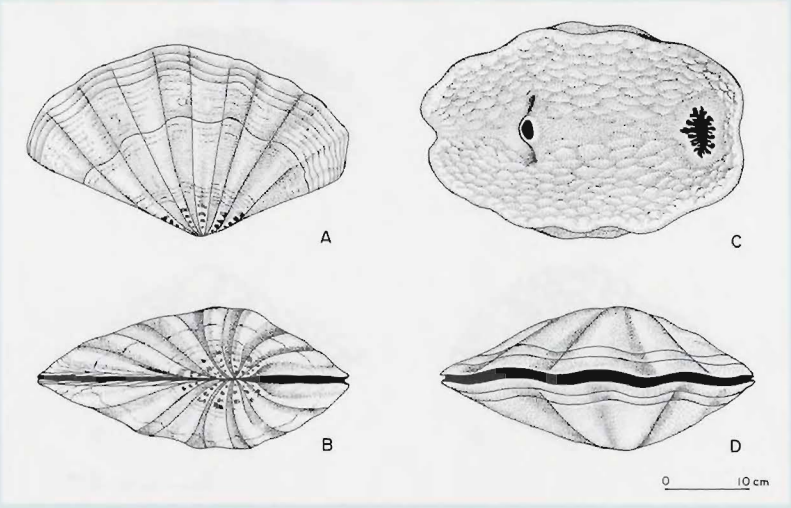
Common name: Boring clam

Description: Smallest species reaching only 15 cm in length; shell white often tinged with pinkish-orange or yellow both inside and outside; usually quite smooth except for closely spaced scales near the upper edge of shell; mantle usually brightly coloured like *T. maxima* but can be differentiated in having a strongly triangular ovate shell (lateral view) and very wide byssal orifice (ventral view).

Habitat: Burrows into coral boulders on the reef-top; only shell margins and mantle are visible.

Distribution: Widespread and common in the Indo-Malay and western Pacific regions, from Thailand to New Caledonia.

Box 1.6 *Tridacna tevaroa*



A. Lateral view. B. Ventral view. C. Mantle view. D. Dorsal view.

Scientific name: *Tridacna tevaroa*

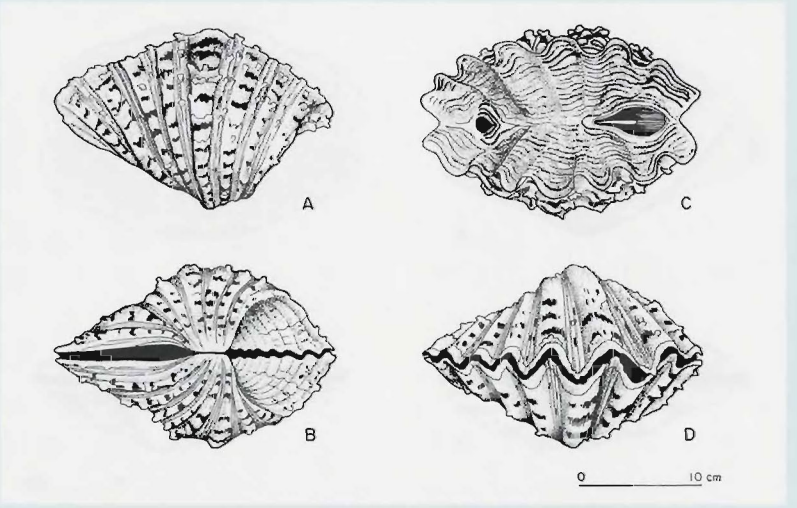
Common name: Tevoro clam

Description: Relatively large, reaching more than 50 cm in shell length, valves semicircular, light with limited thickenings and dark red bands at umbo, upper shell margins undulate, sharp and thin with shallow interdigitations, similar to *T. derasa*; mantle with numerous warty projections and brownish-grey in colour; large white or brown papillae around incurrent siphon.

Habitat: Found at moderate depth (14–30 m) in clear oceanic waters, typically on the outside slope of leeward reefs.

Distribution: Recorded only from Tonga and the eastern (Lau) islands of Fiji.

Box 1.7 *Hippopus hippopus*



A. Lateral view. B. Ventral view. C. Mantle view. D. Dorsal view.

Scientific name: *Hippopus hippopus*

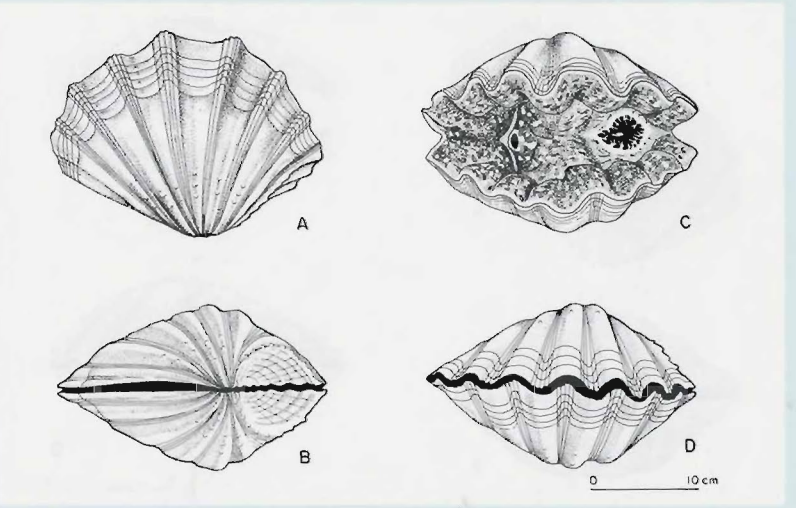
Common name: Rolling clam, strawberry clam, horse's hoof clam, bear's paw clam

Description: Reaches 50 cm in length; shell is thick, heavy, and elongate to triangular in shape with minute scales or spines; some shells possess strawberry blotches; mantle is brown, dull grey or green in colour.

Habitat: Found on sandy areas in coral reefs at depths up to 6 m and on seagrass beds adjoining reefs.

Distribution: Widespread and common in the Indo-Pacific from Thailand to Vanuatu. Recently extinct in Fiji, Samoa and Tonga.

Box 1.8 *Hippopus porcellanus*



A. Lateral view. B. Ventral view. C. Mantle view. D. Dorsal view.

Scientific name: *Hippopus porcellanus*

Common name: China clam or porcelain clam

Description: Thinner and smoother shell than that of *H. hippopus*, usually lacking the strawberry colouration. The mantle is olive green. *H. porcellonus* can be easily distinguished from *H. hippopus* because the incurrent siphon possesses papillae or fringing tentacles.

Habitat: Occurs in sandy portians of coral reefs.

Distribution: Rare, and restricted to the Sulu and South China Seas, Philippines and Palau.

Box 1.9 *Key to the giant clam species (from Lucas et al. 1991)*

There are now eight extant species of giant clams and the key to species given by Lucas (1988) needs to be expanded and modified to incorporate *T. tevoroa*. See Figure 1.1 for the location of the various parts of the clam used in the key.

1 Byssal orifice region of opposed valves with interlocking teeth; distinct region of shell around byssal orifice, outlined by ventral-most pair of prominent radical ribs; mantle, when fully extended, not projecting laterally beyond shell margins ..... *Hippopus* ... 2

Byssal orifice of opposed valves without interlocking teeth; no distinct ventral region of shell outlined by prominent radial ribs; mantle, when fully extended, usually projecting laterally beyond shell margins ..... *Tridacna* ... 3

2(1) Shells thick and strongly ribbed, with reddish blotches in irregular bands; incurrent apertures without guard tentacles (papillae) ..... *H. hippopus*

Shells in specimens less than about 200 mm shell length not thick nor strongly ribbed and with only faint reddish blotches; incurrent apertures with guard tentacles (papillae) ..... *H. porcellanus*

3(1) Shell length of large specimens >550 mm, sometimes greater than 1 m; with about four elongate, interdigitating projections of each distal shell margin, being most elongate and acute in large specimens; shell without scutes, except for some tubular projections near umbo in very small juveniles; mantle brownish, with numerous iridescent blue-green circles ..... *T. gigas*

Shell length rarely >550 mm; without elongate ; interdigitoting projections on eoch distal shell margin; mantle variably coloured, without iridescent blue-green circles 4

4(3) Shell length up to 500 mm, occasionally larger; upper region of large shells ploin, without scutes or strong ribs; hinge usually longer than half shell length 5

Shell length usually > 500 mm; upper shell region with scutes or eroded scutes; hinge equal to or less than half shell length 6

5(4) Rib-like radial folds on shell without coloured patches; mantle without protuberances; incurrent aperture with inconspicuous guard tentacles ..... *T. derasa*

Rib-like radial folds on shell usually striped with coloured patches near umbo; mantle with protuberances; incurrent aperture with conspicuous guard tentacles ..... *T. tevoroa*

6(4) Shell approximately symmetrical about umbo in lateral view, with hinge about half shell length; scutes large and well-spaced both within and between the radial rows; lateral distance between scutes in adjacent rows usually about same as scute width; byssal aperture narrow to moderately wide; not embedded into substrate; mantle usually of subdued and mottled colour; incurrent aperture with distinct guard tentacles ..... *T. squamosa*

Shells usually asymmetrical about umbo in lateral view, with hinge less than half shell length; scutes usually low and often eroded, set close together both within radial rows and between rows; byssal aperture moderately wide to wide; embedded or partly embedded into substrate, mantle brightly coloured; incurrent aperture with indistinct guard tentacles 7

7(6) Shell length <150 mm; shells not strongly asymmetrical about umbo in lateral view; byssal aperture wide; scutes eroded away except near shell margin; occurs deeply embedded in reef substrate ..... *T. crocea*

Shell length of large specimens often >150 mm; shell often strongly asymmetrical about umbo in lateral view; byssal aperture moderately wide to wide; scutes present in substantial part of upper shell region; occurs partially embedded in reef substrate ..... *T. maxima*

There are four basic phases in giant clam culture: the hatchery, the nursery, the ocean nursery and growout phases (Figure 1.2).

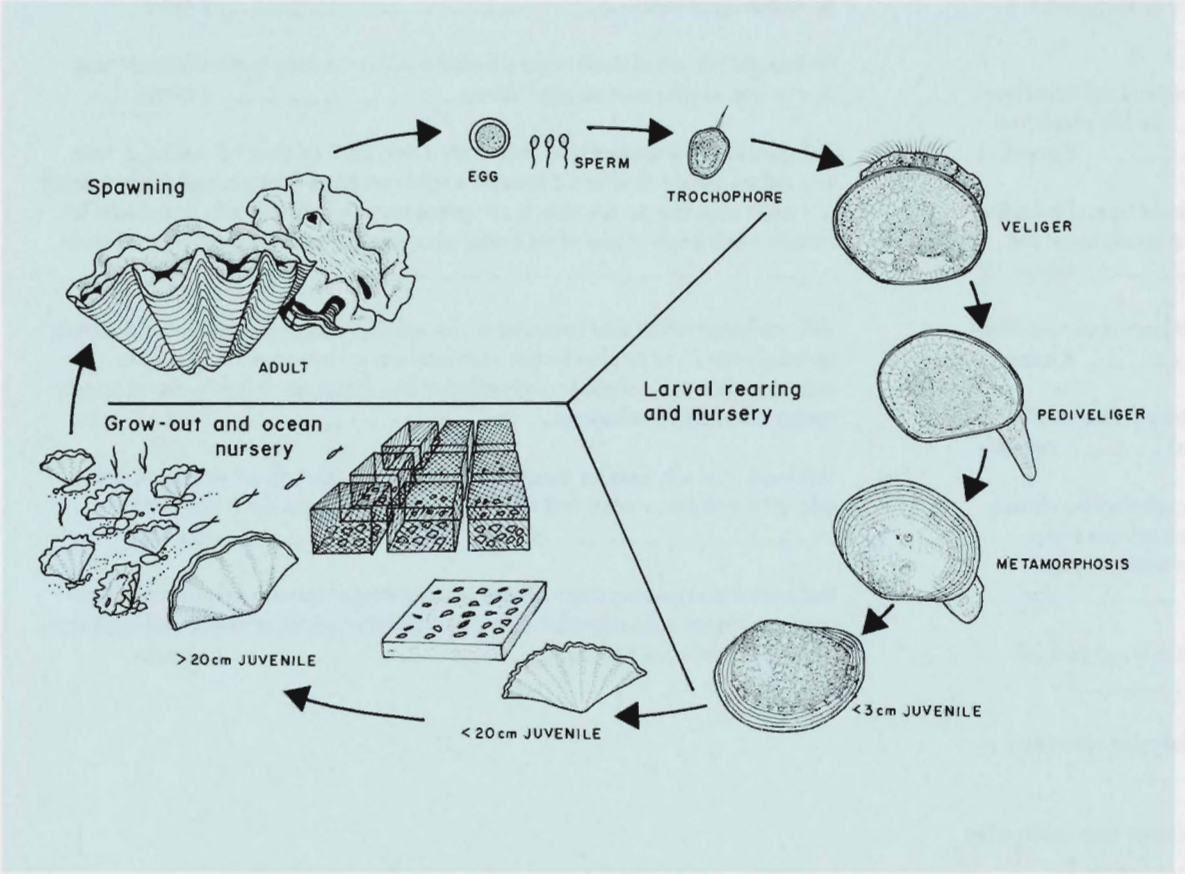


Figure 1.2 Stages in the life cycle and aquaculture of the giant clams

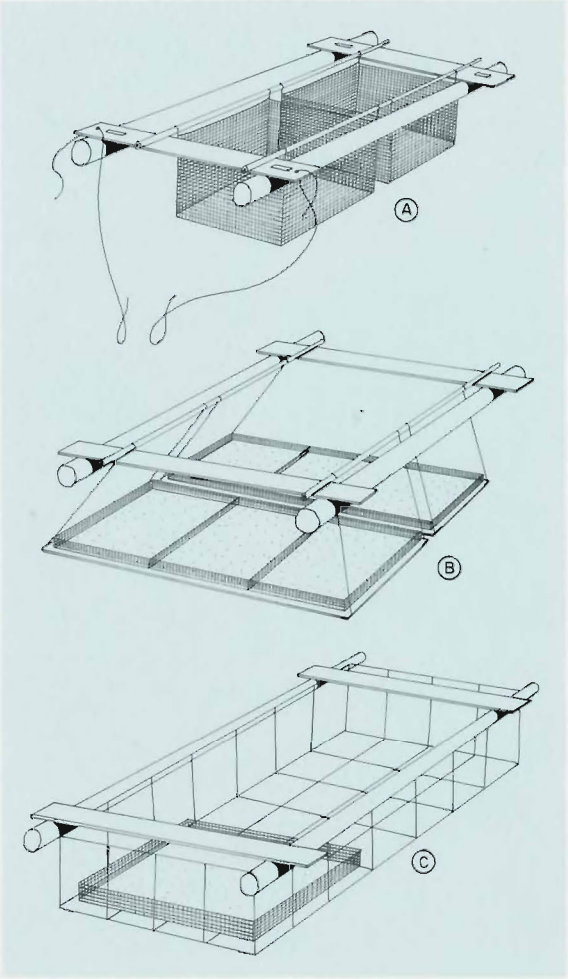


Figure 1.3 Different designs of floating cages used at the Coastal Aquaculture Centre (CAC), Solomon Islands.

1.3.1 Hatchery and nursery phases

The hatchery phase includes: (1) induced spawning of giant clams using macerated gonad suspension or serotonin (neurotransmitter); (2) hatching of eggs for one day in hatching tanks; (3) rearing of clam larvae in larval tanks for 1–2 weeks; and (4) rearing of juveniles for 2–3 months either in land-based tanks or floating ocean cages (see section 1.4).

In the land-based nursery culture, newly settled juvenile clams are placed in tanks with free flowing seawater and aeration. Here, they are allowed to grow to approximately 2 cm, after which they are either allowed to grow to marketable size or transferred to the ocean in benthic cages. For more information on hatchery and nursery culture, please refer to Braley (1992).

1.3.2 Ocean nursery and growout phases

The ocean nursery phase of giant clam culture is discussed in some detail in Chapters 2 and 3 and the growout stage is described in Chapter 4.

1.4 Floating ocean nurseries

In the Solomon Islands, at the ICLARM Coastal Aquaculture Centre (ICLARM CAC), 3.5 mm juvenile clams are transferred to floating ocean nurseries where they are reared to about 35 mm before transfer to benthic cages.

The current designs are shown in Figure 1.3A–C. The essential components are two lengths of 150 mm PVC drain piping with endcaps which provide the buoyancy, joined by two hardwood crosspieces to form a catamaran. Stainless steel strapping was initially used to assemble the catamarans. However, heavy monofilament nylon fishing line is now being used. The resulting structure has relatively high rigidity, can withstand moderate seas and should be adequate for the sheltered lagoonal waters which are envisaged as primary sites for deployment of the system.

Each catamaran has a polypropylene rope bridle with shackles and thimbles. They are usually rigged

in trains of three, shackled to a mooring system which consists of individual anchor blocks, chain and a large float attached to an appropriate length (2.5 × depth) of heavy nylon line.

The catamarans support light ferrocement-based trays in various configurations; either as individual trays (about 0.75 square metres each) suspended from hardwood poles (Figure 1.3A), as platforms suspended from the catamaran to which small (0.42 square metres) trays are individually attached (Figure 1.3B) or as a rigid weld mesh box in which two or three trays are deposited (Figure 1.3C).

These basic designs have been tested in various locations, including the relatively exposed waters fronting ICLARM-CAC, where short choppy waves of around 0.4 m height are often experienced on windy afternoons. For use in sheltered areas there is an obvious opportunity for using bamboo floats, particularly if properly cured and epoxy-coated. In relatively deep water (>3 m) it is unnecessary to have protective covers or sides on the tray. Despite the fact that the hardwood cross bearers are above water-level problems are encountered with shipworm (*Teredo* sp.s) and it is necessary to paint the timbers with epoxy paint.

The optimum sites for floating ocean nurseries appear to be areas of shallow sandy seafloor or seagrass beds, with a moderate degree of exposure to wind-induced waves and good water exchange as a result of strong currents or large tidal amplitudes. The designs shown in Figure 1.3 have withstood severe weather conditions and will suffer few gear failures provided they are regularly inspected and maintained.

The nursery trays are stocked with juvenile clams by submerging the trays in raceways or tanks and stocking them with 2000 3.5 mm juveniles per square metre. The juveniles are left undisturbed for 48 hours for them to establish their byssal attachments, whereafter they can be moved offshore. Omission of this step can result in severe losses of juveniles if they are exposed to moderate wave action before they are properly attached.

Thereafter, husbandry consists of regular checks for predators, principally *Cymatium* species and stylochid flatworms and for control of fouling organisms which can clog the meshes and reduce

water exchange within the tray. Algal overgrowth can be reduced and sedimentation controlled by simply fanning the bottom of the tray and wafting away the unwanted debris. Herbivorous gastropods, such as *Cerithium* species, will also reduce algal growth.

There is no difference in the mortality rates of juvenile clams in unfertilised nursery tanks and in the floating ocean nurseries. However, there is a dramatic change in the rates of growth on transfer to the ocean, with the juveniles in the floating ocean nurseries attaining about 35 mm at 10 months, at which point they can be transferred to benthic ocean nursery cages.

Juvenile clams can be grown in the floating nurseries until they attain a size of 30–60 mm, depending on their location and degree of exposure, and then transferred to benthic cages. Alternatively, it might be possible to retain them in floating cages until they attain a size at which they can safely be placed in enclosures or exclosures.

Chapter 2

The ocean nursery

The ocean nursery phase in the culture of giant clams begins when the juvenile clams are transferred to the sea in protective mesh cages. They are caged for protection against predators until the 'escape size' (about 20 cm) is reached.

The culture site may be subtidal or intertidal. *T. gigas* and *H. hippopus* do well in the intertidal zone while *T. derasa* and the rest are best grown in shallow subtidal sites. The great advantage of intertidal culture is that it is relatively accessible and easy to maintain. The only disadvantage is the greater exposure during tropical cyclones (typhoons) and accessibility to poachers.

The length of time that juveniles stay in the ocean nursery (before transfer to the growout site) is dependent on the clam species and the conditions of the ocean nursery. It may be 2–4 years. Figure 2.1 illustrates an ocean nursery.

2.1 Site selection

Sites that have the following conditions have been found most suitable for optimal growth and survival of clams.

Shallow depth of water. Ocean nurseries can be placed on either lower intertidal or shallow or not too deep (<10 m) portions of the inshore environment. These areas can be exposed or covered during the lowest low tide.

If a protected area with moderate current is found, clams can be placed in the lower intertidal zone. The exposure during extreme low tides minimises the need to clean cages since algal growth is retarded by direct exposure to sunlight. Excessive exposure to sunlight, however, can lead to

bleaching of the mantle and ultimately kill the clam, or if the clams survive, the shells tend to become thickened and stunted in length. Intertidal ocean nurseries have limited application in the Philippines, due to the utilisation of protected areas for docking and other needs. Also, these areas may be exposed at one time or another to the monsoon trade winds.

An alternative is to establish the ocean nursery somewhat deeper where wave action is minimised by the depth of the water. Water visibility must be good (3–7 m) to ensure optimal illumination for photosynthesis. Figure 2.2 shows the possible areas (arrows) within the coastal environment where ocean nurseries can be established.

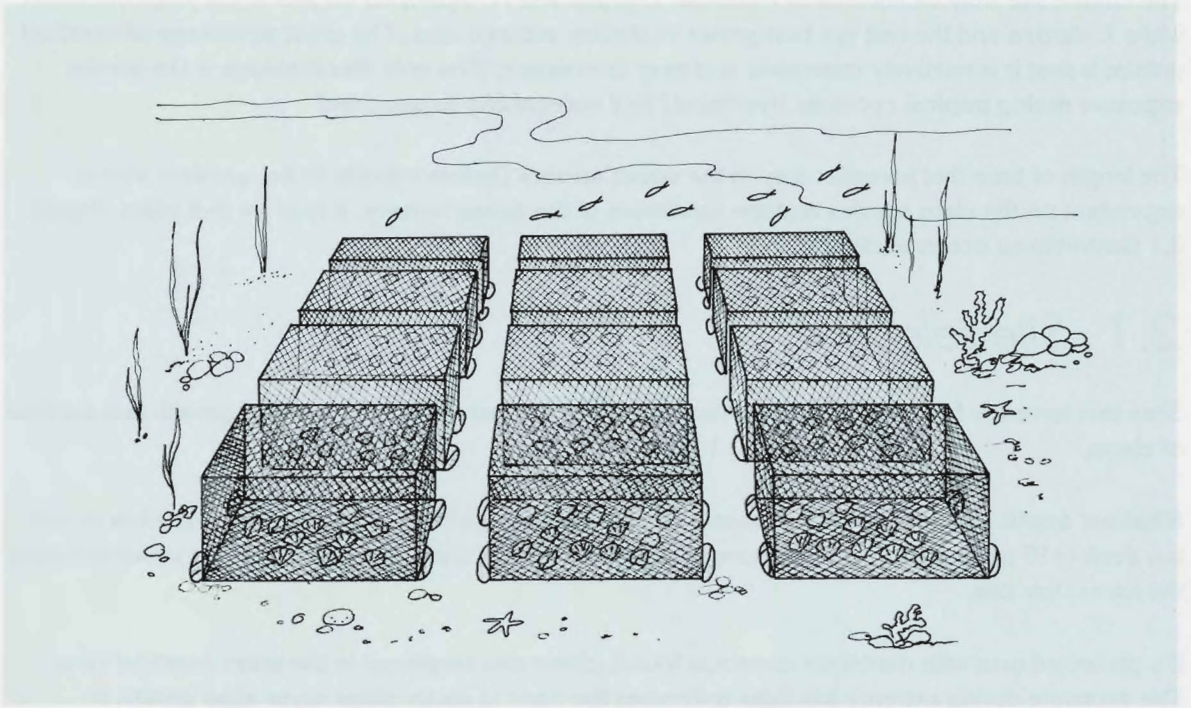


Figure 2.1 Illustration of an ocean nursery

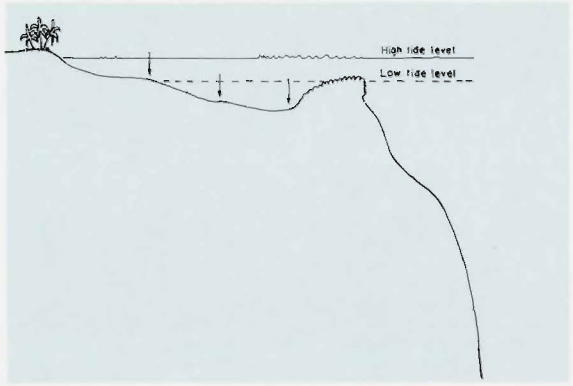


Figure 2.2 Possible areas (arrows) where an ocean nursery may be established.

At Orpheus Island Research Station (OIRS), Australia, where tidal variation is about 3.0 m, scientists from James Cook University (JCU), North Queensland, found that a tidal height of approximately 0.4 m to 0.6 m gave the best growth rates and survival for *T.gigas*. Maximum exposure during spring tides was about 3 hours at this tidal height.

Clear water with high salinity (not brackish). Giant clams require clear water with good light penetration. This is necessary because they have symbiotic algae, *Symbiodinium microadriaticum*, in their mantles which are responsible for providing food for the clams through photosynthesis.

Giant clams have a narrow range of tolerance to changes in salinity. Ocean nurseries should thus be established in **areas away from freshwater runoff**. Heavy rainfall during the rainy season brings high sediment load with river waters reducing light penetration in the water column.

Quiet water with good circulation. Clams thrive in areas with quiet water and good circulation. Good circulation prevents the column of water from stagnating within the nursery,and brings in fresh water with nutrients for the clams.

Strong water movement is caused either by naturally occurring currents or by wave action. Strong wave action can destroy cages, chip clam shells and overturn clams, resulting in the mantle facing downward instead of upward. Water movement due to cyclones (typhoons) and storms can cause similar damage. Further mortality can be caused by sand shifts, which can smother the mantle of the clams or bury them alive.

Areas with naturally occurring strong currents can retard normal growth in shell length and instead, clams grow thick, bulky shells to protect the flesh.

Areas with slow circulating water (as in deep bays) are not advisable for clam culture since the substrate of bays is usually mud and light penetration is correspondingly low.

Sea bed with few or no predators. To ensure clam survival, a site with few or no predators should be chosen. Coral reefs, although natural habitats of clams, have been found to contain many clam

predators such as fishes, crabs and gastropods. In addition, unless a damaged portion of a reef is utilised, the presence of corals will severely limit the size of the ocean nursery.

Sandy-seagrass areas. Sandy areas found near the mouth of bays are also good areas for ocean nurseries as long as the water is clear.

A seagrass bed is another site where an ocean nursery can be established. Although the substrate on which seagrass grows is usually mud and silt, there are some seagrass beds that have a firm, sandy bottom. These beds are usually found fringing the coastline away from bays and composed of several species of the smaller seagrasses.

In the Philippines, at Silliman University Marine Laboratory, (SUML) Dumaguete, and at University of the Philippines Marine Science Institute (UPMSI), Bolinao, the giant clams are successfully cultured on seagrass beds where they have been exhibiting good growth and survival rates.

2.2 Intertidal ocean nursery

2.2.1 Construction of containers for intertidal ocean nursery culture

Boxes

A box (Figure 2.3) is a galvanised steel mesh unit, covered with plastic mesh or chicken wire. In the example shown in Figure 2.3 two pieces of steel mesh (100 mm aperture) were used: a rectangular piece measuring 1.9 m x 0.9 m for the lid and a larger piece measuring 2.3 m x 1.3 m for the base. The large piece allows 0.2 m to be folded up along each edge to give the box its depth. The corners are joined together using either electrical cable or cable ties. Cable ties designed for outdoor applications and preferably without any steel in them to prevent corrosion are used. A plastic mesh is attached outside the steel box frame using the cable ties. The size of the plastic mesh used depends on the possible predators; however, 25 mm aperture appears to work well. A hinge is made from three 90 mm lengths of reinforced plastic tubing approximately 20 mm in diameter. The top wire of the

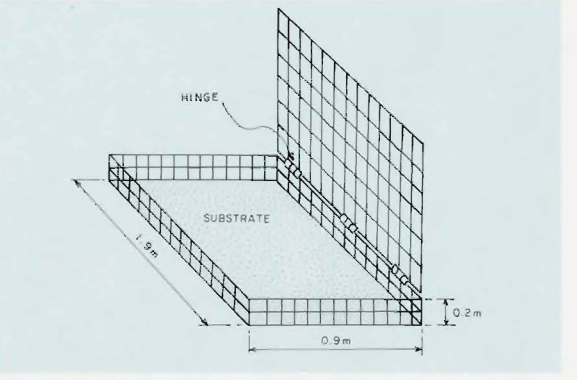


Figure 2.3 Box for intertidal ocean nursery culture

framework is split at the centre of three divisions along one of the longest sides. The same is done on the lid at the equivalent points. These allow the tubing to be slipped onto the wire of the lid and the base, thereby joining them. It is often necessary to make a short slit in the tube to allow it to be moved beyond the split in the wire which can then be taped up. The lid is held closed with short lengths of electric cable.

Similar boxes could be made with a wood frame and chicken wire covering, although chicken wire tends to corrode in the intertidal zone.

Once the box is positioned on the reef-flat or on blocks in sandy areas it can be anchored using fencing pickets and electric cable. A substrate such as coral rubble or gravel is necessary for small clams to attach themselves to. This substrate should be shovelled into the box.

Lines

A line is the extension of the box which uses only the plastic mesh and not the galvanised steel. The structure is derived from the use of galvanised fencing wire which is threaded through the mesh and the pickets (Figure 2.4). Plastic mesh of 12 mm aperture is used for the base and 25 mm aperture for the lid. These are commercially available in 30 m lengths in Australia. One of each is required for a line. Initially, the base is rolled out in the required position. The ends and sides are folded up to the required 0.2 m. The corners are again fixed together using electrical cable or cable ties. With the sides raised a picket can be placed alongside the corners at one end. These can be pounded into the substrate until they are level with the top of the side. Working along both sides of the line, pickets are driven into the substrate every five metres, carefully keeping them the same distance apart as the two end pickets. The galvanised fencing wire is threaded through the mesh at the top of the sides. A second strand is threaded through at the bottom of the side to act as a former as well as a backup in case one breaks. One end is tied off around the end picket and the other end tensioned on itself. It helps to have an anchoring picket driven in at 45° and about a metre beyond the end of the mesh to prevent the end pickets from being pulled out. Electrolysis occurs if there is contact between the wire and the picket. To prevent this, a sleeve cut from reinforced garden hose is used at every contact point. There are two methods of securing the sleeve: (1) by tying the sleeve to the picket using

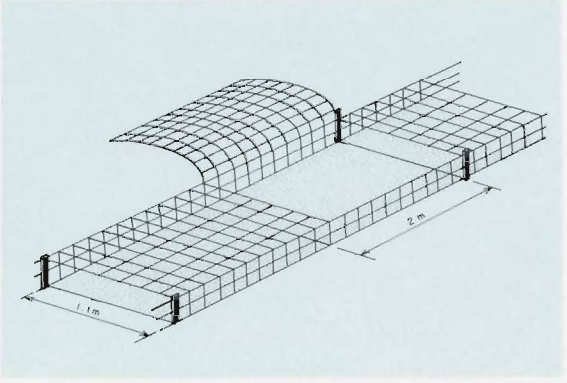


Figure 2.4 Line for intertidal ocean nursery

insulated wire or cable ties; and (2) by drilling holes at the correct height through the pickets with sleeve pushed into it. The latter is more secure.

At this stage partitions are added to divide the line into 15 cells. These partitions are made about 200 mm longer than the width of the base and 100 mm taller than the height. The excess is used to provide an overlap to allow secure fastening. The cell is 2 m long so 14 dividers will be used per line. Cable ties are the preferred method for tying them to the base. When this is done the substrate is introduced, allowing enough to cover the whole bottom to a depth of about 50 mm. The line is now ready for the lid.

To make the lid, the mesh is rolled out along the full length, making sure that it is long enough and sitting straight. Working from one end the windward side is secured with cable ties to the galvanised wire and the mesh of the base. The opening edge is secured using electrical cable, two or three fastenings per cell.

Again, these should be tight and the lid tied down securely. For every second cell a split is cut into the lid running from the leeward edge approximately two thirds across the top. This allows the top to be lifted from two cells at a time. Every divider beneath a cut will need a 100 m piece laid flat along the full length of its divider to prevent a gape from occurring at the cut. The split is joined using a piece of electrical cable passed through the partition.

The trick when working with the plastic mesh is to pull everything tight. It should not be put under too much stress but, if taut, there is less movement during wave action and it is less likely to tear.

2.3 Subtidal ocean nursery

A protected but shallow (2–3 m at low tide) site should be sought for subtidal culture. Clams are placed in trays, cages, enclosures, or a combination of these either on or without substrates.

2.3.1 Cages

Clam cages function (1) to protect clams from bottom dwelling predators such as crabs and carnivorous fishes (e.g. triggerfish) and (2) to retard strong water movement thus minimising disturbance to the clams.

Materials for clam cages depend on the raw materials locally available, i.e., nets (made of polyethylene, nylon or nylon netting material), bamboo, *Leucaena* (ipil-ipil) wood, PVC pipes and others. Cages should be designed and positioned with the least possible resistance to currents to ensure durability. This could be done by lowering cage height (approximately 0.3 m), facing the part of the cage with least surface area to the direction of the oncoming current, positioning the cage behind big coral heads, firmly pegging the cage to the bottom, or creating an artificial barrier.

Experience at SUML, UPMSI and at Micronesian Mariculture Development Centre (MMDC) ocean nurseries has shown that cages have to be regularly cleaned of fouling algae and invertebrates which shade the clams and overly restrict water movements. This can be done by manually scrubbing the cages or replacing and drying them. In intertidal nurseries the cages are regularly exposed during low tides, cutting down algal growth and invertebrate colonisation. The use of antifouling paints on meshes is not recommended as the paint peels off too easily to be effective and is sometimes toxic to the clams.

Below are suggested types of benthic cages and their specific requirements:

Bamboo slat cage

Bamboo slats, such as those used in making the traditional fish trap, can be used for cages (Figure 2.5).

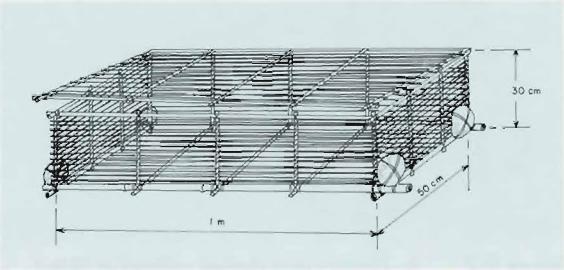


Figure 2.5 Bamboo slat cage

Net and bamboo or Leucaena *cage*

A bamboo base, bamboo strips and *Leucaena* trunks can be used as frames for the cage and a net for covering. The net should be doubled on the bottom part of the cage for durability. A fine mesh net

(10–25 mm mesh) should be used to discourage the entrance of the bottom dwelling predators. The upper surface of the cage is covered with a net of larger mesh (25–40 mm mesh) to allow water movement within the cage. Iron bars can replace *Leucaena* or bamboo as frames for clam cages. They may be more expensive than bamboo but are more durable. The size of the cage depends on the size of the clams. Examples of this type of cage are shown in Figure 2.6.

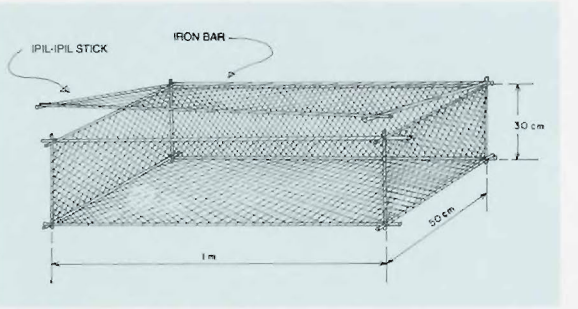


Figure 2.6 Cage made of net and iron bars

Plastic cage

Perhaps the most versatile cages are made from polyethylene like the ones employed at SUML and UPMSI (Figure 2.7A,B). Green polyethylene meshes are available in the Philippines at various mesh sizes (in inches) and 30 x 1 m rolls.

In Silliman, two mesh sizes — 3/4 inch (approximately 20 mm) and 1/2 inch (approximately 12 mm) — were chosen for top and bottom covers, respectively. A 1.5 m length is cut from each roll and a 1 x 0.5 m base is measured from the smaller mesh and a 1.2 x 0.6 m lid from the bigger mesh. Each of the four sides is folded and tied with an electrical cable wire to form a rectangle. Bottom and lids are tied with cable wire at the longer side of the box. The cages are pegged to the bottom using iron bars, with one end hammered to curve back. Iron bars are fitted into the cage fold of the bottom mesh. Two bars plus the added weight of the substrate keep the cage at the bottom.

The plastic cage utilised by UPMSI in Bolinao uses polyvinylchloride (PVC) pipes as frames (Figure 2.7B). PVC pipes have been used since they are more durable than bamboo frames. The case and the lid have the same dimensions and mesh size as at SUML.

The smaller net mesh (10 mm) is used when deploying clams with shell lengths less than 60 mm. These small clams are placed on perforated trays. The bigger mesh (20 mm) is used for clams larger than 60 mm and these clams are placed directly on the cage. The cage is elevated one metre from the bottom and supported by four angle bars to minimise predation from benthic organisms. 'Skirts' made of plastic gallon jars are installed on each post to prevent predation from hermit crabs and other crawling organisms which might prey on the clams (Figure 2.8).

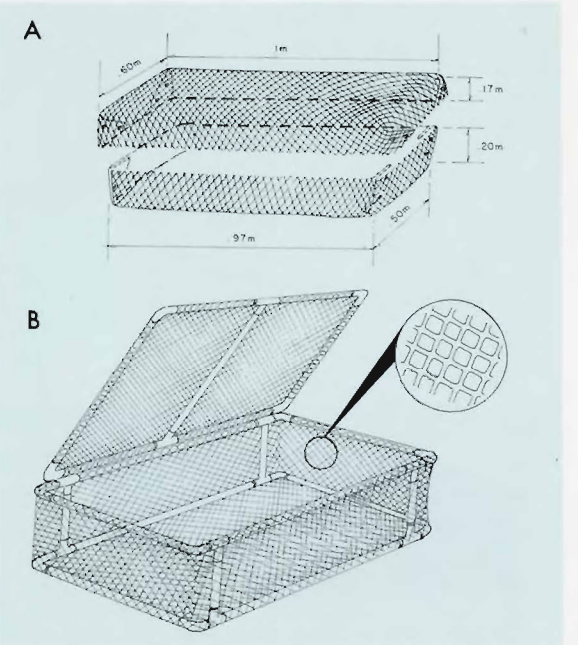


Figure 2.7 A. Plastic cage design utilised at SUML.
B. Plastic cage design utilised at UPMSI

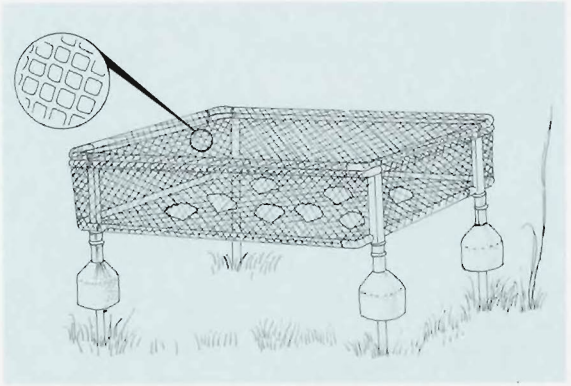


Figure 2.8 Platform design used by UPMSI

PVC-coated wire cages

PVC-coated galvanised wire mesh (14 gauge) is widely used by MMDC (Heslinga et al. 1990). These are made into either square or rectangular cages with two removable parts: top cover and bottom. Figure 2.9 shows an example of a cage made with wire mesh.

Galvanised wire cages

In many countries the cheapest and most easily available cage materials, apart from bamboo or net mentioned above, are a variety of galvanised wire meshes such as 'chicken wire' and various welded wire mesh products. Galvanised wire meshes are used extensively in the Solomon Islands at ICLARM–CAC, in benthic and floating cages (Figure 2.10) and also exclosures.

Advantages over other materials include ease of handling during construction, a good ratio of strength to wire diameter (which means that less surface area is available for bio-fouling) and good bonding between the mesh and cement used for the cage base. The main disadvantage of galvanised meshes is the susceptibility of the wire mesh to corrosion, which limits the life of the mesh to 2–3 years, depending on wire diameter and cage location.

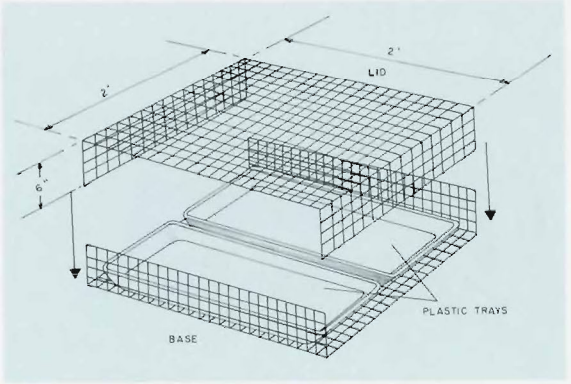


Figure 2.9 PVC-coated wire mesh cage

2.3.2 Platforms

In shifting sandy-seagrass areas as in Bolinao, Pangasinan, Philippines, the clams on the substrate become buried by mounds of sand produced by burrowing organisms. To prevent this, platforms elevated 200–300 mm from the bottom (Figure 2.8) are used. The platforms are made of the same materials as the PVC cages.

2.3.3 Enclosures

Enclosures, fences or pens (Figure 2.11) can be used as protection instead of cages or platforms if the ocean nursery area has most of the requirements mentioned for site selection. Clams do not have to be

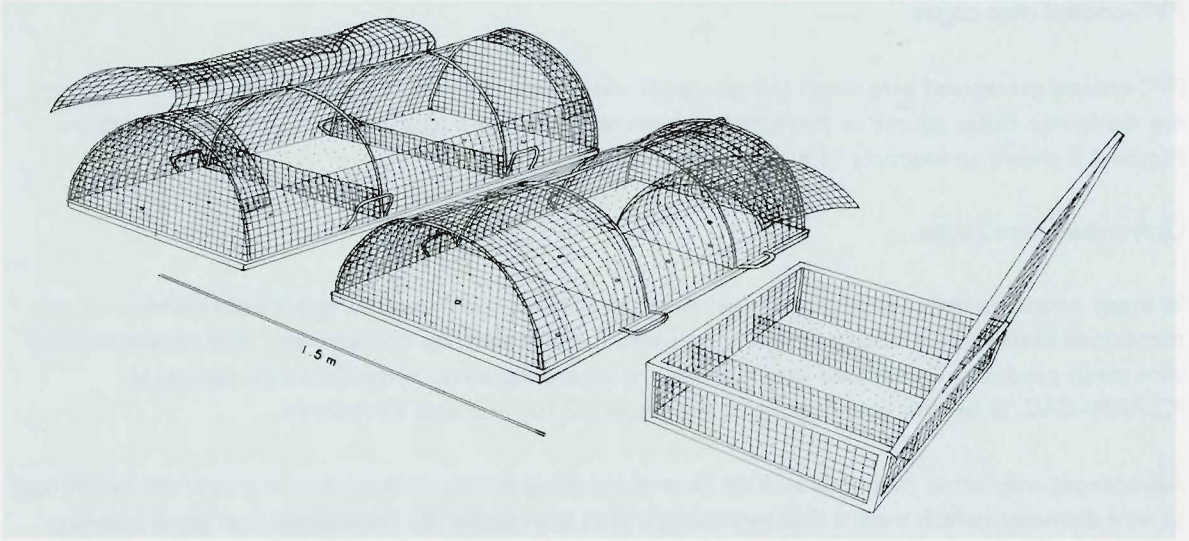


Figure 2.10 Types of galvanised wire cages

caged but rather placed inside a net enclosure held in place by bamboo or wooden (*Leucaena*) posts. The situation is almost similar to the growout phase except for the presence of the net enclosures.

2.3.3 Exclosures

An exclosure is essentially a plastic mesh fence anchored to the substrate and floated from the surface (Figure 2.12A,B). This is slightly different from an enclosure, which does not have floats. The tidal height above the point of the ocean nursery needs to be determined prior to construction so that the exclosure will be taut at high tide but not sink the floats. Once the height above the substrate has been established another 200 mm needs to be added to use as the skirt around the bottom.

For ease, most of the initial construction is best done on land. The netting is prepared in two sections, each 31 m long (a 15 m square unit plus some overlap). The oyster mesh comes in 2 m wide rolls. If

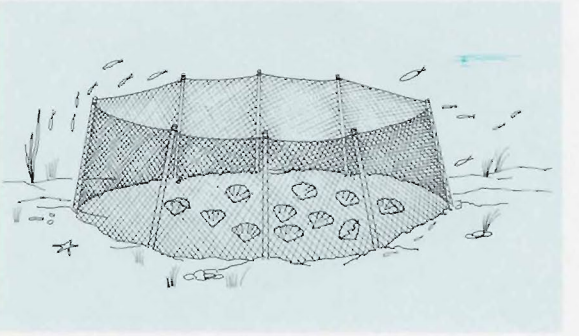


Figure 2.11 Clam enclosure

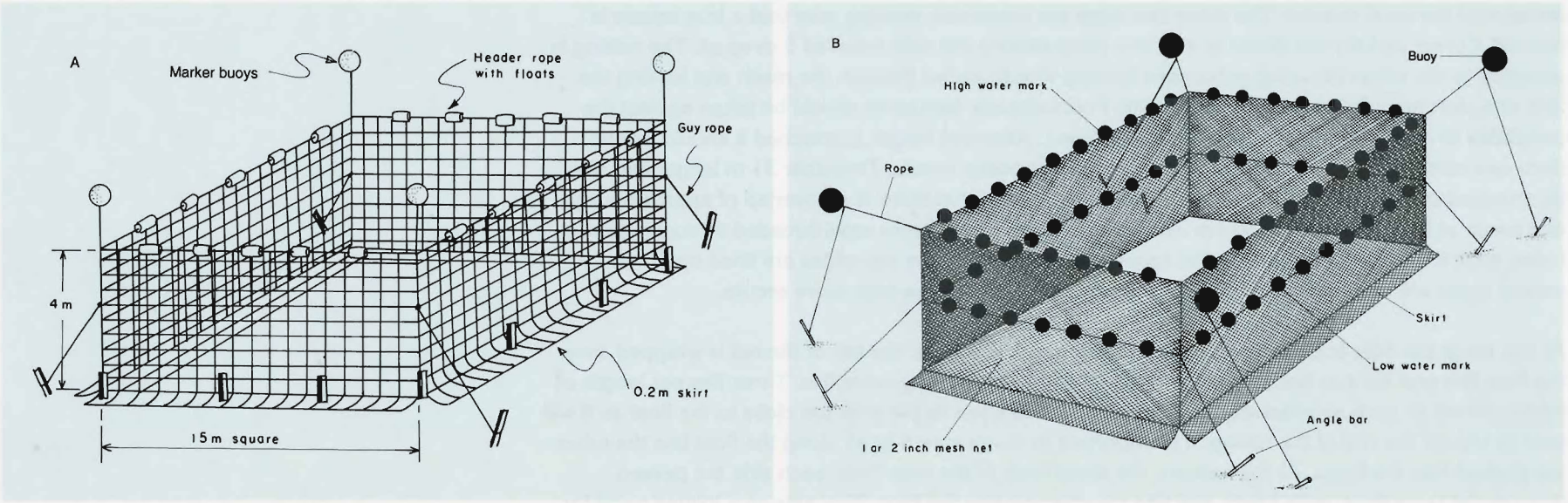


Figure 2.12 A. Exclosure for subtidal ocean nursery – JCU design. B. Exclosure for subtidal ocean nursery utilised at UPMSI.

two widths must be joined this can be done using 3 mm rope woven through every second hole. The mesh should lie flat along the full length, with the ends securely tied. Two 8 mm header ropes can be threaded along the top edge. One rope is positioned close to the edge and the other about 120 mm below it. These are used to attach the float line to the net.

To prepare the float line, a 250 mm ball float with a central hole is used at each comer with 150 mm balls along each side. Each float is separated by a plastic sleeve such as rural grade black polypipe. The length of each piece of tubing is the same, the two end ones are slightly shorter (to accommodate the extra diameter of the end float). Each 15 m side can be made separately and joined after attachment to the net.

During a good low tide, the area for the exclosure should be marked. Two pickets 15 m apart are

driven into the coral or sand. The other two sides are measured, making sure that a true square is formed. Corner pickets are driven in with two more pickets per side installed 5 m apart. The netting is attached to the substrate using galvanised fencing wire threaded through the mesh and leaving the 200 mm skirt lying flat outside the exclosure. Precautionary measures should be taken against the possibility of electrolysis (see Section 2.2.1 on lines). After one length is attached it should extend from one corner along two sides to just beyond the opposite corner. The other 31 m length can also be attached starting where the overlap occurs. This means that there is an overlap of approximately one metre at both joints. The corners can now be joined using 3 mm rope threaded through alternate holes, such that the two meshes lie flat against each other and the top edges are lined up. If two joining ropes are threaded, separated by a small gap, it makes the joint more secure.

At this stage the float line can be attached. Starting at one corner, the top of the net is wrapped over the float line and the two header ropes are attached together using cable ties. Three ties per length of tubing seems to work satisfactorily. Care should be taken not to put a tie too close to the float as it will tend to slip off the end of the tubing. It is important to make sure that all along the float line the tubes are pushed into the floats. At the comers, the loose ends of the rope from each side are passed through the large float, pulled tight and tied together outside the float. This acts as a joining point for the guy rope. After the float lines are in place, the loose ends of the rope are trimmed and sealed. The guy ropes are attached to the corners and tied to a picket placed diagonally off the comer. The guy rope must be long enough not to drag the corner down at the highest tide. If 3 mm rope is tied at substrate height every 3 m to form a grid pattem inside the exclosure the clams can be monitored more easily in 25 squares marked out.

The skirt can have coral boulders laid along it to prevent any benthic predators from crawling under it. It is important to make sure that all the ends are tied off, the ties are all tight and that there are no gaps where the netting has been joined together.

The problem with this design is that during low tide, the net slackens and covers the clam at the periphery, causing shading. A modification of this design is employed in Bolinao (UPMSI) where another row of buoys is added at the height of the low tide level (see Figure 2.12B). With this design, the portion of the net below the low water mark is still upright even during low tides.

Chapter 3

Ocean nursery culture of clams

3.1 Substrates

All clams possess byssal threads for attachment during their early juvenile stages. In later stages, these are retained in some species (*T. squamosa*, *T. maxima*, *T. crocea*) for anchoring to the bottom. The other clam species mainly rely on their shell weight to hold them in place on the substrate.

In the ocean nursery, substrates are used for the attachment of the byssal threads and to prevent the clams from being moved by water currents. Substrates for byssal attachment can be dead pieces of coral, limestone, coral rubble, stones and cement.

Juvenile clams are allowed to attach to substrates in the land nursery before transfer to the ocean nursery. The attachment of clams can also be delayed until placement inside cages at the ocean nursery. However, transfer should only be attempted during calm weather. Clams should be checked daily for possible displacement until attachment is strong enough to withstand currents.

Large pieces of coral rubble and stones may be placed between the juvenile clams in areas of strong water movements to prevent them from being washed about.

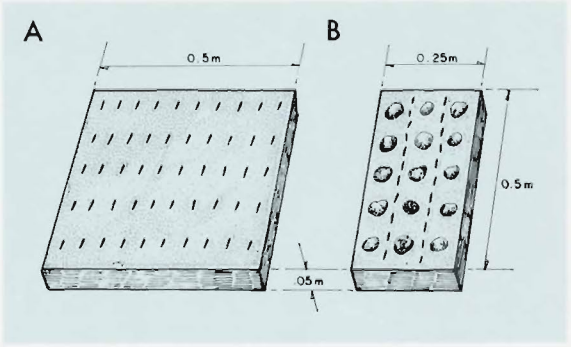


Figure 3.1 A. Concrete block; B. Block with embedded stones

3.2.1 Gravel

Gravel 20–30 mm in diameter can be used for substrate. It is poured or shovelled into the cages or cells to a depth of 50 mm.

3.2.2 Concrete blocks

Concrete blocks (Figure 3.1) offer juvenile clams substrates with no crevices for predators to hide in. Block size depends on the size of cages — a rule of thumb is easy handling and transport. The design

introduced to SUML was 0.5 m x 0.5 m x 3 cm supported by a wire mesh frame embedded in the cement, The surface of each block was scratched with 50 1-mm lines, using a common nail to create lines for clam attachment in the otherwise smooth surface. Juvenile *T. crocea* and *T. maxima* were placed in these and allowed to attach in the land-based nursery before transfer to the ocean nursery.

This was modified at SUML for other species cultured at the laboratory. One modification was making criss-crossing scratches on the blocks. Another modification was embedding gravel (20-30 mm) in the concrete 50 mm apart to create more surface area for clam attachment. The embedded gravel also provides wedges for the juvenile clams with weak byssal attachment. Smaller sizes (5-10 mm) of stones and rubble were also used to cover the entire surface of the block. Smaller areas for attachments are available and clams appear to attach more readily.

Block sizes were designed to fit 2 blocks per 1 m x 0.5 m cage used by SUML. Smaller blocks, however, are easier to handle underwater and fit into cages.

3.2.3 Wire-reinforced concrete

Most cage bases at ICLARM-CAC are constructed of wire-reinforced concrete. A frame for the base is constructed out of appropriate steel or galvanised material such as angle bar. Galvanised chicken wire mesh is stretched taut as possible across the frame and covered with a thin layer of concrete (about 10 mm thick). This technique offers a considerable reduction in weight compared to other concrete designs, allowing larger cages to be made. These bases are commonly used in floating systems at the ICLARM-CAC. Typical sizes of base used at CAC are 1.5 m x 0.75 m and 1.0 m x 0.75 m. In the case of cages made from welded wire mesh folded into the shape of a basket or box, the base of the cage provides the reinforcing as long as cage size does not exceed 1.0 m x 0.75 m.

3.2 Estimating numbers of juvenile clams

Juvenile clams are collected from the nursery tanks by cutting the byssal attachments with a sharp knife. The byssal threads should be cut cleanly and the clam should not be pulled. The clams are then

siphoned out of the tank into an appropriate sized screen and then into a tray. Five to ten piles of 100 clams each, randomly counted, are made. A pile is added to a graduated cylinder with a small amount of seawater and the increase in the volume of water is recorded. The average volume from 5–10 piles is calculated.

For example, for 3500 clams per ocean nursery container:

5.1 mL seawater displaced per 100 clams added (average of 10 measurements)

i.e. 51 mL = 1000 clams

Therefore, for 3500 clams:

51 × 3.5 = 178.5 mL.

Measure clams by starting with a known volume of seawater in a graduated cylinder (say 20 mL) and add clams to 198.5 mL.

3.3 Stocking with juvenile clams

3.3.1 Stocking density

For an intertidal box or cell (of a line), stocking density of about 500 per square metre is recommended. Keeping the density reasonably high helps to control the growth of algae a little, but chains must be monitored closely for the possibility of overcrowding. If more than 75% of the substrate is covered by clams then they must be thinned out because their growth rate will be inhibited. When the clams are about two years old then 30–45 per square metre is the best number for a cell. It is important that if the clams have a seasonal growth pattern they should be thinned prior to summer to allow them to have uninhibited growth during their fastest growth period.

For a cage size of 1 m in length × 0.5 m in width × 0.3 m in height, approximately 100 juveniles (30–50 mm) can be stocked. For bigger juveniles (70–80 mm), fewer clams should be stocked

(30 individuals) for the same cage dimensions. Once the clams start to appear crowded, they should be thinned out (i.e., some clams should be transferred to another cage).

In the exclosure each 3 metre square grid can easily take 90 two year-old clams. Up to 120 can be put into the same area but these will have to be thinned at a later date. It they are left in the exclosure until three years of age they can be moved directly into the growth areas. The alternative is to remove the exclosure leaving the clams in position and setting up the exclosure somewhere else.

3.3.2 Sources of juveniles

Hatcheries that are actively producing juveniles are listed in Box 3.1.

3.4 Transport

For short distances requiring 2–5 hours of travel juveniles must be wrapped in moist cheesecloth or newspaper and placed inside insulated boxes to prevent desiccation and jarring of clams. If transport exceeds 5 hours, clams wrapped in moist cloth need to be placed in plastic bags to which pure oxygen has been added. Pure oxygen can be procured from oxy-acetylene outlets.

At the ocean nursery site tanks should be set up before the arrival of the juvenile clams. If clams are placed inside plastic bags, these should be lowered into the sea water to allow the clams to adjust to ambient temperature. The juveniles are then unwrapped and sprinkled with sea water. When thoroughly wet, they are then transferred to the nursery tanks.

3.5 Measurements of juveniles

It is desirable to keep records of the growth and survival of batches of juvenile clams at least 3–4 times a year in order to discern seasonal differences and the general growth rate to be expected in future batches. The equipment required is basic: a vernier caliper (200+ mm length), made of good

Box 3.1 *Sources of juvenile giant clams*

Species	Location
T. crocea	Australia: Reefarm Hatcheries at Cairns Palau: MMDC at Koror Philippines: SUML at Dumaguete UPMS at Bolinao
T. derasa	Palua: MMDC at Koror Philippines: SUML at Dumaguete
T. gigas	Australia: Reefarm at Cairns Marshall Islands: Clamfarm at Wa'u I. Solomon Islands: CAC at Honiara
T. maxima	Fiji: at Makogai Philippines: SUML at Dumaguete Tonga
T. squamosa	Fiji: At Makogai Philippines: SUML at Dumaguete
H. hippopus	Australia: Reefarm at Cairns Palau: MMDC at Koror Philippines: SUML at Dumaguete Solomon I.: CAC at Honiara
H. porcellanus	Palau: MMDC at Koror Philippines: SUML at Dumaguete

plastic or stainless steel for shell length measurement and a balance that can weigh clams up to 2–3 kg. A measuring board can also be constructed such as is used in measuring standard fish lengths.

A randomly chosen group of 50–100 juveniles which are about 40+ mm shell length may be individually tagged with plastic Dymotag numbers and two-part epoxy glue (Epigen 503 or Aquatopoxy) onto a cleaned and dried area of their shell. These tagged clams can then be placed in the nursery and regularly measured. Individual tagging is superior to taking a random 50–100 clams at each measurement because individual growth increment increased can be followed and the same individuals are measured each time.

When weighing clams, the animal should be inverted on a paper towel for about 10–15 minutes before weights are taken. If the clams are sensitive to drying (especially in the case of small juveniles) the easiest and probably most accurate method is to weigh the clams with mantle cavity water, remove the clam and then get the weight of just the mantle cavity water. The weight of the clam is the difference between total weight and weight of water.

3.6 Records

Records of clam seed, including age, mean size, spawning date and other data should be kept on file. This information is usually indicated in the receipt given by the hatchery.

After the ocean nursery has been operating for a while it will be necessary to move clams into different areas to allow for their increase in size. If accurate records are not kept the age classes will be confused. It pays to establish a program so that age classes are kept in the same general area. If it is important to keep separate spawnings distinct, then it is vital to know where they were originally placed.

If a stocktake (keeping a record of the total stock by counting and recording every clam in each area) is carried out every six months then an accurate account of the number of clams for any one age

group can be ascertained. This enables the farmer to determine if there have been any major losses that have not been accounted for during general monitoring. Also total average weights can be worked out for each age group and for the total farm for determining production rates.

3.7 Monitoring

Monitoring activities of the ocean nursery include the following:

- Cleaning of cages by brushing off algae and other organisms growing inside and outside the cages. This should be done every few days to prevent dense algal growth which will be harder to remove later;
- Removal of predators (see section on predator control in Chapter 5);
- Removal of dead shells and examination of the shells for probable causes of death;
- Thinning out of clams inside cages when they become overcrowded;
- Checking for cage tears, breakage and destruction of anchors;
- Checking of clams after storms, strong wave action,and other occurrences which may affect the clams;
- Stock counting (stocktake); and
- Measuring growth.

Growout phase

4.1 Transfer to growout site

Once the juveniles reach a certain size (about 200 mm) or age where they are presumed able to withstand predator attacks and environmental stresses they are ready for the growout phase of culture. The protective mesh containers or the enclosures in which they have been growing are taken away and these more mature juveniles can be left to grow out to the desired age for harvest (Figure 4.1). Table 4.1 summarises the data gathered from several places indicating the best size or age to transfer clams to growout area.

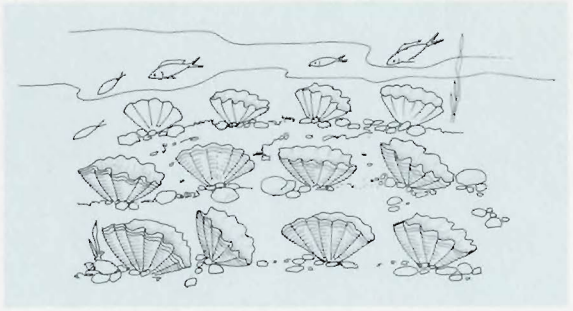


Figure 4.1 Clams during the grow out phase

Table 4.1 Age and size of transfer of different species of giant clams to growout areas

Species	Age (year)	Size (mm)	Ocean nursery	Source site
T. gigas	3	220	intertidal	JCU–OIRS ICLARM–CAC
T. derasa	2.5	180	subtidal	MMDC
T. squamosa	2	150	subtidal	SUML
H. hippopus	3	150	subtidal	SUML
H. porcellanus	3	150	subtidal	SUML
T. maxima	no data			
T. crocea	no data			

4.2 Site selection for growout

See Section 2.1 on site selection for ocean nurseries. The sites may either be adjacent to the ocean nursery or located some distance away from the nursery.

4.3 Sources of juveniles

Juveniles for the growout phase are those from the ocean nursery which have reached 'escape size' (about 200 mm). If available, they may also be purchased from agencies with ocean nurseries and transported to the growout site.

4.4 Transport

The clams can be transported in several ways. One way is by placing the collected animals on their sides in trays and putting them in a shaded area of the transporting vessel. The clams must be sprayed with clean seawater from time to time. Clams can also be transported by putting them in plastic buckets filled with seawater and aerated. If possible, a wedge (i.e., tyres or wood) must be placed at the base of each clam to support it and prevent excessive jarring during transportation.

If transport involves short land travel, clams should be wrapped in moist cloth and placed in insulated boxes. If travel entails more than 5 hours, the wrapped clams are placed in plastic bags and pure oxygen is added.

4.5 Arrangement of clams in growout area

Clams of the same species should be grouped together and separated from other species. They should be placed sufficiently far apart so that the valves will not touch. Each clam should be able to open fully without its valves touching another clam.

4.6 Monitoring

Growout sites should be visited as often as possible, preferably every few days, to see if there are any clams that have changed position, been preyed upon, or have otherwise died. Mass mortality of clams may indicate the occurrence of a disease, parasites, predators, heavy siltation, or other adverse environmental conditions.

Chapter 5

Predators and predator control

5.1 Predators and pests

Most mortality of juvenile clams in a well-sited ocean nursery will probably be due to predators and pests. The success of the whole clam farming operation may well depend on the early detection and adequate control of these predators.

During the ocean nursery phase, juvenile clams are protected by cages from the majority of large and voracious predators. Nevertheless, there are some predators that are not always excluded by the mesh and can kill many clams. These are listed in Table 5.1. The main predators or pests belong to just a few groups and these, together with methods for their control, are described in the following sections.

5.1.1 Ranellidae (= Cymatiidae)

Members of this sea snail family (which until recently was known as the Cymatiidae) are voracious carnivores found throughout the tropics. They occur at almost all giant clam ocean nurseries in the Pacific. Ranellids cause mass mortalities of ocean nursery clams and are probably the most serious biological constraint to the ocean nursery culture of giant clams.

The species most commonly found attacking juvenile clams is *Cymatium muricinum* as reported by Perron et al. (1985) but *C. aquatile*, *C. pileare* and *C. nicobaricum* are also found and appear to attack in the same way. All four species are shown in Figure 5.1. *Cymatium vespaceum* has also been observed consuming clams and may prey in the same way as the previous four species (Perio and Belda 1988). Juvenile *Cymatium* sp. are frequently found in cages for juvenile clams and are

Table 5.1 List of animals known to prey on giant clams

Family	Species
FLATWORMS	
Stylochidae	*Stylochus* sp.
SEASNAILS (GASTROPODS)	
Buccinidae (whelks)	*Cantharus fumosus*
Costellariidae (mitres)	*Vexillum plicarium, V. cruentatum*?
Ranellidae (tritons)	*Cymatium aquatile, C. muricinum, C. nicobaricum, C. pileare, C. vespaceum*?
Fasciolariidae (tulip shells)	*Pleuroploca trapezium*
Muricidae (murexes)	*Chicoreus brunneus, C. microphyllum, C. ramosus, Cronia fiscella, C. margariticola, C. ochrostoma, Morula granulata, Thais aculeata*
Pyramidellidae	*Tathrella iredalei, Turbonilla* sp.
Volutidae (volutes)	*Melo* sp.
OCTOPUS	
Octopodidae	*Octopus* spp.
CRUSTACEA	
Diogenidae (hermit crabs)	*Dardanus deformis, D. lagopodes, D. pedunculatus.*
Gonodactylidae (mantis shrimps)	*Gonodactylus* sp.
Portunidae (swimming crabs)	*Thalamita* spp.
Xanthidae (stone crabs)	*Atergatis* spp., *Carpilius convexus, C. maculatus, Demania alcalai, Leptodius sanguineus, Lophozozymus pictor, Zosimus aeneus.*
FISH	
Balistidae (triggerfish)	*Balistapus undulatus, Balistoides* sp., *Pseudobalistes* sp., *Rhinecanthus* sp.
Lethrinidae (emperors)	*Monotaxis grandoculis*
Labridae (wrasse)	*Choerodon* spp., *Cheilinus* sp., *Halichoeres* sp.
Tetraodontidae (pufferfish)	*Canthigaster valentini, Tetradon stellatus.*
RAYS	
Myliobatidae (eagle rays)	*Aetobatis narinari.*

Figure 5.1 Growth series of the four species of Ranellidae most commonly found preying on juvenile tridacnid Clams. From top to bottom: *Cymatium muricinum*, *C. aquatile*, *C. nicobaricum*, *C. pileare*. Note the different protoconchs, particularly in juvenile shells.

usually distinguished by their thin shells, prominent protoconch (the small larval shell at the top of the juvenile shell) and by their sometimes dense covering of fine hairs.

Adult or subadult *Cymatium* are voracious predators, usually attacking at night and capable of consuming up to 10 juvenile *T. gigas* (30 mm shell length) per week. The snails attack by injecting toxic saliva into the clam through the mantle or byssal orifice, quickly incapacitating even relatively large juvenile clams (up to 200 mm). The dying clam is then rapidly eaten. Ranellids have free-swimming larvae which are capable of drifting in the ocean for months, travelling thousands of kilometres. When a suitable substrate is found the larval snails settle. It appears probable that juvenile giant clams provide such a substrate, inducing settlement. Certainly these snails appear at a size of just a few millimetres within, or close to, juvenile clams in ocean nurseries. The settled snails enter clams at this virtually undetectable size and proceed to feed, sometimes causing blisters on the internal surfaces of the clam's shell. Eventually the juvenile clam succumbs to the feeding ranellid, by which time the snail has grown to a visible size (6–20 mm). The snails then move on to feed on other nearby clams. Because these snails often attack at night and are capable of killing many clams even when very small, they are often not identified as the cause of mortality in ocean nurseries.

5.1.2 Muricidae

Muricid seasnails are well-known pests of cultured bivalves in temperate seas. Muricids are often very abundant in coral and coral rubble substrates. A number of species have been found to prey on juvenile giant clams but have rarely been reported to cause extensive damage (Figure 5.2).

Muricids slightly resemble some members of the Ranellidae, but can be distinguished by their usually more abundant ribs and their lack of any hairy covering. Some muricids such as *Cronia* (*Muricodrupa*) *fiscella* and *Chicoreus brunneus* drill holes through the shells of the juvenile clams through which they feed, while others such as *Chicoreus ramosus*, may attack directly through the gaping valves of the clams. Small clams placed directly on the seabed may be at risk from muricids if the substrate is coral rubble or rock.

5.1.3 Pyramidellidae

Pyramidellids are known parasites of many species of cultured bivalves, including giant clams (Cumming 1988). Two genera are reported to affect giant clams; *Turbonilla*, which has been found in Australia, Philippines, Solomon Islands and Fiji, and *Tathrella*, which has been reported from Palau to Kosrae. Both genera are small (less than 8 mm) and appear white and fragile (Figure 5.3).

Although pyramidellids parasitise all sizes of clams, juvenile clams are more seriously affected. Depending on the number of infesting snails and the size of the juvenile clam, effects range from loss of condition and slower growth to death of the juvenile clam. Usually mortality only occurs after a period of infestation. The infestation may cause the clam to form blisters on the inner surface of the clam shell close to the shell edge or near the byssal orifice. Shell thickening and uneven growth patterns near the inner valve edges may also be present. The mantles of affected clams may be visibly retracted or present dark notch-like scars.

Pyramidellids are found either on the valves of clams, under the clams or on the nearby substrata. Generally they hide in patches of algae or other refuges during the day and come out to feed at night. The snails feed by using their long, flexible snouts to suck the clams' body fluids, either from the edge of the mantle or through the byssal orifice.

These pyramidellids reproduce every 3–4 months, and once a breeding population is established, they can increase their numbers by several orders of magnitude over just a few months.

Figure 5.2 Some species of Muricidae and scavenging organisms commonly found in ocean-nurseries. Clockwise from top left: *Chicoreus brunneus, Cronia (Muricodrupa) fiscella, Pyrene turturina* and *Cronia ochrostoma.*

5.1.4 Flatworms

Some species of turbellarian flatworms are known to be serious pests of cultured oysters and mussels (Littlewood and Marsbe 1990). One species of flatworm belonging to the family Stylochidae has been identified as a serious pest in floating ocean nurseries for *T. gigas* at Solomon Islands (Figure 5.4). These worms are flat and thin and have an irregular shape. Their light-brown to grey colour makes them very difficult to find as they blend in well with clam shells and the cage substrate. They vary in size from a few millimetres to 60 mm in length. Nothing is known about the distribution of the species

Figure 5.3 Pyramidellid parasites (*Turbonilla* sp.) on a juvenile *Tridacna gigas*. Photograph courtesy of John Lucas.

found in Solomon Islands but similar species can be expected throughout the tropics. It is not certain how the stylochid flatworm kills clams, but the flatworm enters the clam either through the byssal orifice or the inhalant siphon, the clam soon dies and appears to be eaten by the flatworm. Flatworms lay their egg masses on the inner surface of the dead shells of clams and other bivalves which may settle on the cage.

Although flatworms are not particularly voracious, they reproduce prolifically and are capable of reaching high densities in clam cages. They then infect nearby cages. High densities of these flatworms are usually associated with increased mortalities of juvenile clams. So far these flatworms have been found only in cages raised off the seabed.

5.1.5 Boring sponges

Some kinds of sponge have been found to bore into the shells of giant clams and other molluscs and can make clams so weak that they either die or are easily killed by predators or diseases. Shell-boring sponges are usually only a problem in older clams which are raised in the ocean where they are resistant to most other pests and predators and therefore not so carefully looked after by the clam farmer.

These sponges are visible on the outer surfaces of the clam's shell as a large number of holes, most obvious on the underside of the clam. The holes vary from 0.5–1.5 mm in diameter and are filled with sponge tissue which may be orange, yellow, green or brown (Thomas 1979). On the inner surfaces of the clam's shell the boring sponge appears as a network of channels of the same colour as the sponge and in seriously infested clams the shell may be blistered or deformed. Other organisms, particularly boring algae and bristle worms may cause similar damage and may be controlled in the same way as boring sponges (Velayudhan 1983).

5.1.6 Other predators

A variety of other animals can prey on clams (Table 5.1). Species of crabs (Families Xanthidae and Portunidae), hermit crabs (*Dardanus* spp.) and fish (such as triggerfish, wrasse and pufferfish) kill

clams by crushing or chipping the shells. Clams are usually protected from these animals by cages or other mesh systems, but predators can sometimes find their way into cages through badly closed covers or even by breaking the mesh, as do stone crabs (Figure 5.5) and triggerfish.

Octopuses are capable of wrenching clams open to eat them. Often empty clam shells from an octopus attack will be found some distance from the cages. Octopuses are able to get into even well closed cages but so far have not presented a major threat to clam farms.

5.2 Predator control

5.2.1 General measures

The most effective way of controlling predation on giant clams is regular and thorough checks of the clam stocks. Because predator occurrence tends to be sporadic or occasional, it is important that the checks be carried out regularly, even though no problems have been detected for a considerable period of time. Checks should be carried out two or three times a week but greater frequency may be necessary if an infestation is detected or a mass mortality of clams has occurred. When clam mortality cannot be attributed to adverse environmental conditions or poor maintenance, dead or dying clams usually indicate recent predator activities. When such clams are found during routine checks, the clams must be thoroughly examined, as well as the empty shells, the cages and the immediate surroundings. Particular attention should be paid to apparently inoffensive, small (3–5 mm) snails. Some predators, like the shell-drilling muricids, may be identified from characteristic markings made on the clam shell that result from the predator's feeding behaviour (Table 5.2). Dying clams should be removed as they may attract more predators or may contain pests that will affect other clams.

Cages should be cleaned regularly. Dead shells, debris, fouling animals and algae should be removed to prevent them serving as refuge or suitable egg-laying substrates for predators. In this respect, the choice of substrate for a cage may be important as gravel and coral rubble may provide such refuge whereas a solid and relatively smooth substrate will not, thereby making predator detection easier. It is advisable to remove all animals from cages except for known grazers such as top shells

Figure 5.4 Turbellarian flatworms (*Stylochus* sp.) from floating ocean-nurseries in Solomon Islands.

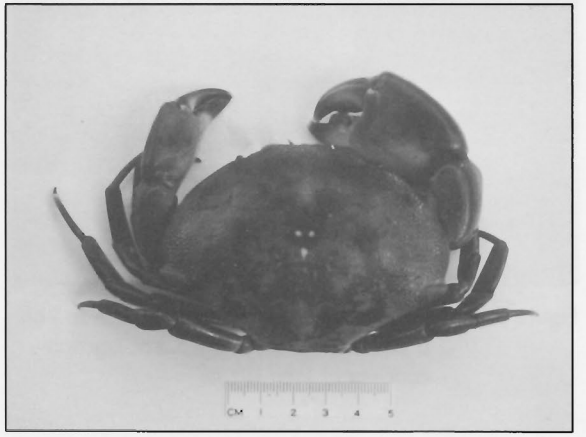


Figure 5.5 *Carpilius convexus*, a xanthid crab capable of preying on juvenile tridacnids.

Table 5.2 Possible causes of predator-related shell damage in juvenile *Tridacna gigas*.

Shell damage	Possible causes
Chipping or crushing	Crabs, hermit crabs, fish
Drilled hole/s	Muricid snails
Blistering on internal surfaces	Pyramidellids Juvenile ranellids
Layering or thickening along valve edge	Pyramidellids
Ligament torn and/or hinge dislocated	Octopus

(*Trochus* spp.) turban shells, and cerithiid gastropods. This is because many of the animals found in cages, although not capable of killing clams, may be scavengers capable of feeding on dying clams (see Section 5.3 on biological control).

These guidelines should be generally applied to control most predators. If severe problems occur with a particular predator, then more specific action is required as outlined in the following sections.

5.2.2 Control of Ranellidae

Regular thorough checks of the cages are essential for the control of these snails. More frequent inspections should be made when infestations of juvenile snails are detected. In rare cases, mortality due to juvenile ranellids settling in clam cages may become unacceptably high, despite intensive efforts at control by the clam farmer. In such cases it may be necessary to relocate the cages, either at a new depth or in a different position with respect to the prevailing currents. In extreme cases, it may be necessary to relocate the whole clam farm.

If a problem is experienced with adult snails entering cages from the surrounding environment it may help to raise the cages above the substrate, using trestles, posts or floating cages.

5.2.3 Control of pyramidellids

Minor infestations can sometimes be controlled by manually removing the parasites. In the case of serious infestations, all the clams from the affected cages should be harvested and scrubbed clean. The cage and substrate should be soaked in bleach and rinsed or at least sun-dried. In situations where such infestations pose a continuous threat, it may be beneficial to take such action on a routine basis (e.g. every three months) in order to prevent infestations recurring. Biological control may also be considered (see Section 5.3).

5.2.4 Control of flatworms

Infestation can largely be prevented by keeping cages clean and free of fouling organisms, thus

depriving the flatworms of refuge and suitable egg-laying substrates. If necessary, clams from affected cages can be harvested and scrubbed and the cages sun-dried. This action can be repeated every 3 months or so if infestations are expected. Serious infestations may be controlled by removing the cage and clams from the sea and briefly spraying the contents with fresh water. This only kills the flatworms as the clams close and do not appear affected by the brief exposure to fresh water.

5.2.5 Control of boring sponges

Infested clams should be kept separate from healthy clams to prevent the infestation from spreading. The sponges can be killed by treating the outer surface of the shell with formalin. The affected areas should be painted with a 1% formalin solution and left out of the water for about one hour after which the shell should be rinsed with fresh water before placing back in the sea. **Caution:** great care should be taken that formalin does not come in contact with the clam's tissues.

5.2.6 Control of crushing predators

Persistent predation by crushing predators may be slowed down or stopped by a variety of methods. Spears may work well for fish, crabs or octopus. The use of tangle nets near the cages may control large crabs, and traps (Figure 5.6) can be used to control some fish and hermit crabs. However, most of these predators should be excluded from clam cages by the use of suitable mesh sizes or by raising the cages from the seabed on posts or trestles. Inverted plastic cones (Figure 2.8) can be placed on these posts to prevent hermit crabs from climbing into the cages.

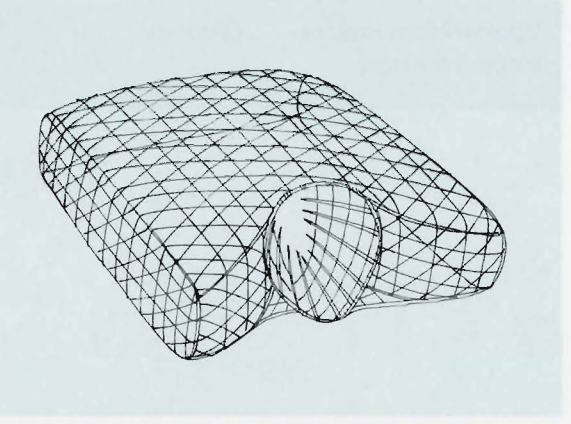


Figure 5.6 Fish trap used at UPMSI to remove some predators.

5.3 Biological control of predators

Certain species of crab, spiny lobster, hermit crab and fish (portunids, palinurids, diogenids and wrasse, respectively) are known to eat pyramidellids and can be expected to consume juvenile ranellids. Unfortunately these same species, if large enough, are also known to eat juvenile clams. Nevertheless, if small species are used or the size of the control agents is rigorously screened there appears to be potential for such biological control.

It has been noted that densities of pyramidellids, juvenile ranellids and flatworms are often lower and, in the case of flatworms, nonexistent in benthic cages. This may be due to the action of benthic organisms feeding on these predators. This suggests that mesh size on benthic cages should be large enough to allow access to such organisms while excluding clam predators.

The species of giant clam being cultured may also affect the level of mortality that can be expected in the ocean nursery. Some clam species may be more resistant to certain predators, or achieve such resistance at an earlier age or smaller size. For instance, *Hippopus hippopus* appears to be more resistant to ranellid and some other predators than *Tridacna gigas* or *T. derasa* (Perron et al. 1985).

Chapter 6

Diseases and parasites of giant clams

While there is a large body of literature on diseases of molluscs, there is little information on the diseases, pathogens and parasites of giant clams. This is probably related to the relatively recent cultivation of the giant clams compared to other molluscs. Except for bacteria and protozoa, no virus, chlamydia, mycoplasma, fungus or neoplasm has been reported in giant clams. The large majority of the deaths in cultured juvenile and adult giant clams have been associated with adverse environmental factors rather than specific infectious pathogens.

6.1 Summary of diseases reported in giant clams

Bacterial disease may be a problem in larval and juvenile stages. Causes include heavily contaminated culture facilities, algae contaminated with bacteria, poor tank hygiene, high nutrient levels in the water and dead clams in the tank. Bacterial disease of juvenile and adult clams is usually secondary to environmental or managerial stress. *Rickettsia*, an intracellular bacterium, may infect clams of all ages but infections may be most heavy in juvenile clams.

The protozoan parasite *Perkinsus* sp., has been seen in both juvenile and adult clams. A *Marteilia*-like protozoan parasite has also been seen in an adult clam. A larval fluke of the metazoan parasite (Bucephalidae) has been found infesting the gonad of adult *T. crocea*. Other parasites have already been discussed in Section 5.1.

In Australia and the the Pacific, winter mortality may cause heavy losses, especially in juvenile clams, where the temperature drops below 20°C. The epithelium of the mantle and gills of *T. gigas* and *T. maxima* may contain banana-shaped, intracellular bacteria of unknown type. In older clams, chilling produces retraction of the mantle followed by secondary infection with bacteria and ciliate protozoa, and finally death.

Heat stress may be a problem for clams kept in small, land-based aquaria or tanks. Prolonged exposure will cause death. Severe heat and cold stress along with insufficient sunlight have been proposed as the causes of bleached mantle in clams, wherein part or all of the siphonal mantle will turn white. Pale siphonal mantles may be associated with a deficiency of nitrogen.

For clams reared in pumped water, gas bubble disease may sometimes cause losses. Air bubbles will appear in the siphonal mantle and small clams may float to the surface. Allowing for temperature and atmospheric gas pressure equilibration of pumped water prior to use will solve this problem.

Excess salinity, i.e. greater than 45 parts per thousand, will cause the clam to close its shell valves during the day. Prolonged low salinity may be associated with swelling of the edges of the siphonal mantle and death.

Details of the life cycles of many of the above infectious agents and parasites are at present unknown.

Because of the presence of the above or other as yet undiscovered infectious agents, the movement (translocation) of clams between farms, regions or countries involves a risk of also moving infectious agents and parasites. Hatchery-reared juvenile clams for translocation between centres therefore need to be disease-free and parasite-free. A quarantine protocol to ensure that the latter is carried out is given in Section 6.3.

6.2 Preparation of specimens for histology

6.2.1 Dissection

The adductor muscle (see Figure 1.1), which holds the clam's shell valves closed, must be cut. This may be done by either (1) anaesthetising the clam, or (2) placing a strong wedge, e.g., of wood, between the dorsal valves while they are open. A long, thin knife or scalpel is slid along the inside surface of the shell valves to cut the adductor and byssal/pedal retractor muscles where they attach to the shell. Once this is done, the tension on the shell valves will be relaxed. The attachment of the siphonal retractor muscles to

the shell valves along the pallial line can then be cut free so that the shell valve can be removed after the hinge ligament is cut or broken by bending the shell valve back and forth.

The above procedure may be repeated for the remaining shell valve so that all the soft tissues of the clam are removed as a unit. The organs of the clams may be dissected out in the following order: the lateral mantle, the siphonal mantle, the gills (ctenidia) and labial palps, the pericardium and heart, the kidneys, the reproductive organ, the digestive organs, the byssal organ and foot, and the adductor muscle.

6.2.2 Fixation

The procedure is as follows: cut sections of each organ or tissue, not more than 0.5 cm thick, using a new scalpel or razor blade. This thickness is important as this is generally the maximum depth into which the fixative will penetrate before the cells deep in the tissue start to degenerate. The area is not as important and a piece of tissue approximately 10 × 10 mm is adequate. If the tissues come from clams 4 months of age or over, place the sections in 10% seawater/formalin (made by measuring 100 mL 40% w/v formaldehyde solution and adding to 900 mL filtered seawater) in a wide-mouthed container for ease of removal when fixed, ensuring that there is 10 times the volume of fixative for each volume of tissue. Tissues from clams under four months of age should be fixed in 3–5% seawater/formalin as the 10% seawater/ formalin will dissolve them.

After the tissues have been in the fixative for 2–3 days, they may be taken out, wrapped in paper or cloth dampened with fixative, and placed in double, strong plastic bags, each sealed with packing tape. The tissues may then be placed in a strong container (e.g., cardboard box) to prevent crushing and mailed or sent by courier to a laboratory for histological processing. Caution should be exercised in handling formalin. Avoid inhaling the fumes or contact with the skin or eyes.

6.3 Quarantine protocol for importing giant clams

The purpose of this protocol (Box 6.1) is to allow larvae or juveniles of giant clams to be translocated

Box 6.1 *Quarantine protocol for country or agency importing giant clams.*

1. Import clams as young as possible (larvae or young juveniles) to reduce the risk of a carrier status.

2. Use a separate batch of facilities (pumps, tanks, etc.) for each batch of clams imported. Identify all these facilities as such in an area where access is restricted to trained, authorised staff only.

3. Each set of facilities must be thoroughly cleaned, disinfected, rinsed and dried prior to use. This is done by subjecting them to a thorough mechanical cleaning with 1% sodium hydroxide solution (10 g NaOH in 1 litre water) using a brush preferably with a pressure spraying machine at a minimum temperature of 60°C. This is followed by soaking for 1 hr in a disinfectant, a chlorine solution (60 mg/L free chlorine) or sodium hypochlorite solution (sufficient amount is added to water so that a residual chlorine concentration of at least 10 mg/L remains after 30 min). After soaking, the facilities are rinsed with 1 µm filtered seawater or freshwater and allowed to dry.

4. All other animals including top shells, fish, etc. must be excluded from the quarantine area.

5. The clams must be maintained in 1 µm filtered seawater and fed with laboratory-cultured algae or artificial diets. If this is not possible, 10–20 µm filtered seawater may be used to allow the entry of phytoplankton. However, this also allows the entry of infective agents (e.g., zoospores of *Perkinsus* sp. which are 4–5 µm in size) and may confuse the issue as to the origin of any pathogen which might appear in the quarantined clams.

6. Filters should be cleaned at least every second day following the protocol in No. 3. Deteriorating filters must be discarded.

7. The outflow water from the quarantine ponds should be disposed of into a land-based sump (e.g., sand trench) to prevent the spread into the local environment of any pathogen or noxious agent that may be present in the imported clams.

8. A daily log (record) should be kept of each batch and should include the following details: cleaning, nutrient applications, any abnormalities, mortalities, water quality, filter changes, water exchange rates, water temperature and weather conditions (e.g., sunshine/cloud/rain).

9. Each batch of clams should be monitored for a minimum of 3 months.

10. Random samples should be collected for laboratory examination at 6 weeks and 12 weeks after the time of arrival of the clams into the importing country or agency. The minimum sample size to detect a 2% prevalence of each infection (with 95% confidence), assuming that the test is 100% reliable, is as follows:

Batch size	Sample size
1 000	140
10 000	145
100 000 or more	150

Each sampling should include gross examination, *Perkinsus* sp. culture and histopathology (preserved in buffered 3–5% seawater formalin).

11. All stress should be minimised to reduce sickness or death which may mask the presence of a pathogen or parasite.

12. A separate set of equipment must be used for each tank/pond (e.g., brushes, nets, etc.). Staff should disinfect their legs and feet with 60 mg/L free chlorine solution before entry into rearing ponds/tanks.

Clams are deemed unsuitable **for release into the local marine environment and should be destroyed if the following are found:**

- Any lesions, e.g. inclusion bodies, or focal necrosis which may indicate the presence of a virus
- Any bacteria, protozoan, metazoan parasite or fungus associated with an inflammatory or degenerative lesion or known to be a pathogen for other marine animals
- Any *Rickettsia* or *Chlamydia*
- Any unexploined lesion
- Any unexplained mortality.

from one region to another with minimal risk of introducing pathogens or noxious agents. Quarantine protocols must be practiced both by the importing country or agency as well as the exporting country or agency. The protocol given in Box 6.1 is that which should be followed by the importing country or agency. For information about the exporting country or agency see Braley (1992).

Chapter 7

Economics and markets

Giant clam farming is not yet a well-established industry but a few firms have already embarked on the commercial production and culture of giant clams. Because this industry is in the early stages of development, there has been little time for commercial data to accumulate. Markets and production techniques are still in the development stage and firms entering the industry have considerable scope for learning by experimentation and by experience. Success in the giant clam industry depends heavily on entrepreneurship, especially in the establishment of markets, and so capital invested in the industry at present should be regarded as venture capital. Nevertheless, effective production techniques have been established and profitable possibilities exist.

7.1 Markets

The main end-markets which have been identified for giant clams are: food (meat), shell and aquarium.

7.1.1 Meat

The main potential market for giant clams is for their meat (whole meat, adductor muscle and mantle) but much must be done to develop this market in more developed countries and to re-establish it where it has 'withered' due to insufficient supply of giant clams from natural stocks. In developed countries, the main current market for giant clam meat prefers *T. crocea*, and to some extent, *T. maxima* in the southern islands of Japan. Currently, it seems that Okinawans consider the meat from other species of giant clams to be a poor to unsatisfactory substitute for meat from these species. No market for giant clam meat has yet been established on the main islands of Japan. Currently, only small remnant markets seem to exist for giant clam meat in Taiwan and Hongkong, and then only for the adductor muscle, especially that of *T. gigas* and *T. derasa*. Substantial potential

markets for giant clam meat exist in New Zealand and Australia among Pacific Islander immigrants. Scope exists in Australia for catering to the tourists. The number of Japanese tourists to Australia is expanding rapidly. Although Japanese mainlanders do not usually know of giant clam dishes, they do try such dishes when they visit the Ryukyu Islands. A substantial ethnic market for giant clam meat would also exist in the USA, especially California and to some extent Hawaii because a large number of Japanese and Pacific Islanders have migrated there. In Hawaii, the tourist trade represents a worthwhile potential market as it does in Guam.

7.1.2 Shells

The market for giant clam shell is well established despite restrictions on international trade in giant clams or their products by CITES (the Convention on International Trade in Endangered Species). However, the shells of some species (*T. gigas* and *Hippopus porcellanus*), are now in very short supply. Overall, the shells of *H. hippopus* seem to be most in demand with *T. squamosa* also of considerable importance in the shell trade (Juinio et al. 1987). It should be noted that some species which are in considerable demand for their meat, e.g., *T. crocea* has little value for its shell. A commercial enterprise needs to consider such factors in deciding which species to farm.

7.1.3 Aquarium

An active market exists for giant clams in developed countries as saltwater aquarium specimens. Maricultured clams are being supplied to the market by MMDC (Palau) and Reefarm at Cairns (Australia). While the size of this market is limited, it represents a worthwhile sideline outlet for a giant clam farm. It is furthermore a working market which can be expected to improve in operations as supplies from mariculture become more regularly available and can be better adapted to customer needs. The main species in demand for this trade are *T. crocea* and *T. maxima* because of their colourful mantles and their small size.

7.1.4 Biological specimens

A small market may also exist for giant clams as biological specimens for scientific demonstration

and experimentation purposes. It is possible that additional end-uses may be found, e.g., extracts from the clam for health or medicinal purposes. Particular attention should be given to the development of high value products from clams. For example, the inside of the shell of some giant clams species can be used for scrimshaw as was done by the ancient Babylonians. Specimens of such beautiful scrimshaw can be seen in the British Museum.

Each of the end-markets may be subdivided into sub-markets. For example, separate markets exist for meat for sushi, for the muscle, for the mantle and the whole meat of the clam minus the kidneys. Species differ in their ability to satisfy these different markets and this should be taken into account in deciding which species to breed and rear and whether to establish a hatchery and nursery aside from a commercial ocean nursery and grow-out.

7.2 Whether to establish a hatchery and nursery

If the business is not already operating a hatchery-nursery, it will need to make a decision about whether to purchase juveniles or to produce them itself. It will have to compare the landed cost of juveniles with the cost of producing them. If only relatively small quantities are required for growing on, it is most economic to purchase these rather than produce these on the farm.

Economies of scope, apart from economies of scale in hatchery-nursery production, appear to exist in giant clam farming. This means that there may be economies involved in multiple or a wider range of activities, e.g., hatchery-nursery operations as well as ocean nursery. For example, if a truck or boats have been purchased for hatchery-nursery, they may be used for associated ocean nursery operations if these occur near the hatchery-nursery. As a result, some economies in capital costs can be achieved. Furthermore, labour employed in hatchery-nursery operations can be jointly used in ocean nursery and growout operations, including surveillance when onshore and ocean nursery operations are located near one another.

7.3 Production decisions

Nursery culture may be achieved in the ocean or kept on land in suitable tanks, containers or ponds. Possibilities for nursery culture on land may be limited by available space, pumping costs of seawater and capital costs involved in pond or container construction. The advantage of land-based nursery culture can be easier protection of clams from predators and diseases. If ocean nursery culture is only for a short period, say until the clams are 2–3 years old, to satisfy aquarium, sashimi or sushi markets, it may be economic. If US$ 2–3 is obtained for 2–3 year-old clams this method could be profitable given a sufficiently large volume of operations (500 000 clams per year).

The altemative is to transfer clams to the ocean at about 10 months old, first of all keeping them protected and subsequently when they reach escape size growing them out unprotected in the ocean. Depending upon the species being cultivated, growout may be subtidal or intertidal. Each has its own advantages and disadvantages and is species specific. Choice of species to farm will be affected by the sites available and by market considerations.

Tridacna derasa and *T. gigas* may be optimally held in the ocean until they are around five years old. This strategy might be expected to maximise the volume of meat production per unit. But in the initial phase of operations this may result in a cash flow problem. The time factor complicates the modelling of the economics of multiperiod production. The longer money is tied up in production the greater the costs involved in terms of interest forgone. Economists allow for this by discounting, for example, by using net present value analysis, or by estimating intemal rates of return on funds employed. This analysis has been applied to a worked example involving the ocean growout of *T. gigas*.

Using the techniques developed in Australia (James Cook University-Orpheus Island Research Station), the following is an assessment of the ocean culture enterprise independent of hatchery–nursery operations. Allowances were made for equipment such as utility truck, workers' accommodation and cost of exclosures and lines. The main operating costs were the cost of seed (assumed to be $0.75 each) and labour.

Assuming that 100 000 clams were placed in the ocean annually, 10 years of growout were found to maximise the firm's net present value, using a 10% rate of interest as a discount factor and considering prices of clam meat of $3–7/kg . Net present value is substantial after 10 years of ocean culture even at on-farm price of as low as $3/kg clam meat. Furthermore, at a lower price per kg for clam meat, a positive profit can be expected.

Internal rate of return analysis using the same data and assuming an on-farm price of $5/kg shows the internal return on ocean culture of giant clams for meat to be approximately 19.5%. These estimates allow for mortality rates for giant clams considered to be realistic by JCU-OIRS.

In practice, adductor muscle and mantle of giant clams may be sold separately. In this case the prices considered above should be regarded as average prices, taking into account the fact that adductor muscle represents about 15% of the whole meat weight. For example, if adductor muscle is paid for at $30/kg on the farm, this would account for a value on average of $4.50/kg whole clam meat. If the price received on the farm for adductor muscle is as low as $20/kg it will account for $3/kg of whole clam meat.

Using these average prices, specific allowance can also be made for loss of weight in processing and transit. It has been shown that up to 40% of the weight of clam may be lost due to 'drip loss' (Parry et al. 1991). This represents a substantial loss. But it is conceivable that methods to reduce drip loss could be developed. Nevertheless, after such weight loss, if giant clam meat sells on the average at $12/kg retail, clam farming may still be economic. Assuming a 100% mark up between clams on the farm and retailing, such a loss would still result in $3.60/kg for the grower of clam meat.

The above example does not allow for possible additional revenue that may be earned by a farm from sales of shell. Also it has been assumed that no transportation costs and/or preparation costs fall on the farmer, or that these are not substantial for the farmer. In any case, it may be most economic to prepare clam meat in central food processing factories, e.g., freezing plants, with minimal preparation being done by farmers. This, for example, used to be the pattern in Fiji using native clam stocks. High valued clam muscle was frozen and exported and the mantle was sold locally in blister packs through retail outlets. The Fijian processor was also involved in preparing and marketing other frozen food items.

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Appendix

Recorded growth rates of giant clams

Growth of five species of giant clams during ocean nursery and growout culture at various locations.

Species	Locality	Growth rate (mm/month)
Tridacna squamosa	San Juan, Siquijor	4.5
	Dumaguete City, Negros Oriental	2.8
	Silaqui Island, Pangasinan	2.3
	Papua New Guinea	6.0
Hippopus hippopus	Apo Island, Negros Oriental	5.3
	Bindoy, Negros Oriental	2.9
	Dumaguete City, Negros Oriental	2.6
	Carbin Cay, Sagay, Negros Occidental	1.3
	Balicasag Island, Bohol	3.8
	Pamilacan Island, Bohol	3.6
	Danajon Bank, Bohol	1.3
H. hippopus (cont'd)	Silaqui Island, Pangasinan	3.6
	Santiago Island, Pangasinan	0.5
Tridacna derasa	Apo Island, Negros Oriental	5.3
	Carbin Cay, Sagay, Negros Occidental	5.6
	Silaqui Island, Pangasinan	3.6
Tridacna gigas	Apo Island, Negros Oriental	6.8
	Silaqui Island, Pangasinan	7.0
	Orpheus Island, North Queensland Australia	7.9

Sources: ACIAR 1986; Crawford et al. 1988; Gomez and Belda 1988; Estacion 1988.