AQUACULTURE
School Projects

3rd Edition

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INTRODUCTION

Aims

This book aims to provide schools with simple low cost equipment that can be used in the teaching of Aquaculture projects to secondary school students.

Projects fall into two broad categories:

a. Those that make equipment needed to house and grow animals and plants used in school based aquaculture systems.

b. Practical exercises for a school workprogram that has been written to either the New South Wales - *Marine and Aquaculture Technology* or Queensland *Marine and Aquatic Practices Syllabi* (see pages 116 - 119 for details).

While the first category may be more applicable to teachers, some students may wish to construct equipment for the school or for their own use at home.

The projects provided are examples only, using materials and methods that the authors have found work.

- They should be seen as starting points, to be modified and improved by the imagination of teachers and their students who will inevitably find alternate materials and better methods.

Permits, policies and procedures

All Educational Authorities in Australia have rules and regulations covering the keeping and care of animals in schools.

- Before you start any school projects you should check your Educational Authority's requirements to see if permits are required.
- You should also check with your state Department of Primary Industries to see if they require you to have licences or permits for your activity (this is particularly important if you are catching wild stock or discharging water).

All teachers should follow the policies and procedures of their employing authority.

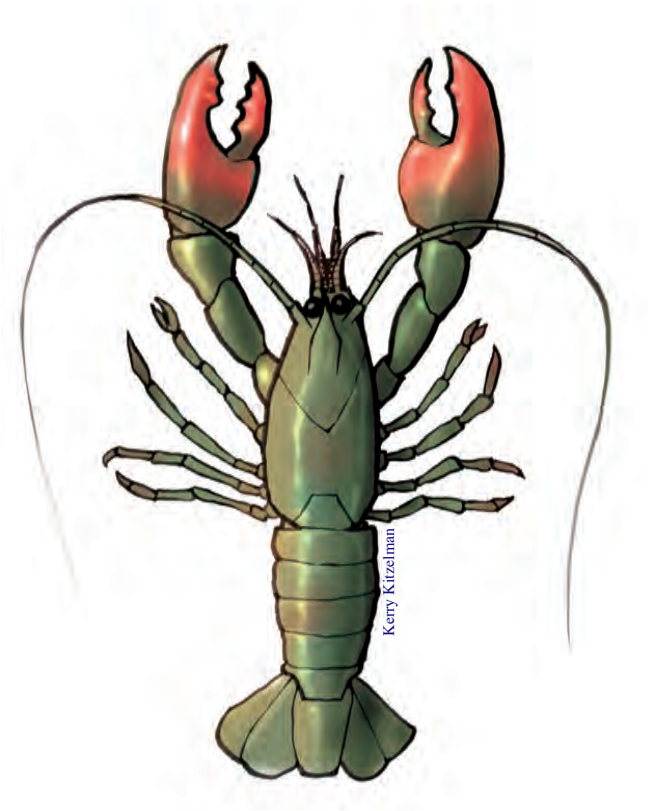
- Ethical procedures specified by community or employers codes of conduct for the handling and keeping of animals should also be adhered to.
- In New South Wales visit:

  www.schools.nsw.edu.au/animalsinschools
- In Queensland visit:

  http://education.qld.gov.au and search the curriculum activity risk assessment documents for handling live animals.

Syllabus coverage

- New South Wales - Marine and Aquaculture Technology
- Queensland - Marine Studies, or Marine and Aquatic Practices Syllabus
- Western Australian Marine and Maritime Technology
- New National Science curriculum, Years 7-10
- All State Agriculture Syllabi



Mick O'Connor

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Figure 5.1 All of the equipment can be found in a local hardware store

PROJECT 1.1 USING POLY PIPE AND FITTINGS

Background

Pipes and fittings needed in many projects can be bought as single items or purchased in bulk from local hardware stores or retail chains. They have been manufactured for home garden irrigation systems, are well made and fit together perfectly giving nice airtight seals.

They are easy to use, a bit like playing with Leggo or Mechano making them ideal for students to design and construct simple aquaculture systems.

Polyethene pipe and associated fittings

Polyethene pipe and associated fittings as shown in Figure 6.1, are ideal for carrying and distributing low pressure air to a wide variety of growth containers and aquaria for school aquaculture exercises.

They are cheap alternatives to the more costly aquarium supplies.

Size

It is important to decide on a size. The projects in this book are based on the following sizes:

- Main lines - 13 mm pipe and 13mm fittings. (This allows interchanging with standard garden hose if necessary)
- Individual air lines - 3 mm pipe and 4 mm fittings. (The 3 mm pipe is a tight fit on the 4 mm fittings and while it may be a little harder to get on it does give the peace of mind that you know it will not blow off)

  On the individual air lines it is always better to use threaded fittings to allow the 3mm pipe to be screwed on tightly.

Materials

13 mm fittings for main air line from pump

- End plug, tap, joiner and elbow T

4 mm fittings for individual air lines

- Screwed adaptor, barbed off-take

Procedure

Try to keep everything standard. 13mm for the main supply pipes and fittings and 3 mm pipe for the individual lines with 4 mm fittings.

There is nothing real hard about designing and constructing this:

Step 1 Design your system.

Step 2 Write down the fittings you need and the lengths of pipe you require.

Step 3 Cut the pipe and place the necessary fittings in position pushing or screwing each fitting home.

Step 4 Use clamps and clips to mount the lines neatly.

Figure 6.1 Common poly pipe and fittings

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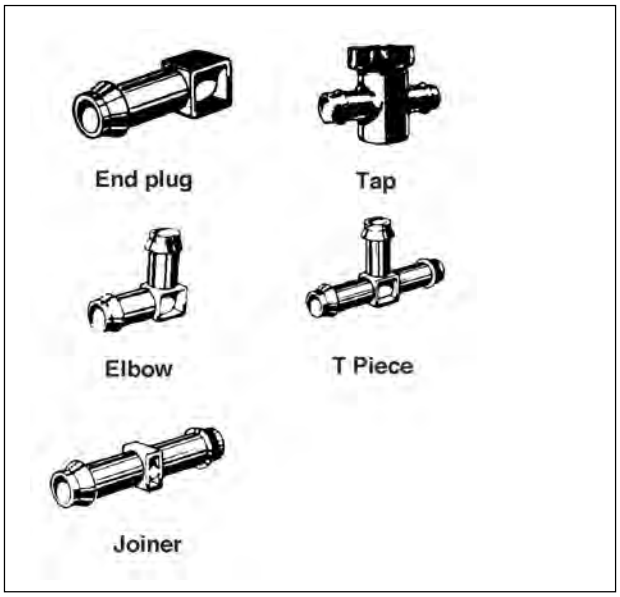
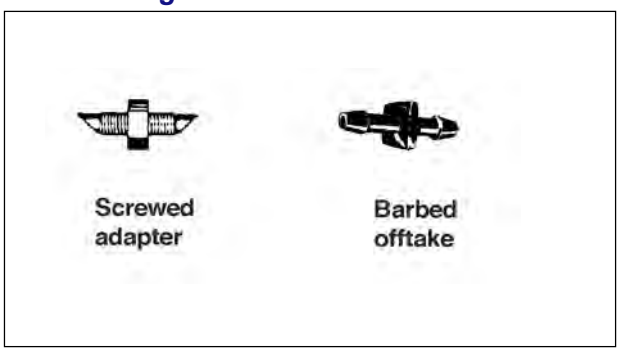
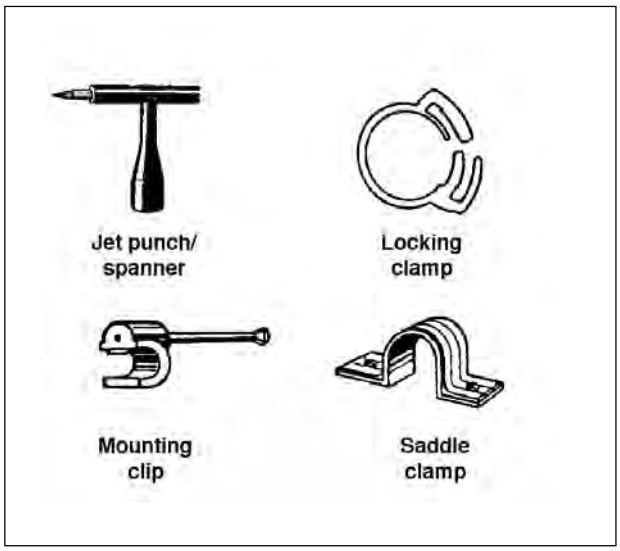
13 mm fittings



4 mm fittings



Hole punch and clamps



Kerry Kitzelman

Figure 7.1 Common poly pipe and fittings

PROJECT 1.2 MAKING AN AIR FILTER FOR ALGAL CULTURES

See page 116 for equipment supplier details.

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Figure 8. 1 Completed air filter

Background

Micro algae like all plants require carbon dioxide (CO_2) for photosynthesis.

- They obtain this gas from the air that is dissolved in the water in which they live.

When micro algae are grown in containers they have to have enough air to provide the carbon dioxide they need.

- This is not a problem when a small amount of liquid is put into a large container - the air can diffuse in and out through a cotton wool or specially designed filter plug as shown in Figure 8.2.

It does become a problem when algae are grown in containers with little free air, providing insufficient carbon dioxide for the algae to make food.

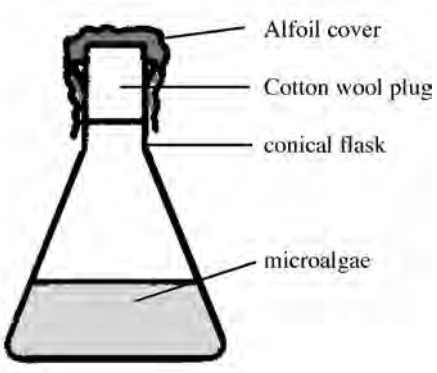

Figure 8.2 A small amount of liquid put into a large container is not a problem for a time
(Illustration Mick O'Connor)

A solution

To overcome this air must be pumped into the container as described in Figure 8.3 (below)

- Normal air will contain minute particles of dust and may also contain micro organisms such as bacteria and fungi that may be harmful to the algae or what the algae will be fed to.
- To minimise the risk of infecting the algal culture, the air being pumped in is filtered. A cotton wool filter is placed on the inlet tube, filtering the air coming in to remove micro organisms. Another similar filter is placed on the outlet tube to prevent microorganisms entering the culture as the excess air escapes.
- The double filter acts as a barrier. Air containing the needed carbon dioxide is provided continuously to the algae allowing them to grow while excluding airborne micro organisms coming back through the outlet tube.

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Project 1.3 over describes how to make this container

Figure 8.3 Algal growth container

Materials

For each filter you will need (See Figure 9.2)

- One 10mm X 100mm PET sample tube with plastic screw top. See page 120 for equipment supplier details.
- One flat bottom 10ml clear polystyrene cylindrical test tube with screw top
- Quantity of 3mm (1/8") clear PVC plastic tubing
- One sterile cotton wool ball
- Hand drill and 3 mm drill bit

Procedure

Step 1 Hold the tube securely and drill a 3 mm hole in the centre of the cap and the bottom of the tube as shown in Figure 9.1.

Step 2 Now unscrew the plastic lid off the sample tube and remove any drill shavings.

Step 3 Sterilise the tube and lid in bleach (see box below).

Step 4 Place a sterile cotton wool ball in the tube (Figure 18.1, Page 18).

Step 5 Insert the required length of plastic into the outlet and inlet holes (Figure 18.1).

Discussion

1. Explain why a filter is needed on the inlet air line.
2. Explain why a filter is needed on the outlet air line.
3. Discuss the benefits of making the filter out of clear plastic specimen tubes that can readily be pulled apart.
4. Explain the reasons for growing micro algae in 'closed' containers rather than open fish tanks.
5. Explain why sterilization is needed for equipment.

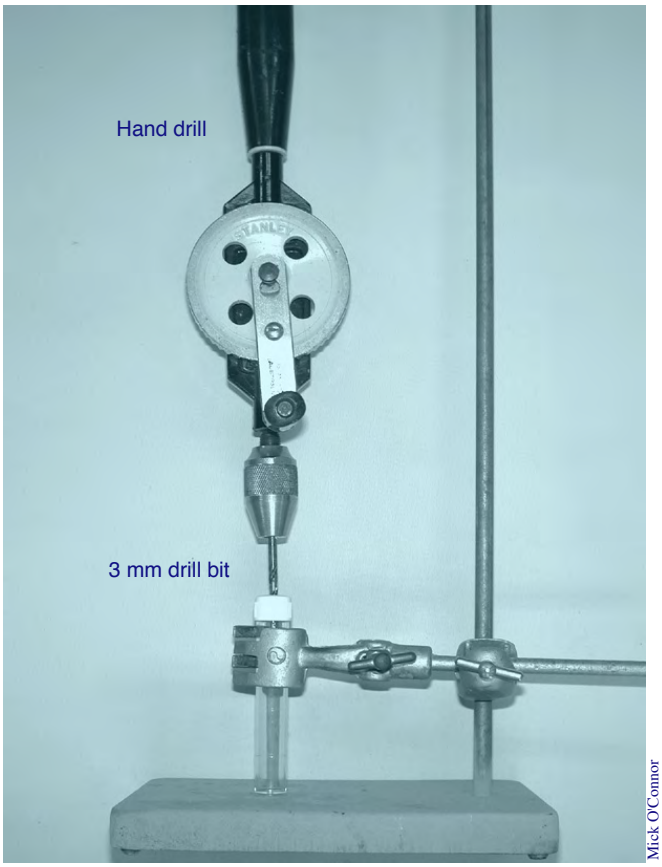

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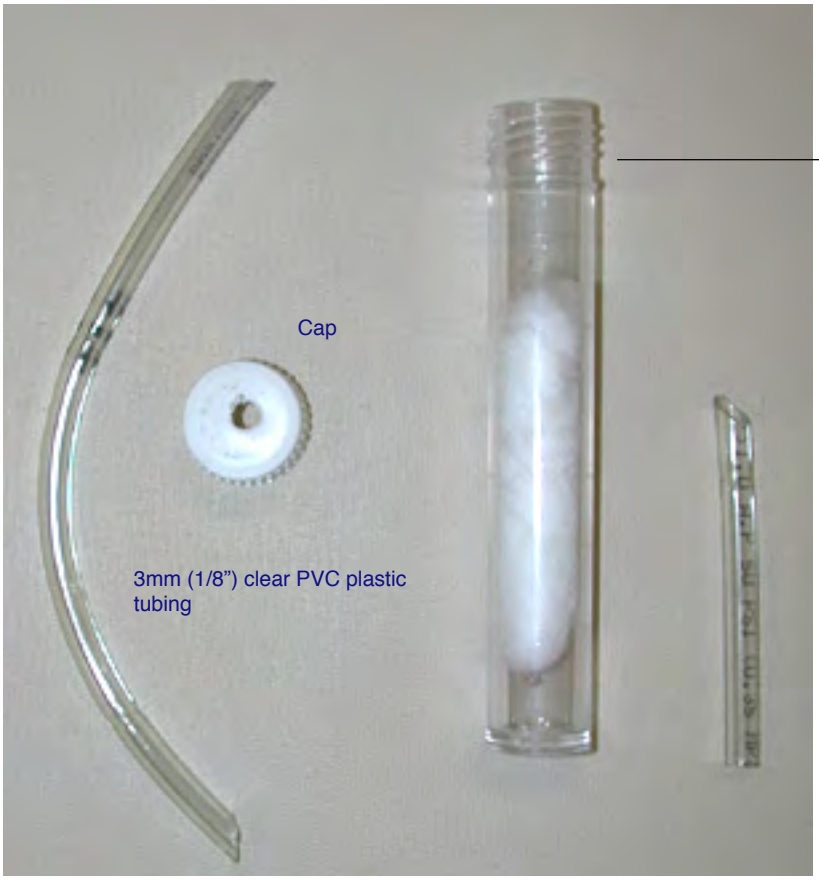
Figure 9.1 Drill a 3 mm hole in the centre of the cap and the bottom



1 flat bottom 10ml clear polystyrene cylindrical test tube with screw top

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Figure 9.2 Parts ready for assembly

Sterilizing

A simple method of sterilization is to use household bleach.

- Follow the instructions on the bottle for nappies - dilution is usually 1/5 dilution
- Fill a container of known volume that will fit all the tubes and lids with water and use one part bleach to five parts water
- Immerse all equipment to be sterilized for one hour.
  - be careful of your clothing with bleach as it can leave nasty white marks on school uniforms!!

PROJECT 1.3 MAKING AN ALGAL GROWTH CONTAINER

Background

Micro algae are small plants which:

- Require light, water and carbon dioxide (CO_2) to carry out photosynthesis and grow.
- Produce a wide range of special fatty acids that eventually end up in fish.
- Are very important sources of food for a variety of marine and aquatic plankton and lavae.
- Live and grow in water, a medium that so many other organisms love to live in.

Problems

Some organisms can be harmful to aquaculture species that algae are going to be feed to, so it is very important to keep them out of the growing vessel.

This is possible on a small scale, eg in a hatchery, but is not possible if the algae are to be grown in an open pond.

Solution

To minimise the risk of infecting the algae with organisms from the air, a closed container is used as shown in Figure 10.1.

- Filtered air (containing carbon dioxide) is pumped to the algae so it can grow by the photosynthetic process.

$$CO_2 + H_2O + \text{energy} \text{ -----> } \text{sugars} + O_2$$

- A readily available sealed access port to allow injection of nutrients is fitted in the top of the vessel.

Containers

See Figure 10.2. These:

- Should be large enough to grow the amount of algae required
- Made of a sterilisable material
- Made of clear PET plastic or glass to allow light to come into the algae

Materials

- One PET bottle with plastic screw lid
- 3 mm hand drill
- Quantity of clear PVC 3 mm plastic tubing
- Two air filters
- Golf tee

See page 116 for equipment supplier details.

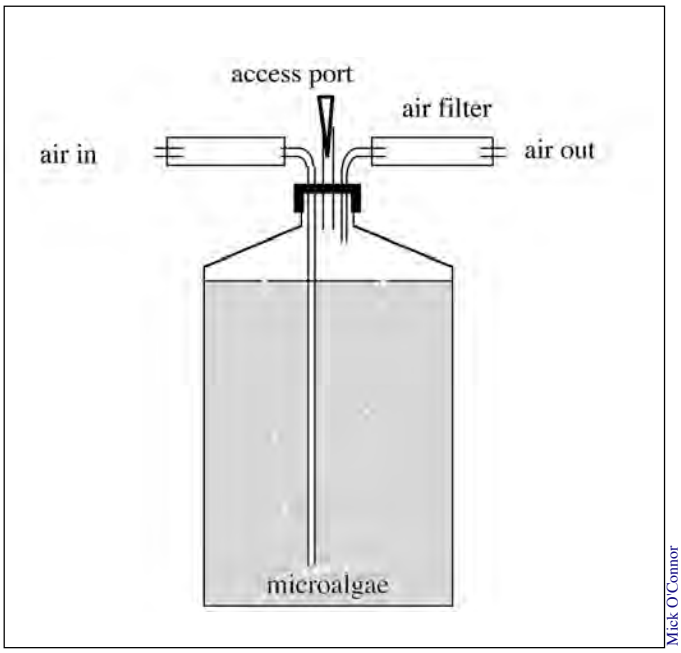

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Figure 10.1 A closed container is used to minimise the risk of infecting the algae with organisms from the air

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Figure 10.2 Materials required

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Figure 11.1 Finished algal growth container

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Figure 11.2 Step 1: Hold the bottle securely and drill three 3 mm holes in the cap

Procedure

Step 1 Hold the bottle securely and drill three 3 mm holes in the cap (Figure 11.2).

Step 2 Unscrew the plastic cap and remove any drill shavings.

Step 3 Sterilise the bottle, tube, golf tee and lid. A simple method is outlined in the box below.

Step 4 Fit the inlet filter and outlet filter in two of the holes.

Step 5 Cut a piece of tube 3 cm long and place it through the third hole.

Step 6 Place the sterile tee in this 'access port' to seal it.

Step 7 Now insert the air filter from Project 1.2 as should in Figure 11.1.

Discussion

1. Explain why the container should be made from clear transparent material.
2. Discuss the advantages and disadvantages of glass and plastic containers.
3. List some containers that are readily available at low or no cost.
4. Discuss the advantages of having an access port.
5. List some procedures that could be used to stop infection through the access port.

Sterilizing

A simple method of sterilization is to use household bleach.

- Follow the instructions on the bottle for nappies - dilution is usually 1/5 dilution
- Fill a large tub of known volume with water and use one part bleach to five parts water
- Immerse all equipment to be sterilized for one hour
  - be careful of your clothing with bleach as it can leave nasty white marks on new jeans!!

PROJECT 1.4 MAKING A LIGHT CUPBOARD

Background

Micro algae are plants. They require light, water and carbon dioxide (CO_2) to carry out photosynthesis and growth.

The best conditions for growth vary slightly between species. Tropical species for example like a slightly higher temperature than those found in cooler waters.

A 'light cupboard' is the easiest way to monitor and control the conditions while the algae grow.

It has the advantage that you can control the type of light, the amount of light, the temperature, the air supply and any vermin that may take a liking to your cultures.

The most simple and in expensive cabinets are made from existing cupboards - laboratory cupboards are fine.

- Daylight fluorescent tubes are used to supply the wavelengths that the algae need.
- They are much cheaper than 'grow' tubes and do the job well. A timer is needed on the lights.
- These micro algae like a photoperiod of 12:12 ie 12 hours light: 12 hours dark if grown in small containers up to about three litres.
- For bigger containers the light period is extended to 16 hours so the photoperiod is 16:8.
- Temperature should be kept in the range of 20 -24°C.

Materials

- Cupboard approximately 1700 mm X 450 mm X 750 mm
- Carpenters tools
- High gloss white paint
- 13mm poly garden irrigation tube plus fittings
- Two fluoro tubes
- One powerboard and 240 volt timer

Procedure

See Figures 12.1 - 12.3.

Step 1 Replace any particle board with ply as particle board will fall to pieces if it gets wet.

Step 3 Fit one central shelf.

Step 3 Paint the interior with high gloss white paint.

Step 4 Fit two daylight fluoro tubes - one to supply light to the top shelf and the other to supply the bottom shelf.

Step 5 Cut a series of 50 mm holes for ventilation and cover with flyscreen.

Step 6 Replace the doors with a clear front screen if it is to be a feature.

Step 7 Run the main air supply lines under the cupboard top and shelf at the front (out of sight).

- Take it through one end with an elbow at the bottom and a T at the top to go to the main air line.

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Figure 12.1 Base of cupboard

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Figure 12.2 Cupboard approximately 1700 mm X 450 mm X 750 mm

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Figure 12.3 113mm poly garden irrigation tube plus fittings and cupboard with lights

Growth light cupboard

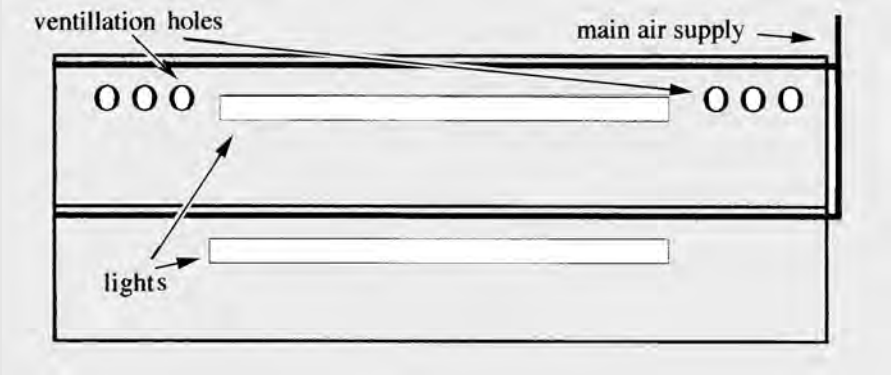

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Figure 13.1 Finished cupboard

Discussion

1. For your microalgal species list the optimum range for the following conditions:
   - Temperature
   - Light
   - Photoperiod
   - Nutrient conditions
2. Discuss the advantages and disadvantages of using wood instead of steel to make a light cabinet.
3. List some suitable methods of keeping the cabinet cool in summer and warm in winter.

PROJECT 1.5 MAKING A SMALL GLASS AQUARIUM

Background

A small rectangular glass aquarium is simple to construct and will provide hours of enjoyment during constructing, setting up and watching the operation as you keep your plants and animals.

Formula for the glass

Refer to Figure 14.1 where A is the length, B is the width and C is the height of the side.

- BASE: A x B
- SIDE: A x C
- ENDS: C x (B + 2 x thickness of glass being used)

The ends will glue to and cover the ends of the sides and bottom

Using neutral cure silicon glue

- If you haven't used silicon in a chalking gun before read the information in the box below.
- Practice makes perfect so try the gun and silicon out on some cardboard or old glass sheets.

Materials

- Pre-cut glass from an aquarium shop
  - this is usually sold as kits or glass cut to your requirements from a local glazier or window maker
- Neutral cure silicone glue (Eg, Sellys wet area silicon adhesive) and chalking gun
- Plastic sheet and two x 2 litre plastic drink bottles full of water.

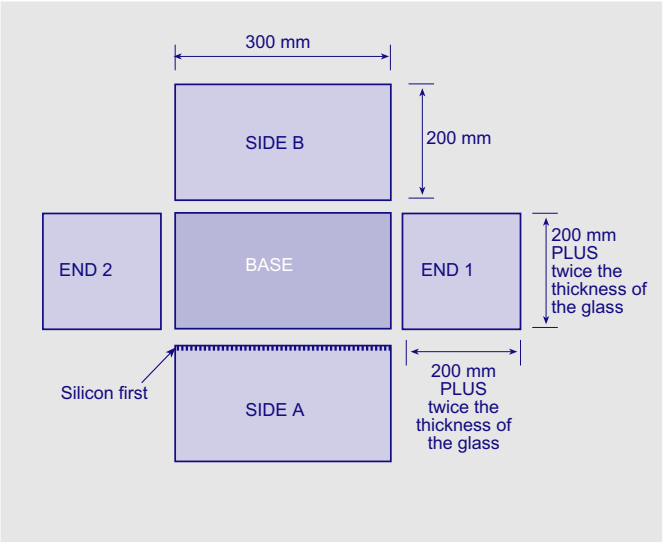

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Figure 14.1 Glass pre-cut in pieces from an aquarium shop

Using silicon - must be neutral cure

- Material Safety Data Sheet at http://www.selleys.com.au/Selleys-Wet-Area-Silicone/default.aspx
- Thoroughly clean and dry surface from oil, dirt and grease with Mineral Turpentine firstly and then Methylated Spirits.

  Allow to dry completely before applying silicone.
- Cut the seal at the top of the cartridge and cut the nozzle at a 45° angle, to the size of the gap opening. Fit the nozzle to the top of the cartridge.
- Insert the cartridge into a caulking gun
- Hold the caulking gun at a 45° angle.

  Press the nozzle opening against the joint, apply steady pressure to the caulking gun and extrude the silicone sealant forcing the sealant into the gap and move in a pushing motion, along the joint in one smooth action.
- For a smooth finish, smooth with a finger dipped in mineral turpentine. Excess sealant must be cleaned up before the sealant skins in 5-10 minutes.

  Wipe excess away with a cloth dampened in mineral turpentine. Avoid smearing on surrounding surfaces, see handy tip.
- Sealant will cure fully in 72 hours. As this point any unwanted sealant can be removed by trimming with a sharp blade, but avoid under cutting the seal.

Handy tips

Sellys

- When first applying product, extrude a small amount onto a piece of scrap material to ensure a smooth continuous flow of silicone sealant.
- Work in sections that can be completed before sealant skins.

Safety tips

- Uncured product may irritate eyes. If in eyes, flood the eyes for at least 15 minutes and seek medical advice.
- Avoid contact with skin, if contact occurs, wipe off immediately and wash with detergent. It may irritate sensitive skin.
- Avoid breathing in vapours.
- Ensure you use a well ventilated area as product releases methyl ethyl ketoxime while curing (poisonous to fish).
- For advice, contact a Poisons Information Centre (AUS 131 126; NZ 0800 764 766) or a doctor.
- For further detail on the safe use of this product, please refer to the Material Safety Data Sheet, which can be downloaded from this site above.

Procedure

Step 1 Place the protective plastic sheet on the bench.

Step 2 Lay the base on the protective plastic sheet.

Step 3 Using the glue gun and your finger as shown in Figure 15.1, run an even bead of silicon around the side edges of the base.

Use your finger as a guide to keep the bead even and pressed against the glass edge.

Hint

- You may want to use some masking tape along the glass to get an even bead.
- As soon as you finish take of the tape.
- Any excess on your hand can be washed off with turps or soapy water.

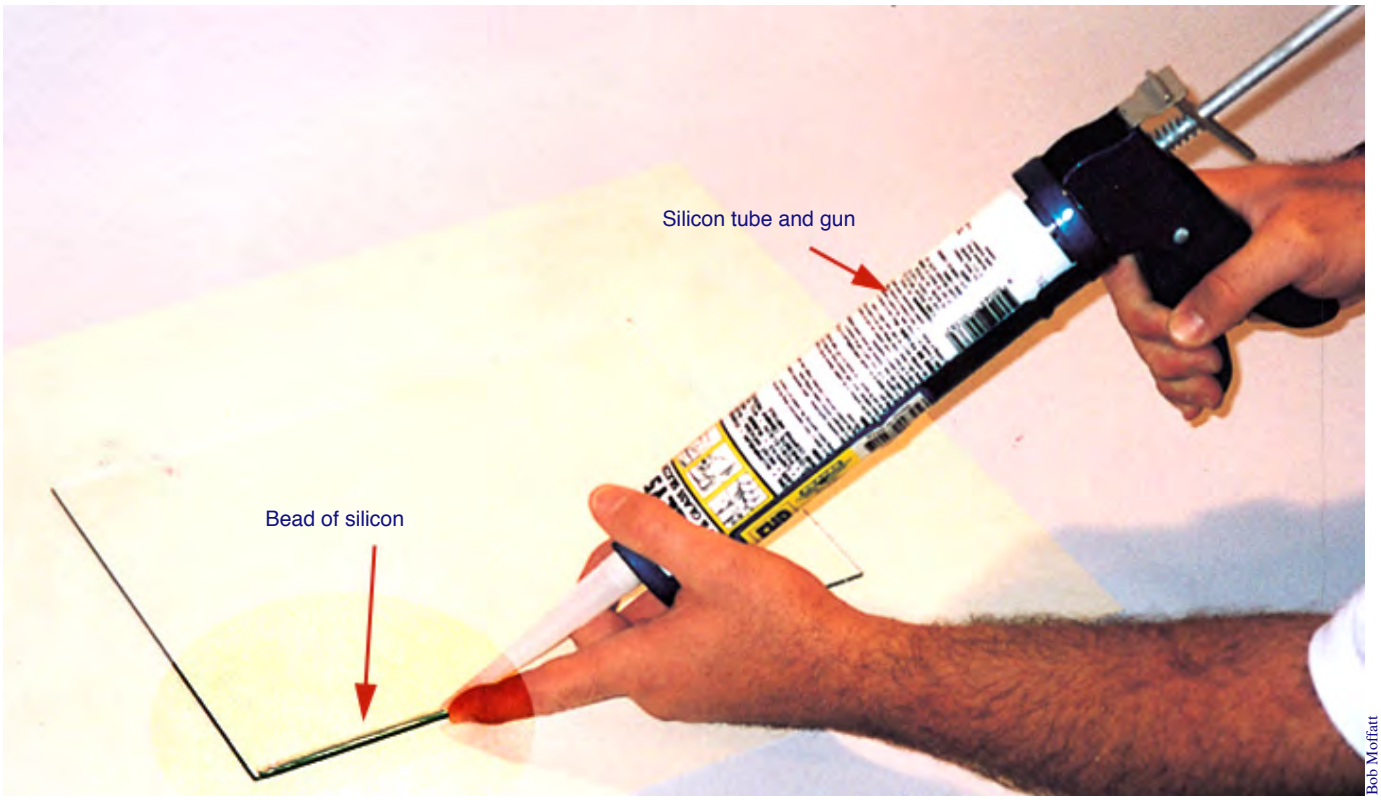

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Figure 15.1 Run a bead of silicon along the edge of one side

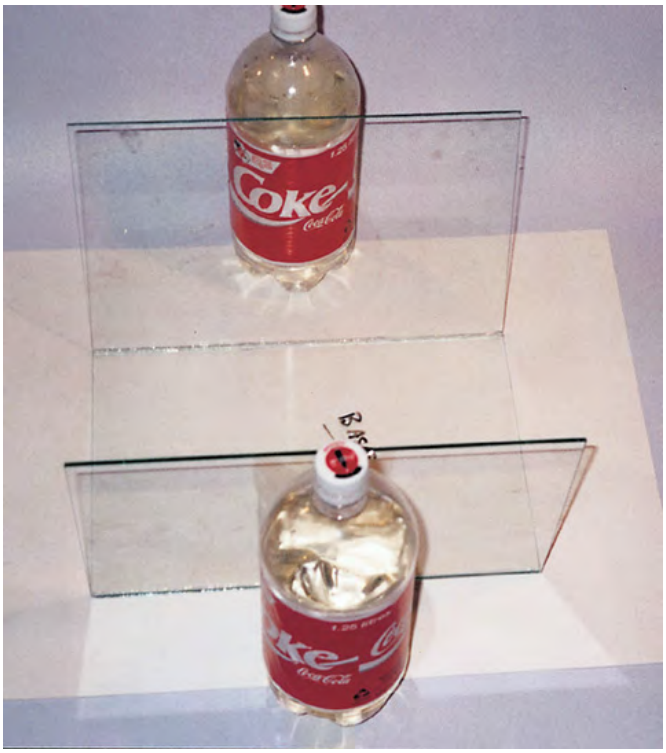

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Figure 15.2 Carefully move the glued section towards the edge of the base so that it just touches the base's edge

Step 4 Place the 2 litre bottle of water against the glass side to hold it in position while you do the other side as shown in Figure 15.2.

Hints

The secret of success and neatness is

- Don't use too much silicon glue.
- Wipe off excess and clean up with turps before it sets.
- Use masking tape if your hands are not steady, but pull it off before the glue sets and allow time to complete additional steps.
- Practice using the chalking gun.

Turn over for steps 5 - 9

Step 5 Take one end glass and run a neat bead of silicon around the two sides and bottom as close as you can to the edge.

See Figure 16.1.

Step 6 Make sure the two sides are standing perpendicular to the base.

Step 7 Carefully bring the end with its glue bead against the sides and bottom, again gently squeezing the bead to make contact and fill gaps that would later leak.

Step 8 Using a small set square, make sure the end is perfectly positioned and tape it to the sides using strips of masking tape on the internal and external surfaces.

- The tape is removed when the silicone sets.

Step 9 Repeat the procedure for the other end.

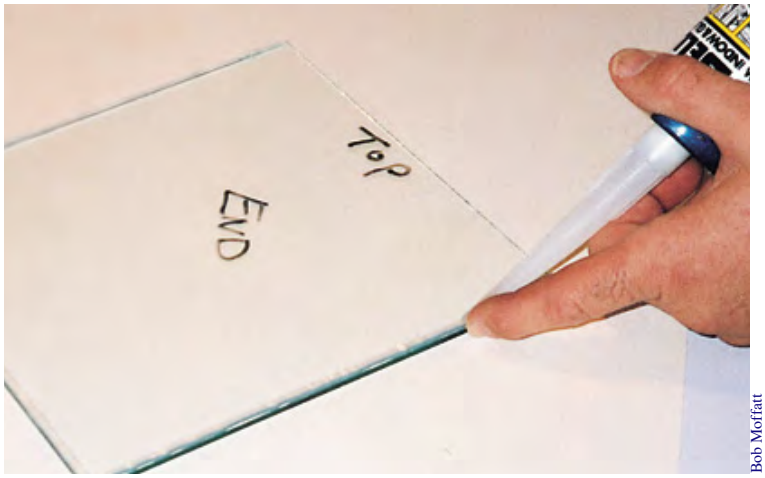

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Figure 16.1 Step 5: Run a bead of silicon along three sides of end 1

If all else fails

You can always buy small aquariums from an aquarium shop.

These come with a set of instructions, glass aquarium, under-gravel and filter hosing and a small pump.

Aquarium shops also come with loads of advice especially if you have never kept an aquarium before.

Aquariums on the internet

Building aquariums is one of the most popular sites on the internet.

Just google aquariums and you can be sure to find out what you want


Bob Moffatt

Figure 16.2 Place four pieces of masking tape at the edges you will be gluing, ready to hold the ends when finished

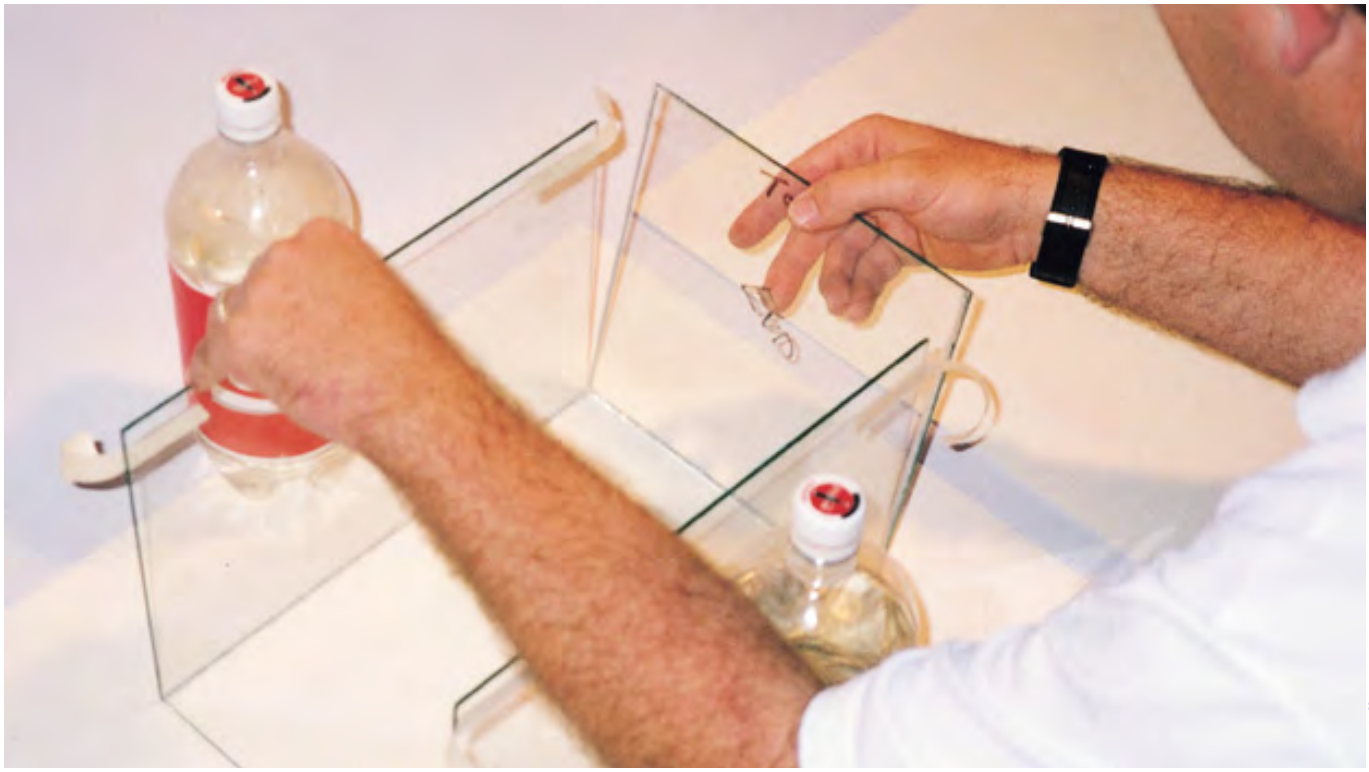
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Figure 16.3 Tape the end pieces in position

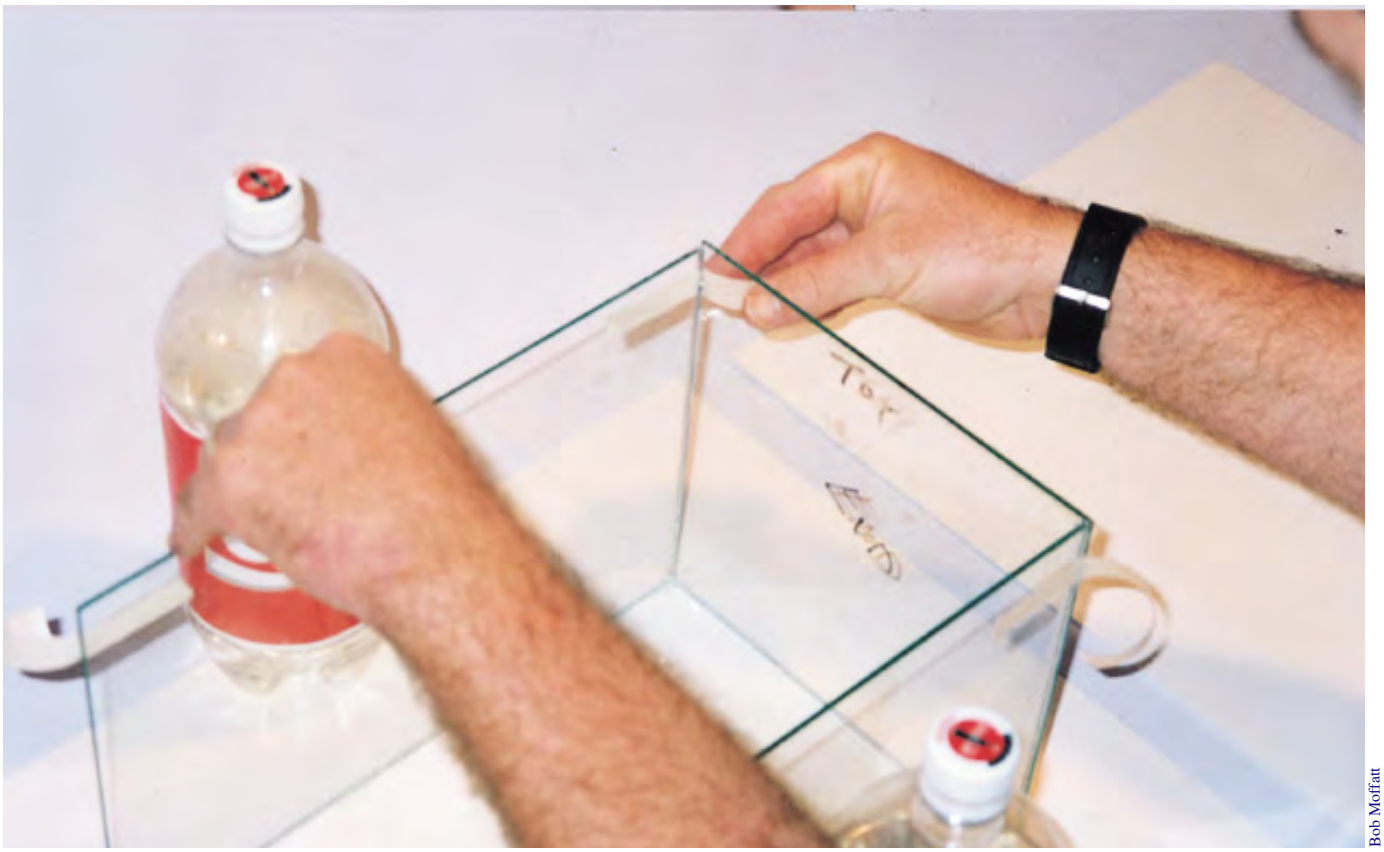

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Figure 17.1 Finished project

PROJECT 1.6 MAKING A PLASTIC TUB AQUARIUM

Background

Readily available plastic storage tubs make great aquariums.

- They are cheap, easy to make and come in a variety of sizes and colours.
- The only problem is that you cannot see through the sides of them - this can be overcome.

Keep in mind that an aquarium is simply a container in which marine and aquatic species are kept away from their natural environment.

Whilst the container is important it is not as important as the processes that go on in it or that are associated with it to maintain good water quality for the species being held.

This exercise makes a 40 litre aquarium using a 50 litre stackable Nylex storage tub.

Filtration and aeration are up to you - a sponge filter works really well in this aquarium (See project 2.12).

The old saying "an inch of fish to a gallon of water" was a good rule.

- 10mm of fish for every two litres of water is an excellent conversion allowing 200mm of fish to be kept in this aquarium.
- It is ideal for keeping small fish.

Material

- One Nylex 50litre stackable crate
- One piece of perspex sheet 190mm X 250mm X 170mm
- One tube of neutral cure silicon
- Fine sandpaper and sanding block
- Stanley knife
- Drill and 8mm drill bit
- Jig saw
- Nylon nuts and bolts or threaded section

Procedure

The idea is to place a clear window in one end of the crate without weakening the crate.

Step 1 Locate the stacking wedges on each end of the crate.

Step 2 Select the wedge that protrudes - ie extends on the outside of the crate. In the crate shown in the photo this measures 200mm X 260mm X 170mm.

Step 3 Mark out a 150mm X 210mm X 120mm shape on the outside of the wedge.

Step 4 Using a drill and a jig saw neatly cut out the window.

Step 5 Smooth carefully with Stanly knife using a scraping action.

Step 6 Cut out a 190mm X 250mm X 160mm piece of perspex using the jigsaw - careful, perspex scratches easily.

Step 7 Smooth the cut edges carefully with a sandpaper.

Figure 18.1 Materials

Step 8 Stand the crate on its end, cutout end down.

Step 9 Run a neat bead of silicon on the perspex and glue it on the inside of the crate to form the window.

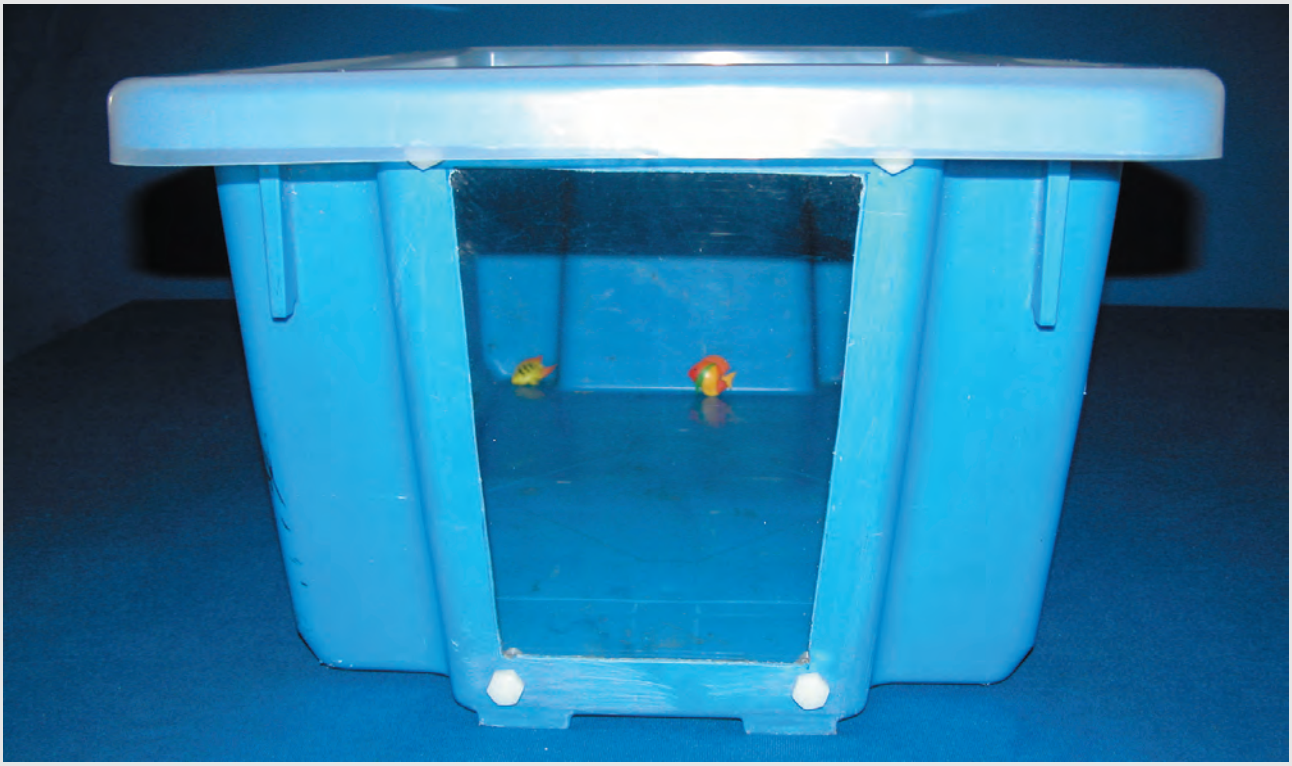
Step 10 Place some weight, (a few dive weights) on the perspex to assist the glue as it dries to give an even seal.

Step 11 When dry trim excess silicon with a sharp blade.

- While measurements will vary with the crate size and the size of the window required, the general rule is, the window hole should be 20mm smaller on every side than the perspex window.
- The window is always fitted to the inside to take advantage of the water pressure to assist in sealing.

Step 12 The viewing panel can be made more secure by fastening with four - six nylon bolts.

10mm of fish for every two litres of water is an excellent conversion allowing 200mm of fish to be kept in this aquarium.



Finished project

Mick O'Connor

Figure 19.1 Stages of construction

PROJECT 1.7 MAKING A CRAYFISH TANK

Background

All Australian native crayfish species love to find ways of escaping.

- Stories abound of crayfish walking around classrooms at night terrifying cleaners and dieing in inaccessible spots behind cupboards leaving a smell that seems to linger on and on.
- They can climb airlines and use the right angles in glass aquaria as ladders to get out.

The trick is to make a container that is escape proof and at the same time can be used for mating, hatching and growing out - such a container does exist.

20 litre plastic containers (commonly called polycubes) used for all manner of material from vegetable oil to detergent to agrochemicals are ideal (Figure 20.1) .

- Ones that have contained poisons naturally should not be used unless they can be cleaned and any leached poison neutralised chemically.

These containers have no seams, have rounded corners and have a smooth surface that is impossible for crayfish to climb. They fall backwards off the curved surfaces as well. Best of all they cost nothing.

Mick O'Connor

Figure 20.1 Polycube

Materials

- 1 X 20 litre plastic polycube
- Hand drill, 12 mm drill and 3 mm drill
- Jig saw
- Marking pen and tape measure
- Quantity of 3 mm clear plastic PVC tubing

Procedure

Step 1 Completely clean the polycube removing any labels.

Step 2 Position it on its side so the handle is vertical.

Step 3 Place the cap at the TOP.

Step 4 Mark out a 250 X 150 square in the centre of the upper surface.

Step 5 Drill 4 neat 12mm holes in the corners of the square.

Step 6 Cut the square out with a jig saw - it is the main access.

Step 7 Drill a 3mm hole in the centre rear of the top surface - on most polycubes there is a flat round reinforced section that is the spot - this is the airline entry hole.

Step 8 Use a knife or blade or sandpaper to smooth the cut edges.

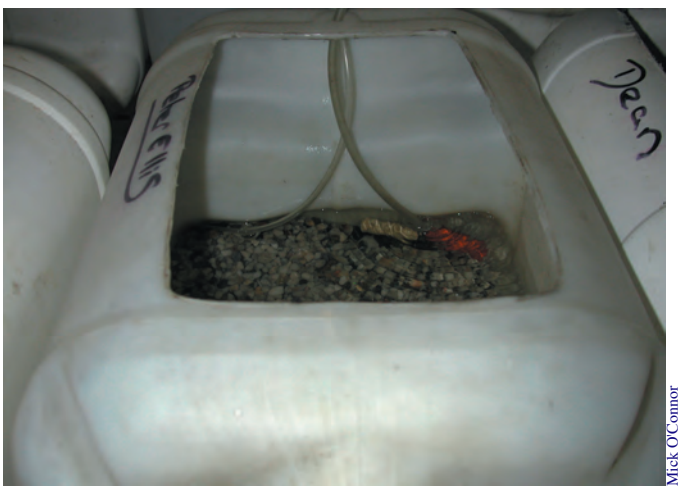
Step 9 If you have a lot to do, make up a 250 X150 metal frame out of a piece of 150 purlin. Cut the lips off, put a handle on it heat it up on the barbie and melt through the container - very quick and neat!!

Step 10 You can also melt the air hole through with a hot tripod leg.

Step 11 Add stones, shelter pipe and water to about 50mm depth.

Mick O'Connor

Figure 20.2 Construction



Mick O'Connor

Figure 20.3 Finished project

PROJECT 1.8 MAKING A FLOW THROUGH CRAYFISH TANK

Flow through systems

Yabbies can be grown in isolation or in small groups to allow more detailed study, using individual containers.

Growth trials, monitoring moulting and breeding can all be accomplished in small containers (See Figures 21.1 and 21.2).

Unfortunately the smaller the container the lower the water volume and the harder it is to maintain good water quality.

Flow through systems, where the water is continually circulated through the yabby container may overcome many of these problems. In these systems the same amount of water leaves the system that enters it, keeping the level and the water volume within the container constant.

Two simple types of flow through systems are:

- Self contained flow through
- Battery flow through

Type A. Self contained

These systems consist of a large tank with smaller individual containers for the yabbies within it (Figure 21.1 and 21.2).

The containers have holes in their bases to allow water to flow out. A small submersible fountain pump in the large tank is connected to a spray bar over the top of the containers and delivers water to them at the same time aerating the water (Figure 21.3).

The water level in the containers is controlled by the water level in the main tank. The depth of water in the containers may be adjusted by raising and lowering them eg by putting bricks under them.

Materials

- Large tank
- Separate growing containers. This will be determined by the size of the animal and what is to be done with them.
  - The separate containers may be plastic flower pots or polycubes
- Submersible fountain pump
- 13 mm poly pipe and 3mm drill
- Clean gravel for the bottom

Procedure

Step 1 Drill a series of 3 mm holes in the bottom of the individual containers.

Step 2 Place some gravel in the bottom of each and place them in the bottom of a large tank.

Step 3 Position a small fountain pump in the large tank.

Step 4 Cut 13 mm poly pipe and using the necessary fittings, make a spray bar or a network of spraybars to fit over the individual containers.

Step 5 Place a small hole in the 13mm pipe in the centre of each container to deliver the water.

Step 6 Adjust the water level in the large tank to suit.

Step 7 If you need greater volume in the large tank, place the smaller containers in the bread crate and lift it by placing a few bricks under it.

Mick O'Connor

Figure 21.1 Small yabbies being grown in flower pots in self contained flow through system

Mick O'Connor

Figure 21.2 Larger yabbies being grown in polycubes in self contained flow through system

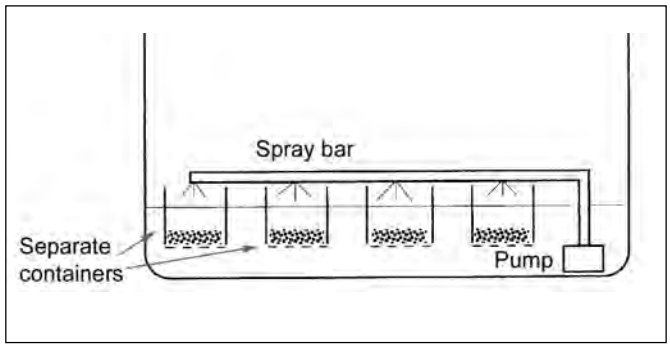


Figure 21.3 Spray bars
Mick O'Connor

Type B: Battery flow

The larger battery flow system consists of individual units completely separate from each other.

The number of units can be increased or decreased at will. The units are stacked on a rack where the top units are supplied with water via a spray bar.

The water level inside each individual container is controlled by a stand pipe allow containers to have different levels and different volumes if required.

The stand pipe (see Figure 33.1 page opposite) delivers overflow water to the tank below it through an overflow delivery pipe, the water eventually flowing through the battery under gravitational force.

Eventually the overflow water is collected from the lowest level containers, directed to a sump and then, using a submersible fountain pump, is pumped back to the spray bars to repeat the process.

It is important that the water circulates both through the system but also within each individual container.

Inlet water must be directed well away from the standpipe, preferably at the other end of the container, so that there is a flow and mixing within each container.

Materials

- Rack or stand (Figure 22.2)
- Poly cube yabby containers
- Polycube tap
- 13 mm poly pipe
- 13 mm right angle poly elbow
- Submersible fountain pump

Procedure

Step 1 Make a crayfish container from a poly cube as shown in the previous exercise.

Step 2 Turn the container over and locate the tap bung.

Step 3 Unscrew the bung.

Step 4 Take a 12mm drill bit and drill a hole in the blank wall which the bung was covering.

Step 5 Screw in the tap.

Step 6 Cut a length of 13mm poly pipe 300mm long and push one end of it over the tap outlet.

Step 7 Fit a right angle elbow to the other end and put another 100 mm (length may vary) piece of 13 mm poly pipe on the elbow as a drop pipe.

Step 8 Turn the container upright and fit a stand pipe.

- The length of this pipe will determine the water level inside the container.
- For small yabbies a piece of nylon flyscreen will need to cover this standpipe opening to prevent escape.

  Nylon flyscreening and cable ties work well!

Mick O'Connor

Figure 22.1 Materials required

Summer cooling (optional)

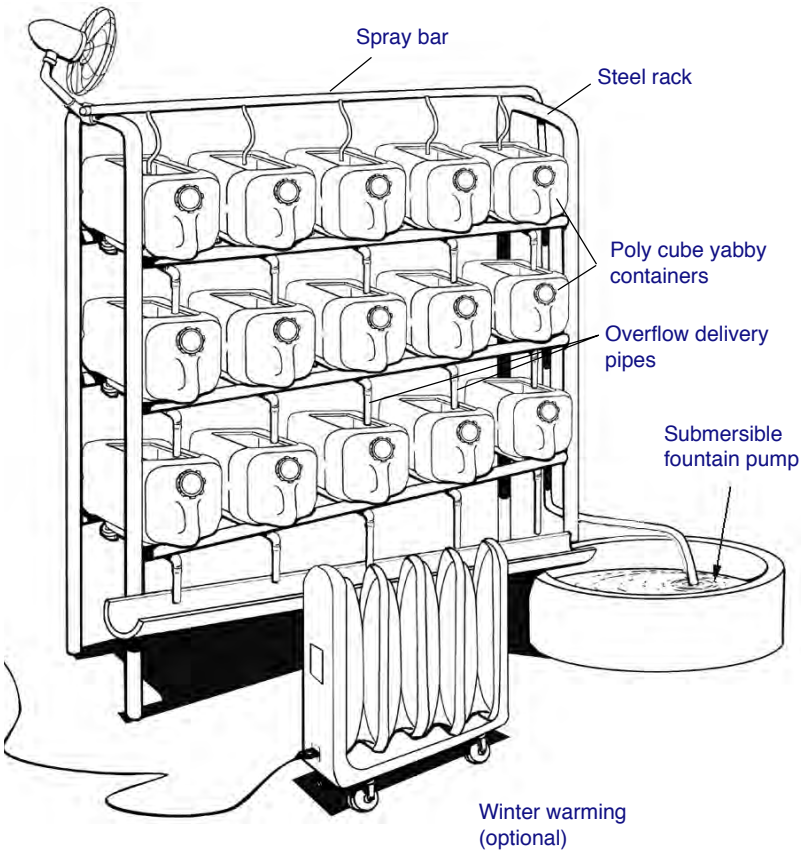
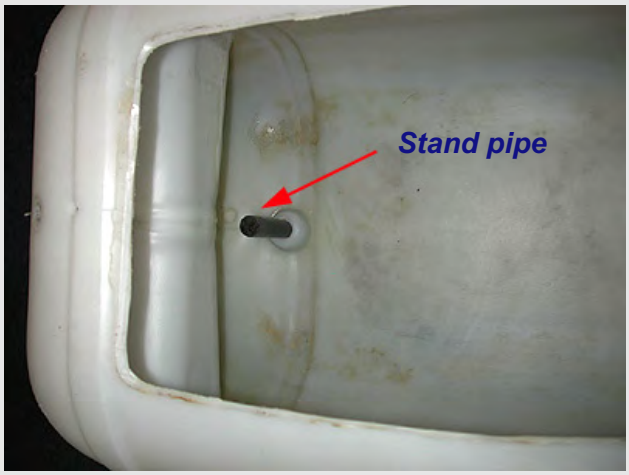


Figure 22.2 A battery flow through system

Kerry Kitzelman



Standpipe tube fitted into hole

Drilling hole for standpipe

Individual tank on rack showing overflow delivery tube

Individual tanks on rack

Finished tank

Mick O'Connor

Figure 23.1 Stages of construction

PROJECT 1.9 MAKING AN ARTEMIA HATCHERY

Background

In a hatchery, artemia and their eggs should be kept in 'suspension' with their food and constantly aerated to supply the oxygen they need as they hatch.

The hatchery, which can also be used as a growth container, consists of an inverted conical container aerated from the bottom of the cone. This design keeps the hatching cysts circulating in the water and will avoid 'dead spots' where dissolved oxygen may be low and carbon dioxide may be high.

Conical containers also allow easy separation of cysts and nauplii after hatching (Figure 24.1).

- All the steps needed to hatch artemia can be done in this container.
- Inverted plastic (1 or 2 litre) soft drink bottles with their bottoms cut off make excellent containers to hatch small amounts of artemia and are an inexpensive alternative to glass or fibreglass containers.
- This design also keeps mature and growing Artemia 'mixed with their food making it easier for them to eat.

A 4mm poly screwed adapter is screwed through the bottle lid to take a 3 mm plastic airline. The tube is connected to an aquarium aerator to give gentle aeration and is looped through the two holes in the top of the vessel to prevent siphoning.

Materials

- Two clear plastic 2 litre soft drink or water bottles
- 4mm poly screwed adapter
- 3mm clear PVC plastic tubing

Procedure

Step 1 Select two appropriate 2 litre plastic soft drink containers.

Step 2 Soak the label off the bottles and wash thoroughly with clean water.

Step 3 Rinse the bottle 2 or 3 times in clean detergent free water - all chemicals MUST be removed from the bottle.

Step 4 Using scissors cut off the bottom of one of the bottles. This bottle will become the 'container'.

Step 5 Cut the other bottle in half. It will be the 'stand'.

Step 6 Drill a 5mm hole 10cm from the bottom, up the side, of this 'stand bottle'.

Step 7 Using a poly jetpunch/spanner place a hole in one of the bottle caps.

Step 8 Screw in to this hole the 4mm screwed adapter and attach the airline.

Step 9 Screw the lid back on the container bottle, invert it into the stand bottle and feed the airline out through the hole in the stand.

Step 10 Punch two 4mm holes apart in the 200 mm apart at the top (cut end) of the container bottle with a hole punch (see Figure opposite).

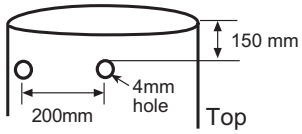


Illustration Bob Moffatt

Step 11 Thread the airline through these two holes before connecting to the aerator. This will ensure the airline remains above the surface of the liquid and will prevent siphoning out the aerator.


Mick O'Connor

Figure 24.1 Materials

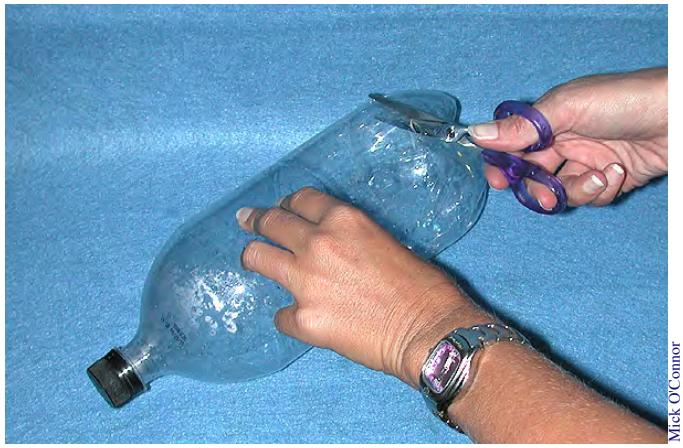
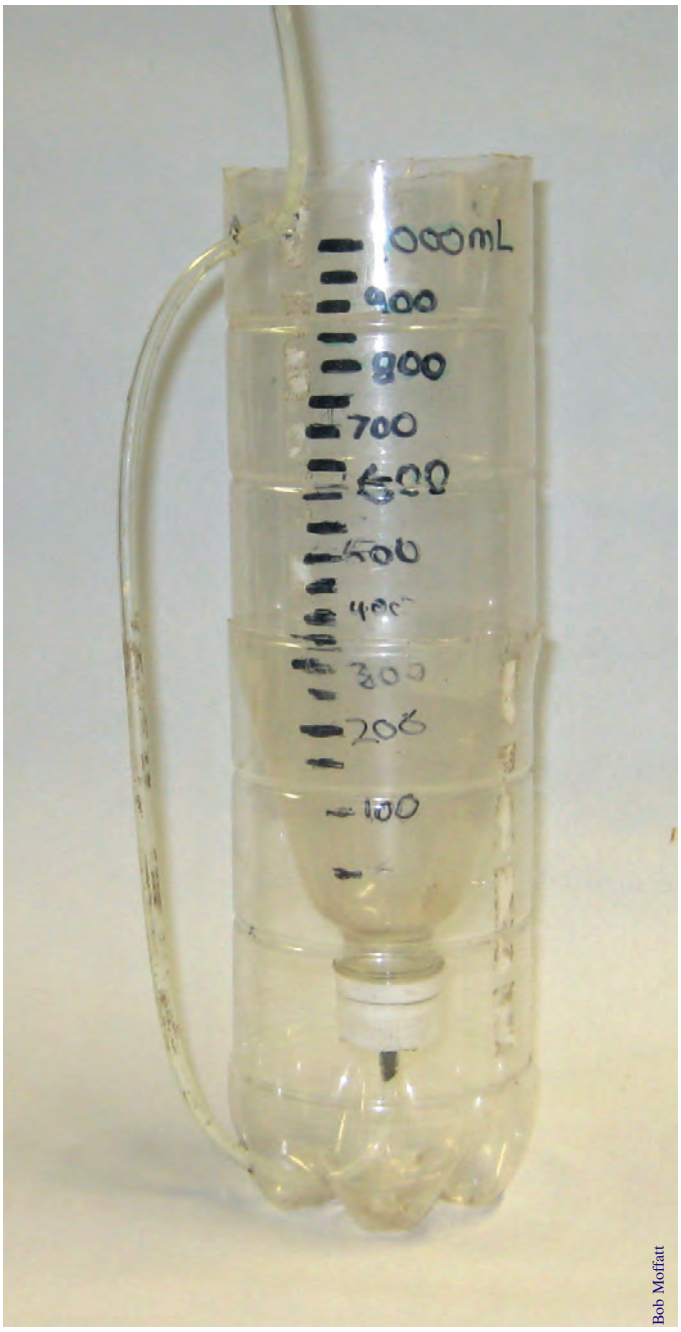
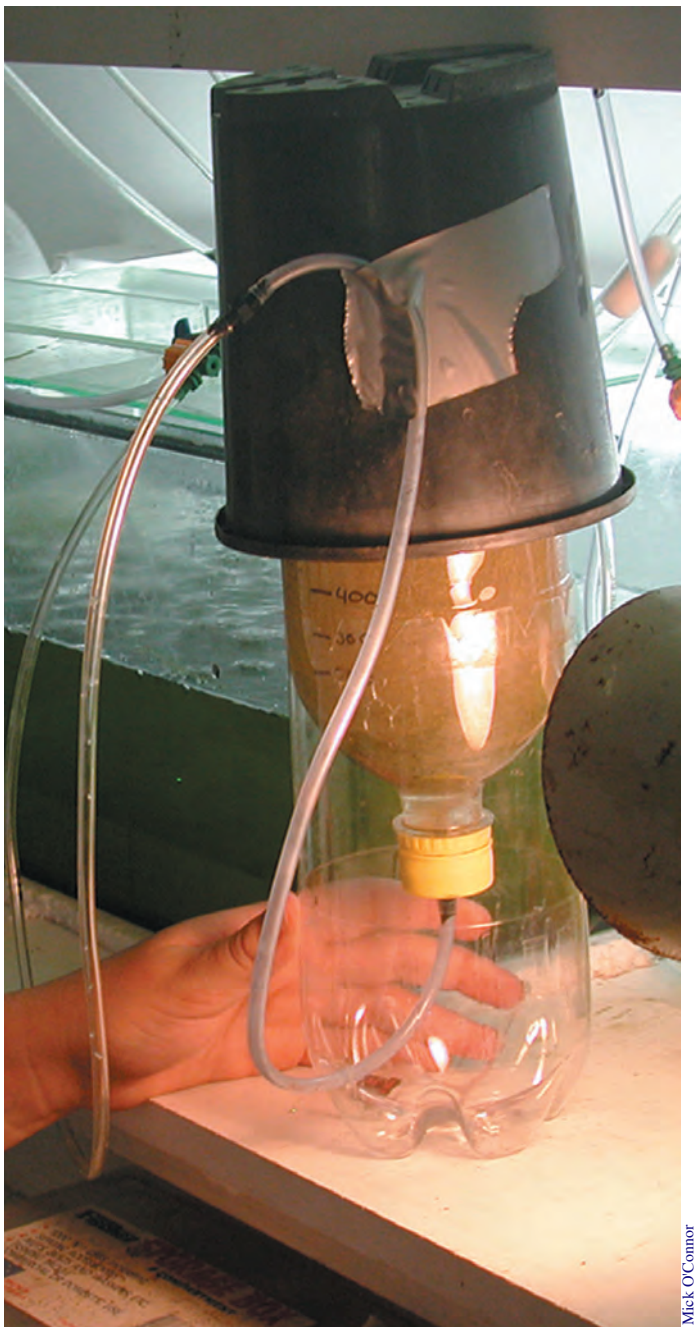

Mick O'Connor

Figure 24.2 Step 1



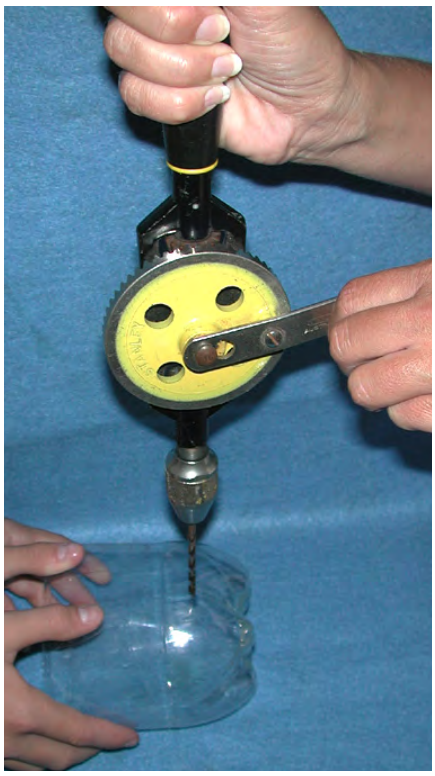
Bob Moffatt

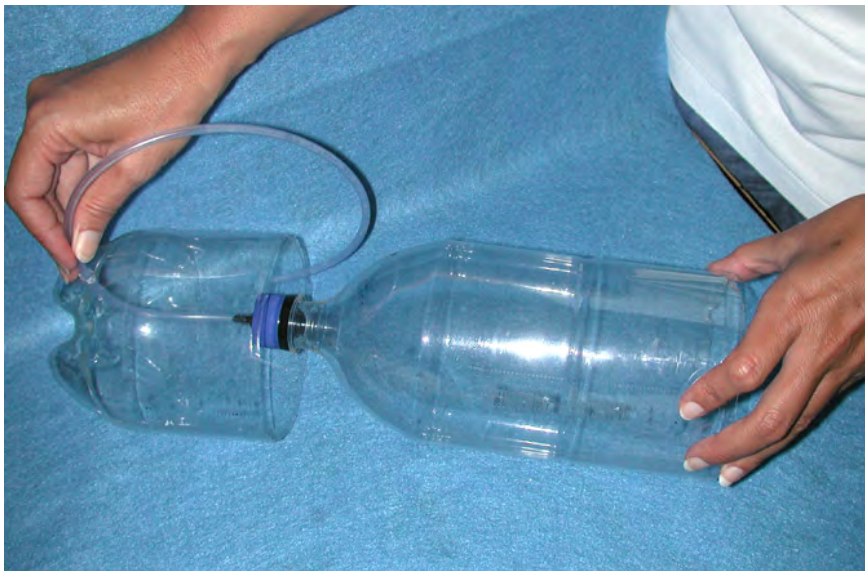


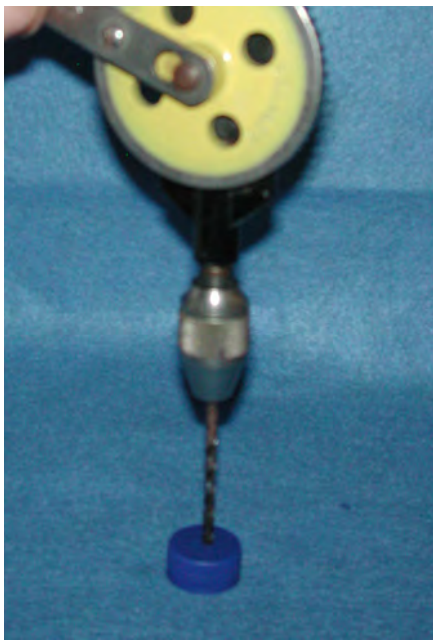
Mick O'Connor

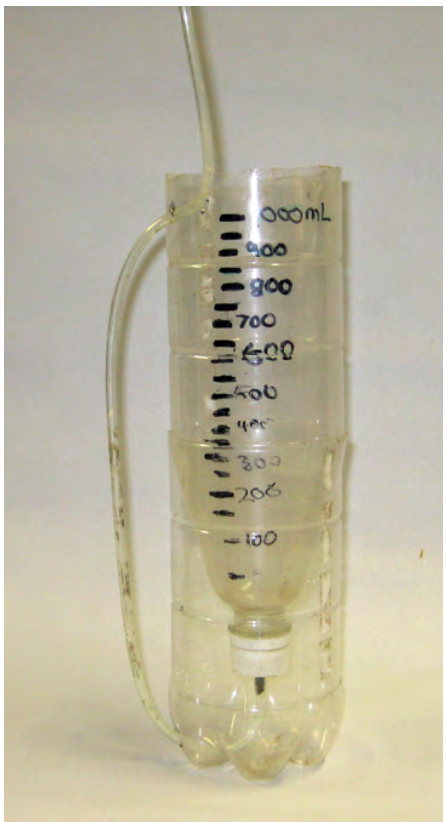
Mick O'Connor

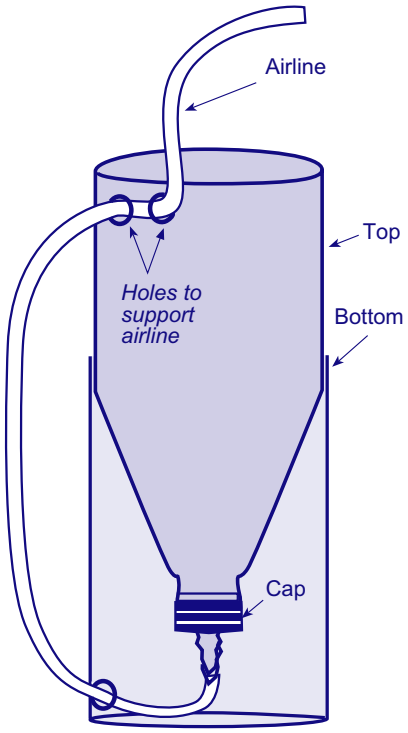
Figure 25.2 Hatcheries in use











Photos Mick O'Connor, Illustration Bob Moffatt

Figure 26.1 Stages of construction

PROJECT 1.10 MAKING A LARGER GROW OUT TANK

Background

There are a variety of plastic tubs ranging from large storage bins to plastic cattle troughs that make ideal grow out tanks for fish and crayfish. They are cheap and easy to modify for aquaculture.

- Some, like cattle troughs, are designed to hold water and need no strengthening, but they are more expensive (See page 120 for supplier details)
- Black polythene storage bins are cheap but tend to bulge at the top when filled - this is easily rectified

This exercise makes a 180 litre grow out tank using a 220 litre Black polythene storage bin. The biofilter as described in Project 2.11, coupled to a small submersible fountain pump, is recommended for this type of tank.

Materials

- 1 x 220 litre Black polythene storage bin.
- 1 x broom handle
- 1 x metre of 25 mm black poly pipe
- 4 x large cable ties (nylon rope will suffice but is not as professional)

Procedure

Step 1 Fill the tub with water.

Step 2 Check if the top edges bulge outwards.

Step 3 Mark the points of greatest bulge.

Step 4 Drill a 8mm hole through the reinforcing lip at this point on each side.

Step 5 Empty the tub.

Step 6 Cut a broom handle to fit across on top of the lip.

Step 7 Slide it inside a piece of 25mm poly pipe the same length.

Step 8 Drill an 8mm hole in each end to line up with the previous holes drilled at the bulge point as shown in Figure 27.2 below.

Step 9 Fix each end using large cable ties. Trim the ties.

This strengthening spar can also be used as a support for the biofilter (Figure 27.3).


Mick O'Connor

Figure 27.1 Materials


Mick O'Connor

Figure 27.2 Step 8

Mick O'Connor

Figure 27.3 Finished tank

Other grow out tanks
Ballina SHS

Figure 28.3 Finished tank

Water quality

Water quality is a term used to describe the condition of water and its suitability for a particular use. Eg drinking, swimming, fishing, irrigation or in our case, as a medium for growing plants and animals.

Water quality describes the chemical, physical, and biological characteristics of water. Specific characteristics used to provide a objective measure of the water's quality are the:

- Amount of particulate matter suspended in the water (turbidity)
- Concentration of salt (salinity)
- Concentration of dissolved oxygen (DO)
- Concentrations of plant nutrients nitrogen and phosphorus
- Concentrations of aquatic waste eg ammonia
- Level of fecal coliform bacteria from human and animal wastes

Pesticides, herbicides, heavy metals and other chemicals may also be measured to describe the water quality and assess its suitability for particular uses.

Projects

This following projects are designed to provide some insight into how water quality is assessed

Project 2.1 Making a turbidity tube

Project 2.2 Testing water for turbidity

Project 2.3 Making a Secchi Disc

Project 2.4 Testing water for total dissolved solids

Project 2.5 Making a salinity hydrometer

Project 2.6 Testing water for Nitrates

Project 2.7 Testing water for Nitrites

Project 2.8 Testing water for Ammonia

Project 2.9 Testing water for Dissolved oxygen

Project 2.10 Testing water for pH

Waterwatch

Waterwatch is a national community water quality monitoring network that encourages all Australians to become involved and active in the protection and management of their waterways and catchments.

The Waterwatch network is made up of individuals, community groups and school groups who undertake a variety of biological & habitat assessments and physical & chemical tests to build up a picture of the health of their waterways and catchments.

By monitoring their local waterways over time community members can determine if the health of the waterway and surrounds are improving, declining or being maintained.

If you are interested in joining a Waterwatch group or perhaps starting up your own group the contact details of the facilitator closest to you is available at

waterwatch.org.au

Bob Moffatt

PROJECT 2.1 MAKING A TURBIDITY TUBE

Background

See page 40 - Testing water for turbidity

Materials

- 700mm of 25mm internal diameter acrylic tube
- Small piece of acrylic sheet
- Acrylic glue
- Engraver
- Permanent marker or black paint

Procedure

Making a turbidity tube is fairly easy

Step 1: Remove the drill from a hole saw whose internal cut is the same as the internal diameter of the tube or slightly larger.

Step 2: Hold the acrylic sheet in drill press vice, and using the hole saw cut out a circle that will become the bottom of the tube.

Step 3 : Using fine sandpaper finish the disc to form a perfect fit inside the acrylic tube.

Step 4: Use an engraver to place a 1cm X 1cm cross in the centre of the disc.

Step 5: Paint or colour the cross black with the marker.

Step 6: Using the acrylic glue fix the disc in the bottom of the tube to make a watertight seal.

Step 7: Using the engraver and the marker or paint , place marks with the units on the outside of the tube at the distances from the bottom as shown in the diagram.

Notes:

1. NTU scale is not linear and estimations between the marks may be inaccurate.
2. If the 700mm tube is too long for convenient packing/ storage/transport, it can be cut in half and a tight fitting male and female pull apart joint can be made at the cut using a lathe as shown in Figure 39.3.

   The joint should be tight to avoid parting as the tube is filled with water.

Commercial availability

You can purchase a commercially made turbidity tube from your State Water Watch Co-ordinator

Google waterwatch

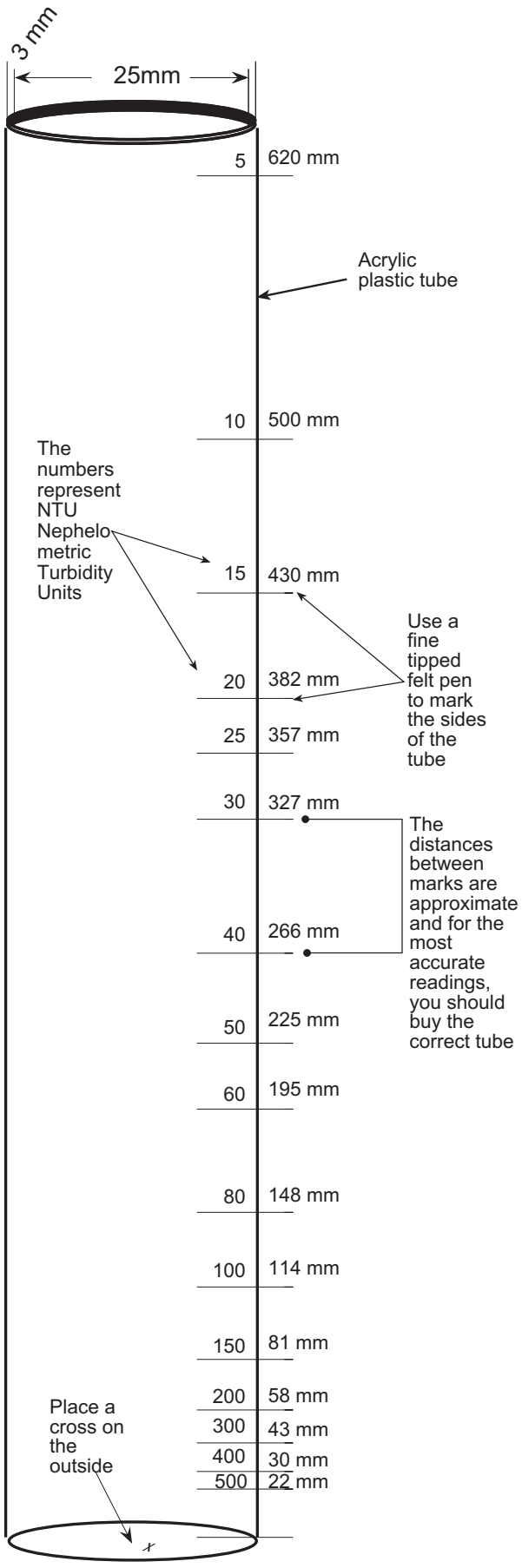
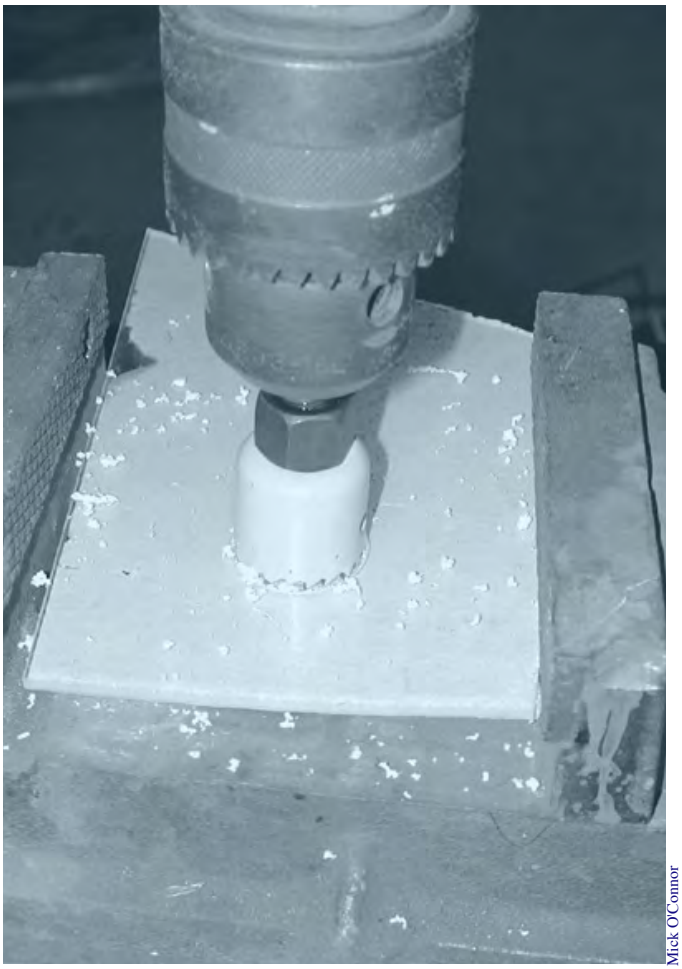


Figure 30.1 Turbidity tune measurements
Illustration Bob Moffatt

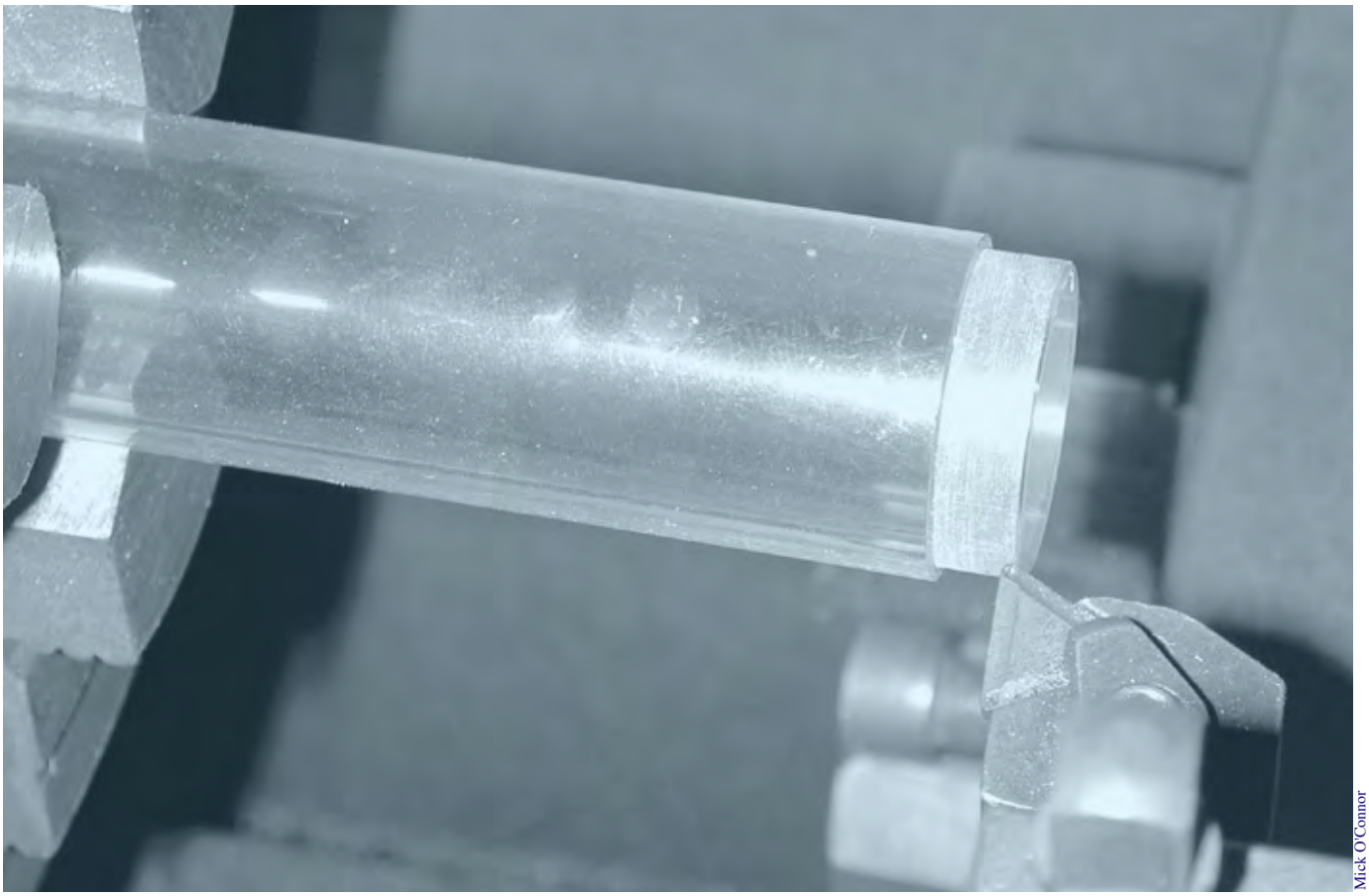


Mick O'Connor

Figure 31.1 Cutting out the acrylic bottom for the turbidity tube

Mick O'Connor

Figure 31.2 Finished turbidity tube



Mick O'Connor

Figure 31.3 Machining the male end of the turbidity tube that has been cut in half to reduce storage space and make it easy to carry. This machining is done on a lathe.

PROJECT 2.2 TESTING WATER FOR TURBIDITY

Background

'Murkiness' is the first and most obvious feature people notice about any body of water.

Clear water is seen as a 'good' feature while muddiness is seen as 'bad' . To avoid confusion we should be aware of the terms:

- Turbidity is the measure of the light scattering properties of water.
  - The light is scattered by suspended (held up) matter in the water and so turbidity depends on the amount, size and composition of this suspended matter - things like clay, silt, organic matter, plankton and other microscopic organisms.
  - Turbidity is measured with a turbidity tube in nephelometric turbidity units (NTU).
- Suspended solids refers to the mass of un-dissolved solids suspended in the water.
  - Suspended solids are filtered dried and weighed and recorded as milligrams of solids per litre of water (mg/L).
- Clarity is a measure of how clear or transparent the water is.
  - Clarity is measured using a Secchi disc and is recorded as centimetres (cm).
  - It depends on both colour and light scattering and is usually referred to as visibility by snorkellers and SCUBA divers.

You must decide on which one you will measure and that will depend upon the aquatic environment you are studying.

- For riverine systems, turbidity or suspended solids are the most appropriate features to measure
- For wetlands, estuaries and marine systems: clarity, measured using a Secchi disk, is the most appropriate feature to measure. In this case we are measuring turbidity.

High turbidity does not make water 'look good'.

- The high levels of suspended solids causing it can prevent light penetration needed for aquatic plant growth, can interfere with fish breeding, can smother habitats.
- High turbidity levels can clog gills in fish and macroinvertebrates, affecting their growth and survival.
- High levels can make it difficult for sight-feeding predators, such as bass, pike, and trout to find their food, while other species like carp, which do not depend on sight to feed, are able to survive in the murky conditions.

Materials

- Turbidity tube

Procedure

Step 1. In normal sunlight slowly fill the turbidity tube while looking down into it vertically until the cross on the bottom disappears.

Step 2. Note the reading on the tube at the water level.

Step 3. Record the NTU value.

Mick O'Connor

Figure 33.1 Testing water for turbidity

PROJECT 2.3 MAKING A SECCHI DISC

Background

Water quality can be measured using a secchi disc.

Clarity is a measure of how transparent the water is. It depends on colour, the amount of light as well as the presence of suspended matter.

As water clarity decreases, these things can happen:

- Water loses its ability to support a diversity of aquatic organisms because the light penetration is low.
- Waters become warmer as suspended particles absorb heat from the sun. Warmer water reduces the amount of dissolved oxygen which can affect fish and plant populations.
- Less light can penetrate water depths which effects photosynthesis.
- The suspended particles can sink and cover such things as fish eggs, burrows of crabs or prawns in larval or sub adult stages.
- In areas where coral reefs are close to land, sediment can the corals causing death and subsequent loss of habitats for reef creatures.

A Secchi, or visibility, disc is used to measure the transparency of the water column.

- The disc is 20 cm in diameter with two white and two black quarters. It is lowered into he water until the observer cannot distinguish between the black and white sections.
- The depth is measured in centimetres and gives the 'visibility distance'. It is a convenient absolute measurement and allows comparison between water columns.

Materials

- 20 litre plastic paint drum lid
- Black permanent marker pen
- Cup hook
- Lead weight
- Plastic tape measure
- Drill and suitable bit

Procedure

Step 1 Using a compass mark out a 20cm diameter circle on the lid. On many lids one of the moulding marks is the correct size or so close to it that it can be followed.

Step 2 Carefully cut the circle out with a pair of tin snips.

Step 3 Divide the circle into four quarters.

Step 4 Using the black permanent marker colour in two opposing quarters black as shown.

Step 5 Place the lead weight on the underside. Glue it in the centre of the disc with silicon.

Step 6 Using a suitable drill and drill bit drill a hole through the centre of the disc into the lead.

Step 7 Screw the cuphook into this hole.

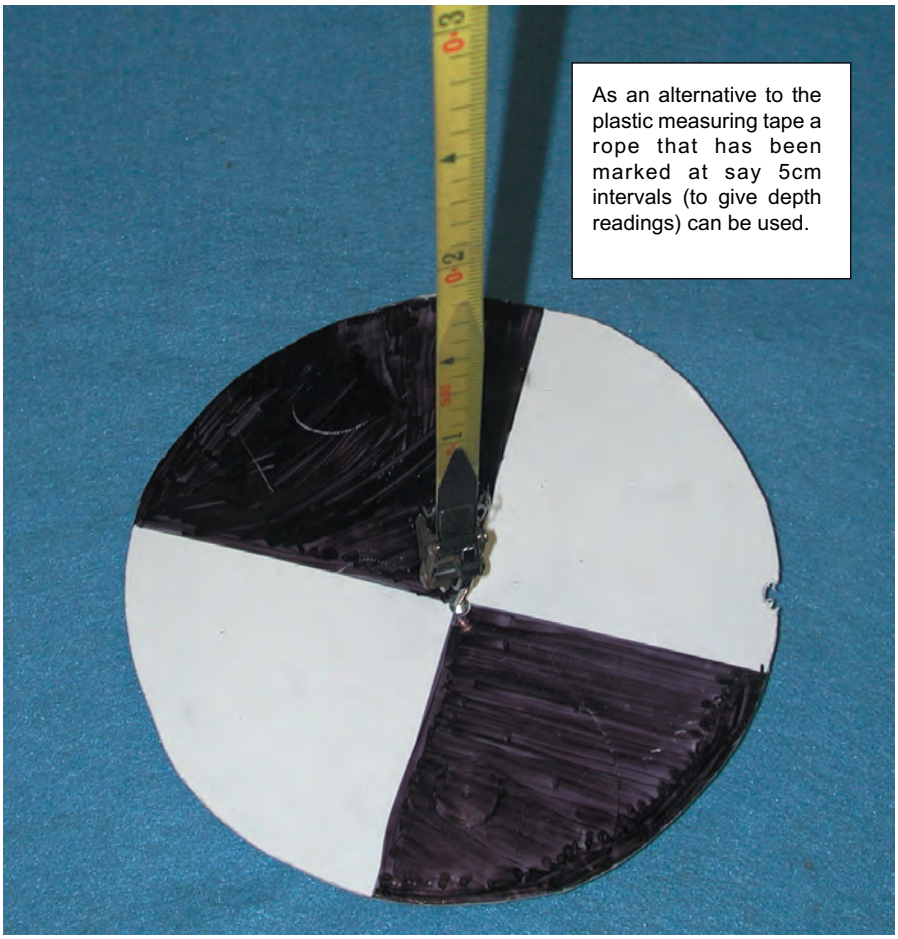
Step 8 Attach the tape to the cuphook.

Mick O'Connor

Figure 34.1 Screwing the cup hook into through he disk into the lead weights

Mick O'Connor

Figure 35.2 Materials



As an alternative to the plastic measuring tape a rope that has been marked at say 5cm intervals (to give depth readings) can be used.

Mick O'Connor

Figure 35.3 Finished secchi disk

PROJECT 2.4 TESTING WATER FOR TOTAL DISSOLVED SOLIDS

Background

Total dissolved solids (TDS) and Total suspended solids (TSS) are tests that can be used to help determine water quality.

Total Dissolved Solids (TDS) describes anything in the water that you cannot see.

- It is defined as "the combined content of all inorganic and organic substances contained in a liquid that are present in a molecular, ionized or microgranular suspended form."
- It will pass through a 2 micrometer filter.

Total Suspended Solids (TSS) will not pass trough a 2 micrometer filter and can be separated dried, weighed and expressed as a weight: volume ratio or concentration.

- These small solids are too small to see. They are usually colloids and stay in suspension.

Measurement

Two methods to measure total dissolved solids are:

a. Gravimetry

b. Conductivity

Gravimetric

Gravimetric methods involve evaporating the water from a sample to leave a residue that can subsequently be weighed with a precision analytical balance.

- It is the most accurate method although time-consuming.
- It is accurate where the solids are ionic or high melting point organics but inaccurate if low boiling point organic molecules, which will evaporate, are present in the water sample.

Conductivity

Electrical conductivity of water is directly related to the concentration of soluble ions in the water.

- Electrical conductivity is measured with conventional conductivity meter that has been standardised with laboratory TDS measurements.
- It is accurate to within 10%.

Gravimetric measurement

To measure the total solids simply collect a sample of water and boil off all the water and weigh what remains.

Step 1 Filter the water through a 2 micrometer filter to remove any suspended solids.

Step 2 To measure Total Dissolved Solids boil off the water from what you have filtered and weigh what remains. To do this you will need a good balance that weighs to 1/1000 of a gram.

- This is called a milligram balance and is very expensive.
- Usually the science section will have one such balance but if not you will need to use plan B.
- Here you just use a larger volume and divide by 10)

Step 3 Collect 100 mLs of water sample (1000mLs if you are going to use plan B).

Step 4 Take a 300 mL (1.5l) beaker and dry it for one hour in a 103^{0}C oven.

- Remove the beaker with tongs, allow it to cool and weigh it to the nearest .0001g (0.01g).
- Record the weight as W1.

Step 5 Add the sample (100 or 1000mLs) to the beaker.

Make sure all materials have been transferred.

To do this you use a wash bottle containing distilled water and squirt water into the bottle so that the sample bottle is clean.

Step 6 Evaporate the sample, dry the beaker overnight and the resulting residue in the 103^{0}C oven.

- Don't touch the beaker with you hands or anything that may give a false reading.
- Now weigh the beaker and record as W2.

Step 7 Subtract W2 - W1, multiply by 1,000,000 and divide by the number of mLs in the original sample to give the ppm.

TDS meter measurement

Step 1 Check that the battery and conductivity probes are in good order and clean.

Step 2 Place the probes in the water sample and depress the 'on' button.

Step 3 Keep the button depressed and read the TDS vale on the digital display.

Commercial availability

Your State Water Watch Co-ordinator will have a supplier list of where you can buy TDS meters

Just google *waterwatch*

Mick O'Connor

Figure 37.1 Evaporating water to recover the dissolved solids

Mick O'Connor

Figure 37.2 Recovered solids are weighted on a balance

Mick O'Connor

Figure 37.3 TDS Meter

Results table

Sample	W1	W2	ppm

Bob Moffatt

PROJECT 2.5 MAKING A SALINITY HYDROMETER

Background

A hydrometer is an instrument used to measure the specific gravity (like density) of a liquid or solution.

Hydrometers work on Archemedes Principle and can be made from anything that floats vertically.

- The denser the liquid, the more upthrust from the liquid displaced by the hydrometer and the higher it will float.

Hydrometers can be used to obtain very accurate measurements of salt concentration in water samples.

- Simple hydrometers can be used to determine salt concentrations of solutions in g/L.

Materials

- Plastic transfer pipette (specifically non graduated narrow stem polyethylene transfer pipette 15cm which can draw of 3.5 ml (ie suck up) Product code: A1503 Supplier: Livingstone International School Supplies (see Page 120)
- Matches/lighter
- Fine dry sand
- Standard salt solutions 35g/L 30g/L 25g/L 20g/L 15g/L 10g/L 5g/L in 500mL measuring cylinders
- Distilled water in large beaker (4 L)
- Curved pointed tweezers
- Fine permanent marker pen
- Salt
- 500mL volumetric flask

Procedure

Step 1 Two thirds fill the bulb of the pipette with fine dry sand. Use a scooping motion with the end of the pipette through the sand or using a folded paper (see Figure 45.1 opposite).

Step 2 Holding the open end of the pipette between thumb and forefinger, place the pipette into the beaker of distilled water (0g/L salt).

Step 3 Add or remove sand until the pipette floats with about 25mm of the 'stem' above the water line.

Step 4 Remove the pipette and hold the open end over a match flame until it melts. Blow out the match and use it, or the tweezers, to seal the open end of the pipette. (Don't use you fingers – molten plastic burns!!)

Step 5 Check for leaks then place back into the beaker of distilled water.

Step 6 Using the pair of curved pointed tweezers grip the pipette and place it into graduated cylinder with the 0g/L solution.

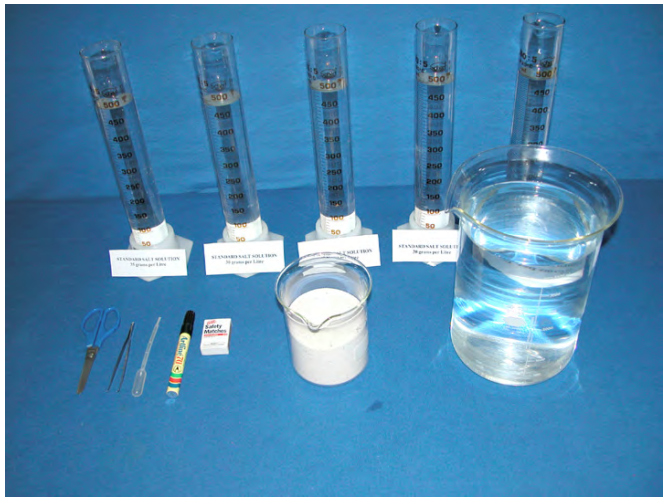
Step 7 Now grip right on the waterline (meniscus) and carefully lift it out taking care not to let the tweezers move.

Step 8 Using the fine tipped permanent marker, place a "0" mark between the tweezers - the waterline.

Step 9 Repeat this procedure with all the salt concentrations, marking carefully the point where the pipette floats in each of 5, 10, 15, 20, 25, 30 and 35 g/L solutions.

Notes:

1. Many organisms live in the sea which has a salt concentration of 35 grams of salt per litre of sea water.
2. Fresh water streams have very little salt in their water, often less than one gram per litre and pure water has no salt at all. Fresh water organisms die if placed in salt water.
3. Estuarine species that live in brackish water are able to tolerate a wide range of salt concentrations.



Mick O'Connor

Figure 38.1 Materials

Making the standard salt solutions

500mL measuring cylinders work well for the graduation process, they are deep and sufficiently wide at the top to allow work with the tweezers, so it makes sense to make up 500ml solutions

You need to make up standard salt solutions having concentrations of 35g/L 30g/L 25g/L 20g/L 15g/L 10g/L 5g/L (see table below).

Step 1 For each concentration halve the quantity of salt.

Step 2 Weigh this out using an accurate balance.

Step 3 Place into a 500ml volumetric flask.

Step 4 Add 300 mL of distilled water to completely dissolve the salt.

Step 5 When the salt is dissolved add distilled water.

These solutions can now be delivered to the measuring cylinders.

Final concentration g/L	Mass of salt required to make 500 mls
35	17.5
30	15
25	12.5
20	10
15	7.5
10	5
5	2.5

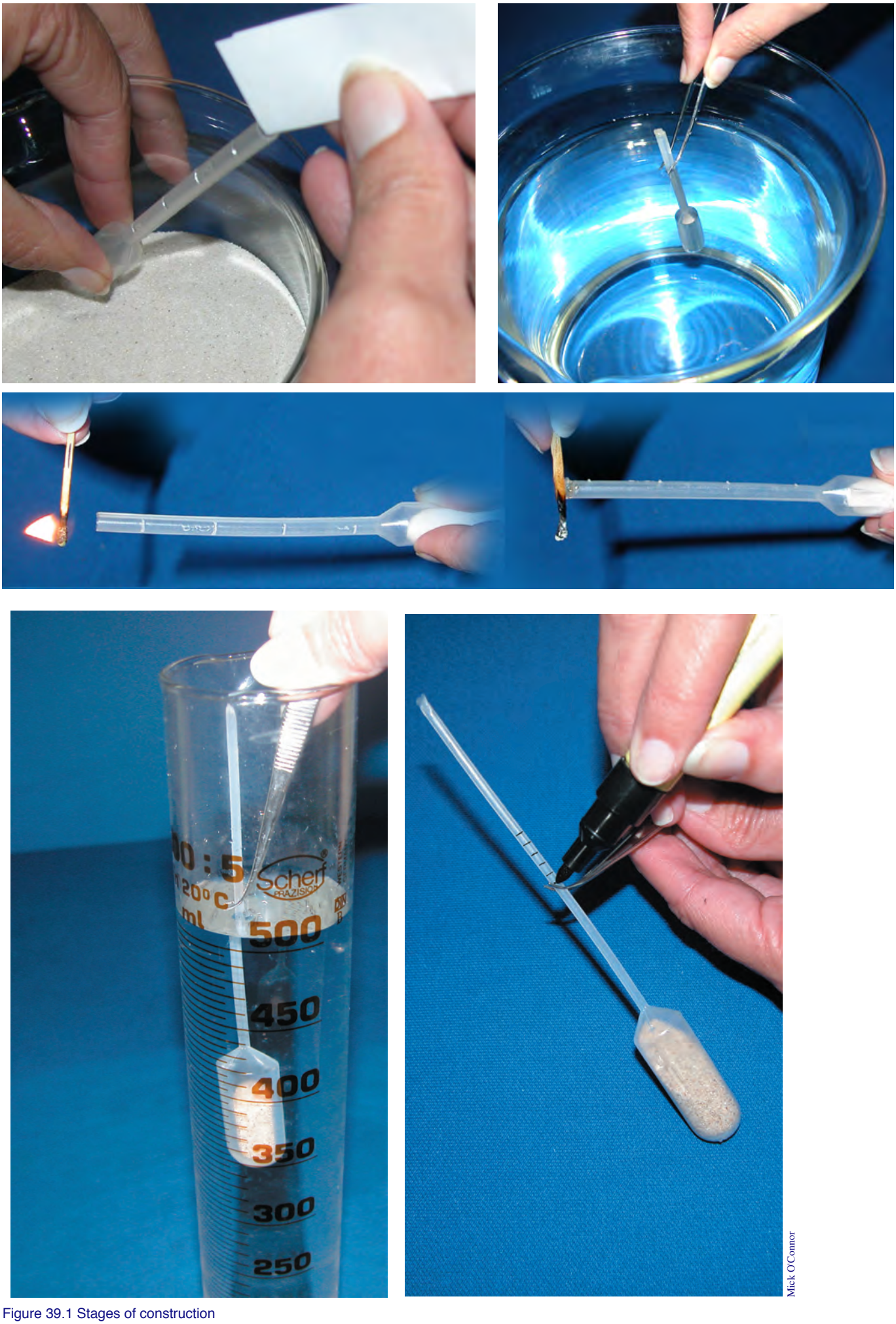


Figure 39.1 Stages of construction

PROJECT 2.6 TESTING WATER FOR NITRATES

Background

Nitrate ions (NO_3^-) are produced in an aquarium as a result of deadly nitrite ions (NO_2^-) being oxidised by bacteria.

The chemical reaction which occurs is shown below:

$$\underset{\text{(Nitrite)}}{NO_2^-} + H_2O \rightarrow \underset{\text{(Nitrate)}}{NO_3^-} + 2H^+$$

The nitrite ions NO_2^- form as bacteria oxidise ammonia excreted by aquatic and marine animals.

Nitrobacter species of bacteria were given credit for this conversion in aquaria however recent research suggests that *Nitrospira sp* may be responsible for nitrite conversion to nitrate. The conversion occurs in the biofilter of the aquarium.

Historically, nitrate was considered harmless to fish as it is far less toxic than ammonia or nitrite. While lethal levels may be in the area of 1000 ppm, the effects of lower levels on long term health of aquatic and marine animals are not well understood.

Certainly the sensitivity of different species to nitrate levels varies, and it is a good idea to avoid any potential long term effects on general health, growth and breeding ability by keeping levels low.

Nitrate levels should be kept below 50 ppm to avoid long-term health effects, but levels below 25 ppm should be aimed for. Fish which have been aquarium bred for generations are more likely to tolerate nitrates better than wild captured fish.

Natural environment

In a healthy natural aquatic environment there is little or no detectable nitrate. Nitrate accumulates in an aquarium in proportion to the number of stock and is a good indicator of general water quality.

As nitrate levels increase, water quality decreases.

- The quantity of permissible nitrate varies with the particular aquatic life being cultured.
- For systems growing fish, acceptable levels are 20 to 40 ppm.
- Many species of marine fish will accept higher levels of 80 to 100 ppm with little effect on their health.
- It is recommended that when levels climb above 40ppm steps should be taken to lower the nitrate and avoid stress.

For reef systems much lower levels of nitrate are needed. A level of 5ppm or less is optimum. Again, many invertebrates will tolerate higher levels but once these levels climb above 20ppm many of the more sensitive invertebrates will be injured or die.

It has been shown that small quantities of around 5ppm of nitrate are of assistance in culturing marine plants.

The easiest method to lower nitrate is to change water. Regular water changes should ensure that nitrate levels stay within the acceptable limits.

- Recommended minimum water change requirements are 25% per month. If water changes do not keep the nitrate at acceptable levels then your stock load maybe too great for your aquarium system.

Aquasonic

Figure 40.1 Nitrate kit

Denitrification filters can lower nitrate successfully, but this must be closely monitored to avoid dangerous gases that can harm aquarium inhabitants.

Marine plants and algae can use some nitrate as nutrients and are a great way to control nitrates.

Materials

- Aquasonic (TM) Nitrate test kit (Figure 40.1) or similar

Procedure

Step 1 Rinse a clean test tube and it's cap several times in water to be tested.

Step 2 Fill the test tube to the 5, 10 or 20ml mark. When testing 5 or 10ml samples, distilled water has to be added to reach the 20ml mark. The test has to be timed.

Step 3 Add 7 drops of reagent 1.

Step 4 Add 1 tablet of reagent 2, cap and shake vigorously (tablet takes approx. 2 minutes to segment).

Step 5 Open test tube and leave it stand for 5 minutes.

Step 6 Add 7 drops of reagent 3 and 7 drops of reagent 4, cap, and invert several times.

Step 7 Open test tube, wait 3 minutes, then compare with the colour chart. Place the test tube on the white portion of the colour chart, below the closest matching colour. View vertically to compare the test with the colour chart.

Step 8 On completion of the test, rinse clean the test tube and cap.

The water quality monitoring tables on Pages 107-111 allow you to record your data over a number of weeks.

Figure 41.1 Nitrate kit and colour chart (see page 47 for colours)

Bob Moffatt

PROJECT 2.7 TESTING WATER FOR NITRITES

Background

Nitrite ions NO_2^- form as bacteria oxidise ammonia excreted by aquatic and marine animals.

The bacteria responsible for this conversion were always thought to be the *Nitrosomonas species*, but recent research seems to suggest that the *Nitrosococcus species* may be the true ammonia-oxidising bacteria in aquariums.

The concentration of nitrite ions in the aquarium must be zero! Nitrite ions attack the nervous system, the liver, spleen, and kidneys of fish.

Even very low nitrite concentrations (as low as 0.1 ppm) over a prolonged period can cause long term damage to aquatic animals.

Nitrite causes stress for fish at around 0.5 ppm, while levels exceeding 10-20 ppm are usually lethal. Nitrite binds to the pigment haemoglobin in the fishes blood making it useless as a oxygen carrier.

Fish can suffocate even if aeration is good and the dissolved oxygen levels in the tank are high.

Symptoms of nitrite poisoning include gasping for air at the surface and rapid gill movements - symptoms that are often mistaken for a shortage of oxygen.

Nitrites will not accumulate to dangerous levels in a balanced tank and conversely, accumulating nitrite is a sign of an unbalanced tank.

Water changes are essential in diluting nitrites, but only helps if the tank is balanced - frequent water changes will not only effect the short-term nitrite concentration, but all other parameters as well. It may temporarily save your fish....BUT the problem has to be fixed.

In a recirculating filter systems, as the biological filter develops, a nitrite condition occurs where nitrite ions accumulate in solution. If the biological filter is being artificially developed (by chemical or bacterial treatment) then no live stock should be cultured until a nitrite reading of zero is achieved - the time taken for this can be reduced artificially by inducing the growth of *Nitrosococcus spp*.

Materials

- Aquasonic (TM) Nitrite test kit (or similar)

Procedure

Step 1 Rinse a clean test tube several times in the water to be tested.

Step 2 Fill the test tube to the 5 mL mark with the water to be tested.

Step 3 Add 3 drops of Nitrite Reagent A.

Step 4 Add 3 drops of Nitrite Reagent B. Cap the test tube and invert twice.

Step 5 Wait a minimum of 2 minutes, but no longer than 3 minutes, to compare the test with the colour chart. After this time the test will continue to colour, but further colouration is irrelevant.

The water quality monitoring tables on Pages 107-111 allow you to record your data over a number of weeks.

Bob Moffatt

Figure 43.1 Nitrite kit

PROJECT 2.8 TESTING WATER FOR AMMONIA

Background

When carbohydrates are metabolised by animals the end products are carbon dioxide and water.

When proteins are broken down, a very toxic amine group (NH_2) is cut off by a process of 'deamination', before the remaining carbohydrate skeleton is metabolised. Your Chemistry Teacher will explain this to you.

- This nitrogen containing group must be stabilised and must be excreted - commonly as urea, uric acid or ammonia, depending on the organism.

Aquatic organisms usually excrete ammonia

- Ammonia has a small molecular size and high solubility in water, making it an ideal molecule to excrete for aquatic animals.
- It rapidly diffuses from any surface in contact with water and moves away from the organism quickly as it dissolves in the surrounding water.

Fish lose the ammonia mainly through their gills with a small amount excreted through the kidneys. Some fish do make small amounts of urea but it is very small relative to the ammonia they produce. All aquatic invertebrates and laval amphibians excrete ammonia.

The ammonia level in an aquarium containing fish should always be zero (ie, undetectable by conventional test kits).

- Low levels of ammonia (0.1ppm- a level too low to be detected by many kits) can be disastrous for fish.
- Ammonia poisoning will cause haemorrhaging and destruction of mucus membranes, the gills are particularly likely to be damaged, and may appear reddened.
- As with nitrite poisoning and low dissolved oxygen levels, fish under ammonia stress may appear to gasp for air at the surface, and show rapid gill movement.
- Higher levels, of several ppm, will generally kill fish.

Ammonia poisoning

Ammonia poisoning is more likely to occur in the alkaline pH range.

The proportion of ammonia (NH_3) dissolved as gas and the ammonium ion (NH_4+) in solution present in an aquarium depends mainly on pH, and to a lesser extent temperature.

- At alkaline pH, more of the toxic ammonia will be present while at acidic pH, more of the less toxic ammonium ion will be exist in solution.
- Remember that these two will probably be in equilibrium and the ammonium ion may revert to ammonia.

In a well balanced aquarium, ammonia is oxidised by Nitrosomonas and Nitrosococcus bacteria to form nitrites.

Thankfully these bacteria work well and keep the levels low - it is usually human intervention in their work that upsets the balance.

$$NH_3 + 2\,H_2O \rightarrow NO_2^- + 7\,H^+$$

Protein issues

When caring for aquarium animals you should be conscious of protein that can be broken down in their aquaria.

Fish are not the only source of protein and hence ammonia becomes an issue.

Fish waste, uneaten food and decaying plant matter will all contribute to the level of ammonia in the tank.

The secret is to keep enough ammonia-converting bacteria to ensure that ammonia levels do not rise to detectable levels.

Poor maintenance and bad practices can cause ammonia spikes and kill fish. Some of these include:

- Over-feeding produces an excess of protein and as breakdown bacteria deaminate it ammonia is released.
- Filter failure, or lack of maintenance will not remove wastes and or food particles.
- Over-enthusiastic cleaning or disinfecting of 'biological' filter media kills the Nitrosomonas and Nitrosococcus bacteria responsible for the removal of the ammonia
- The addition of a large number of fish at the same time produces an 'overload' of waste - there are not enough bacteria to remove the ammonia.
- Use of medications may kill those bacteria responsible for the conversion of ammonia to nitrite ions.

In these situations it really is the bacteria that cannot physically cope with their increased workload. In time they will reproduce to build their population to cope with the demand.

If fish appear unwell, testing for the presence of ammonia should be a priority.

Materials

- Aquasonic (TM) Ammonia test kit or similar

Procedure

Step 1 Rinse a clean test tube several times in the water to be tested.

Step 2 Fill the tube to the 5mL mark.

Step 3 Add 1 tablet of reagent 1.

Step 4 Add 4 drops of reagent 2 and 4 drops of reagent 3.

Step 5 Shake to dissolve the reagent tablet.

- This takes little more than 1 minute.

Step 6 Now time the test.

- Wait 10 minutes to compare the results with the colour chart on the box.

Step 7 After the test rinse the test tubes with plenty of water.

Notes

1. Ammonia tests in salt water will cause some cloudiness in the test tube due to salt precipitation, but this will not affect the test results.
2. Handle reagents carefully. MSS at http://www.aquasonic.com.au

The water quality monitoring tables on Pages 107-111 allow you to record your data over a number of weeks.

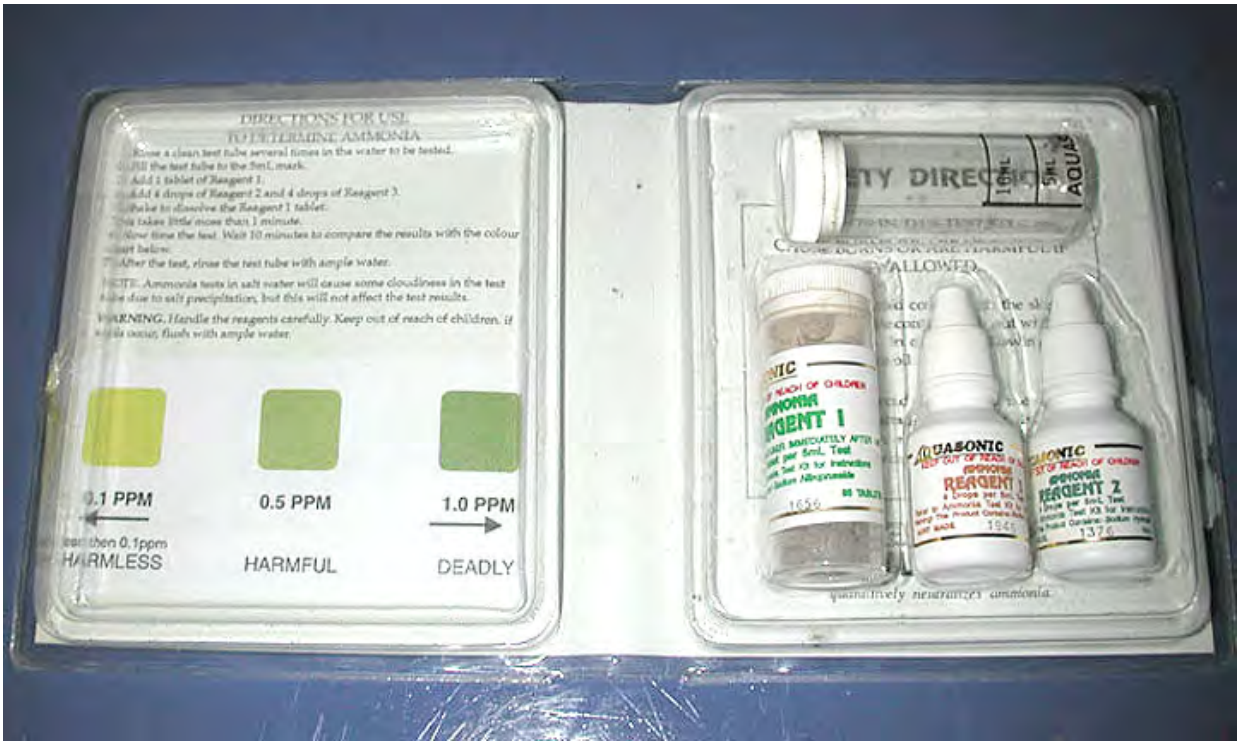


Figure 45.1 Ammonia kit
Bob Moffatt

PROJECT 2.9 DISSOLVED OXYGEN TEST

Background

All animals require oxygen for respiration. Terrestrial animals get oxygen from the air. Aquatic and marine animals must get their oxygen from oxygen that has been dissolved in water.

The amount of air that dissolves is determined by the temperature of the water and is usually small.

Dissolved oxygen in water available to fish is much less than that available to land animals.

This dissolved oxygen can be measured chemically or by meters.

Materials

- HACH dissolved oxygen kit
- Temperature and salinity meters
- Safety goggles and rubber gloves
- Seawater sample
- http://www.vic.waterwatch.aus.net/manual

Procedure

Suggestion - Wear goggles and rubber gloves during the test.

Step 1. Collect a seawater sample in a bucket.

Step 2. Measure the temperature and salinity using the meters supplied.

Step 3. Now submerge the DO bottle in the water. To avoid trapping bubbles in the bottle, incline the bottle slightly and look into the bucket to see that there is no air in the bottle. Gently stopper the bottle while it is still under the water.

Step 4. Tap down the chemicals in sachet number 1 and shake its contents into the DO bottle.

Step 5. Repeat for sachet number 2 in the same bottle.

Step 6. Collect a small amount of sample water in the square mixing bottle and use it to wash out any chemicals in the neck of the DO bottle.

Step 7. Re-stopper the bottle carefully, tipping the small quantity which escapes into the waste bottle.

Step 8. Hold the bottle and stopper, move away from the group and rotate the bottle 3 times to dissolve the chemicals in it.

Step 9. Allow the sample to stand for 5 minutes.

Step 10. Tap down the chemicals in sachet number 3.

- Remove the stopper from the bottle and add the chemicals from the sachet.
- Carefully re-stopper the bottle and rotate.
- The floc, (the yellow–brown), should dissolve and a yellow colour will develop if oxygen is present. This yellow solution is called the prepared sample.

Step 11. Fill the plastic tube with the prepared sample then empty the tube into the square mixing bottle.

Bob Moffatt

Figure 46.1 Dissolved oxygen kit in use

Step 12. Open the sodium thiosulphate solution by reading the instructions on the neck of the bottle.

- Add sodium thiosulphate solution drop by drop to the bottle, swirling to mix after each drop.
  Hold the dropper vertically above the bottle and count each drop as it is added.
- Continue to add drops until the sample changes from yellow to colourless.
- Record the number of drops on your result sheet.
- Each drop is equal to 1 mg/L dissolved oxygen.
  You may like to add 1 drop more after the yellow colour goes clear.
  If there is no further clearing after adding this last drop, don't count it!

Step 13. Use the graph over on the facing page to establish the maximum oxygen solubility.

- Locate the level of salinity on the left-hand scale, the temperature on the right-hand scale and draw a line connecting these 2 points.
- Record the value of maximum solubility from the centre line.

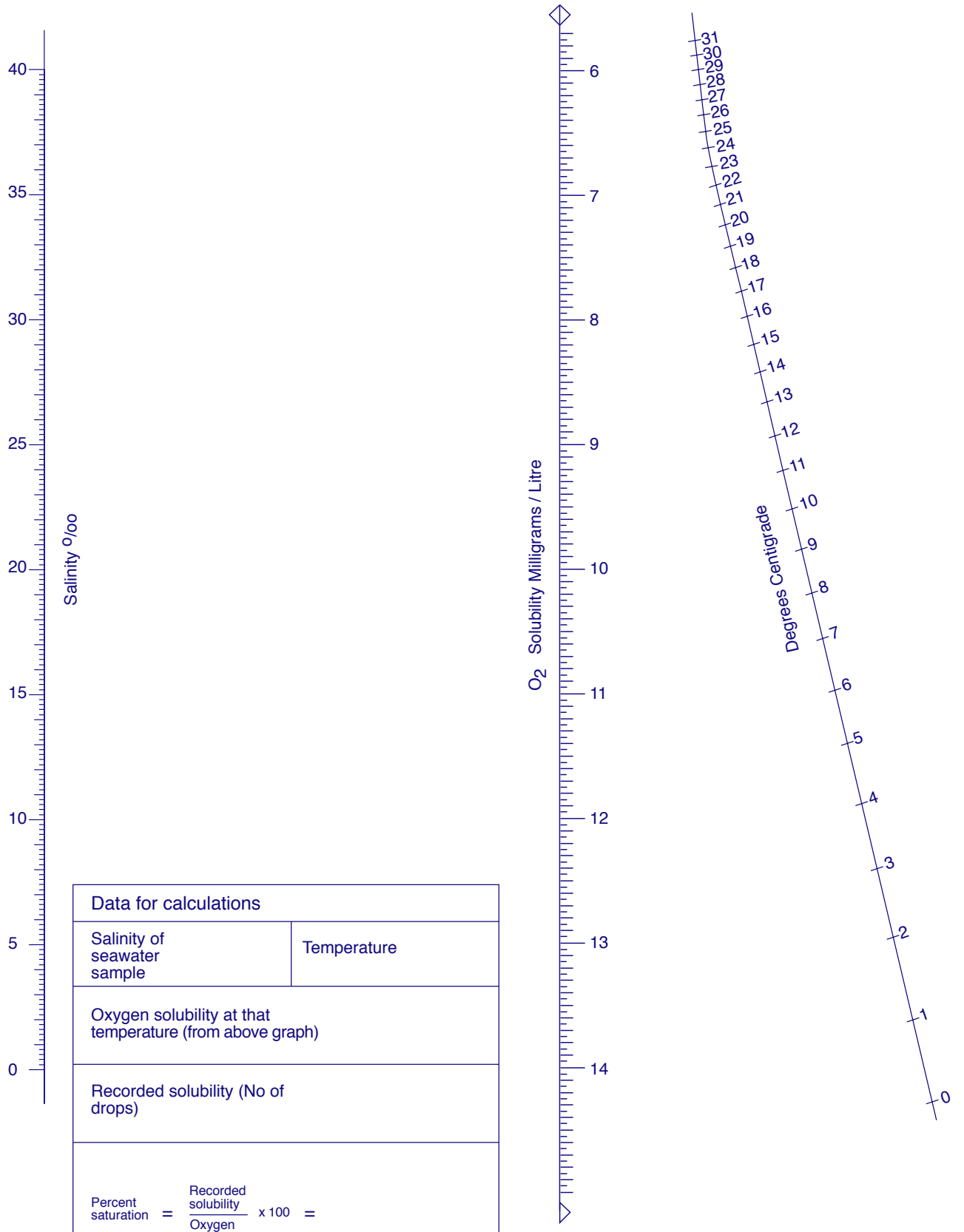
Step 14. Divide measured dissolved oxygen value by this value and multiply by 100.

- Record the value as a per cent saturation.

Discussion

1. State the percentage saturation of the sample measured.
2. List some of the factors that may cause the % saturation to fall.
3. Explain the importance of high dissolved oxygen levels to marine and aquatic organisms.

After Whipple and Whipple (1911) based on measurements by Fax (1909) in American Journal of Oceanography



Data for calculations	
Salinity of seawater sample	Temperature
Oxygen solubility at that temperature (from above graph)	
Recorded solubility (No of drops)	
Percent saturation = Recorded solubility / Oxygen solubility at that temperature x 100 =	

The water quality monitoring tables on Pages 107-111 allow you to record your data over a number of weeks.

Figure 47.1 Dissolved oxygen conversion table and graph
Illustration Bob Moffatt

PROJECT 2.10 TESTING WATER FOR pH

Freshwater pH background

pH is the measurement of the hydrogen ion concentration of a given sample of water.

From this measurement it can be determine whether the water is acid, neutral or alkaline.

- The scale extends from 0 to 14. A pH between 0 and 7 is termed acid, and a pH between 7 and 14 is termed alkaline or basic (Figure 48.2).
- As fish live in a narrow pH range, an indicator must be selected that works best in that range.
- The indicator in the kit below has a pH range from 6.0 to 7.6 and is therefore suitable for most freshwater aquaria.
- At or below pH 6.0 the test sample would colour yellow At or above pH 7.6 the test sample would colour blue.

pH is not a constant factor and is subject to continual change due to influences, like bacterial breakdown of waste, causing acidity or calcium and magnesium compounds like shells or coral that cause alkalinity.

Under normal aquarium conditions, because of fish and plant waste, the water would drift to acidity.

Materials

- Aquasonic Freshwater pH Kit (see Figure 48.1 below) or similar

Procedure

Step 1 Rinse the clean sample tube several times with the water to be tested.

Step 2 Half fill the tube with water to be tested.

Step 3 Add three drops of indicator solution and swirl.

Step 4 Compare with the colour chart.

Discussion

1. Explain what pH is and give some other examples like the ones shown in Figure 48.2.
2. Explain why narrow range indicators are used to test aquaculture water rather than use universal indicator.
3. Identify at least one more accurate method to test pH (than the use of indicators).
4. Explain what a buffer is
5. Discuss how low pH may affect marine life.
6. List safe methods that could be used to lower pH.

Bob Moffatt

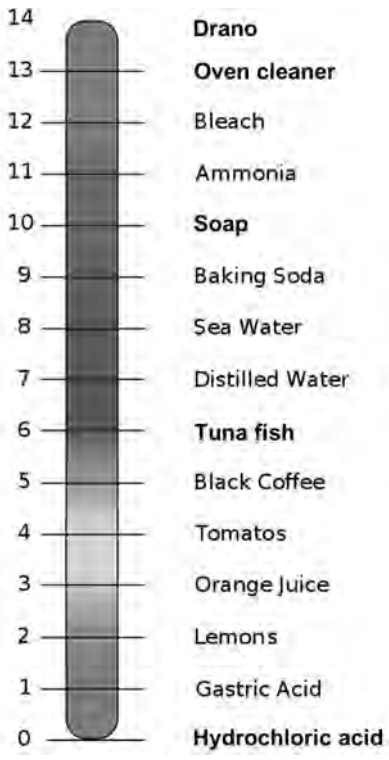


Figure 48.2 Some typical pH values

Background - saltwater pH

The natural pH of the oceans is around 8.1. This is also the recommended pH of a marine aquarium, with levels slightly above or below this acceptable.

The pH of the oceans is buffered by carbonate ions - sea water having a hardness of about 125 ppm carbonate.

Again as marine fish live in narrow pH range an indicator must be selected that works best in sea water's range.

The indicator in this kit has a pH range from 7.5 to 8.7 and is therefore suitable for most salt water aquaria.

Biological filters will tend to produce acids and lower the carbonate ions in solution.

Large quantities of carbon dioxide will also tend to lower the pH. Carbon dioxide will dissolve in water to produce carbonic acid. It is important when measuring pH that the CO_2 in the water be in equilibrium with the CO_2 in the air.

Materials

- Aquasonic Marine pH Kit (see Figure 49.2 below) or similar

Bob Moffatt

Figure 49.2 Marine pH kit

Procedure

Step 1 Rinse the clean sample tube several times with the water to be tested.

Step 2 Fill the tube to the 10 ml mark with water to be tested.

Step 3 Add ten drops of marine indicator solution and swirl.

Step 4 View vertically on the colour chart in natural light and read immediately.

The water quality monitoring tables on Pages 112 - 115 allow you to record your data over a number of weeks.

Tropic marine

Tetra marine

Figure 49.1 Some pH kits

PROJECT 2.11 MAKING A BIOFILTER

Background

Biofilters are 'living water cleaners'.

They are simple, require little maintenance and rely on the action of 'friendly' micro organisms, particularly bacteria, to maintain high water quality.

As aquatic and marine animals eat and digest food, they produce solid faeces and ammonia as waste products.

Excess food, dead organisms and plant material in aquariums also decay to produce ammonia.

- This ammonia is poisonous to animals that excrete it, and must be removed.
- Beneficial bacteria are used to convert the ammonia to nitrites which are in turn converted to nitrates by other bacteria.
- Nitrates in normal levels are harmless to freshwater fish and can be used by aquatic plants living in the aquarium.

A working biofilter will convert toxic ammonia into harmless nitrates, without chemicals or your assistance.

Biofilters mimic what happens in nature. They remove waste material using living organisms.

In natural aquatic and marine ecosystems animals and plants cycle minerals, carbon dioxide and oxygen between them.

Biological filtration uses this same principle although the plants are smaller and beneficial bacteria are used to remove waste.

A good biological filtration system needs

- A place for the bacteria to grow called a substrate.
  - Bacteria will grow on almost anything.
- The secret is to have a substrate that has a large surface area and that is not poisonous to the organisms that will grow on it or to the animals that are in the aquarium.
- Oxygenated water (bacteria need oxygen to reproduce and grow).
  - Any aquarium with good aeration and good water flow over the substrate will provide sufficient oxygen for all organisms to grow and reproduce.
- Ammonia for the bacteria - this is their food.
  - The animals in the aquarium will provide sufficient ammonia, however it must get to the bacteria on the substrate.
- A pump, such as an inexpensive submersible fountain pump, is an ideal way to circulate the water over the substrate.
  - The size of the biofilter will vary with the tank size.

This exercise describes how to make a large biofilter for a 600 litre grow out tank .

It can be downsized using your imagination to what ever size you would like.

Materials

- Two or three stackable crates
- 4 metres of 20 mm PVC pipe
- 2 PVC 20 mm right angle bends and 1 PVC 20mm end cap
- 2 metres of 25 mm clear PVC flexible plastic pipe
- Submersible fountain pump with 20 mm outlet
- Plastic milk/drink bottle tops
- Tools
  - Jig saw
  - Drill
  - 3 mm drill bit
  - 25 mm hole saw
  - Hack saw
  - Tripod and Bunsen burner

Mick O'Connor

Figure 50.1 Materials

Bob Moffatt

Figure 50.2 Completed project

Part A. Making the substrate

One of the best cheapest and easiest to obtain bacterial substrate for schools is plastic drink bottle tops (Figure 51.1) - recruit an army of students to collect them for you.

The plastic is food grade plastic safe for holding human food. No poisonous chemicals are present and no harmful chemicals leach out of this plastic. It is an ideal medium for bacteria to grow on and will not poison the fish.

Step 1 Collect as many plastic bottle tops as you need to fill your crates.

Step 2 Using a Marine Studies class, a heat mat, a tripod and a Bunsen burner, heat up the bottom of one of the tripod legs and burn a neat hole through all the tops. This will prevent water laying in the tops and stagnating (Figure 51.1).

Step 3 Place the tops in a net bag eg onion bag and wash in detergent and rinse five or six times in clean hot water.

Repeat the same number of rinses in cold water.

Step 4 Place in a container of cold water to soak for 48 hours and rinse again in clean cold water.

Part B. Making the biofilter container

Stackable crates are used to hold the bottle tops. Water is delivered to the top crate by a submersible pump. The water is sprayed through the air in the uppermost crate which is cut down to provide good air flow. It is then allowed to percolate down through the bottle tops and return to the tank..

Step 1 Mark out one of the crates as shown in Figure 51.3.

Step 2 Use the 25 mm hole saw to cut two holes in the top container as shown to take the spray bar.

Step 3 Using the jig saw cut the crate (Figure 51.4).

Step 4 Drill as many 3mm holes evenly distributed across the base as you wish to give even and free water flow into the underlying crate (See Figure 58.1 over).

Step 5 Drill the bottoms of all other crates to be used in this filter.

Figure 51.3 Making the biofilter container

Mick O'Connor

Figure 51.1 Top of crate

Mick O'Connor

Part C. Making the spraybar

The spray bar fits into the top cut down stackable crate. Its size depends on the size of the crate.

Step 1 Measure a length of 20mm PVC pipe 50mm longer than the width of the two 25mm holes you have cut in the top crate.

Step 2 Drill a series of 3 mm holes in a line, on this pipe approximately 20mm apart - this will be the spray area.

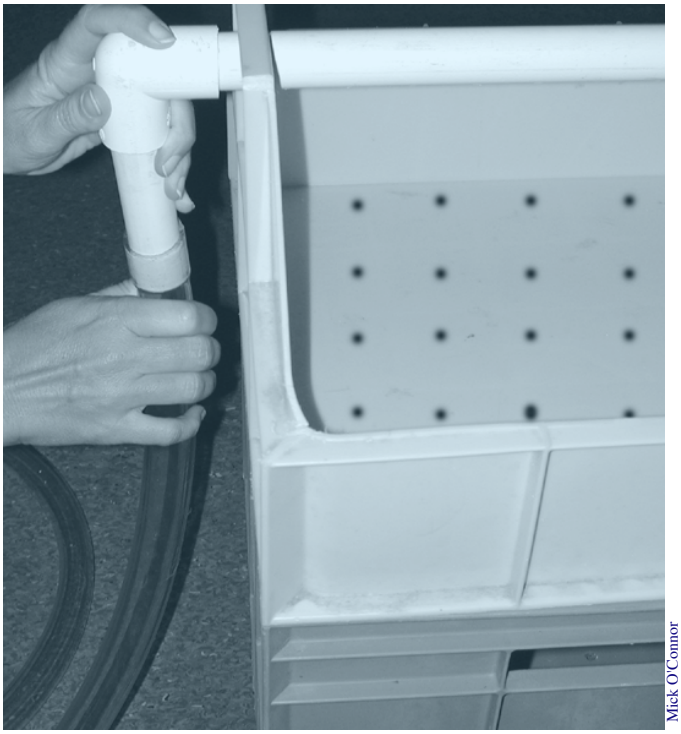
Step 3 Cut a piece of 20mm PVC pipe 100mm long.

Step 4 Assemble the spray bar by placing it in the holes on the crate - push on the end cap on one end, and the elbow on the other so that the holes and elbow point down.

Step 5 Push the 100 mm piece of 20mm pipe into the other end of the elbow ready to take the flexible pipe from the pump.

Mick O'Connor

Figure 52.1 Fitting the spray bar



Mick O'Connor

Figure 52.2 Connecting the delivery pipe

Part D. Plumbing the pump

The submersible pump or its intake should be located as far away from the filter as possible to increase the circulation within the tank. The easiest way to do this is to locate the pump at the opposite end of the tank to the biofilter.

Step 1 Cut 150 mm length of the clear flexible PVC tubing and fit over the pump outlet.

Step 2 Cut a length of 20 mm PVC pipe long enough to go from the pump outlet to directly under the biofilter.

Step 3 Place one end of this pipe into the free end of the clear flexible PVC on the pump outlet, and place the other end into the right angle elbow.

Step 4 Cut a 100 mm lengths of 20 mm PVC pipe, and place it into the 90 degree elbow.

Step 5 Cut a length of 25 mm clear flexible PVC tubing to go from the elbow on the spray bay to the elbow coming from the pump.

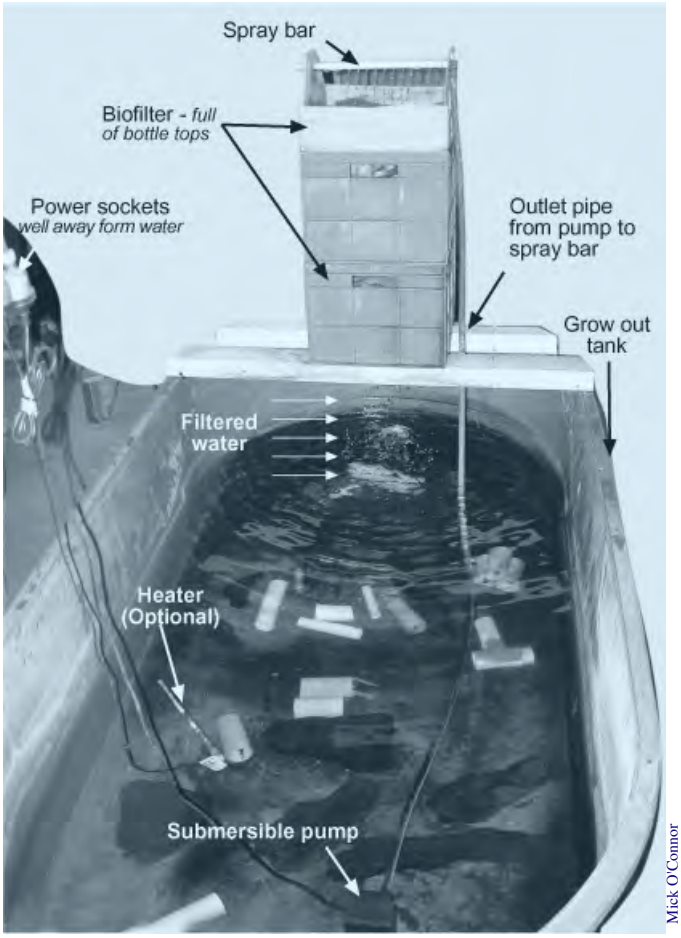
Part E. Operation

(See Figure 52.3 below)

Step 1 Attach the outlet plumbing pipe from the submersible pump to the spray bar.

Step 2 Place the pump in the tank. Make sure all power sockets are secure and cannot fall into the water and switch on.

Step 3 Adjust the spray bar to give even flow and direction into the substrate.



Mick O'Connor

Figure 52.3 Finished project

Figure 53.1 Finished project

Figure 54.1 Finished project

PROJECT 2.12 MAKING A SPONGE FILTER

Background

There are a number of filters that will keep the water clean for you but they come at a cost if you decide to buy manufactured ones.

Most filters are easy to make using common inexpensive materials and a sponge filter is one of the easiest to make and works well.

- The sponge will filter out solids from the water improving its clarity.
- However at the same time it will act as a medium for the growth of microorganism which break down the solids and remove soluble ions such as ammonia and nitrates from the water, ie it will act as a biofilter.

Sponge filters are cheap, effective and easily maintained. Two types are:

- Horizontal
- Vertical

The measurements given here are suited to a 400mm deep tank.

You can vary these to suit your requirements.

- The horizontal filter gives better results than the vertical because it filters bottom water and discharges water away from the filter - this effect can be magnified by placing a ninety degree elbow on top of the airlift.

Any sponge forms a habitat for mainly anaerobic organisms and will act as a biofilter.

- To wash it, squeeze it out on the garden, and wash in a bucket containing a few litres of water taken from the tank.
- Do not sterilise by washing in bleach or detergent- this will not only kill beneficial organisms in the filter but may also transfer harmful chemicals to your tank..
- Sponge filters should be washed out once a month.

Mick O'Connor

Figure 55.1 Horizontal sponge filter

Procedure

To make any sponge filter you must first cut the sponge into blocks.

200mm X 75mm X 75mm blocks are ideal. They can be cut using an electric breadknife. Second task is to cut a cylindrical hole down the middle of the block to take the intake tube that will be connected to the airlift.

Part A Cutting the sponge into blocks

Materials

- Sponge sheet
- Three sided wooden jig to hold the sponge and measure its depth (measurements suited to your requirements)
- Electric breadknife

Procedure

Step 1 Place the sponge in the jig and using the electric breadknife cut the sponge horizontally (Figure 55.2).

Step 2 Using the measure on the jig cut the strip into blocks, as shown in Figure 55.2.

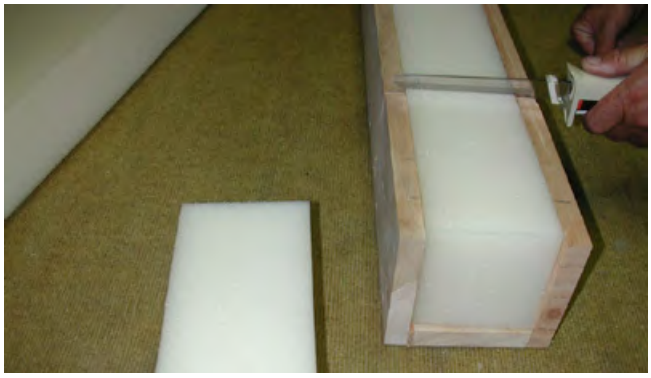


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Figure 55.2 Part A Cutting the sponge into blocks

Part B Boring a centre hole in the block

Step 1 Make a 'sponge hole cutter' by sharpening one end of 300mm length of 20mm PVC pipe and cut serrations into the edge using a hack saw as shown in Figure 56.2.

Step 2 Wet the outside of the hole cutter and using finger and thumb pressure rotate to cut into the sponge.

Step 3 Remove the centre from the hole (Figure 56.2).

Part C Making the horizontal filter

Materials

- 400 mm of 20mm PVC pipe
- Sponge block 200mm X 75mm X 75mm with centre hole stopping 1 cm from the end ie hole does not go right through the end of the sponge block
- Perspex sheet as wide as the sponge block and as long as it + length of the end + 50mm. ie in this case 325mm long and 75mm wide
- 20 mm PVC push in female to screwed male end socket
- 20 mm PVC push in female to screwed female end right angle.
- Tools:
  - drill press,
  - 30mm hole cutter,
  - 12mm drill bit
  - jigsaw
  - heat gun

Procedure (see page opposite)

Step 1: Cut a 30mm hole on the centre of the perspex 25mm from one end.

Step 2: Mark the perspex 150 mm from the same end and heat with the heat gun.

Step 3: While soft bend it at right angles 150 mm from the end.

Step 4 Cut a length of 20mm PVC pipe 200mm long.

- Drill 12mm diameter holes along the length of one 30mm apart right through both sides.
- Then rotate the pipe ninety degrees and drill another series of holes in between the ones you have just drilled.

Step 5 Screw the 20 mm socket into the 20mm elbow.

Step 6 Push in the horizontal and vertical pipes.

Step 7 Place the sponge over the intake pipe.

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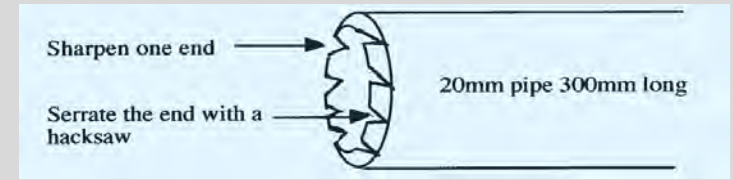
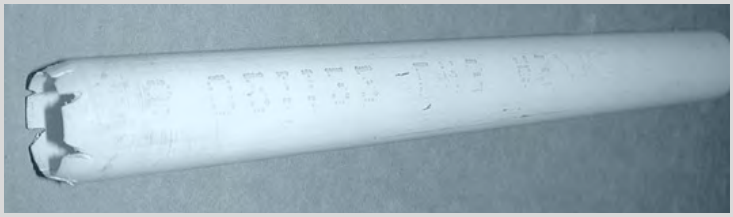
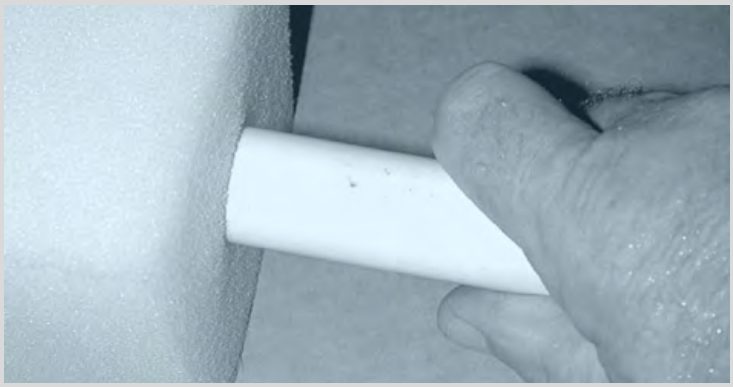
Figure 56.1 Materials for making a horizontal Sponge Filter







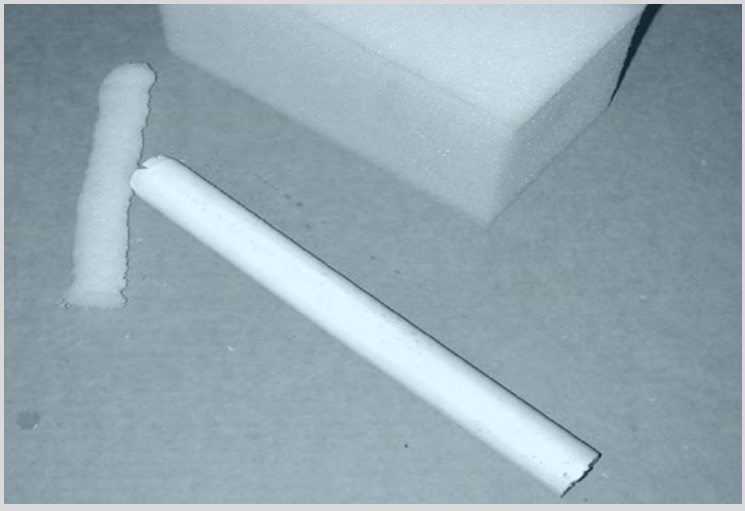


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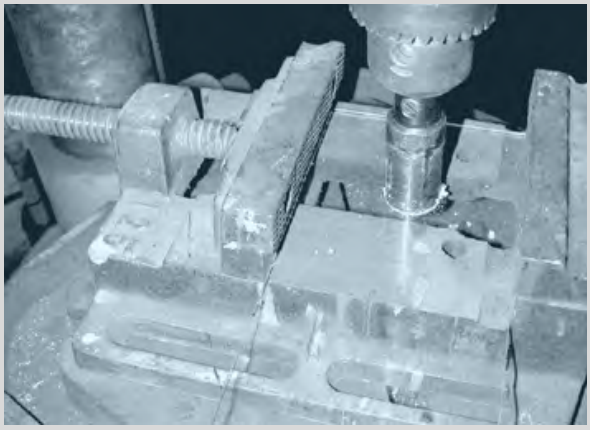
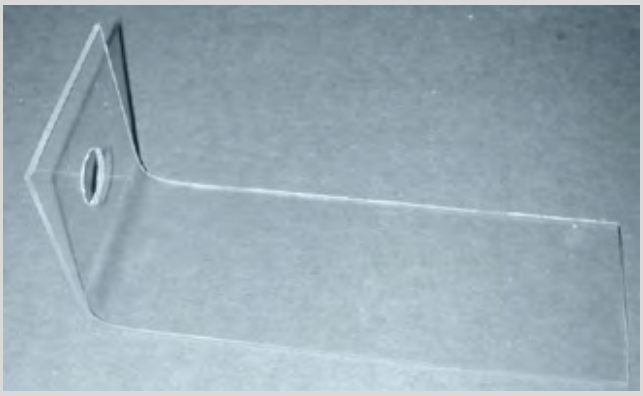
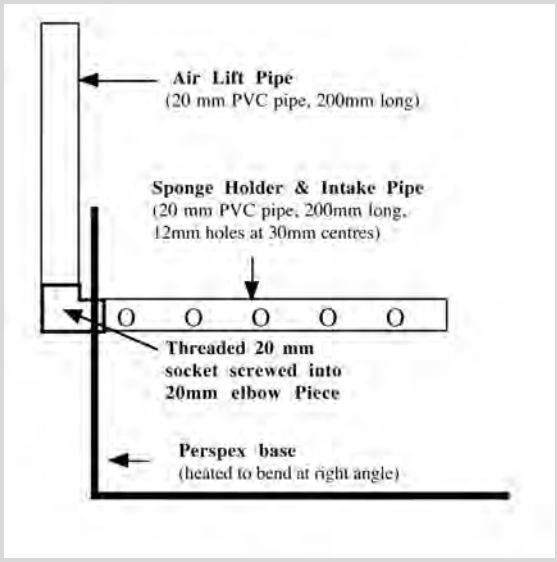
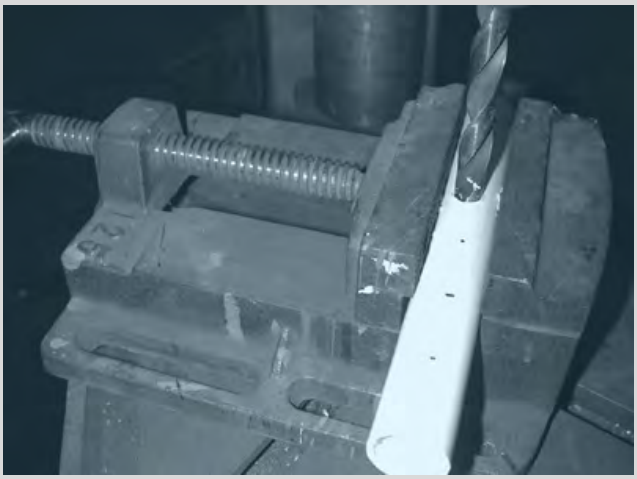
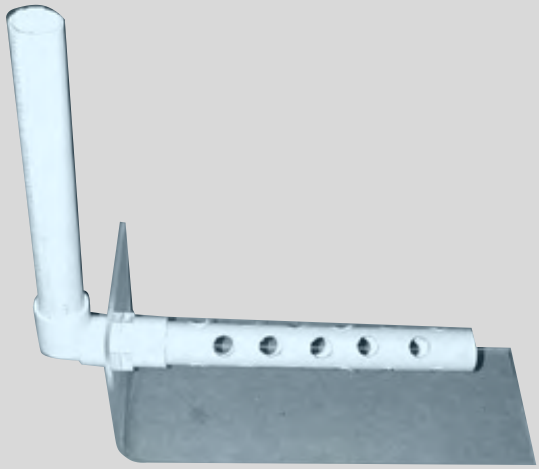
Figure 56.2 Construction methods

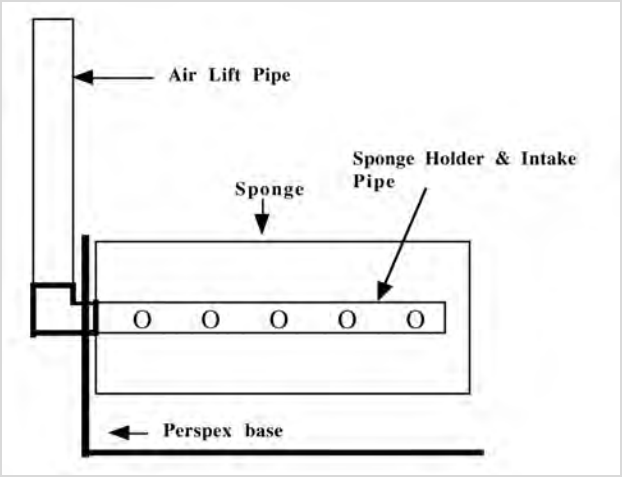












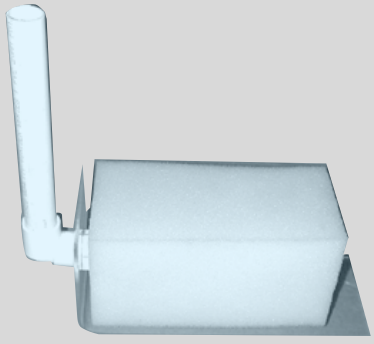


Figure 57.1 Construction methods

Part D: Vertical sponge filter

Materials

- 400 mm of 20mm PVC pipe
- Sponge block 200mmX 75mmX 75mm with centre hole cut all the way through it
- Perspex sheet 150mm long and 75mm wide
- 20 mm PVC push in female to screwed male end socket
- Tools: drill press, 28mm hole cutter, 12mm drill bit, jigsaw

Procedure

Step 1 Cut a 28mm hole on the centre of the perspex.

Step 2 Screw the female/threaded male socket into the hole in the perspex.

Step 3 Make a lift /intake pipe as described previously but 50 mm longer. Do not drill in this 50mm section. It carries air and water out the top of the filter.

Step 4 Push the lift pipe into the socket.

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Figure 58.1 Materials

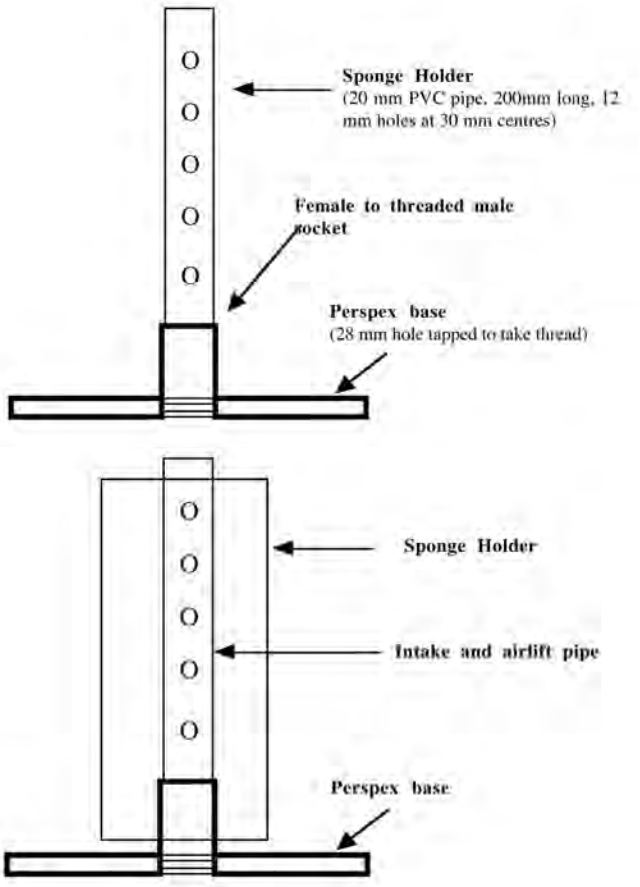
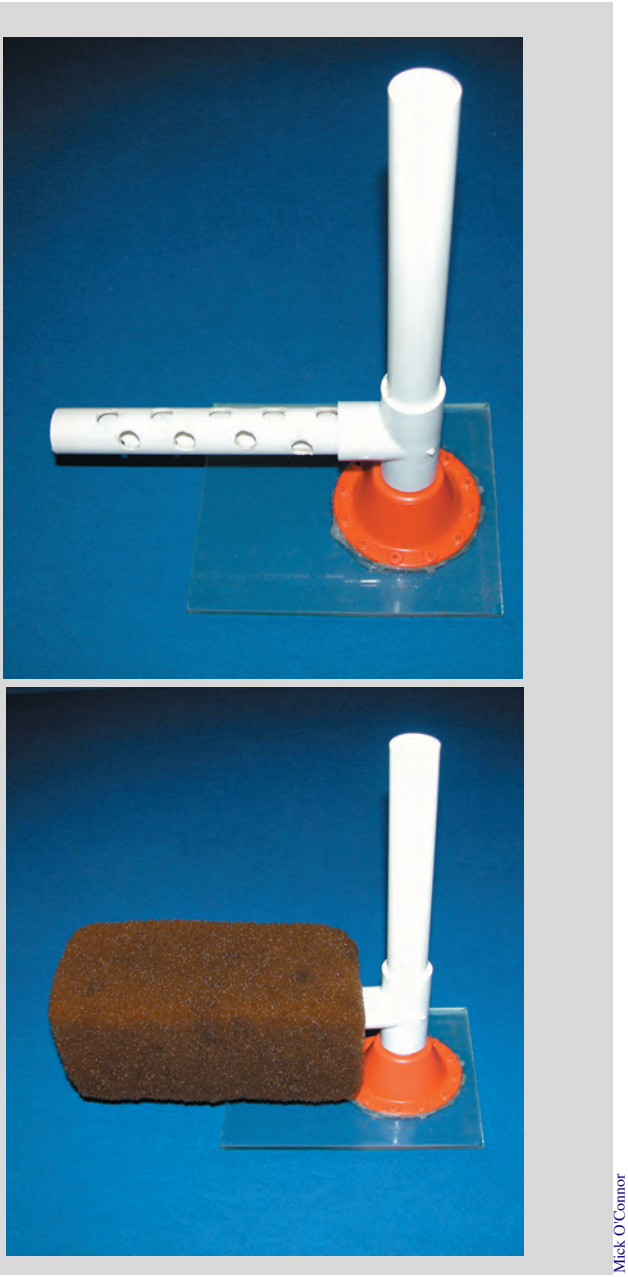


Figure 58.2 Construction details
Illustrations Mick O'Connor



Mick O'Connor

Figure 58.3 Finished alternative stand

Alternative stand *(Figure 58.3)*

A 20 mm conical electrical pipe makes and excellent stand. It is not as attractive as the perspex stand but avoids the need for screw fittings.

The orange stand is glued to a piece of glass to stabilize it and the 20 mm tube pushes straight into the stand. See colour photos page 10.

PROJECT 2.13 MAKING AN UNDERGRAVEL FILTER

Background

Undergravel filters consist of a slotted plate under a bed of gravel in the bottom of an aquarium.

They are used to provide continuous circulation of aquarium water either by introducing a stream of air bubbles into a lift tube or using a powerhead to pump the water from underneath the gravel and up through a lift tube back to the top of the tank - see Figures 59.1 and 59.2.

Air lifts are usually favoured since they circulate the water and at the same time aerate it.

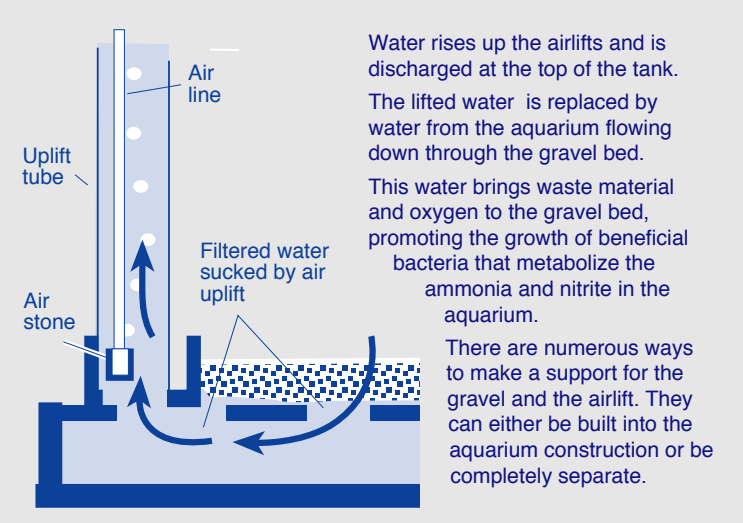
The water must be oxygenated to allow the bacteria on the gravel to reproduce and grow as well as to keep the fish healthy. Proper aeration and good water flow over the gravel containing the bacteria will circulate the ammonia (produced by the fish) over the beneficial bacteria for them to convert to nitrites which will be further converted to harmless nitrates.

Undergravel filters act as mechanical and biological filters. As the water is drawn into the gravel, solid wastes are also drawn in and remain there until they are broken down. Biological filtration occurs as beneficial bacteria colonise the gravel and neutralize ammonia and nitrites as the water passes through the gravel bed.

Undergravel filters can be purchased commercially or can be constructed from simple materials.

Commercial undergravel filters have:

- A hard slotted plate which is raised slightly off the bottom of the tank to support a layer of gravel and allow free water flow under it.
- Air lift riser tubes at each end to take an airstone or water pump (a "powerhead") to move water up the riser tubes.



Bob Moffatt

Figure 59.1 Aquarium air lift

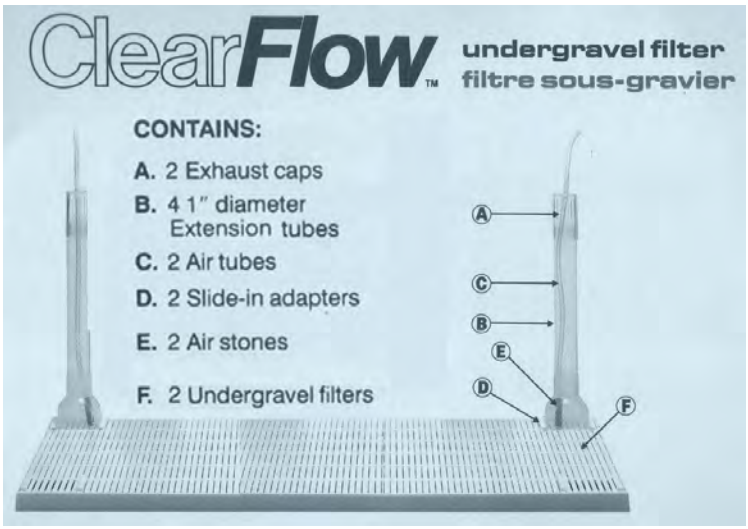


Figure 59.2 Commercial filter
Clearflow

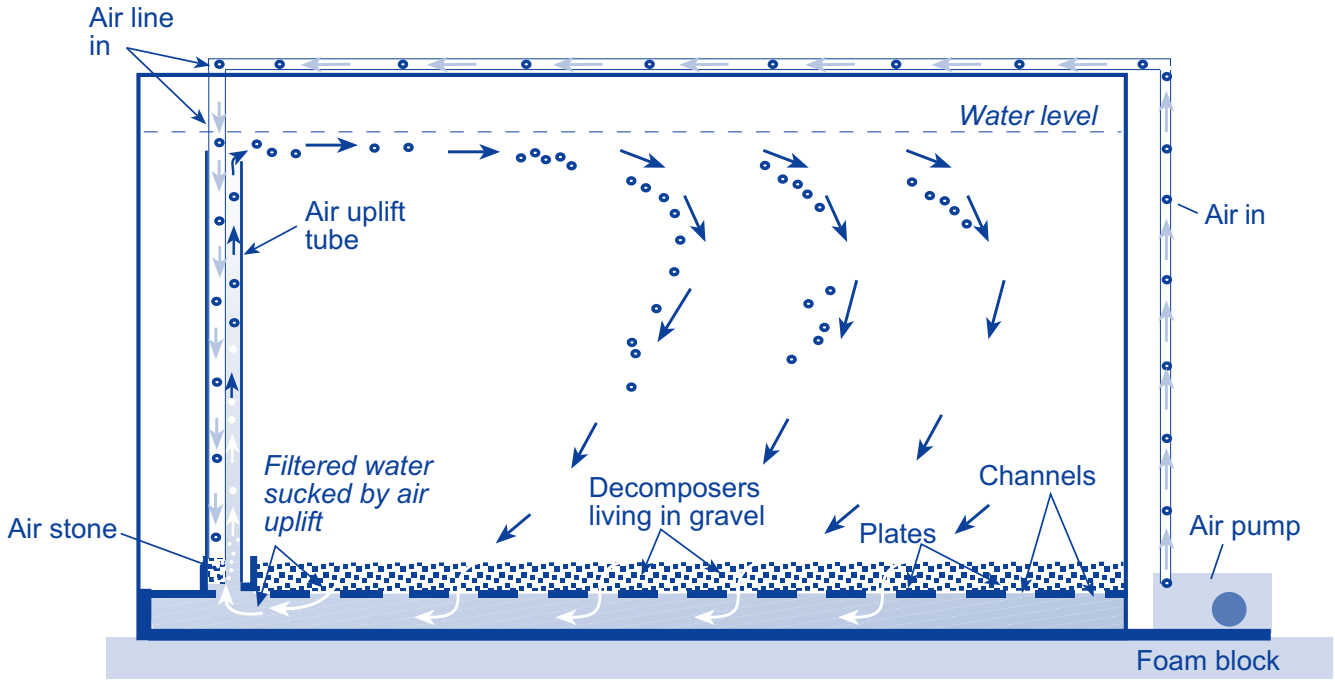


Figure 59.3 How an undergravel filter works
Bob Moffatt

Filtration

Biological filtration organisms are usually the algae, crustacea, worms and bacteria that grow in the under-gravel filter, (see previous page).

- As water passes over them, they take out the soluble animal wastes for food.
- Bacteria, algae and worms can be grown on plastic, gravel, glass beads or anything that will not release into the water substances that will poison your animals.

As fish produce ammonia, the bacteria convert it into nitrite and then into nitrate.

- Nitrate is less harmful to fish than nitrite.
- Plants can use both nitrite and nitrate, however often nitrate builds up, and a 10 per cent water change is needed when nitrate levels get past one part per million.

Another benefit of this filter is that any solids in the water are drawn into the gravel and remain there either until they break down or become part of the living filter.

Cleaning

The undergravel filter needs to be cleaned to remove debris that accumulates in the filter tubes.

The best way to do this is to move the fish, remove the water and gravel to another tank and then wash out the filter.

Remember to keep the gravel because it contains your microorganisms and do not use disinfectants or cleaners because **residual** chemicals can pollute your tank.

Bob Moffatt

Figure 60.1 Aquarium undergravel filters

Bob Moffatt

Figure 60.2 An airlift and gravel bed

Part A: Making a undergravel filter as part of aquarium construction

These are very effective filters that direct the water circulation through a series of channels on the base of the aquarium - these channels are not found in most commercial filters.

Materials

- Glass
- Glass cutter
- Neutral cure silicon glue

Method

Step 1 On a sheet of paper the same size as the aquarium base, draw out the channel pattern as shown in Figure 61.1 below.

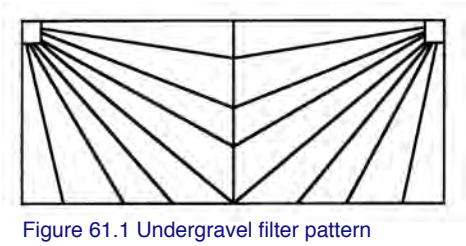


Figure 61.1 Undergravel filter pattern
Bob Moffatt

Step 2 Lay the glass base of the aquarium on this sheet.

Step 3 Cut strips of glass or perspex 25mm wide to match these line lengths.

Step 4 Glue them in position with neutral cure silicon glue as shown in Figure 61.2.

Step 5 When the sides have been glued together, glue a further two vertical strips in the corner to make an airlift 'box'.

Step 6 Cut a piece of shadecloth to cover the base and add 50 -100 mm of gravel or shell grit for the medium on which the bacteria will grow. Shell grit is good in that it acts not only as a medium but as a buffer keeping the pH between 8.2-8.4.

Bob Moffatt

Figure 61.4 Positioning the air lift

Part B Making a undergravel filter as free standing unit

The advantage of 'free stand alone' units is that they can be changed from one aquarium to another.

- The materials and construction of them is limited only by ones imagination.
- 20 mm PVC pipe with elbows and T pieces connecting drilled pipe is the simplest.
- Shade cloth then covers the pipe network and supports the gravel bed.

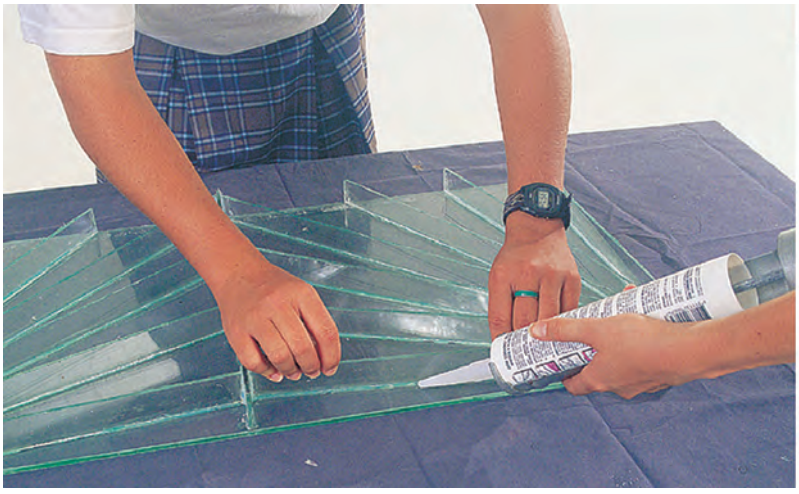
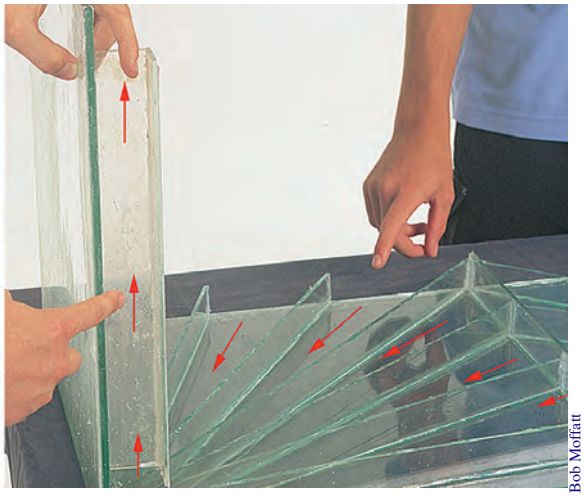


Figure 61.2 First cut out glass strips and glue them into a V pattern



Bob Moffatt

Figure 61.3 Make the air uplift tube by cutting pieces of glass and gluing this onto the side of the tank

PROJECT 2.14 MAKING AN ALGAL SCRUBBER

Background

An algal scrubber is an 'artificial lawn' that removes dissolved phosphate and nitrate ions from aquariums.

- They are often called 'algal turf lawns' and are very simple yet highly effective structures consisting of a screen or mat over which the aquarium water is pumped.
- Algae grow on the screen fertilised by the nutrients in the water.
  - Their growth and hence the removal of nitrates and phosphate from the water is accelerated by shining grow lights or daylight fluorescent lights onto the mat.
  - When the algae builds up on the mat it is simply scrapped off and put on the garden.

A big advantage of algal scrubbers is that they can be located away from the main tank, eg in sumps, on roofs, outside buildings, in other rooms, while performing their cleaning task.

They, like all good water treatment systems, mimic what happens in nature.

Other advantages of algal scrubbers are that they;

- Stop algal growth in tanks.
- Reduce and almost eliminate nitrate and phosphate.
- Increase dissolved oxygen levels and pH.
- Reduce the need for water exchanges.
- Are cheap and effective.
- Work equally well in salt and fresh water.

Ten square centimetres of algal scrubber surface area will keep five litres of water clean at normal fish stocking rates (25cm of fish to 5 litres of water).

There is enormous variation in the design of algal scrubbers only limited by your imagination. It depends:

- If you want to make them a feature, or hide them.
- How much water you want to treat.
- The amount of space you have.

Two basic algal scrubber designs are vertical and horizontal.

Type A Vertical

Materials

- All materials for Making a Biofilter Project 2.11 including spray bar and plumbing.
- Rigid plastic/perspex sheet
- Drill and 6mm drill bit
- Cable ties
- Shade cloth
- Scissors
- Fishing line

Procedure

All that is needed is a spray bar an algal mat and lights - how the bar is supported is up to your imagination.

It can be in a bucket, have attractive glass or Perspex supports above a tank or make the spray bar self supporting with the mat hanging off it.

This procedure describes a method using the frame for a biofilter made in Project 2.11.

Step 1 Follow the procedure for making a biofilter Project 2.11 but instead of drilling the holes in the bottom of the crates remove them completely.

Step 2 Cut large rectangular holes in two sides of the crate to allow light to shine in onto the mat.

Step 3 Cut a frame for the shade cloth from the plastic or Perspex as wide as the spray bar and as long as the crates are deep. Holes can be cut in this sheet to make an open frame to increase aeration or they can be left as a sheet.

Step 4 Drill holes around the edges.

Step 5 Cover the frame with shade cloth and sew it or use small cable ties to complete the mat.

Step 6 Using large cable ties attach the mat to the underside of the spray bar.

Step 7 Align the lights so they shine onto the mat.

Switch on the pump and lights.

Type B Horizontal

This scrubber is essentially the same as the vertical scrubber except it lies horizontal.

The lights are above the matt and only the upper surface operates.

It has that advantage that with the correct selection of the tub, crayfish can be added to graze on the algae. Also they do like the odd nibble on the shade cloth!

Materials

- All materials are the same as for the vertical scrubber.
- A shallow tub is also needed with a drain tube.

Procedure

Step 1 Make the scrubber mat to fit inside your tub.

Step 2 Fit the spray bar across one end of the tube using the sides as support.

Step 3 Attach the mat to the spray bar using cable ties.

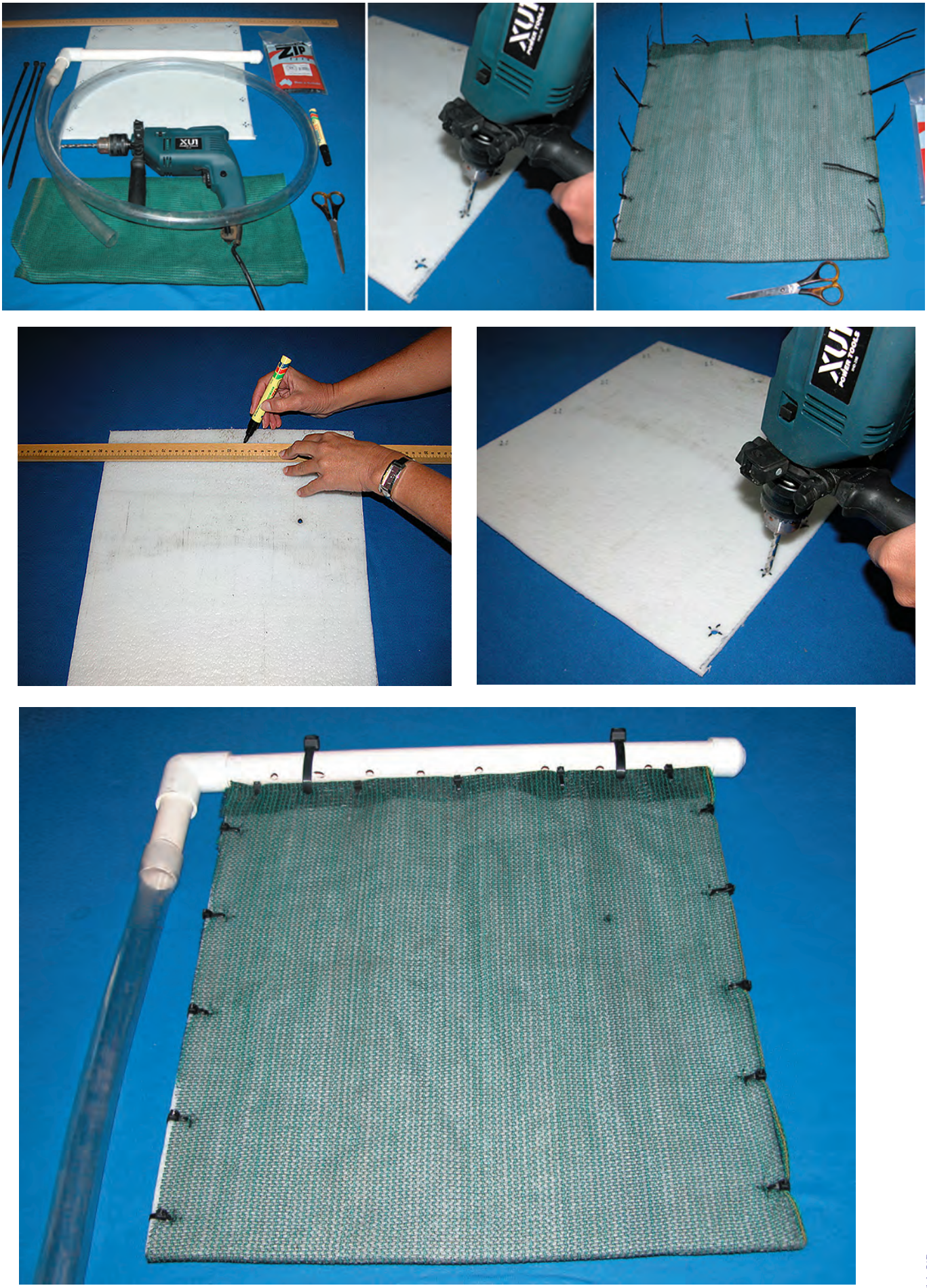
Step 4 Mount the mat so it slopes to the other end of the tub.

Step 5 Drill a hole in the drain end of the tub and attach the drain pipe.

Step 6 Mount the tub above the tank on a slight angle to facilitate drainage and return of water back to the tank.

Step 7 Mount the lights above the tub.

Step 8 Turn on the pump and the lights.



Mick O'Connor

Figure 63.1 Construction methods

PROJECT 2.15 MAKING A WATER SAMPLER

Background

Water samples are taken when testing equipment cannot be carried into the field or when complex chemical or biological tests needing laboratory facilities are required.

Samples are often taken at different depths.

- The samples need to be taken from that depth only.
- It is no good throwing a bottle overboard attached to a rope if you need a sample from 10 metres!

The secret is to keep the bottle closed until the sampling depth is reached, then open the bottle to let the water in, wait until the bottle is full, close the bottle and bring it back to the surface.

This will prevent the sample being contaminated by water entering the bottle on the way down or on the way up.

Materials

- 32mm PVC electrical conduit
- 20mm PVC water pipe
- Rubber stopper to fit inside the 20mm PVC pipe
- Water bottle
- Silicon glue
- Knife or hack saw
- Heat gun
- Drill 3mm and 12mm drill bits

Procedure

Step 1 Cut the 32mm PVC pipe to the length you desire say 3 metres.

Step 2 Cut the 20mm pipe 20cm longer than the 32mm pipe.

Step 3 Take the water bottle and make sure the top is firmly screwed on.

Step 4 Using the heat gun soften about 20 mm of the end of the 32 mm PVC pipe.

Step 5 Holding the water bottle carefully push its lid up inside the softened tube and allow to cool.

Step 6 When the pipe has cooled remove the bottle with its lid. Leaving the lid on the bottle carefully cut the top part of the lid off – this is where water will enter.

Step 7 Wipe the outside of the lid with silicone glue and push it back inside the tube waiting 24 hours for it to set.

Step 8 Using the drill and 12mm bit carefully drill a hole RIGHT THROUGH the 32mm PVC pipe above the top. These are the entry holes for the sample and is taken as the zero point for the sampler.

Step 9 Using a tape measure and a permanent marker, mark the hole as zero and then draw a scale in 25mm increments up the length of the tube highlighting the metre and half metre points.

Operation

Step 1 Attach the empty sample bottle.

Step 2 Place the 20mm tube inside the 32mm tube so that it covers the inlet hole.

Step 3 Lower the bottle to the required depth.

Step 4 Lift the inner 20mm pipe and allow the bottle to fill.

Step 5 Lower the inner pipe to seal the bottle.

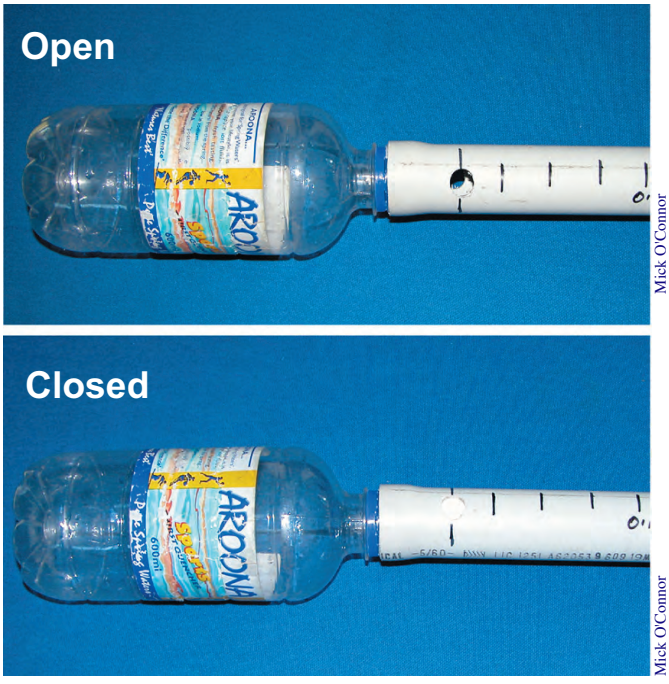
Step 6 Raise the sampler and remove the bottle.

Step 7 Screw on a new bottle and repeat the procedure at other depths.

- Both pipes can be cut into manageable sections and joined on site.
- Keep in mind that the inner 20mm pipe will need to be joined with dowel to maintain its 20 mm diameter.

Mick O'Connor

Figure 64.1 Materials



Mick O'Connor

Figure 64.2 Finished project

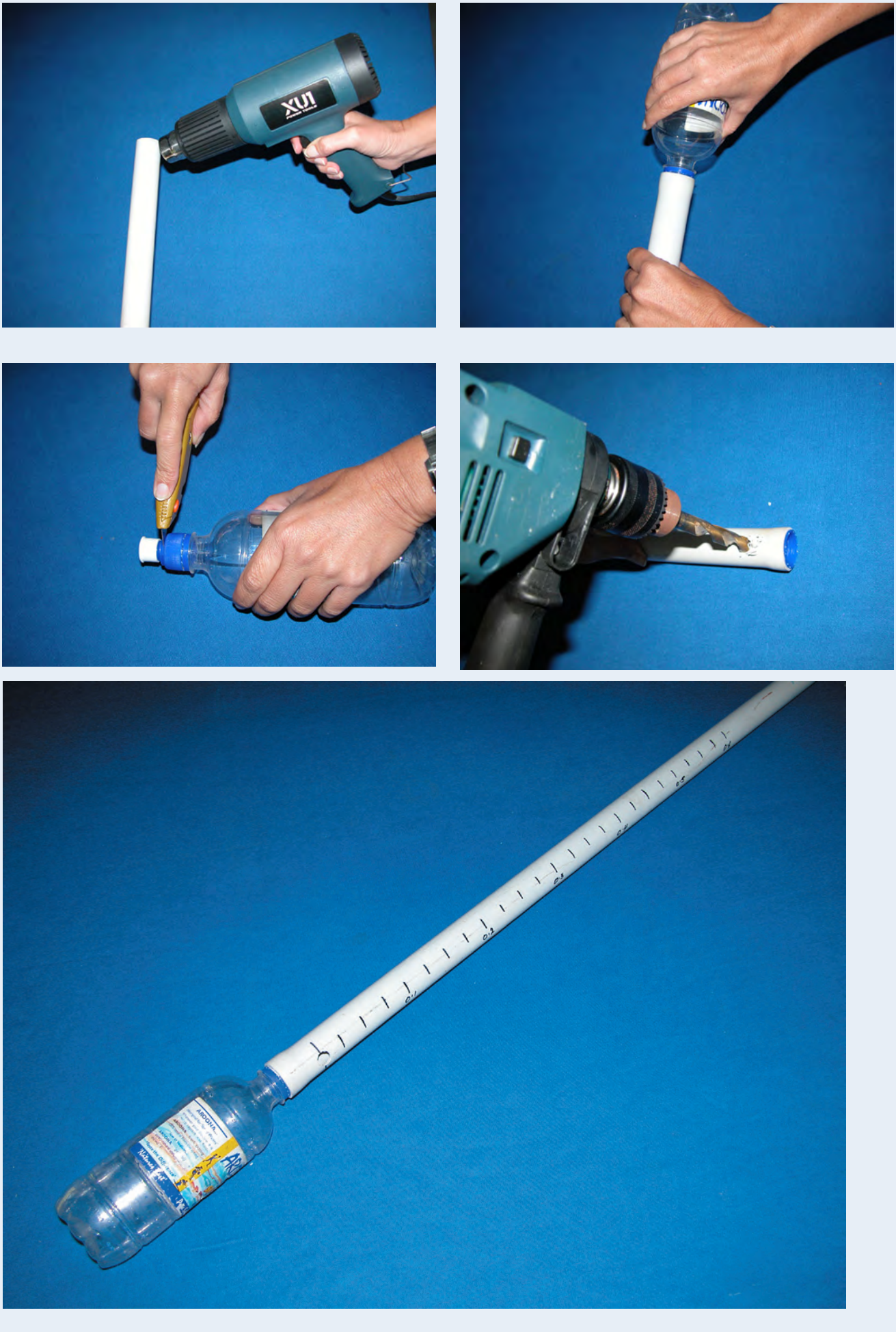


Figure 65.1 Construction methods

PROJECT 2.16 MAKING A SYPHON

Background

The best way to remove sediment from the bottom of aquaculture tanks, including aquariums is to use a syphon as they do not stir up the sediment and hence do not further contaminate the water with suspended solids.

The problem is that many aquaculture tanks and aquaria have a 'medium' on their bottom such as gravel or sand.

- Many use this medium as a 'bed filter' as solid waste and excess food collects on the bed and is drawn into it by the water circulation pattern.
- Anaerobic (and some aerobic) organisms then break this material down overtime.

The filter, and hence water quality, can be assisted by the removal of surface solids using a syphon that removes the waste but not the medium.

A straight syphon tube draws the water too rapidly and will remove both the waste and the base medium. The water velocity on the intake end must be reduced to remove only the less dense waste and leave behind the stones sand etc.

This is most effectively done by securing a 15 cm fitting that is approximately ten times the diameter of the syphon pipe to the inlet end of the syphon. (The garden hose/drink bottle syphon is so effective it should be patented).

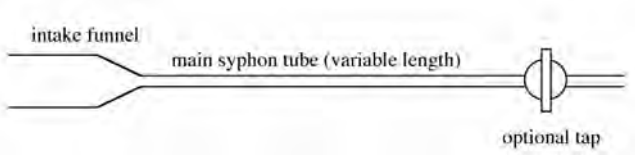


Figure 66.1 Construction
Mick O'Connor

Materials

- Water bottle with leakproof lid (Figure 66.2)
- 13 mm garden hose
- 13 mm tap (optional)

Mick O'Connor

Figure 66.2 Materials

Procedure

Step 1 Remove the outer fitting of the bottle mouth piece exposing the flange.

Step 2 Cut the garden hose to the desired length and push the flange into one end of the garden hose.

Step 3 Carefully cut the bottom out of the bottle making sure that the cut is straight and not jagged.

Step 4 Fit the tap if you are making the delux model.

Note

If the diameter of the bottle is too large it can be reduced by shrinking it with a heat gun before cutting - Figure 66.4.

Mick O'Connor

Figure 66.3 Finished siphon

Mick O'Connor

Figure 66.4 Shrinking

PROJECT 3.1 WHAT'S INSIDE DAPHNIA

Background

Daphnia or water fleas are a very common crustacean found in pond water or pools of water after rain (Figure 67.1). They do not like fast moving or polluted water and are very important in the aquatic food chain, forming an important food source for young and adult fish and both immature and mature insects.

Daphnia have a complex muscular system which tend to hide some of the smaller internal structures however most organ systems can be easily seen under the microscope.

Notable features are: (see also Figure 67.2)

- A long, bent, dark-coloured intestine.
- A simple football-shaped heart located behind the head on the dorsal side - its beat can be counted.
  - Daphnia has no actual blood vessels - its colourless blood plasma is directed throughout the animal by a series of very small membranes called mesenteries.
  - Its heart rate varies with water temperature - a good exercise is to alter the temperature and observe the changes to Daphnia's heart rate.
- A compound eye formed by the fusion of two eyes
- Eggs
- Antennae

Daphnia move through the water in a series of "hops" produced by rapid strokes of its feathery paired antennae. Reproduction is by parthenogenesis, ie eggs develop without fertilization in the brood chamber and hatch there as fully developed young.

- They develop during the year, in most habitats, and only females are produced.

Daphnia populations peak in spring and autumn, beginning when the water temperature rises to approximately 12°C.

- During these times, special "sexual" males and females may be produced, usually in response to a variety of environmental circumstances, males copulate with specialized females who produce haploid eggs.

Materials

- Microscope
- 1xml transfer pipettes
- Cavity slide and coverslip
- Daphnia anatomy diagram

Procedure

Daphnia are transparent so looking at them is in fact looking into them.

Step 1 Cut the tip off a 1 ml transfer pipette.

Step 2 Take a drop of water containing daphnia using a 1 mm transfer pipette.

Step 3 Place the daphnia in a cavity microscope slide.

Step 4 Place a cover slip over the cavity.

Step 5 On low power locate a daphnia.

Step 6 Locate the internal organs shown in Figure 67.2

Figure 67.1 Daphnia

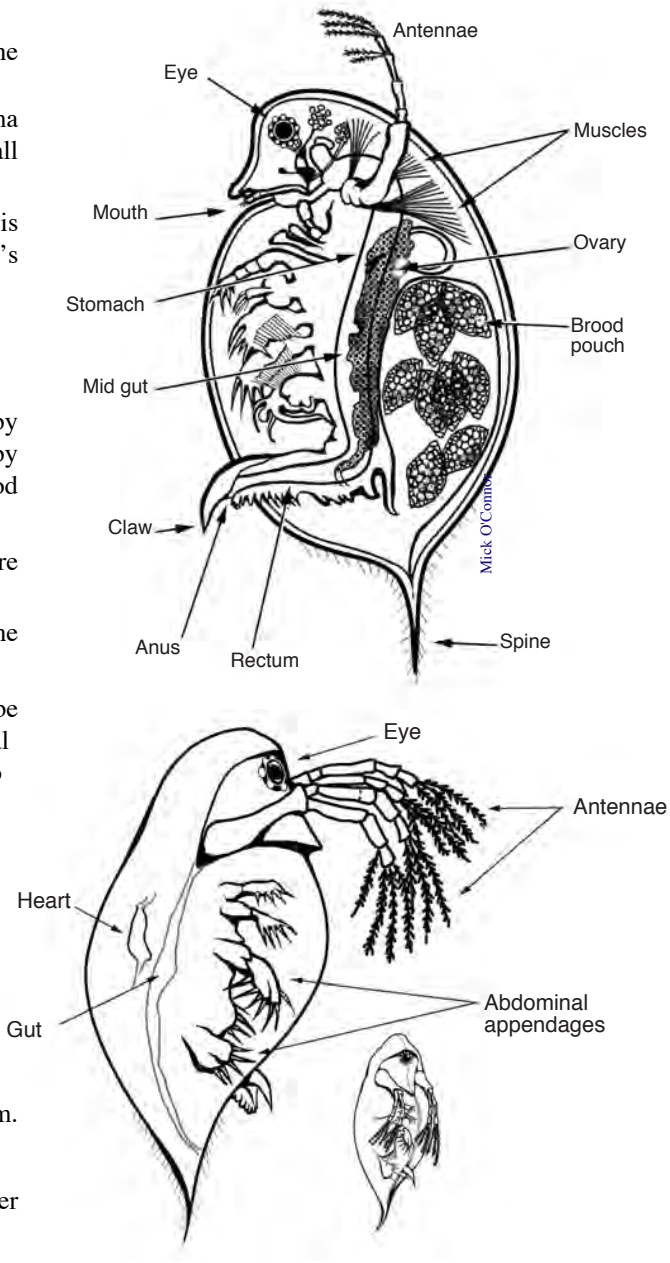


Figure 67.2 Daphnia female (left) and male (right)
Illustration Kerry Kitzelman

PROJECT 3.2 WHAT'S INSIDE ROTIFERS

Background

Rotifers are brackish water microorganisms used in the first-feeding of marine larval fin fish and crustaceans.

The word "rotifer" literally means "wheeled animals" and about 2,500 species of rotifers have been found in freshwater, brackish water, and seawater around the world.

Rotifers are a valuable first live feed in aquaculture.

- They can be produced in very large numbers very quickly and are actively sought by the stock being fed.
- Rotifers are not very nutritious by themselves but they are great feeders and are able to carry vital nutrients in microaglae to the laval fish and invertebrates that eat them.
- It is vital to understand the importance of providing them with a nutritional diet - they are great 'carriers'.

Rotifers are interesting animals:

- They are extremely small animals about 60-300 μm in size and hatch in twelve hours and are sexually mature at 18 hours.
- Their life-span of the females at 25°C is 6-8 days and males live for two days.
- They possess a pseudocoelom, a gut, a digestive system possessing both a mouth and an anus.
- Their internal organs lie within the pseudocoelom. (The fluid that fills the pseudocoelom serves as a hydrostatic skeleton. The fluid also acts as a medium for the internal transport of nutrients and wastes).
- They have bilateral symmetry, sexual dimorphism and reproduce quickly through the production of an egg sac.
- Female rotifers produce eggs which hatch within 12-hours.

Some features

Brachionus is one of the most common genera among the known 2,500 rotifer species. The genus is an important zooplankton species as a primary live food source for the early life of both marine and freshwater animal species.

- The body is comprised of four regions: head with corona, neck, body, and foot.
  - The foot is an appendage that extends from the body ventrally and possesses two toes.
- Other notable features include:
  - Cilia, which are small hairs that circulate water and nutrients towards the mouth.
  - A stomach and intestines, were the most nutrition is held for later consumption by fry and filter-feeding organisms.
  - A foot , which is utilized to temporarily attach themselves to the substratum.
  - A corona, which is a lip that are lined with the Cilia.
  - The mastax, a muscular pharynx containing a complex set of hard jaws.
  - A pseudocoel which surrounds the external organs and the digestive tract.

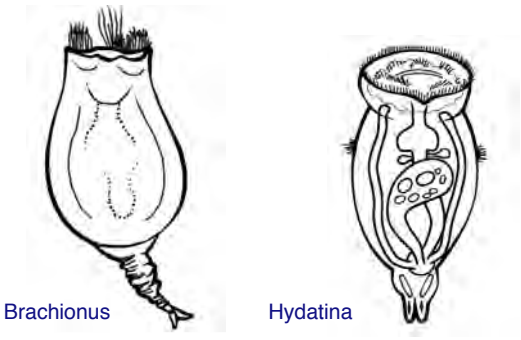


Figure 68.1 Two types of rotifer
Illustration Kerry Kitzelman

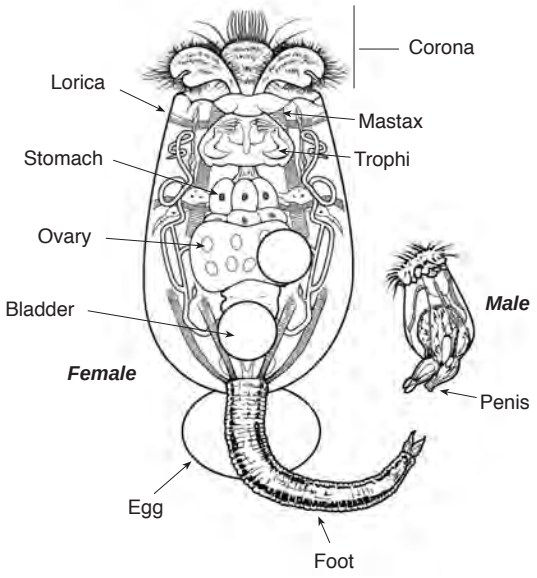


Figure 68.2 Rotifer anatomy
Illustration Kerry Kitzelman

Procedure

Rotifers are transparent. So looking at them, is in fact, looking into them!

Step 1 Take a drop of water containing rotifers using a 1 mm transfer pipette.

Step 2 Place on a clean microscope slide.

Step 3 Place a cover slip on the drop.

Step 4 Using a tissue reduce the amount of water around the cover slip.

Step 5 View the rotifer using the low power objective lens.

Step 6 Locate the internal organs shown on the diagram of *Brachionus plicatilis* above.

Discussion

a. Explain what is meant by the term 'carrier'. (see page 110)

b. Discuss the need to feed fish lava rotifers rather than feed them microalgae.

c. Research some of the predators of rotifers.

PROJECT 3.3 WHAT'S INSIDE ARTEMIA

Background

Also known as brine shrimp, Artemia are small shrimp (crustaceans) that live in salt water - the saltier the better!

They are found in large numbers in the warmer and drier regions in bodies of water which have been concentrated by evaporation or have high salt concentrations due to factors other than evaporation.

Artemia have amazing powers of adaptation that allow them to withstand a wide range of salinities from fresh water to almost saturated salt water.

- They have the most efficient osmoregulatory system known in the animal kingdom allowing them to change their bodies to suit these different salt concentrations.
- It is this ability to live in waters of such high salinity that excludes predators, that allows it to survive so well.

Artemia are filter feeders, injesting everything in the size range 1-40 micrometers, ie, they are not selective .

- If its in the water and if it is the right size they take it in and eat it - they are not choosy.!! They eat micro-algae, bacteria, protozoa and detritus (dead and decaying material).

They have two large compound eyes. They have a brain which is fed nervous impulses from a ventral nerve cord running the length of their body.

The circulation system is open with a heart pumping blue blood trough major vessels.

The gut is a continuous tube that runs the length of the body allowing digestion and absorption to take place.

Metabolic wastes are excreted through a maxillary gland at the base of the 'head'.

Reproduction

Reproduction is sexual.

- Male and female mature brine shrimp in favourable conditions will pair off and mate.
- Large quantities of eggs are often laid by the females and will float to the surface of the water.
  - Under favourable conditions the eggs hatch into a free swimming nauplii which will turn into adults in about 8 days. Mature females produce about 300 nauplii every four days.
  - Under harsh conditions such as low oxygen and high salt the female produces a very tough cyst able to withstand the harsh conditions.

    It floats and is blown ashore where it dries. Females can produce up to 75 cysts per day.

Materials

- Dissecting microscope
- 2 x 1 ml transfer pipettes
- 1 x Petri dish
- Artemia anatomy diagram (Figure 69.1)
- Red food dye (optional)

Procedure

Artemia are transparent so looking at them is in fact looking into them. To view artemia follow these steps.

Step 1 Cut the tip off a 1 ml transfer pipette.

Step 2 Take a drop of water containing Artemia using a 1 mm transfer pipette that has had the tip cut off it to avoid damaging the Artemia.

Step 3 Suck up a few Artemia from a culture using the pipette.

Step 4 Place them in the centre of a petri dish in as little water as possible one artemia per drop.

Step 5 Using an uncut pipette remove as much water as possible so the artemia cannot swim around.

Step 6 Locate the internal organs shown on the diagram below.

Step 7 It may help to increase the contrast by placing the petri dish on a coloured background, or place some red food dye diluted 10:1 with water in the bubble.

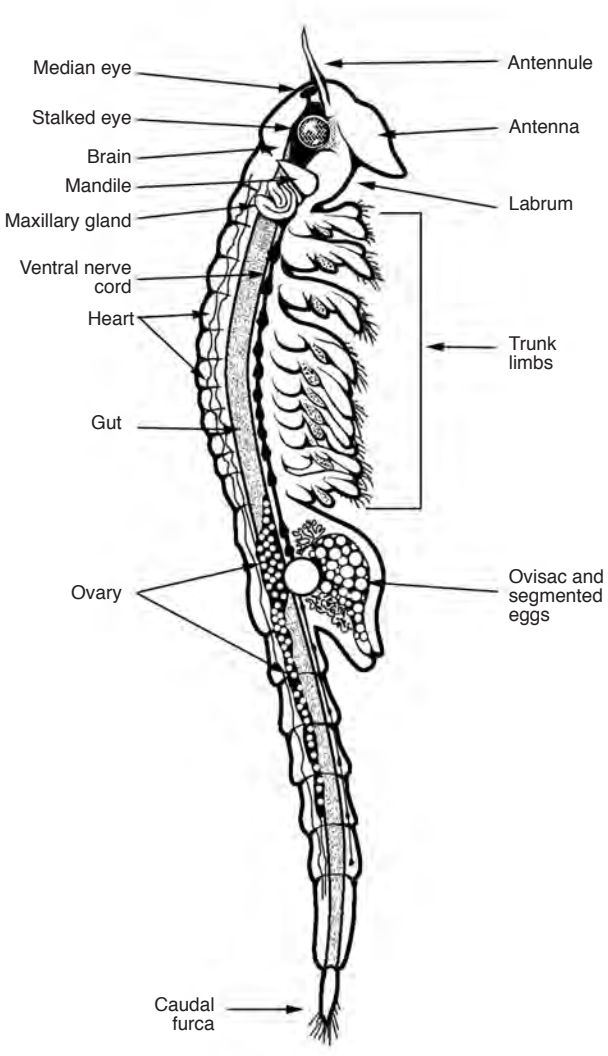


Figure 69.1 Artemia anatomy
Illustration Kerry Kitzelman

PROJECT 3.4 EXTERNAL FEATURES OF A FISH

Background

Fish have the following characteristics:

- A slimy, usually scaly skin
- Are all poikilothermic -the temperature of their body depends upon the temperature of the environment
- Are all aquatic
- Fertilization is usually external; eggs are usually laid, but some bear their young alive
- They breathe by means of gills
- Paired fins are present
- The tail is muscular and forms the main locomotory organ.

Materials

- A fish (preferable dead)
- Diagram of the external features of a fish (below)

Procedure

If you have access to Silver Perch then the diagram below will be good for you. If not it does not really matter since the characteristics of all fish are all the same. Shape and proportions may differ but you should be able to recognise the features on your fish.

Step 1 Compare the features on the diagram with your fish.

Step 2 Complete the diagram on the page opposite.

Discussion

a. Relate the shape of the fish's body to the viscosity of the water it must move through.

b. Discuss any colouration and relate this to the fish's chance of survival.

c. Compare and contrast the structure of the caudal and dorsal fins.

d. Explain why it is hard for a fish to swim backwards.

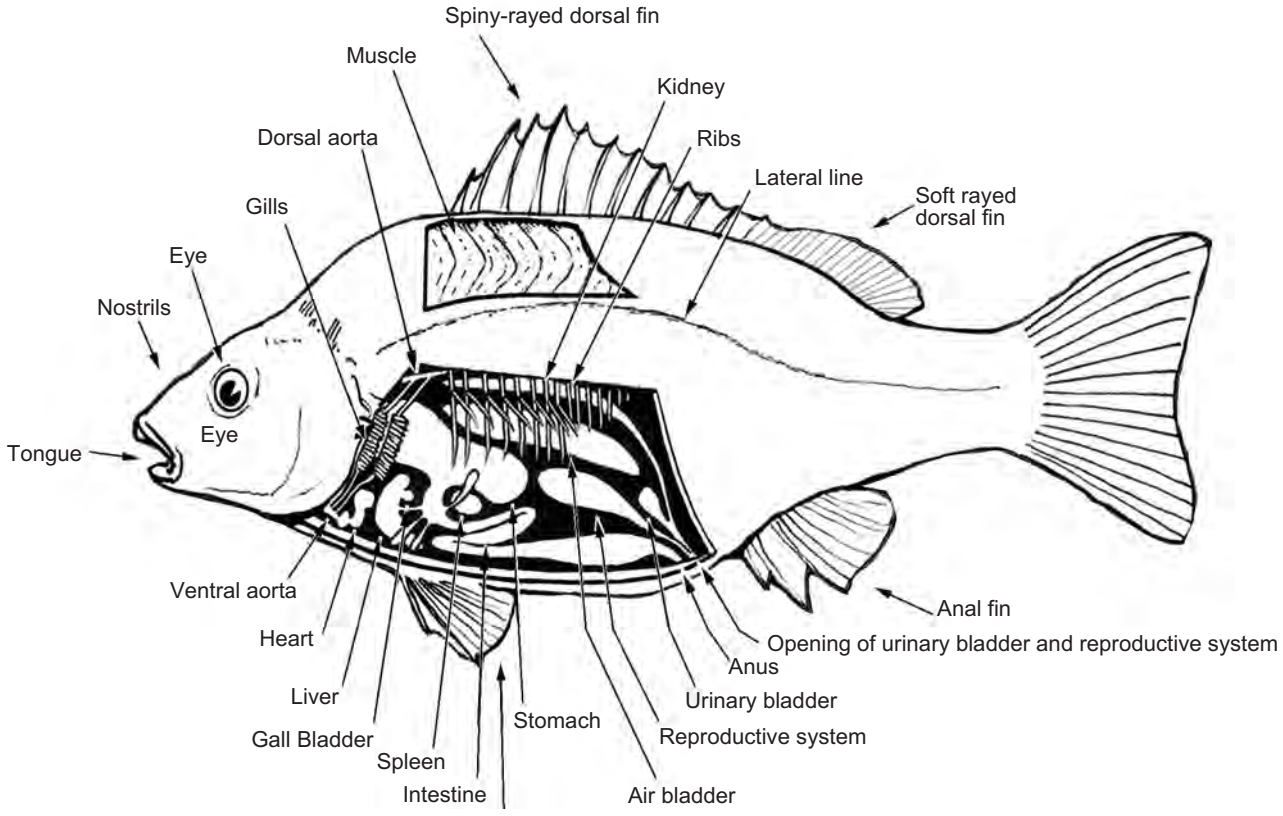
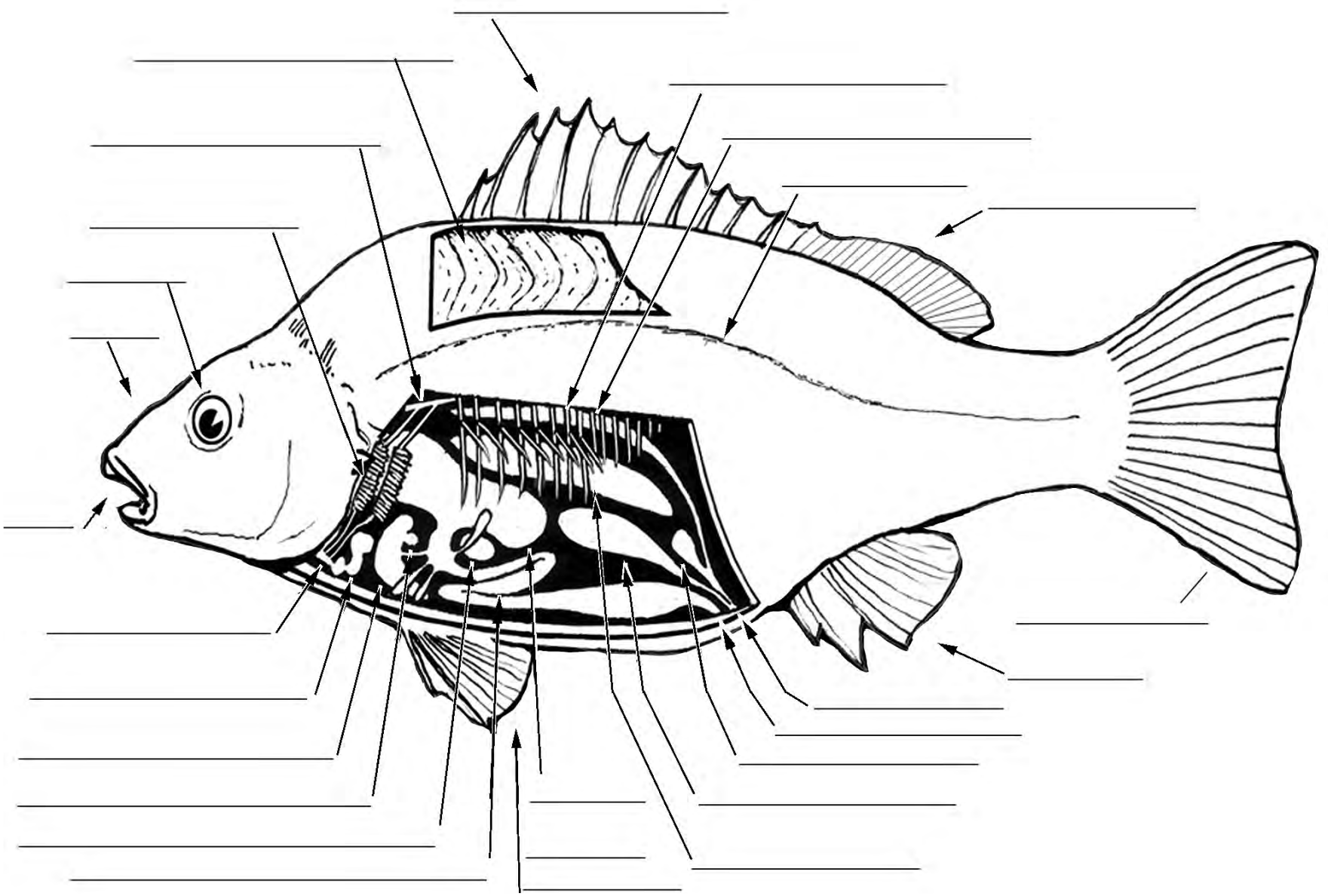


Figure 70.1 Parts of a fish
Illustration Kerry Kitzelman

Students name _____ *Teacher* _____

Label the illustration below

Illustration Berry Kitzelman



PROJECT 3.5 SEA MULLET DISSECTION

Materials

- Sea mullet, dissecting equipment, magnifying glass, dissecting tray, gloves, pencil and notebook.

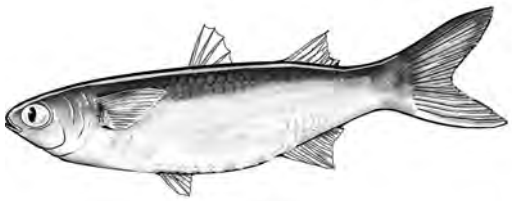


Figure 72.1 Mullet
Illustrations Kerry Kitzelman

Procedure

Take your fish, tray, paper and instruments and follow the instructions below.

Step 1. Using the outline illustration over, make a drawing of your fish identifying the following: Eye, mouth, nostrils, operculum (gill cover) and pectoral, dorsal, ventral fins, anal and caudal fins.

Step 2. Under the drawing
- a. Make a description of the shape of the fish and write an explanation of how this shape may assist the fish in water.
- b. Describe the colour of the dorsal surface (top surface) of the fish and compare it with the bottom (ventral surface).

Step 3 Hold the fish up above your head and look at its ventral surface against the ceiling as a background. Put it on the grey lab bench and look down on its dorsal surface.
- a. Explain how this colouration may give an advantage to the fish.
- b. Compare this colouration to that of a camouflaged fighter jet.

Step 4. Identify on your drawing which fins act as:-
- a. "Brakes"
- b. "Reverse gear",
- c. "Stabilisers",
- d. "Steering gear",
- e. "Power plants'.

Step 5. Identify the type of body covering on this fish and describe the feeling of the fish's surface. Explain the function of the mucous coat. (Ref 3rd para page 95).

Step 6. Look at the diagrams of caudal fin shapes below and the tail of your fish. Write the name of type of caudal fin that mullet has on your diagram. How does this compare with other tails?

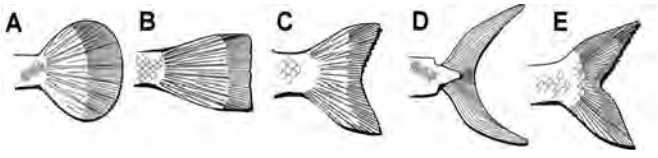


Figure 72.3 Caudal fin types: a - rounded; b - truncate; c - emarginate; d - lunate; e - forked.
Illustrations Kerry Kitzelman

Step 7. Scales. Take a few scales of the fish and using 100X magnification examine them and draw what you see.

Select one row of scales and count the number of scales in that row. Record the number.

Using the diagram below identify the type of scales and write this on your diagram.

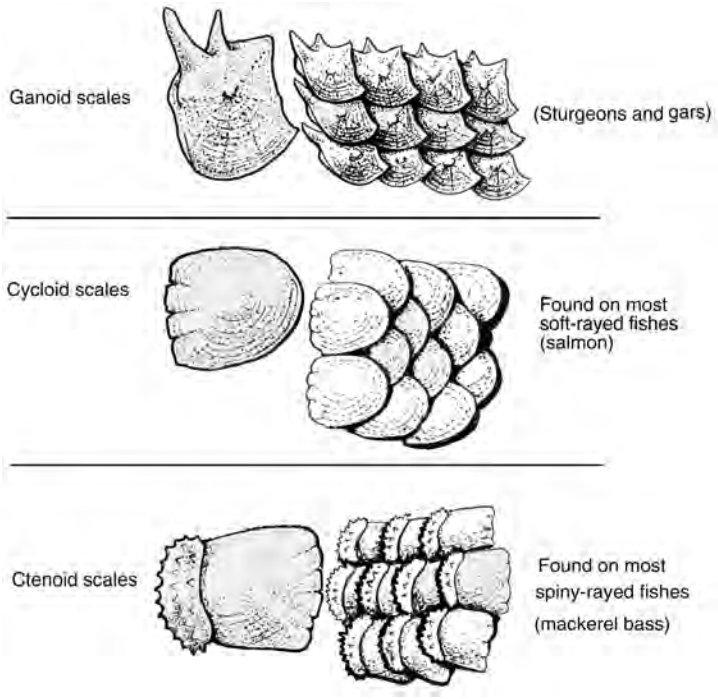


Figure 72.2 Three types of fish scale
Illustrations Kerry Kitzelman

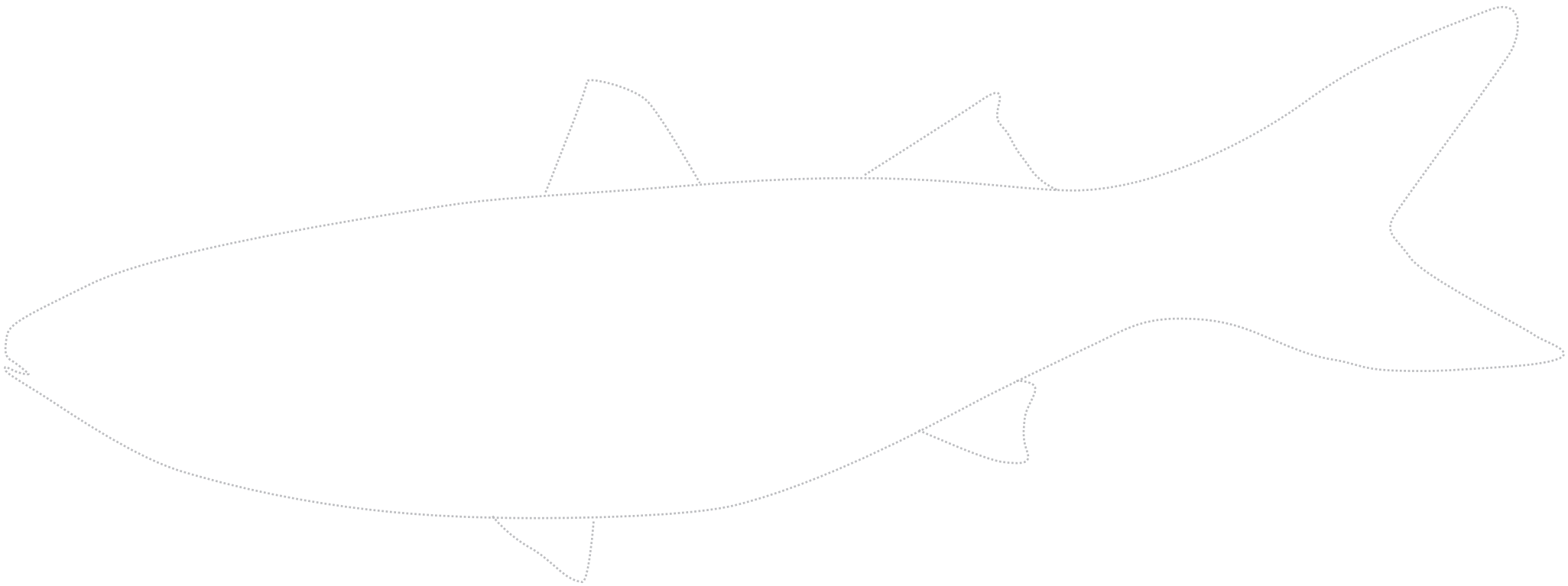
Step 8. Examine the body openings and mark the following on your diagram: Mouth, nostrils, anus and gill slits or cover.

Step 9. Examine the mouth. Pull out the mouth and examine carefully. Compare the fish's lips with yours. Write down the diet of this fish based on its mouth parts.

Step 10 Examine the gills. Turn the fish on its back (dorsal side) and cut with a pair of scissors through the thin area joining the head to the abdomen on the ventral side.
- a. Bend the neck back and carefully break it. With your index finger remove all the gills in one piece then place them in a petri dish.
- b. Investigate the gill to see if they are separated or stuck together and record your findings.
- c. Predict what they may look like in the fish in water.
- d. Place the gills in a beaker of water to test your prediction. Record your findings.
- e. Cut out a small section of gill and place it under the binocular microscope. Draw what you see.

Students name _____ *Teacher* _____

Illustration Bob Moffatt



PROJECT 3.6 EXTERNAL FEATURES OF A CRAYFISH

Background

Crayfish have the following characteristics:

- Hard exoskeleton made of chiton
- All are poikilothermic -the temperature of their body depends upon the temperature of the environment
- Are all aquatic
- Fertilization is usually external; eggs are usually laid, and held by the female under her tail while development occurs
- Breathe by means of gills
- Body in two parts - cephalothorax and abdomen
- Five pairs of legs one pair modified to large nippers
- Two pairs of antennae
- Large compound eyes

Materials

- One crayfish
- Diagram of the external features of a crayfish (Figure 74.1)

Procedure

Step 1 Compare the features in Figure 74.1 with your crayfish.

Step 2 Tick off those features you are able to find.

Step 3 Note the endings on all legs.

Step 4 Examine the mouthparts and relate their structure to the crayfish diet.

Step 5 Explain why it is hard for a fish to swim backwards but very easy for crayfish.

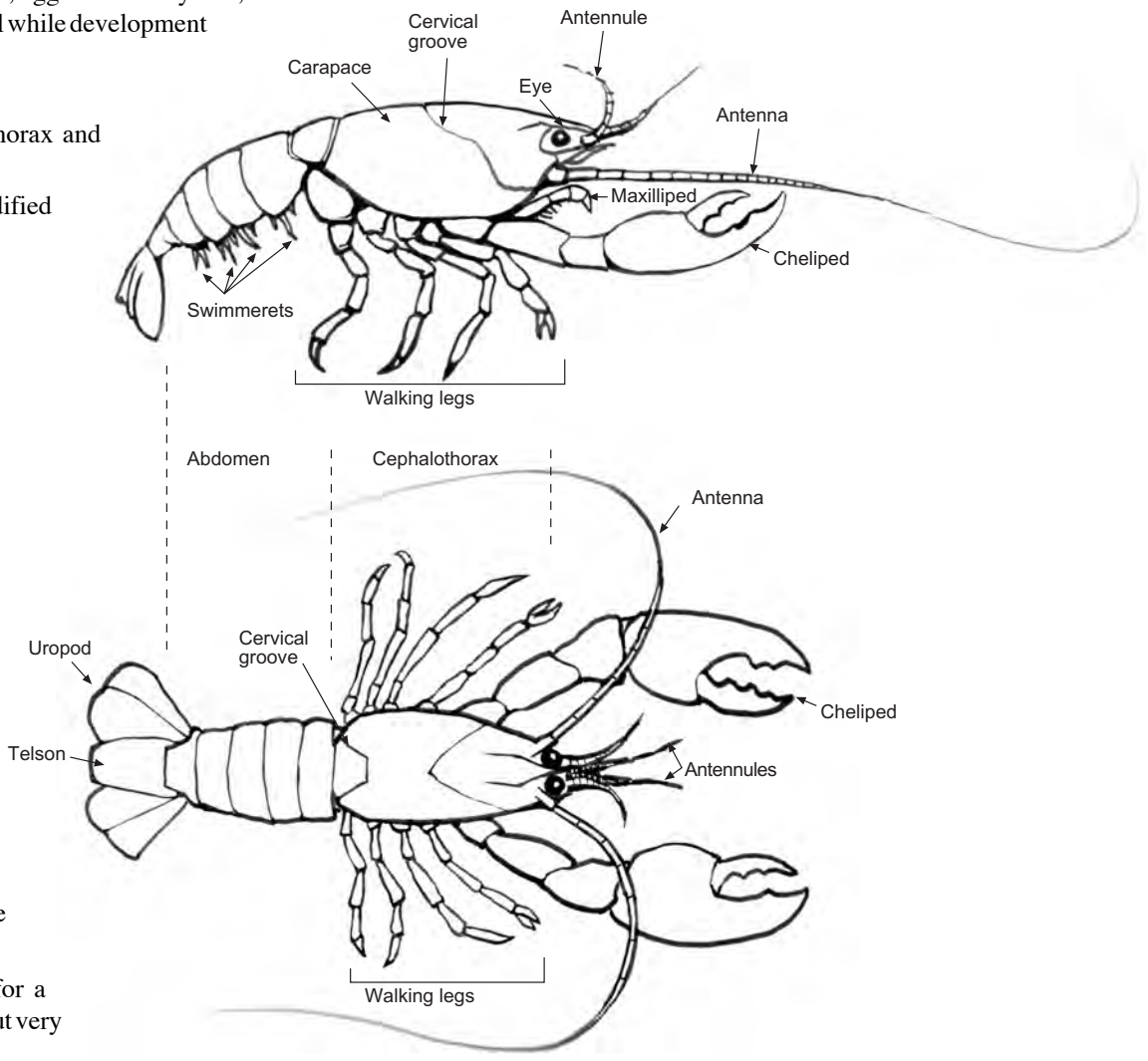


Figure 74.1 Crayfish external features
Illustrations Kerry Kitzelman

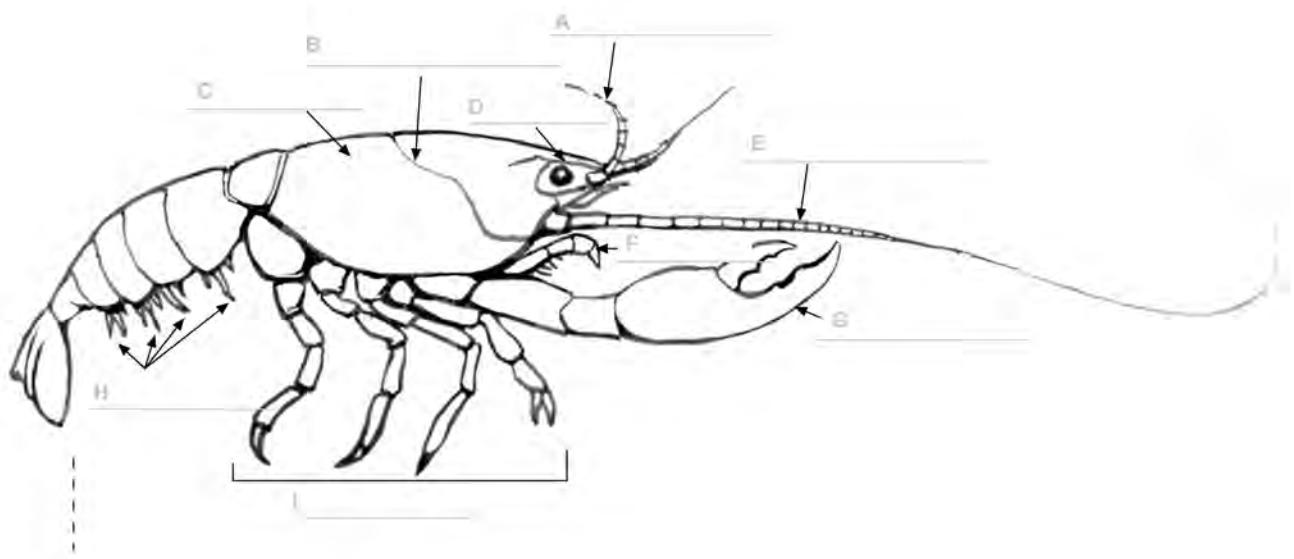
Discussion

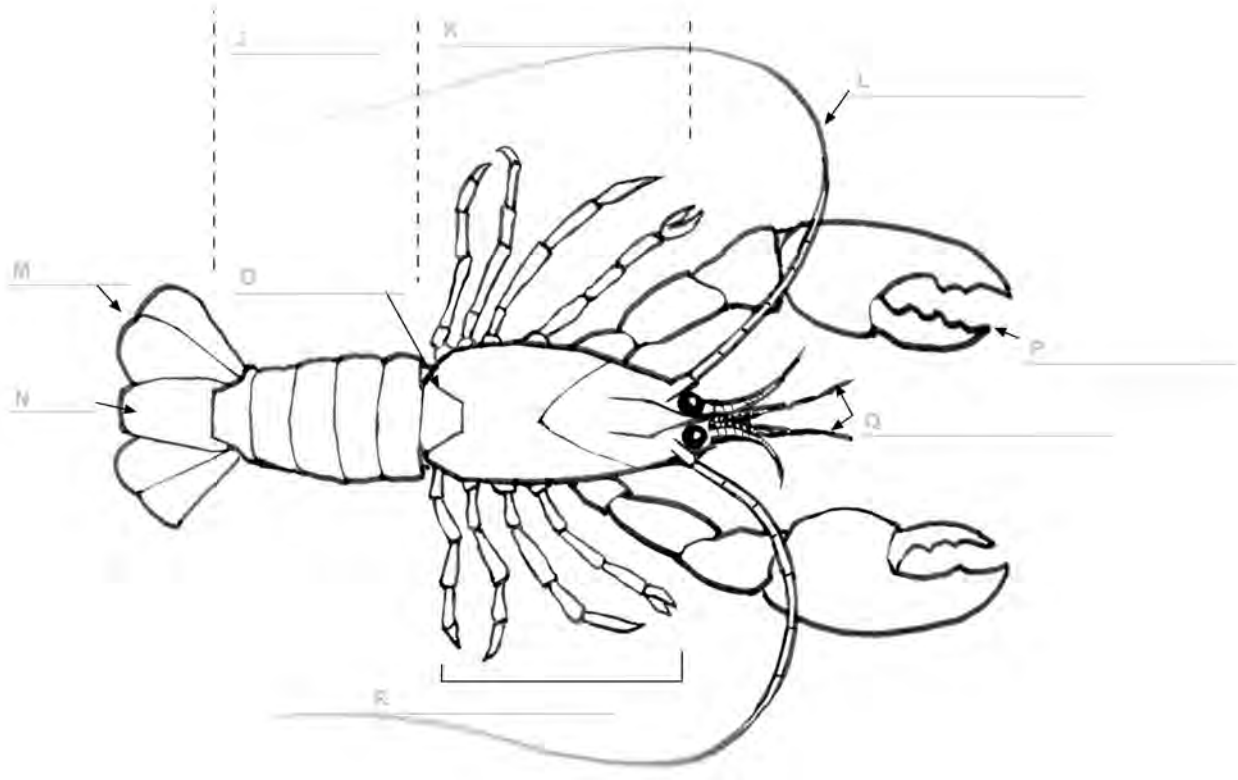
1. Discuss the advantages and disadvantages of having an exoskeleton.
2. Account for the claws on the legs of your crayfish.

Students name _____ *Teacher* _____

Label the body parts A - R on the diagrams below

Illustrations Kerry Kitzelman





PROJECT 3.7 MAKING A FISH MEASURER

Background

Fish are measured for a variety of reasons.

Amateur and professional fishers are only allowed to take certain size fish and are fined heavily if they are caught with undersized specimens - measurement to them is very important!

Fish biologists and marine researchers need to keep accurate records on sizes and masses of fish. They catch, measure, tag and release the species under study and later repeat the process. The information gained provides valuable knowledge about the species . Knowledge that may protect and conserve that species while allowing it to be harvested for food.

In aquaculture the length and weight of a fish are used to measure its growth rate. The effects of factors such as temperature oxygen levels nutrition and disease can be studied using length and mass measurements.

Materials

- Perspex strip 800mm X 80mm
- NSW or Qld Fisheries Fish Measuring Sticker (available free of charge from bait and tackle shops as well as from Fisheries Offices
- Jig saw if the perspex has to be cut to size and a heat gun

Procedure

Step 1 Using the jig saw cut a perspex strip 800mm X 80mm.

Step 2 Heat the perspex 80mm from one end using the heat gun until it is soft.

Step 3 Bend a right angle at around the 80mm mark and hold until firm.

Step 4 Allow the perspex to cool.

Step 5 Place the perspex on a flat surface with the bent 80mm perpendicular (pointing to the ceiling).

Step 6 Peel the backing strip off the sticker and place the zero mark against the upright.

Step 7 Fix the sticker carefully from the zero mark end to avoid air bubbles.

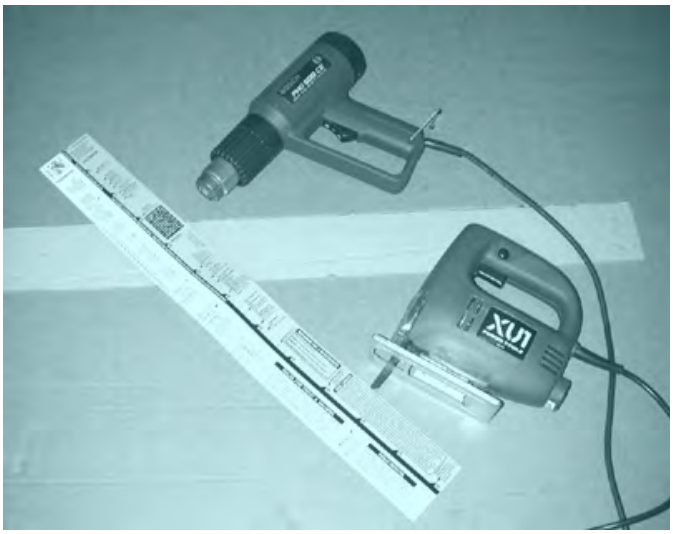
Using the measurer

The advantage of this type of fish measurer is that it can be kept wet and it allows quick measurements to be taken.

This is very important in aquaculture and in catch and release or tagging operations where stress to the fish must be kept to a minimum while it is out of the water.

By placing the 'nose' of the fish against the upright very rapid measurements particularly length measurements can be taken.

The measurer can be modified for fin , profile or other measurements if needed - simply increase the width by three to 240mm and stick three stickers down side by side.



Mick O'Connor

Figure 76.1 Production methods

PROJECT 3.8 MAKING A CRAYFISH RESTRAINER

Background

Crayfish have big nippers and can bite!!... and when they bite they hurt. Fortunately for us they have a nice little groove on the top of their cephalothorax that can be used to hold them down with a unique restrainer.

This restrainer will hold the crayfish down once they have been taken from their tank.

- The easiest and best way to catch crays is with a net.
- Handling involves approaching from the tail end grasping the cephalothorax towards the abdomen using the thumb and forefinger.
- Pick the crayfish up and turn your hand over so you are looking at its underside.
  - This makes it harder for them to nip you! (If they do latch on put your hand and the cray back in the water because they will usually let go).
- The restrainer is used to put downward pressure on the cray, preventing them from walking off when you put them face down on the desk, in a dish or on a measure grid.

Materials

- Plastic coated coat hanger
- Pair of pliers
- Masking tape

Procedure

Step 1 Cover both jaws of the pliers with masking tape to avoid damaging the plastic coating on the wire as you bend it.

Step 2 Straighten the coat hanger and cut a 200mm section from it.

Step 3 Measure and mark with a pencil 20 mm, 40 mm and 60 mm from one end.

Step 4 Use the pliers to make an approximate 120 degree angle bend at the 20mm mark..

Step 5 At the 40mm mark use the pliers to bend to bend the wire back on itself.

Step 6 Use the pliers to straighten the 'handle' at the sixty mm mark to make a flattened 'Y' shape restrainer.

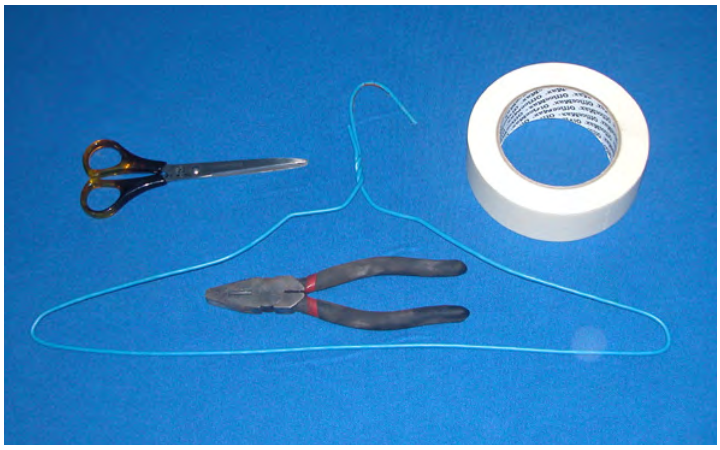
The flattened Y shape fits into the groove on the cephalothorax.

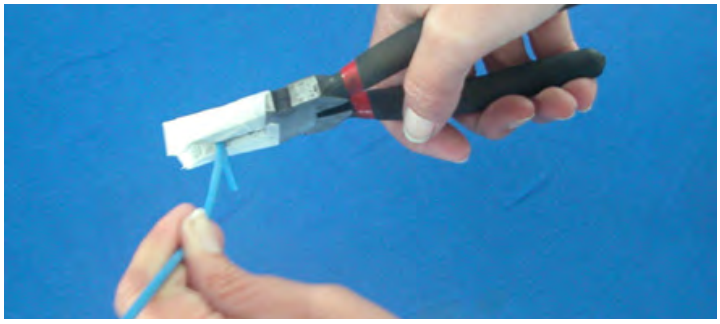
Hint

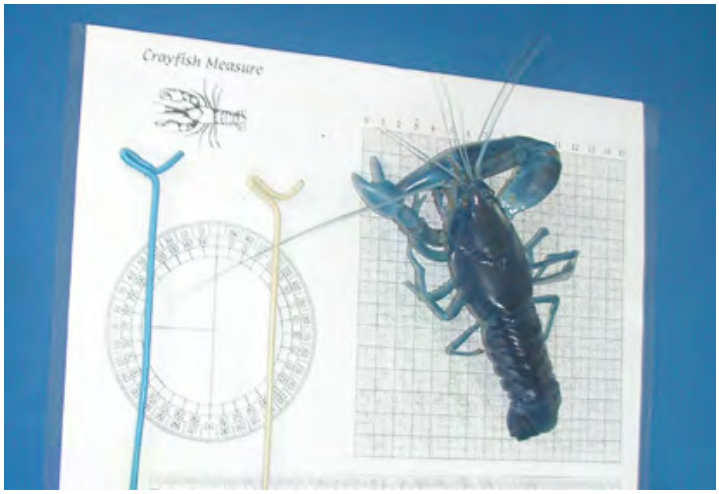
When picking up crayfish approach them from behind, grasping the cephalothorax towards the abdomen and using the thumb and forefingers.

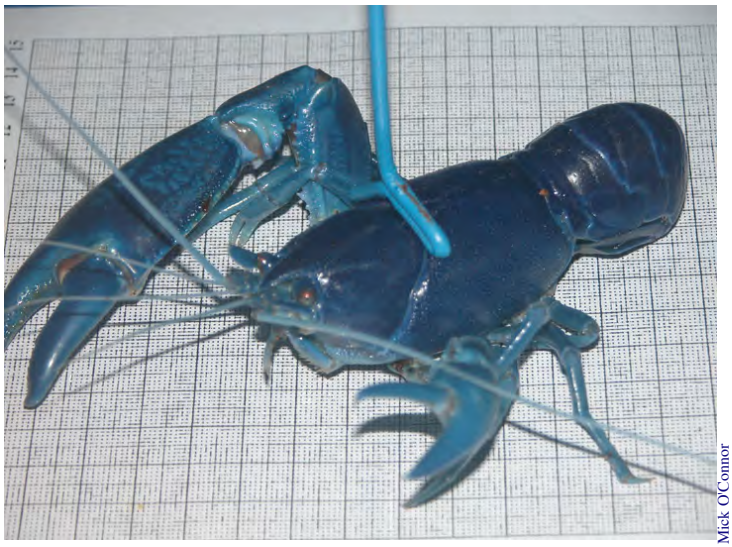
Pick the crayfish up and immediately turn it over so you are looking at its belly. This makes it harder to bite you.

The restrainer is designed to put downward pressure on the crayfish and prevent it from walking off when you put them face down on he desk, on a measurer or on a disk.









Mick O'Connor

Figure 78.1 Production methods

PROJECT 3.9 MAKING A CRAYFISH MEASURER

Background

Most of the monitoring exercises performed with crayfish involve measurement. Crayfish are sometimes a little hard to measure - they don't like to be picked up - they want to walk about when placed on any surface out of water and they bite!

The quickest and easiest way to measure them is to place them on a surface that has measurements on it - graph paper is ideal. All you have to do is protect it from getting wet!

The yabbies are placed on this and held still with a little downward pressure on the cephalothorax (See Figure 79.1)

Materials

- Metric Crayfish Sizer Master Sheet
- Laminator / contact / perspex / plastic
- Plastic coated coat hanger if you are worried about being bitten (See Project 3.8)

Procedure

Step 1 Photocopy the Metric Crayfish Sizer on the page over.

Step 2 Laminate it or cover with perspex, glass plastic or some other waterproofing.

Step 3 Decide what measurements you will take for the exercise.

Step 4 Now place the crayfish on the waterproof sizer.

Cut and bend a plastic coated coat hanger to make a holder for the crays (See Project 3.8).

Hold the crayfish still with a little downward pressure on the cephalothorax.

Step 5 Take the measurements you require and record them appropriately.

Discussion

1. Record your measurements in a table.
2. Graph the change in length of the various body parts after each moult.
3. Discuss the uses of the external features illustrated on page 72.

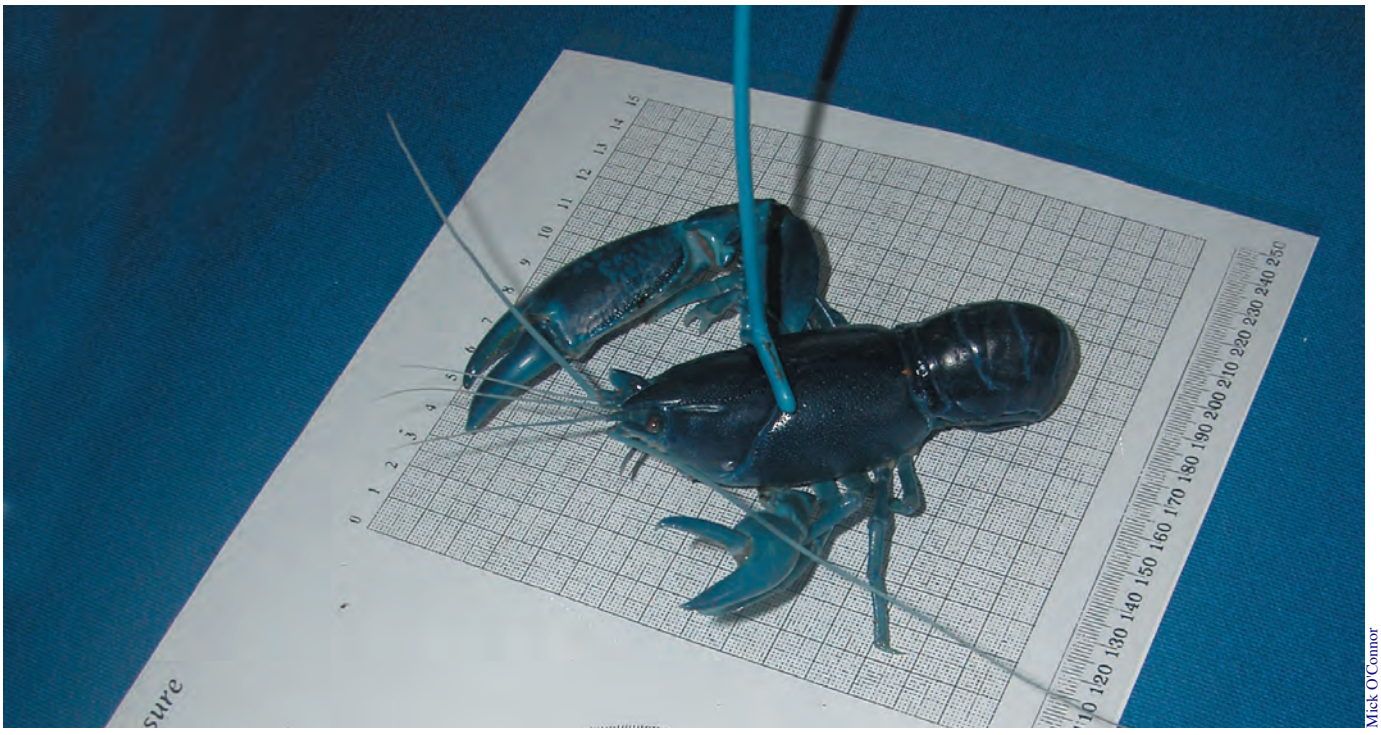
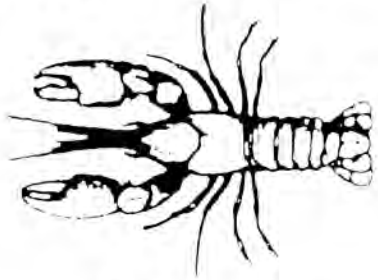
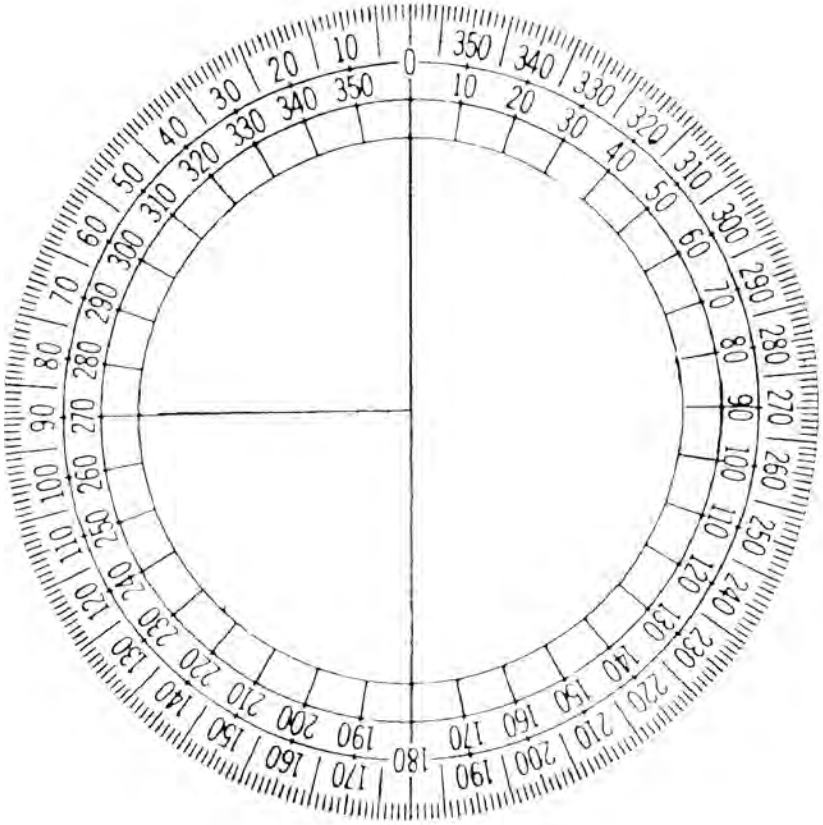


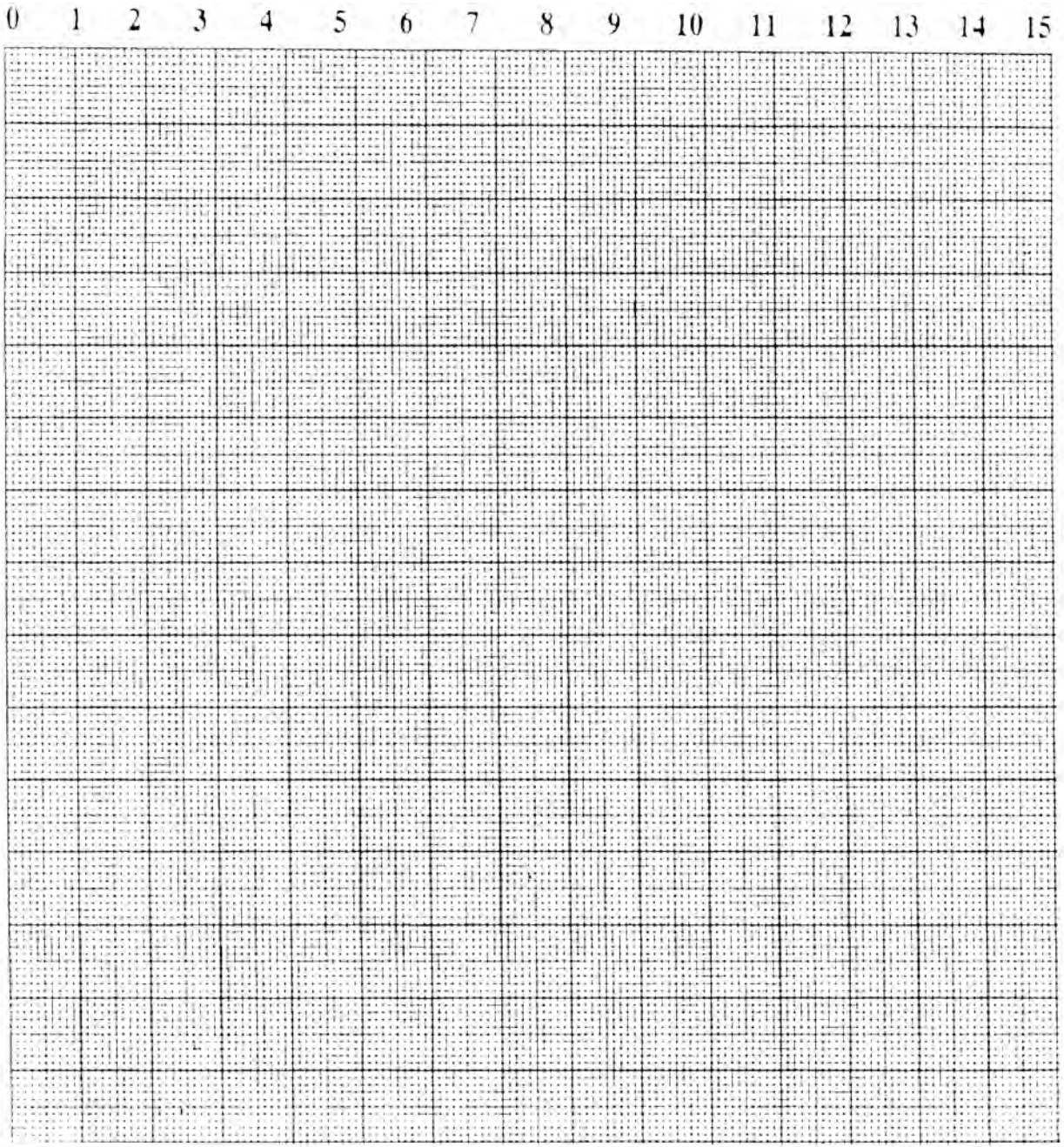
Figure 79.1 Finished project

Crayfish Measure

Illustration Mick O'Connor







PROJECT 3.10 SQUID DISSECTION

Adapted from LaRosa, E. (2000). Middle school science. From www.middleschoolscience.com with questions by Adam Richmond Clontarf Beach SHS

Materials

- Squid, dissection equipment and tray

Procedure

Read the instructions below and make drawings of your dissection in your note book.

Step 1 Identify the tentacles, head, eye, siphon, mantle and fins.

Step 2 Open the mouth and look at the *radula* (the squid's tongue). Remove the *beak* then draw and label the squid's mouth parts.

Step 3 Using tweezers grasp the *brain* by going through the mouth and the long nerve attached to it. Carefully remove and examine them.

Step 4 Make sure the squid is siphon-side up. Cut carefully through the *mantle* only with scissors. Lay the mantle flat in the dissection tray.

Step 5 Using Figure 81.1, find and examine the following - *intestine,* , *stomach* (glistening white), *gill* (feathery), and *3 hearts*.

Step 6 Find the silvery, black *ink sac*.

Step 7 Locate the hard *pen* near the squid's fin. Grasp it and pull it out. Now use this to pierce the ink sac, and write your name in the squid's ink (Figure 81.2).

Step 8 Remove and examine the *eye* . Remove the *cornea* (film like), and the *lens* (hard, silvery pearl-like structure). Sketch and label it.

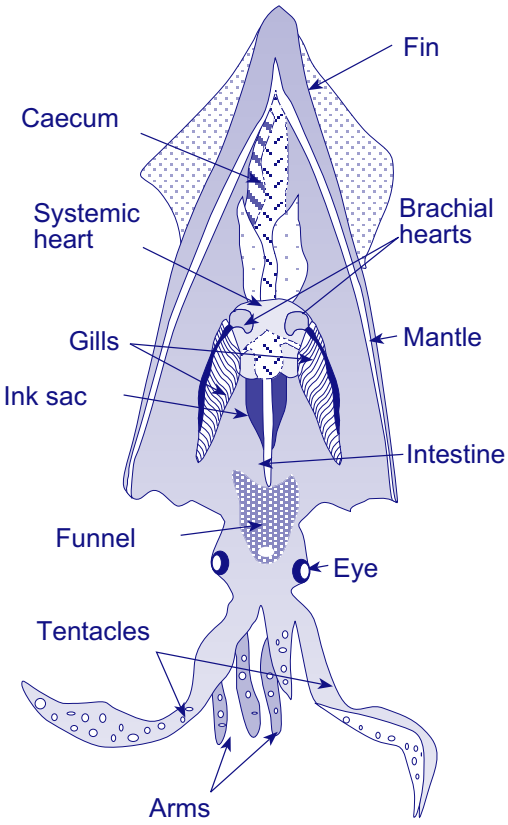
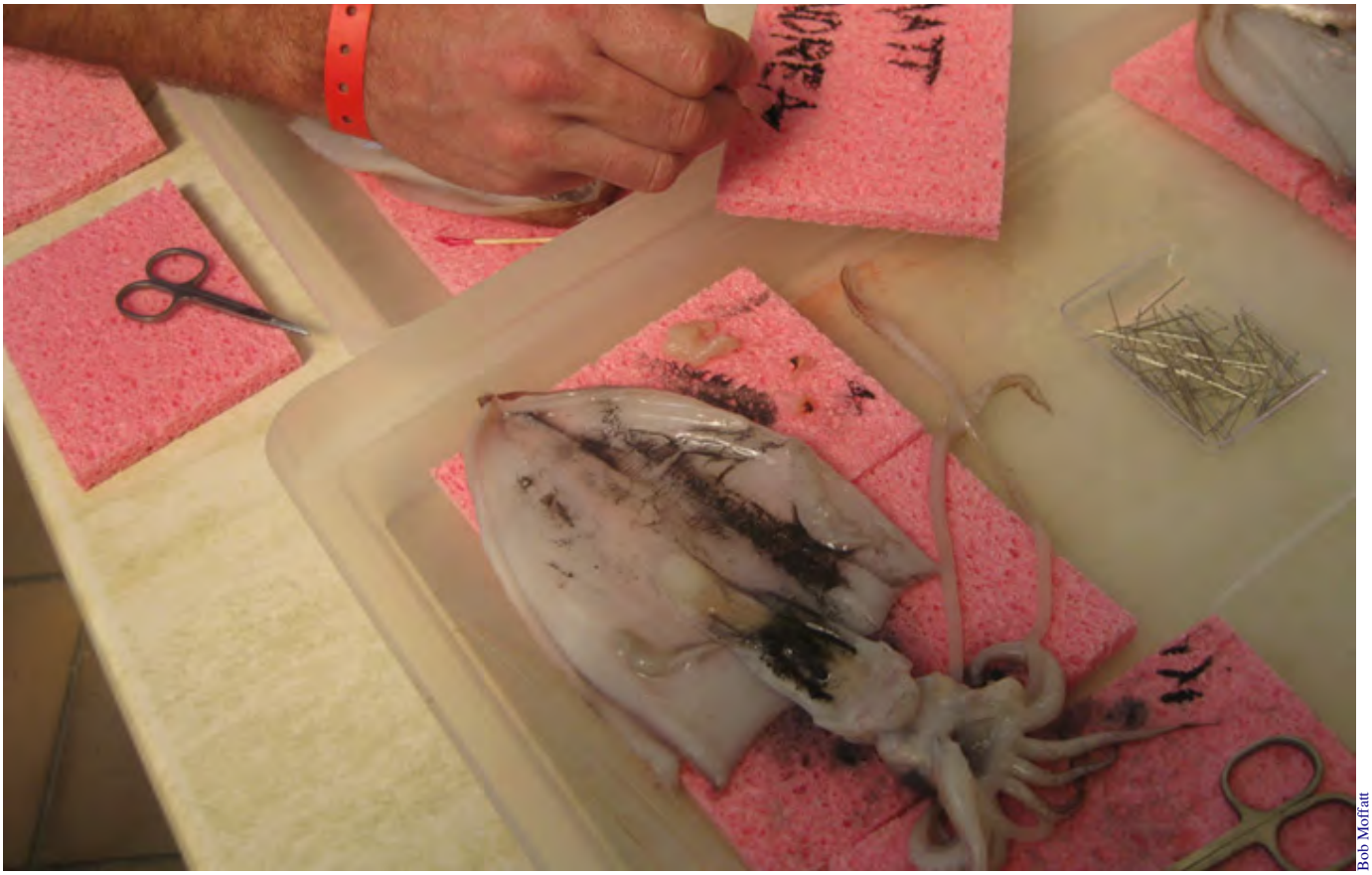


Figure 81.1 Squid biology
Illustration Bob Moffatt



Bob Moffatt

Figure 81.1 Dissection

PROJECT 3.11 PRAWN DISSECTION

Background

Prawns are made up of nineteen body segments grouped into the head thorax and abdomen as shown in Figure 82.2.

Each segment may possess one pair of appendages, although in some prawns these may be reduced or missing.

The head and thorax is covered by a tough skin (called a carapace) which protects the internal organs and the gills.

The section of the carapace that projects in front of the eyes is called the rostrum.

- The head has six appendages. These are the antennules, antennae, mandibles, first and second maxillae. The head also bears the (usually stalked) compound eyes.
- The thorax has mouth parts and walking legs with special names.
- The abdomen has swimming legs and a big tail fan which helps the prawn escape its prey.

Sexing prawns

- In prawns, sperm are produced in a gonopore from the base of the 5th walking leg (Figure 82.1).
- From here they are picked up by a tubular organ called the petasma located on the first swimming leg and transferred to a female opening called at thelycum where they fertilise the eggs.

  The thelycum is located in the mid section of abdomen opposite the 5th walking leg.

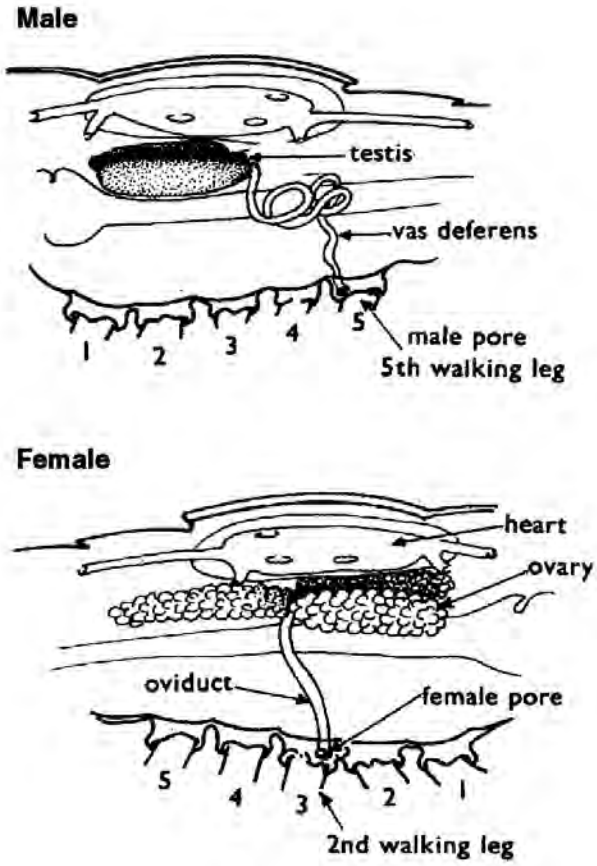


Figure 82.1 Sexing a prawn
(Illustration Steven Byers)

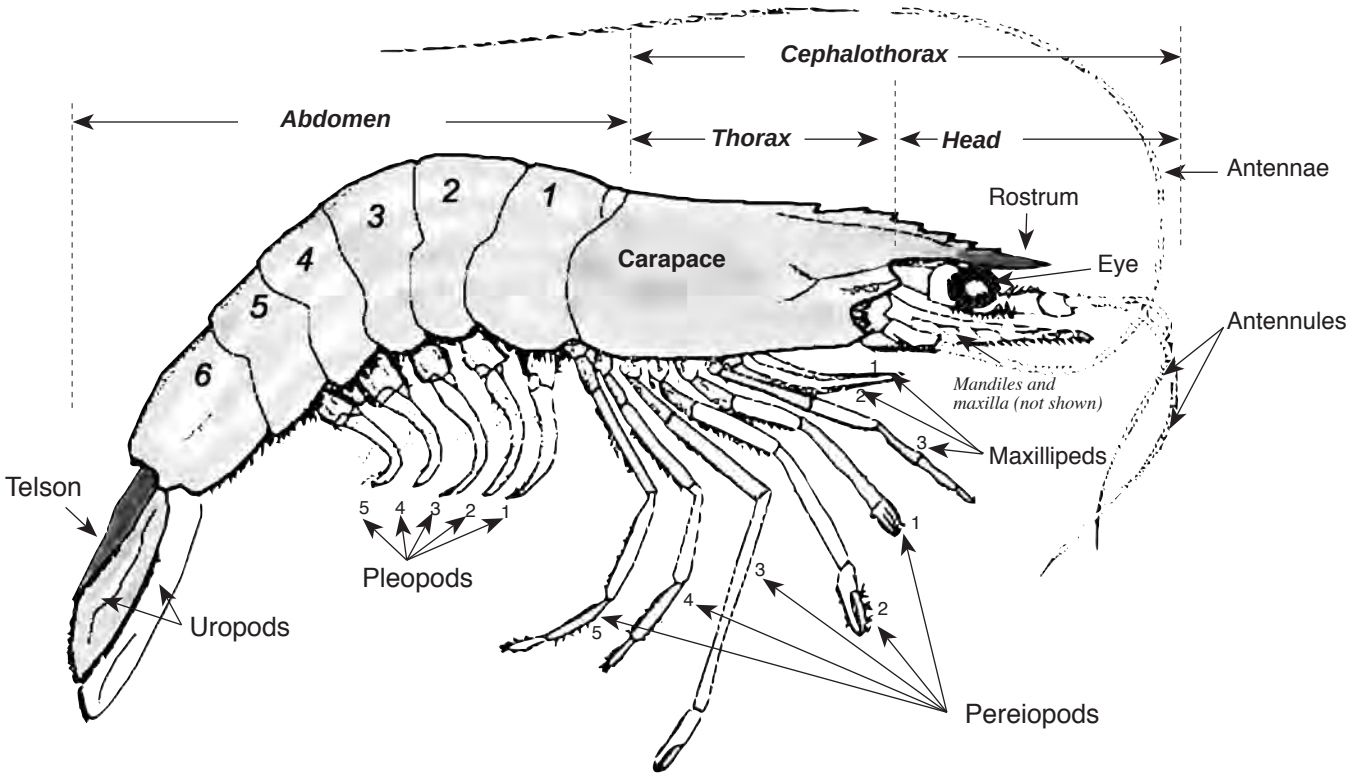


Figure 82.2 Prawn external features
(Illustration Bob Moffatt and Steven Byers)

Procedure

Step 1 Look at the abdominal region and the illustration below. Now identify the sex of your prawn.

Step 2 Carefully remove all the appendages and stick them on a piece of A4 paper.

Take photographs of each appendage and prepare a class talk on the appendages of a prawn.

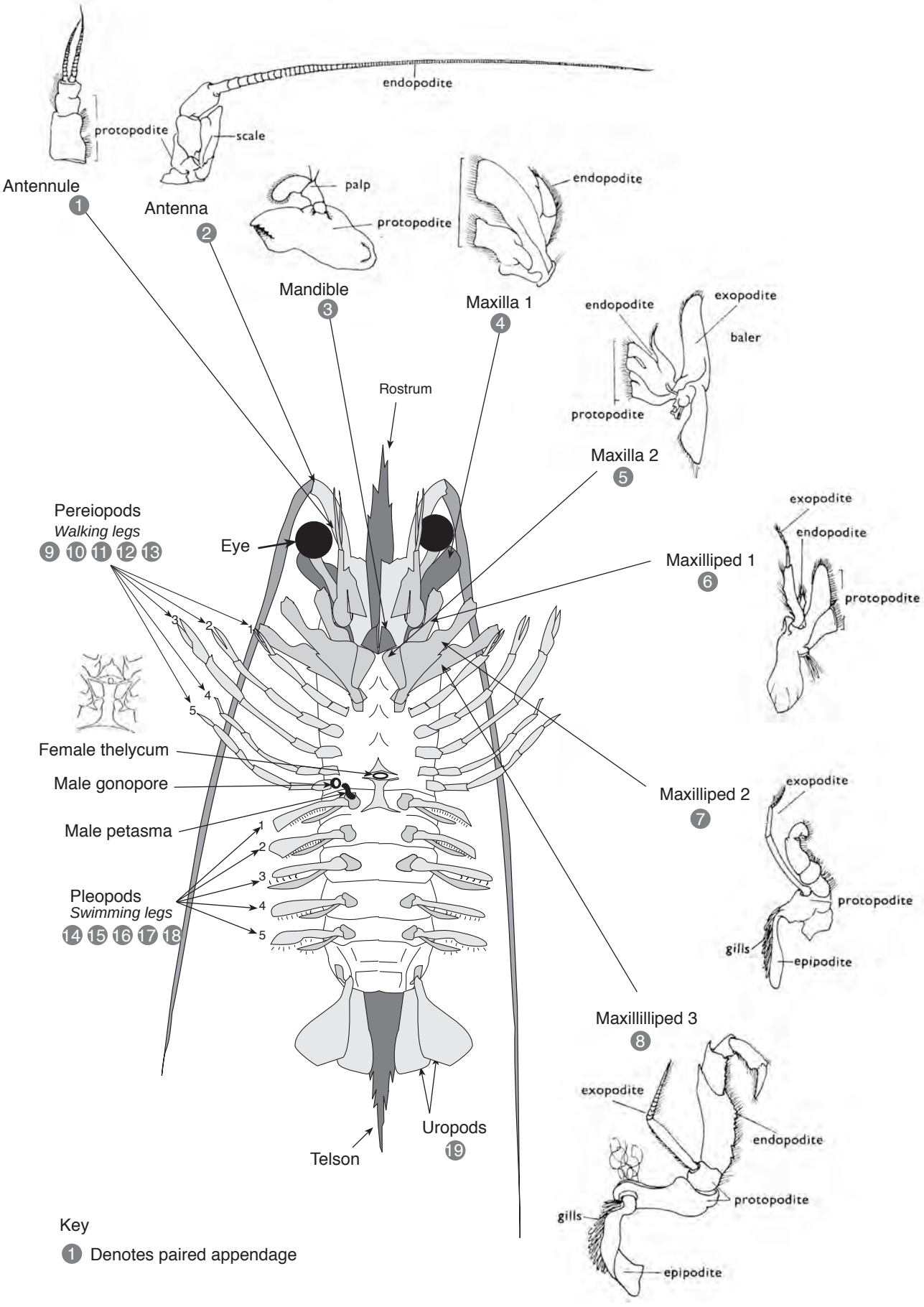
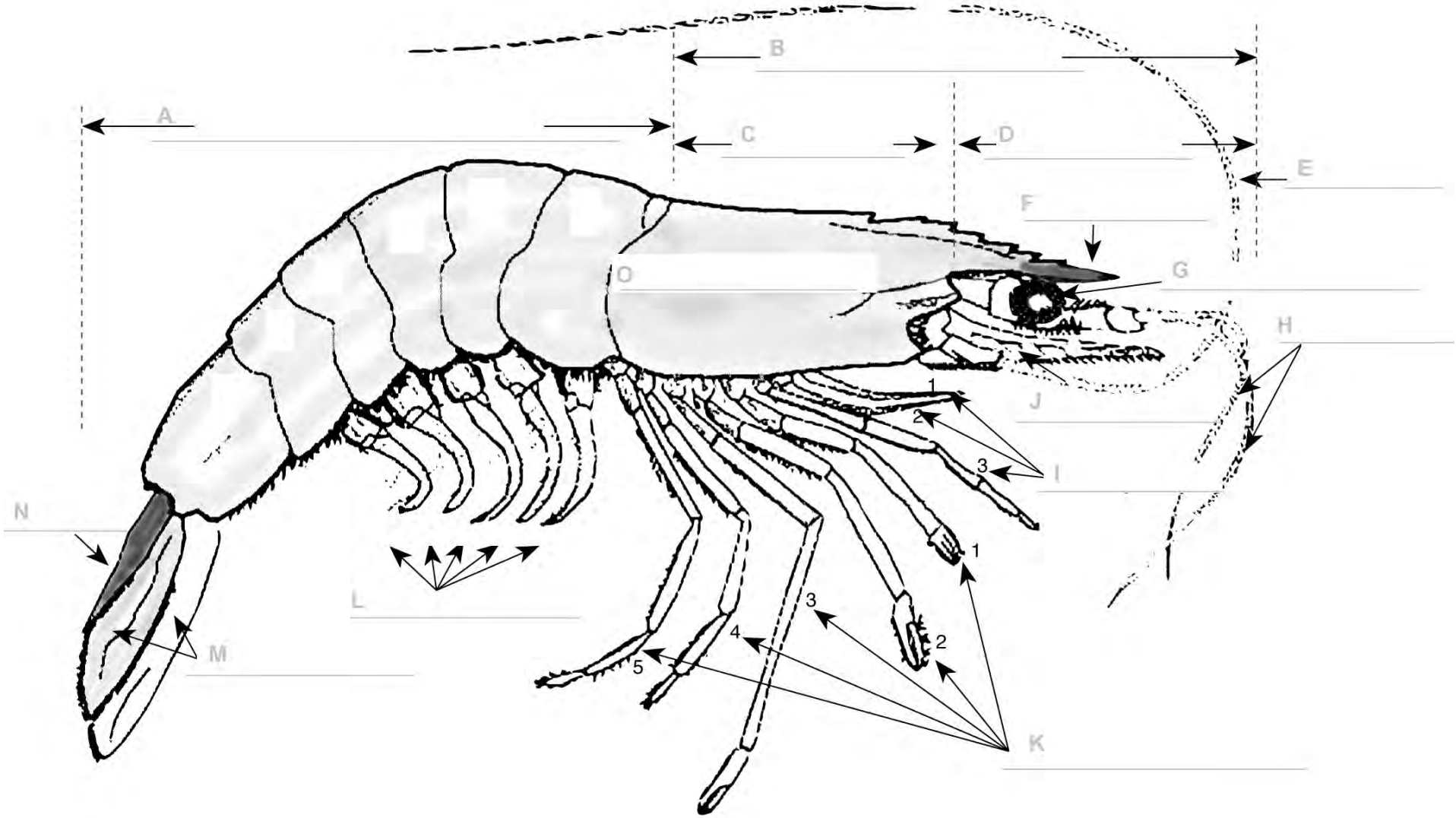


Figure 83.1 External features of a prawn - ventral view
(Stylised illustration Mark Moffatt, Mouthparts by Dr. Tom Hailstone 1967)

Students name _____ *Teacher* _____

Label the body parts A - L in the diagram below

Illustration Bob Moffatt



PROJECT 4.1 GROWING ALGAE

Background

Microalgae are phytoplankton, and like all plants, require warmth, light, carbon dioxide and water for photosynthesis and minerals for the thousands of reactions that occur within their cell or cells. They produce a range of essential fatty acids and other molecules needed for a wide variety of organisms further up the food chain.

Omega-3 fatty acids such as Eicosapentaenoic acid (EPA) and docosahexaenoic acid (DHA) play a vital role in many metabolic processes in animals including us. While popular magazines portray fish as the producers of these fatty acids, they do not make them at all, but obtain them from the microalgae they consume.

Much of the world's crude oil supplies originated in microalgae! They are extremely important producers in the marine food chain and like many plants are easy to grow if the conditions are right.

Conditions for growth

Light: Fluorescent grow tubes emitting in the red and blue ends of the spectrum are ideal light sources. Light should be provided for at least 18 hours each day – many hatcheries leave the lights on continuously to maximise growth.

Carbon Dioxide: Constant aeration is required to provide an ample supply of carbon dioxide . It also prevent clumping and sedimentation of the algal cells. The mixing, not too violent, will also ensure that all cells are exposed to light and nutrients.

Temperature: Between 20-24°C

Salinity: Slightly lower than seawater 20-25g/Litre

pH: In the 7-9 range with optimum growth around 8.

Nutrients: Guilliards F2 solution or Aquasol with additional iron

Materials

- Bunsen burner
- 250ml erlenmeyer flasks
- 3 litre erlenmeyer flasks
- Aquasol fertiliser or Guilliards F2 solution (Guilliards F2 solution can be purchased ready for use, from Algaboost or made up using CSIRO's modified recipe
  - Available in ready made concentrate www.algaboost.com.au
- Ferric citrate/citric acid solution (0.5mls of the solution made for Guilliards F2 added per litre of seawater when Aquasol is used)
- Sterilised diluted sea water to 20g/litre
- Algal growth containers (Project 1.3)
- Light cupboard (Project 1.4)
- CSIRO microalgae sites - methods and recipes for all media (see details page 116)
  www.marine.csiro.au/microalgae

Notes

Like many plants these algae are relatively easy to grow. CSIRO's Marine Laboratories, Microalgal Research Centre in Hobart are the experts, and are extremely helpful with advice. Their website has a detailed list of recipes and procedures for growing these algae.

They also have stock solutions of a huge variety of microalgae which they supply to schools at reduced educational rates

Three aspects need to be considered when growing the algae:

a. Making the growth medium
b. Keeping pure stock cultures
c. Growing the algae in large containers for feeding to animals

Making the growth medium

These algae are marine plants and grow in salt water – they do best in water with a salinity of between 20-25g/litre (seawater is 35g/litre).

Step 1 Filter a clean volume of seawater.

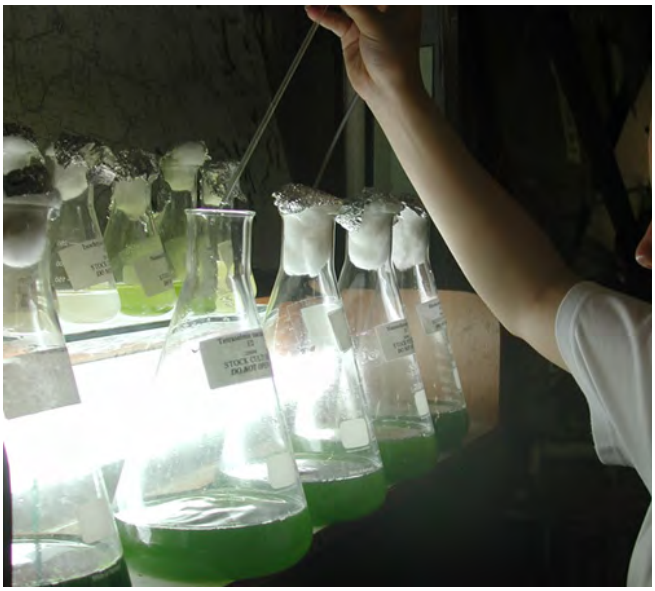
Step 2 Dilute the seawater – for every 660 mls of seawater add 340 mls of demineralised water (This will give a salinity of around 23g/litre).

Step 3 Sterilise the water in an autoclave or by boiling for 15 mins – add sterile demineralised water to return the volume if necessary.

Step 4: Bring the water to between 20-24°C by letting it stand in a sterile sealed container, a room in that range until the temperature is reached.

Step 5 Add the Guilliards F2 solution as per the CSIRO or manufacturer's instruction or alternatively add Aquasol at the rate of 80 mg of Aquasol per litre of seawater. (Dissolve the 80mg in demineralised water first, then add to the diluted seawater).

Step 6: Store the sterile medium ready for use.



Mick O'Connor

Figure 85.1 Maintaining stock solutions

Maintaining stock cultures

The greatest threat to the algae is contamination by bacteria - they can infect the container and grow quicker than the algae.

Good sterile practice is essential and if you have a laminar flow cabinet, transfers should be done in that with regular flaming of instruments and container openings.

Pure cultures of algae can be obtained by schools at non commercial prices for educational purposes from CSIRO's Marine Laboratories - see page 116.

The cheapest and easiest way to purchase the stock cultures is by 20ml test tube of starter culture. The starter culture comes with its own instructions.

- It is used to inoculate 150mls of growth sterile growth medium in a 250ml sterile flask.
- This is then used to inoculate 2 litres of sterile growth medium in a 3 litre flask which is known as the 'working culture'. It is from this culture that the plastic growth containers are inoculated to avoid contamination of the original master stock.

Growing algae in large containers for feeding to animals

- Project 1.3 outlines the steps needed to construct an alga growth container.
- Project 1.4 outlines the steps needed to make a light cupboard.

Using a lamina flow cabinet, or a fume cupboard with a UV light or a dust free room

Step 1 Three quarters fill the growth container with the sterilised sea water.

Step 2 Fertilise with the correct amount of Guilliards F2 solution or the Aquasol solution with added iron.

Step 3 Add 40 mls of algae from the working stock flask.

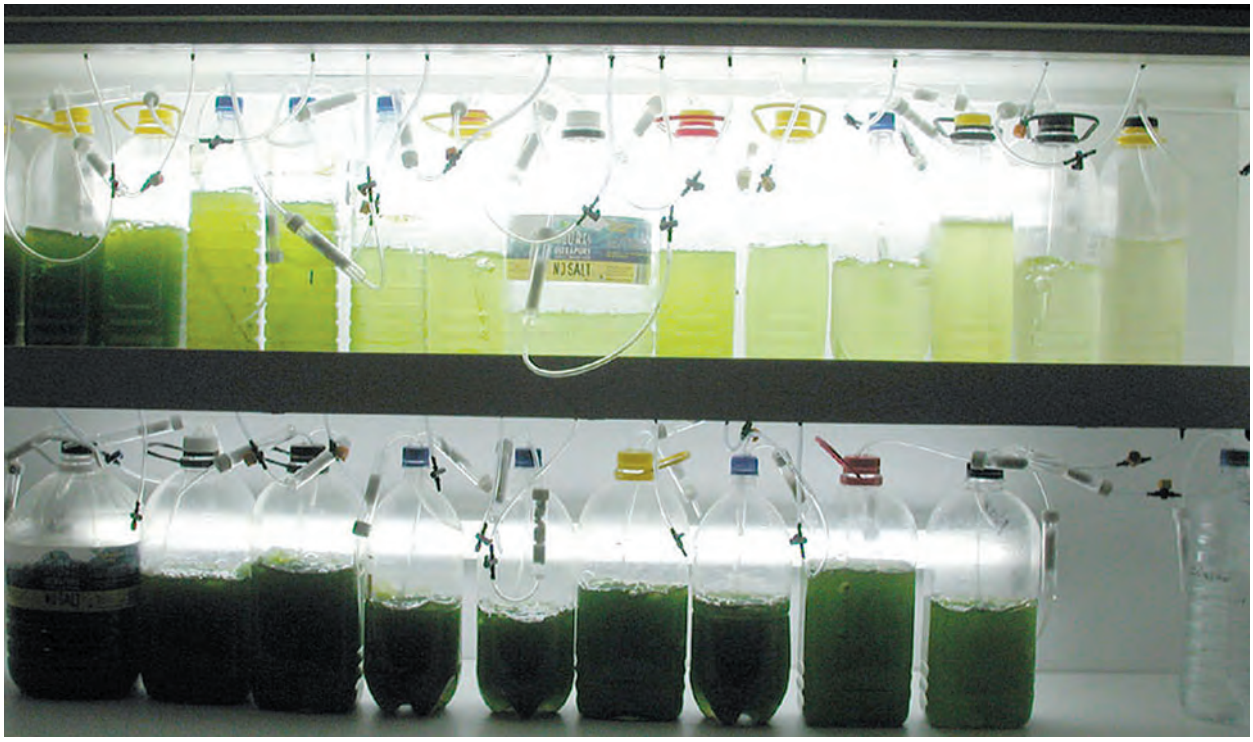
Step 4 Place in the light cupboard, attach to the airline and switch on the light.

Step 5 Turn on the air and adjust to a fine even flow.

Mick O'Connor

Figure 86.1 adding algae from work flask to the growth container

Mick O'Connor



Mick O'Connor

Figure 86.3 Producing algae in the light cabinet

PROJECT 4.2 GROWING ARTEMIA

Background

Artemia also known as 'brine shrimp' and 'sea monkeys' are small branchiopod crustaceans found throughout the world, with two genus found in Australia – one, *Pararartemia* , is found only in Australia.

Artemia are well adapted to withstand a wide range of saline conditions from almost fresh water to saturated salt solution.

They have the best osmoregulatory system in the animal kingdom and their success as a species lies in their ability to live in waters whose salinity kills their predators.

Artemia populations are most common in warm dry regions in where evaporation is high and given the right conditions artemia are easy to grow.

- They thrive in water whose salt concentration has been increased by evaporation or has become salty for other reasons.
- The Great Salt Lake in Utah, USA is the 'mother' of all lakes for brine shrimp, with salt concentrations almost twice that of sea water.
- At certain times of the year, long clearly visible lines of dense concentrations of atremia stretch across the lake – they are harvested and sold throughout the world.
- In Africa and Chile, flamingos strain the pinkish red brine shrimp from the water with their curved bills.

  Their carotene-rich artemia diet colours their feathers an attractive dark pink and the absence of the brine shrimp in the diets of these birds held in zoos explains their white rather than pink colour.

Materials

- Large shallow tub or aquarium
- Fresh sterilised sea water with a salinity of 35 grams of salt per litre
- Microalgae for food
- Aerator
- Light

Procedure

Step 1: Set up your growth container. Almost any non metallic tub can be used.

The key considerations are those that apply to all aquariums – the needs of what is being grown.

Artemia require good oxygen levels provided by gentle aeration; warm temperatures and a good source of food that does not send the water 'off'. Microalgae placed into the container live and kept alive by lights are by far the best.

- The container should incorporate all these – plastic tubs or glass aquariums are ideal.
- A filter will remove atremia, eggs, naupalli, and algae… no filter!!
- Commercially grown artemia are often grown in illuminated tubs in racks.

Mick O'Connor

Figure 88.1 Artemia trays

Step 2: Set the temperature in the range of 25-30 °C: The warmer the water, the faster their growth and reproduction rates.

- Room temperature is OK but it will take an extra week for the artemia to complete their life cycle – four to five weeks instead of three or four at warmer temperatures.

Step 3: Collect fresh clean sea water (salinity 35 parts per thousand) sterilise, settle and filter it before use – this will get rid of organic matter and natural organisms some of which may be harmful to your culture.

Step 4. Set up the aeration – atremia do like oxygen levels above 2 mg/L Growing brine shrimps do not like the rather violent aeration/agitation used in the hatcheries to prevent rafting.

- Gentle aeration through a stone, sparge bar or sponge will work well.

Step 5. Set up grow lights above the growth tank. The light is necessary to keep the algal food alive as you feed the artemia.

- The animals will eat large quantities of food as their densities increase.
- The secret is a combination of controlling densities and providing plenty of food without excess.

Step 6. When the tank is in place, add the water and let it come to the right temperature, add microalgae flowed by atremia.

- Switch on the aeration and lights and watch the atremia grow.

PROJECT 4.3 GROWING ROTIFERS

Background

Rotifers are brackish water microorganisms used widely in the aquarium and aquaculture industry as a ' first-feed' source for larval fin fish and invertebrates.

- They are about 150-300 µm in size have a short life span and reproduce quickly. They have little intrinsic nutritional value but are important carriers of nutrients particularly those found in microalgae.
- Rotifers are enriched by feeding them on the microalgae before they are in turn fed to livestock.

Brachionus plicatilis and *Brachionus rotundiformis* are two marine rotifer species that can be grown successfully.

The biological characteristics that makes them ideal food for fish larvae and invertebrates are:

- small body size and round shape,
- slow swimming speed and their habit of staying suspended in the water column,
- easily enriched with external nutrients resources, eg from microalgae,
- high reproduction rate and high density cultures.

Brachionus plicatilis has been most widely used as an essential food for raising marine fish, prawns and crab larvae because it can cope with the marine environment.

- Its use as a live food source for marine fish larval rearing started 43 years ago in Japan.
- The Chinese now consider rotifers to have more advantages than Artemia nauplii giving higher survival rates of prawn larvae when used as food.

Growth conditions

Containers

Rotifers can be grown in any container that provides the requirements to cope with their high rate of metabolism. They have short lives and live 'fast'! On a small scale they can be grown in the same containers as used to hatch artemia. The conical drink bottle vessels with gentle aeration from the bottom are ideal- gentle aeration provides good food and water circulation and the absence of fine bubbles in the air flow prevents clumping or rafting of the rotifers. Any vessel with gentle aeration is fine – buckets, tubs, aquarium tanks, and plastic bags can be used for larger scale production.

Salinity

Optimum growth 20-25 g/litre however rotifers will grow in salinities ranging from 15-35g/Litre.

Water temperature

Optimum growth 23-28°C. around room temperature is fine and the rotifers grow well.

pH

Slightly alkaline Optimum 7.0 - 8.0 Try to keep around 8.

Dissolved oxygen

Above 1.0 mg/L. The high metabolic rate of these animals mean they feed constantly and must be fed every four hours; they reproduce quickly and they consume a lot of oxygen in the process. Continual gentle aeration is needed to keep DO levels high, to keep the culture mixed and to prevent rafting of the animals.

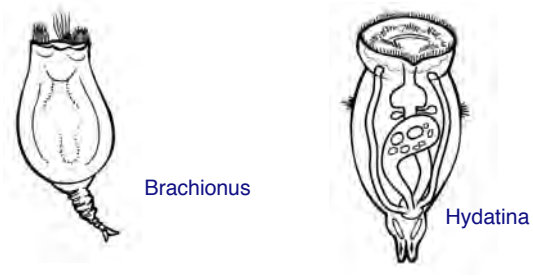


Figure 89.1 Rotifers
Illustrations Kerry Kitzelman

Light

Rotifers do not need light – they are attracted , but don't need it.

Foods

Microalgae - *Nannochloropsis*, is the best although *Nannochloris, Dunaliella, Tetraselmis*, and yeast are also used as food.

Because of the high metabolic rate and their inability to store fat, rotifers must feed constantly. Algae are added to the water until it takes on a green tinge.

- This must be maintained at all times by adding more algae – never let the culture run out of food – the rotifers will go into shock and die.
- Feed them every four hours or at least check them every four hours to see if the algae are still present.

Materials

- Drink bottle growth containers
- Microalgae or yeast for food
- Rotifer culture

Procedure

Step 1: Set up your inverted bottle growth container.

Step 2: Fill with dechlorinated warm water 23 - 28°C.

Step 3: Add microalgae.

Step 4: Add the rotifer culture.

Step 5: Turn on the air supply.

Remember:

- Rotifers do not need any light, and will do best in the dark.
- Dark may kill the algae and send the water off.
- Rotifers have a very high metabolism and need to eat every 4 hours.
- Do NOT let rotifers ever run out of food.
- Use a microscope to measure rotifer densities.
- Use trial and error to find an equilibrium how much algae to feed and how many litres of rotifers to remove each day without affecting culture densities.
- If the tank maintains its light green you have it right.

PROJECT 4.4 GROWING DAPHNIA

Background

Daphnia are crustaceans closely related to artemia. They are known as 'water fleas' because their jerky movements through the water resemble fleas hopping on land….. BUT THEY ARE NOT FLEAS!!

Daphnia are found in ponds, dams, creeks and streams, in fact, their presence is often an indicator of good water quality, ie, they are an 'indicator species').

- They are used widely in the aquarium trade as 'nutrient carriers' ingesting microalgae that small fish or fry that would not normally eat, and then being fed to these animals with the algae in their intestines or converted into their tissue.
- Daphnia can also be put directly into fish tanks and will remain alive in the tank until eaten. Unlike non living food they will not spoil the water.

Daphnia can be raised in anything from two litre plastic drink bottles to old fridge liners to children's swimming pools. Success does however depend on realising that there are a few restrictions, For example:

- Daphnia do not like fine aeration – they have a double walled carapace made of chiton - small bubbles in the air stream become trapped in between the walls causing lethal problems for them.
  - Dissolved oxygen needed by the daphnia for respiration comes from diffusion through the surface, so a growth container with a large surface area to volume ratio is ideal. Large plastic containers like the ones pictured are ideal (See Figure over).
- pH of the water should be slightly alkaline in the range 7-8.5 (this can be adjusted using sodium bicarbonate).
- Temperatures should be slightly cooler than normal room temperature in the range 18-24 celsius.
- Food for the daphnia must be in suspension – floating - they are filter feeders using leaf like legs to make water currents that bring food particles to the oral (mouth) grove.
  - The food must be small enough to fit in their oral groove (up to about fifty micrometres).

Materials

- 'Pure' daphnia culture
- Growth trays
- Sodium bicarbonate
- Clean water dechlorinated water

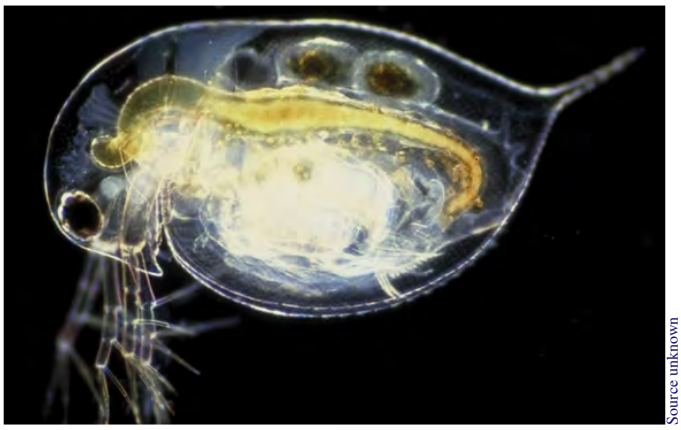
Procedure

Step 1 Allow the water to be used to come within the correct temperature range.

Step 2 Half fill the containers with water and adjust the pH if necessary using the sodium bicarbonate.

Step 3 Place the daphnia culture in a plastic bag and float in the water to allow water temperatures equilibrate.

Open the bag and release the daphnia.



Source unknown

Figure 90.1 Daphnia

Step 4 Daphnia need a light period (photoperiod) of at least 10 hours light each day.

Feeding notes

1. Daphnia will feed on very small organic matter eg detritus, yeast, bacteria and micro algae.
2. Remember they are filter feeders and ingestion rates are directly dependant on concentration of food in the water column.

   Like any filter feeder, daphnia find it difficult to pump a lot of water for little food. – the food must be plentiful and must be in the column NOT on the bottom.
3. Old time Daphnia growers used horse manure that had been aged for a week, as food suspended (inside a cotton bag) into the growth container.

   The scientific reasoning behind its success is that daphnia love the decaying organic matter and the organisms that break it down, particularly the coliform bacteria found in the gut of mammals. Horse manure was found to be better than cow sheep or goat poo!
4. Dried yeast, dried malted milk, wheat bran and 'just boiled' lettuce leaf and of course microalgae all make good food.
   - The organisms found in hay or grass infusions also are good food but care must be taken to avoid harmful organisms from the infusion being fed to the livestock with the daphnia.
5. Food should be prepared in a separate container to the growth container and then fed to the daphnia.
   - Remember there is no aeration in the growth container and the introduction of large amounts of organic matter will take the available oxygen from the water suffocating the daphnia.
6. Daphnia will clear the water as they feed – it is important to keep the feed up to them.
7. Harvesting should always leave sufficient daphnia to continue reproducing – never take more than 20% of the daphnia out of the culture.
8. Careful watch should be kept on water quality and exchanges made regularly as required using a siphon and stocking or 100 micron filter to return any daphnia to the growth container that get sucked up in the siphon.

Figure 91.1 Growing trays

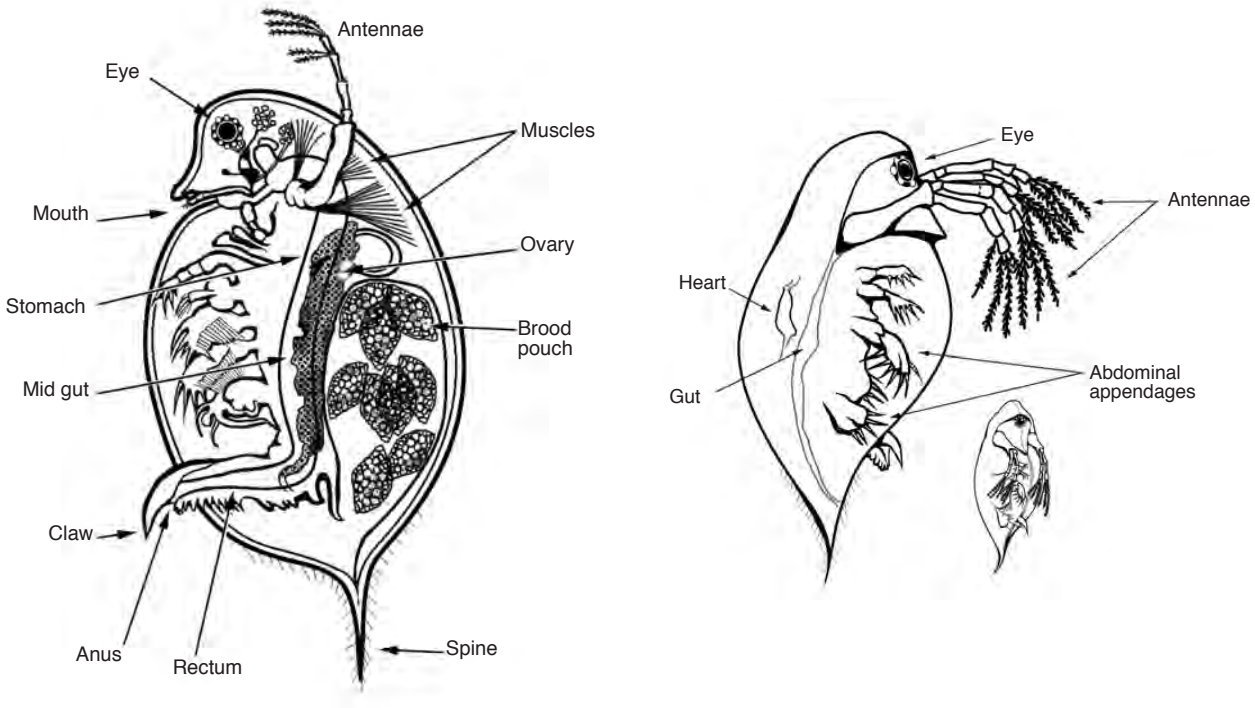


Figure 91.2 Daphnia biology
Illustrations Kerry Kitzelman

PROJECT 4.5 GROWING WORMS

Background

Even in a cartoon, the humble worm is the most common food used to catch fish. Worms, grubs, maggots and larvae that have worm shapes are favoured by many fish species, and are used and sold widely as fishing baits. Many plastic lures used by anglers resemble the shape, colour and action of worms.

Blood worms, beach worms, tiger worms, red wrigglers, Indian blues and common earthworms are loved by many fish, and are easy to grow as a source of high protein fish food. Whole, cut and minced worms are used as an excellent food for growing fish.

This project gives some ideas for successfully raising worms - you should try local worms with your fish first to see which they like, then use the procedures given in this exercise to grow those worms.

Materials

- Shallow stackable plastic tubs, foam fruit box, wooden box
- Bricks or stand

Procedure

Feeding worms to your fish will depend on the type and size of the worm you are growing and the size of the fish you are feeding.

You will need two plastic stackable tubs.

Step 1: Drill 6 mm diameter holes in the bottoms for drainage, and in one of the lids for ventilation, in a 50 mm square grid pattern.

Step 2: Drill a single row of ventilation holes 25mm down from the top rim of the tub and 50mm apart in a ring air around the sides and ends.

Step 3: Put the intact lid on the ground (ie the one with no holes), and put place four bricks on it, one in each corner. This will support the farm, allow it to drain and is to keep the worms from getting out into the ground.

Step 4: Put one of the tubs on the bricks.

Step 5: Line the tub with wet newspaper. Half fill it with "bedding" materials - a mix of damp compost, shredded paper (watch poisonous inks), leaves, straw, peat moss or hay. Add a small amount of grit - handful of sandy loam soil to help the worms' digestion.

Spread worms to the surface of the bedding material - they will burrow down, and cover them with nice loamy soil.

Step 6: Place the second tub on the soil inside the first tub, so that its bottom is firmly in contact with the soil. Worms will move up from the lower tub as they use the food and breed in the lower tub.

Step 7: Add some wet wadded newspaper or shredded paper. Mix in kitchen scraps - bread coffee grounds, tea bags, and vegetable trimmings, but not too much meat and no fat and then cover with a little bedding material.

Then cover the lot with a wet hessian bag, old blanket or other natural fabric to prevent drying out and to prevent insects entering.

Step 8: Maintenance

- Put the lid with ventilation holes on the top tub and add some weight to prevent rats and other scavengers getting in.
  - Keep the worm farm as wet as a soaked sponge.
  - Keep an eye on the pH. Do not let it become acidic as it will allow ants and cockroaches to take up residence.
- Feed the worms by adding kitchen scraps and wet paper to the top bin once or twice a week, or as often as needed. Make sure the contents are always moist. Rotate the bins every few months when the top worm bin has plenty of worms, and the food in the bottom bin has been eaten.
- The worms initially placed in the bottom bin grow and reproduce then move up through holes to get to the food in the top bin. When all the food is used up in the bottom bin, that bin is emptied and rotated to the top. The old top bin will become the new bottom bin, setting up a continuous cycle.

2. Locating the farm

- Worm farms can be kept indoors in the garage, shed, laundry but most people keep them out of the house!
- If the farm is kept outdoors it should be kept out of direct sunlight, where it will remain cool and moist.

Feeding the worms

Worms will eat:

- Most fruits and vegetables (except citrus fruits, onion, garlic and chilli)
- Tea bags/tea leaves
- Coffee grounds and filters
- Egg shells
- Newspaper, cardboard, egg cartons and pizza boxes (shredded and soaked)
- Hair
- Old flowers and small amounts of garden waste

Worms don't like (ie. avoid feeding these to your worms):

- Onions, garlic, citrus fruits (oranges, lemons, limes, mandarins) and chillies - worms breathe through their skin and these are too acidic for them.
- Meat
- Seafood
- Dairy products (milk, cheese)
- Oil
- Too much bread, pasta and rice

Commercial worm farms

These can be purchased from hardware stores or nurseries. To set them up just follow the manufacturers instructions.

Handling live animals

The old saying 'once bitten twice shy' rings true because when handling live animals you not only need to look after the well being of the animal, but you also need to look after yourself!

Consideration for animals

Any handling of wild animals will cause them some stress, but the 'escape' mechanisms for aquatic and marine animals is often very strong and can result in mechanical damage to the animal.

Work on the principle that just as *you* don't like being held under water, aquatic animals don't like being held in air. For example, fish have a mucus coat on them that must be protected when removed from water; their eyes generally protrude and are easily damaged and scales tear off easily when caught on nets and containers.

Consideration for yourself

Many fish have spiny fins and bony plates around their gills and heads. Crayfish have nippers and spikes and a lot of animals have teeth that can inflict terrible damage.

The chances of painful punctures, bites or lacerations is high if you are careless or do not understand the animal you are handling.

As a rule of thumb, a fish's mouth or a yabby's claw is no place to be putting your hands unless you know exactly what you are doing. (Remember bream can crush oyster shells with their mouths!)

All education authorities in Australia have developed documents that outline guidelines for handling animals in schools. These documents must satisfy all national and state legislation that governs the treatment and use of animals for educational purposes.

Teachers and students should be familiar with and adhere to these guidelines particularly those relating to aquariums and aquaculture.

In New South Wales visit www.schools.nsw.edu.au/animalsinschools and in Queensland http://education.qld.gov.au and search the curriculum activity risk assessment documents for handling live animals.

After you have read all the documents and are ready to handle your livestock, these hints may help you.

Crayfish

- They escape backwards with a massive inward flick of their tail.
- To catch them with a net put the net behind them then place your hand in front of them – not too close to the nippers though they will flick back into the net!
- To pick them up press down gently on the top cephalothorax and pick them up with the thumb and forefinger grasping the rear sides of the cephalothorax. Turn your hand over when you pick them up to look at the underside
  - if they latch on to your finger with a nipper, just bear the pain and place back in the water – they will let go
  - if they have latched onto a net, put them back in water with the net and they will release
  - they will continue to 'flick' while you are holding them to 'escape' your grip so be ready!

Prawns

- Prawns are nice animals to handle but….
  - they have a big spiny rostrum. The prawn doesn't come swimming at you with it , however people do use it to spike themselves when handling prawns.
- The easiest way to handle prawns is with a small fine woven net, but remember, like crayfish, they escape backwards.

Fish

- Fish will flip about as soon as you try to remove them from the water. They are trying to escape by swimming and by driving their spikes into you.
- If you are catching them in a tank, cut down the size of their swim (escape) area by using a removable partition and remove them using an 'environet'. This is a net with no knots because it will not:
  - remove the mucus from the fish,
  - damage protruding eyes,
  - split fins (including tails) or remove scales.
- Handle with a moist fine woven glove that will not damage the scales or remove mucus.
  - If you get spiked place your hand in hot water until the sting goes and then apply a disinfectant.
  - Consult your doctor if you have any doubts.

Oysters

- There are no attack or escape mechanisms for oysters – they just shut their shells!
- However the shells are sharp and can cut deeply into your skin.
  - Often more than one oyster shell is present making the outside of the shell dangerous.
- The safest way to handle oysters is with a good pair of gloves – even the professionals use gloves!

Bob Moffatt

Figure 93.1 Take care when handling live animals

PROJECT 5.1 DETERMINING THE SEX OF ARTEMIA

Background

Artemia spp or brine shrimp are small shrimp that live in salt water.

The adults are approximately one centimetre in length with the females slightly larger than the males.

It isn't too hard to tell the males from the females.

- Males have large "claspers" (over-developed antennae) on their heads
- Females have two large egg sacs in their tail area.

Materials

- Binocular (dissecting) microscope
- Plastic disposable petri dish
- Two one millilitre polyethylene transfer pipettes
- Adult artemia

Procedure

Step 1 Take one of the pipettes and using a pair of scissors cut the tip off so there is no constriction.

- This will be used to suck up Artemia without damaging them.

Step 2 Suck up a few Artemia from a culture using the pipette.

Step 3 Place them in the centre of a petri dish in as little water as possible.

Step 4 Using the uncut pipette remove as much water as possible so the Artemia cannot swim around.

- It may be more convenient to place one Artemia in its own drop of water around the dish so that you can look at each one individually.

Step 5 Focus on the lower magnification first and then switch to the higher one if needed

- Red food colouring diluted 1:10 with water may help provide contrast and may make the features easier to see.

Step 6 Use the diagrams from the page opposite to identify the sexes and count the number of males and females.

- Record this in your note book or as a class result on the board.

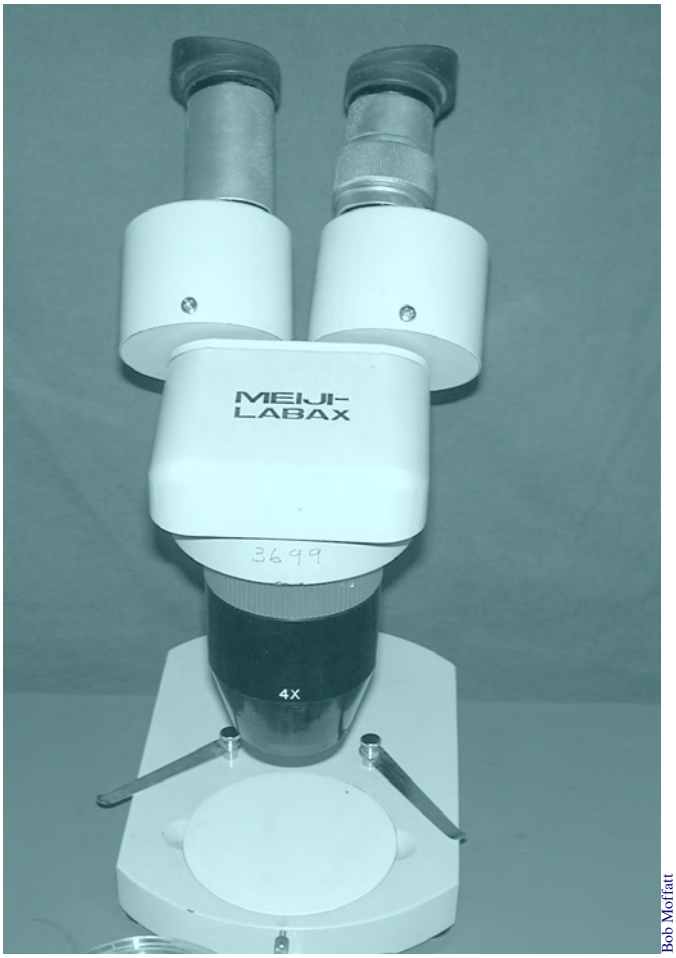
Questions

Answer the following questions in class or in your notebook.

Q1. Describe a simple method to identify the sexes of large artemia in a growth container.

Q2 . Explain why the artemia cannot move in the bubble.

Q3. Explain why a plastic petri dish is used rather than a glass one.



Bob Moffatt

Figure 94.1 Binocular microscope

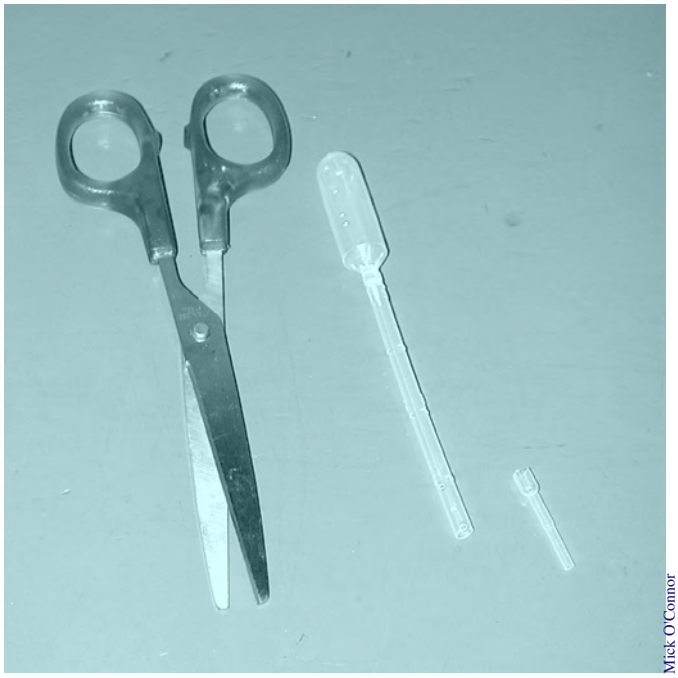

Mick O'Connor

Figure 94.2 Equipment

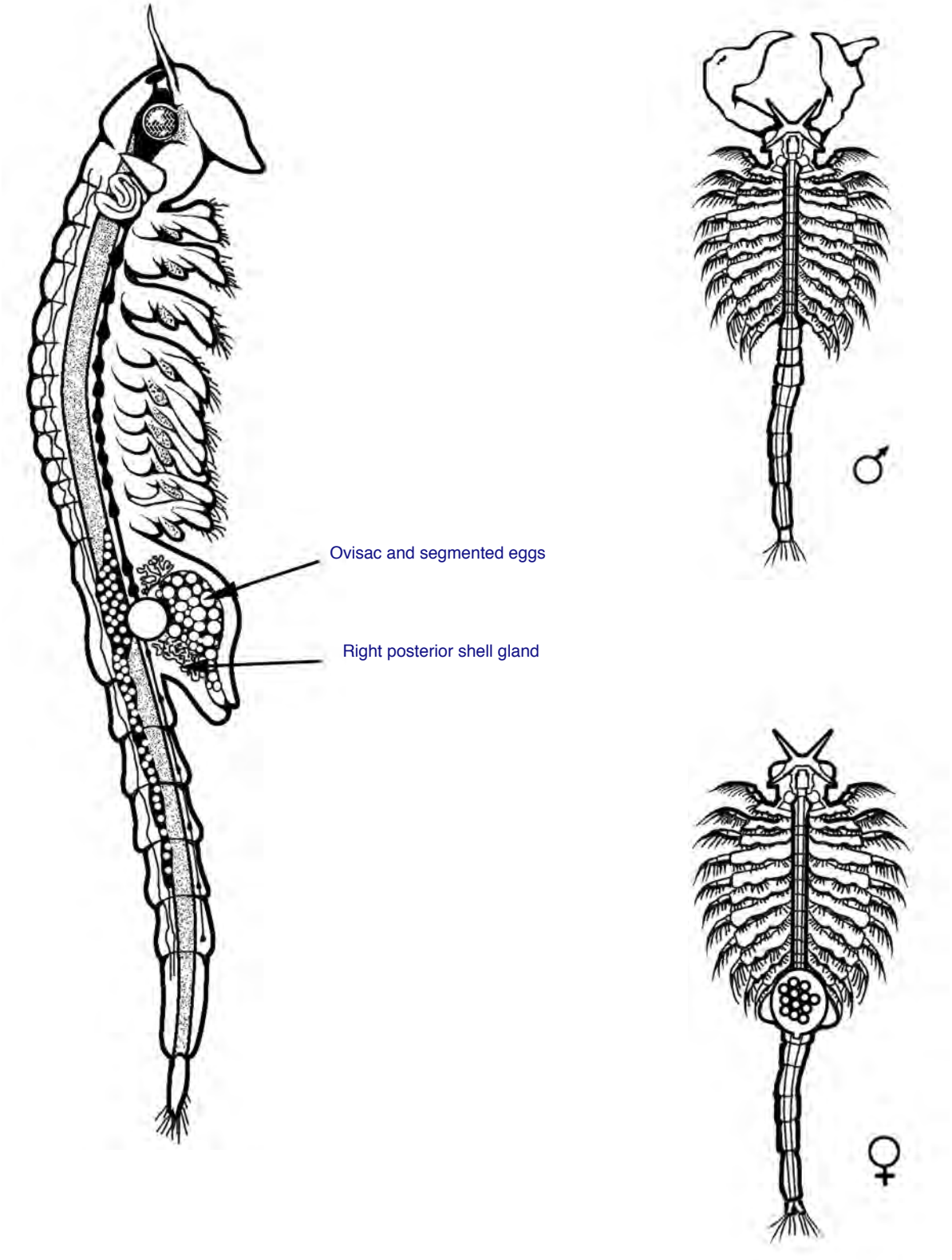


Figure 95.1 Artemia external features
Illustrations Kerry Kitzelman

PROJECT 5.2 DECAPSULATING ARTEMIA

Background

Artemia survive in nature by encysting - putting embryos into a state of suspended animation inside a hard shell or 'cyst' . The cysts are dehydrated.

The diagram below shows a cross section through a fully hydrated cyst .

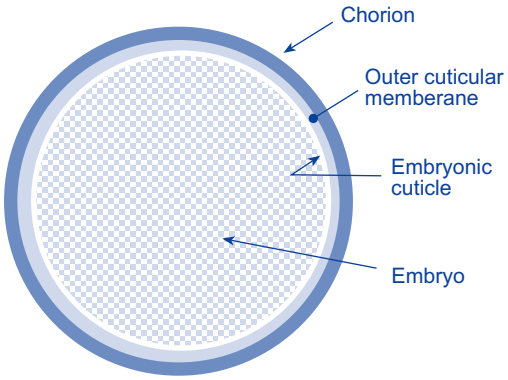


Figure 96.1 Artemia egg

(Bob Moffatt, After National Key Center for Teaching and Research in Aquaculture, Tasmania)

- The chorion, also known as the 'capsule', is a shell made up of lipoproteins reinforced with haematite (iron ore) which gives the cyst its brown colour. It is this indigestible shell that causes gut problems when eaten by many fish larvae.
  - This must be removed before the 'egg' will hatch.
  - It is removed by oxidising it with a 2.6% sodium hypochlorite solution without affecting the embryo in a process known as decapsulation.
- The outer cuticular membrane is directly under the chorion and allows the movement of small molecules only such as O_2, H_2O and CO_2.
- The embryonic cuticle is like a plastic bag , a transparent and elastic membrane surrounding the embryo.
  - During incubation the embryo comes through this like a tear drop.
- The embryo is the young artemia in a state of suspended animation - a bit like hibernation, alive but going nowhere.
- Decapsulation removes the chorion or shell to give the embryos a better chance of hatching.

  Other advantages of decapsulation includes that it:
  - Does improve the hatching rate.
  - Removes the need to disinfect cysts,
    - this is done by the hypochlorite solution.
  - Allows decapsulated cysts to be fed directly to stock without worrying about gut complications caused by ingesting the chorion.
  - Removes the need to separate the shells from the hatching artemia before feeding them to stock.

  However, if you get it wrong, you will kill the embryos and nothing will hatch!!

Decapsulation involves three stages:

Stage 1: Hydration of the dehydrated cysts.

Stage 2: Dissolving of the chorion with sodium hypochlorite.

Stage 3: Washing the cysts with demineralised water to remove any chlorine remaining from the hypochlorite solution.

Stage 1: Hydration of the cysts

Cysts are in a dehydrated state and resemble a dried pea - have a look under the microscope before you start.

Hydrating them will cause them to swell, becoming spherical and allowing the sodium hypochlorite to do its job of oxidising the chorion (Figure 96.2).

Figure 96.2 Dehydrated cyst Hydrated cyst

(After National Key Center for Teaching and Research in Aquaculture, Tasmania)

Materials

- Artemia hatchery made in Project 1.9
- Household bleach with a 4% (40g /L) sodium hypochlorite concentration
- Distilled or demineralised water
- Plastic spatula
- Electronic scales
- 100-130 micrometre filter or stocking or coffee filter

Procedure

Refer - Figure over

Step 1. Fill the artemia hatchery up to the 350 mL mark with distilled water.

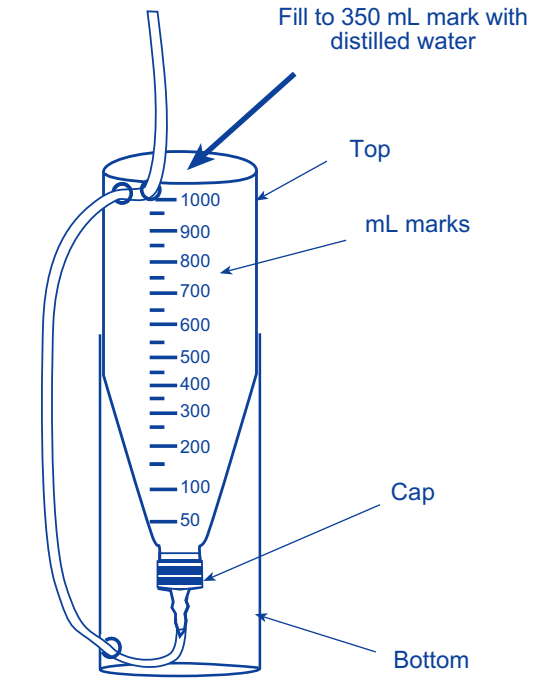
Step 2 : Using the spatula and the beam balance weigh out 5 grams of dehydrated cysts.

Step 3: Tip the 5 grams of dehydrates cysts into the water in the hatchery.

Step 4: Turn on the air supply to give an even stream of small bubbles that will keep the cysts circulating but will not force them violently to the tom and make them stick to the sides.

- Push any back that get stuck on the sides with the spatula.

Step 5: Aerate for 2 to 3 hours. The cysts are now ready for decapsulation.



Notes

1. Decapsulated cysts can be purchased from K mart and aquariums shops.
2. However a better result and a greater educational experience is gained by performing the decapsulating process

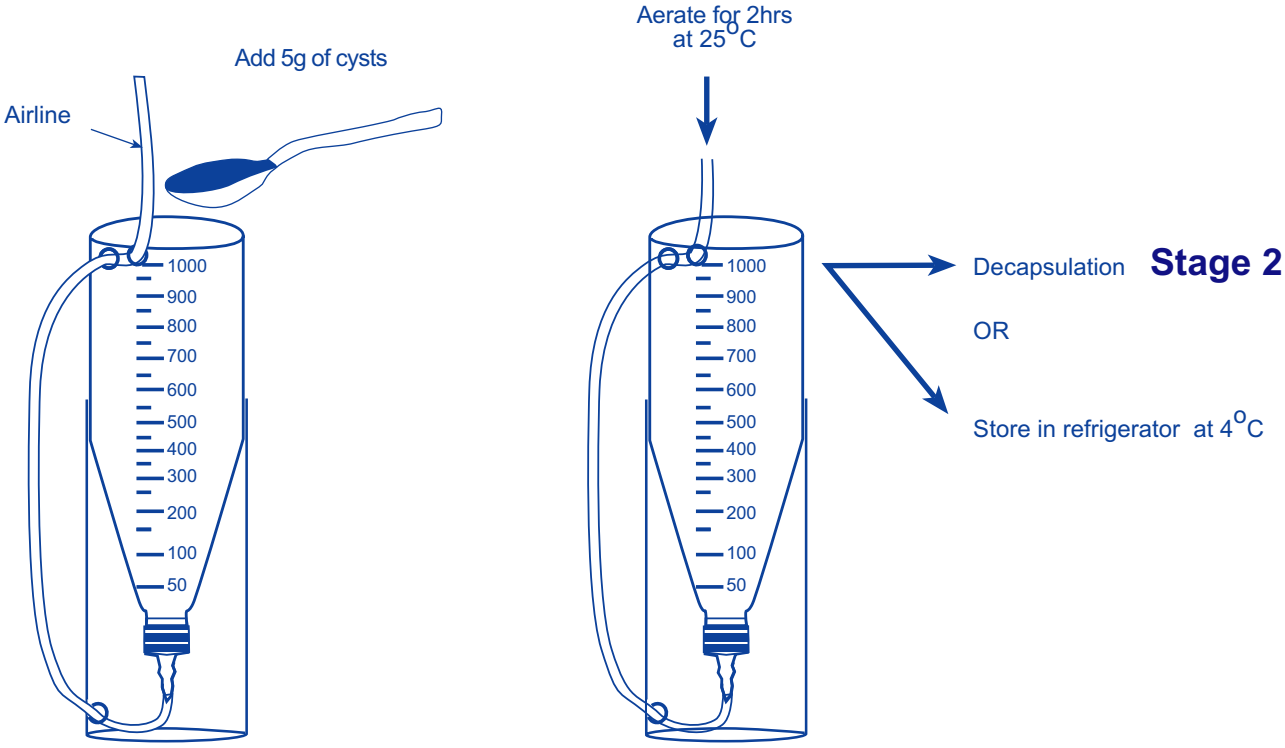


Figure 97.1 Stage 1: Hydration

(Illustration Bob Moffatt, After National Key Center for Teaching and Research in Aquaculture, Tasmania)

Stage 2: Decapsulation

(Refer Figure 98.1)

Cysts are immersed in a 2.6% sodium hypochlorite solution for about five minutes to dissolve the chorion capsule.

Care must be taken not to harm the embryo with too strong a bleach solution or leaving them in the solution for too long.

Fortunately the cysts change colour as the capsule dissolves, from dark brown to grey to white and finally to orange.

- Note that the amount of water used will depend on the strength of the bleach used.
- The formula is 1000 - (2600/figure value of the bleach concentration). Examples:
  - if the bleach is 4% then
    1000-(2600/4) = 350 mls;
  - if the bleach is 3.5% then
    the water needed would be 1000-(2600/3.5) = 257 mls.
- The (2600/bleach concentration) gives the volume of bleach to be added during decapsulation to make 1 litre of 2.6% hypochlorite needed to dissolve the chorion.

Procedure

Step 1 Place the hatchery with the 5 grams of hydrated cysts in the 350 mls of water on the bench so that it cannot fall over.

Step 2 Carefully add 650 mls of 4% household liquid bleach (the solution is now 2.6% sodium hypochlorite).

Step 3 Turn on the air supply and adjust to ensure there is an even supply of small bubbles that will keep all the cysts moving through the sodium hypochlorite.

- Use a plastic spatula to remove any stuck to the sides.

Step 4 Keep a close check on the colour. Be vigilant from 4 minutes onward.

Step 5 View some cysts under a dissecting microscope .

- They should be spherical in shape and orange all over.

Step 6 When the decapsulation has been completed wash all cysts immediately in clean water.

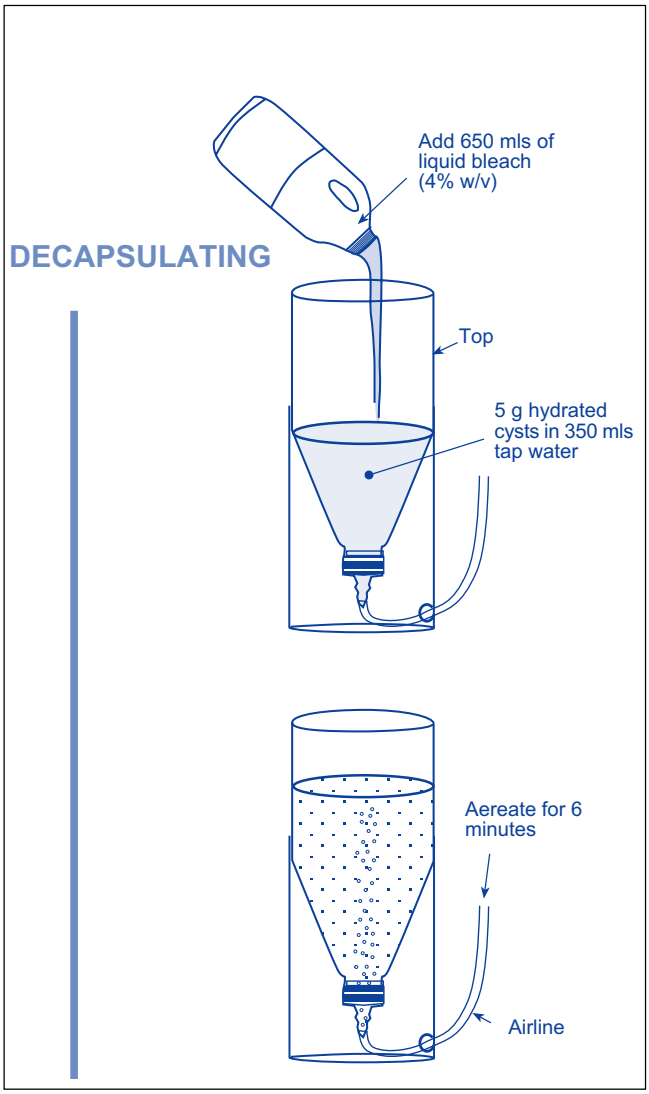


Figure 98.1 Stage 2 Decapsulation

(After National Key Center for Teaching and Research in Aquaculture, Tasmania)

Stage 3: Rinsing

(Refer Figure 98.2)

Rinsing with clean water is essential to stop the chemical oxidation process that decapsulates the cysts.

- If it is not done immediately the oxidation will continue and kill the embryo.

Procedure

Step 1 Put all the cysts into a 100-130 micrometre filter. (Stocking or coffee filter is fine).

Step 2 Run clean tap water over the cysts for at least five minutes or until there is no smell of chlorine.

Step 3 The decapsulated cysts are easily damaged without their shell.

They should be hatched immediately or placed in a plastic container and put in the fridge - BUT they will only last for a few days.

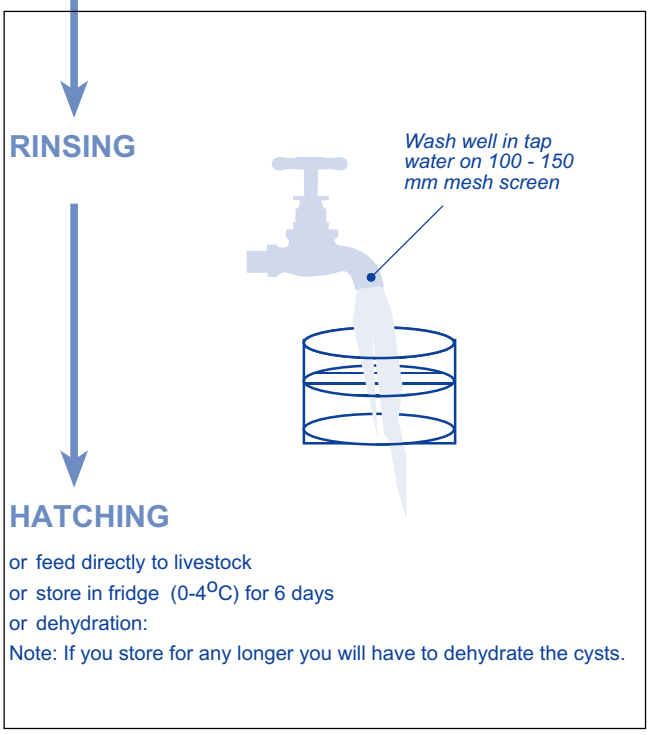
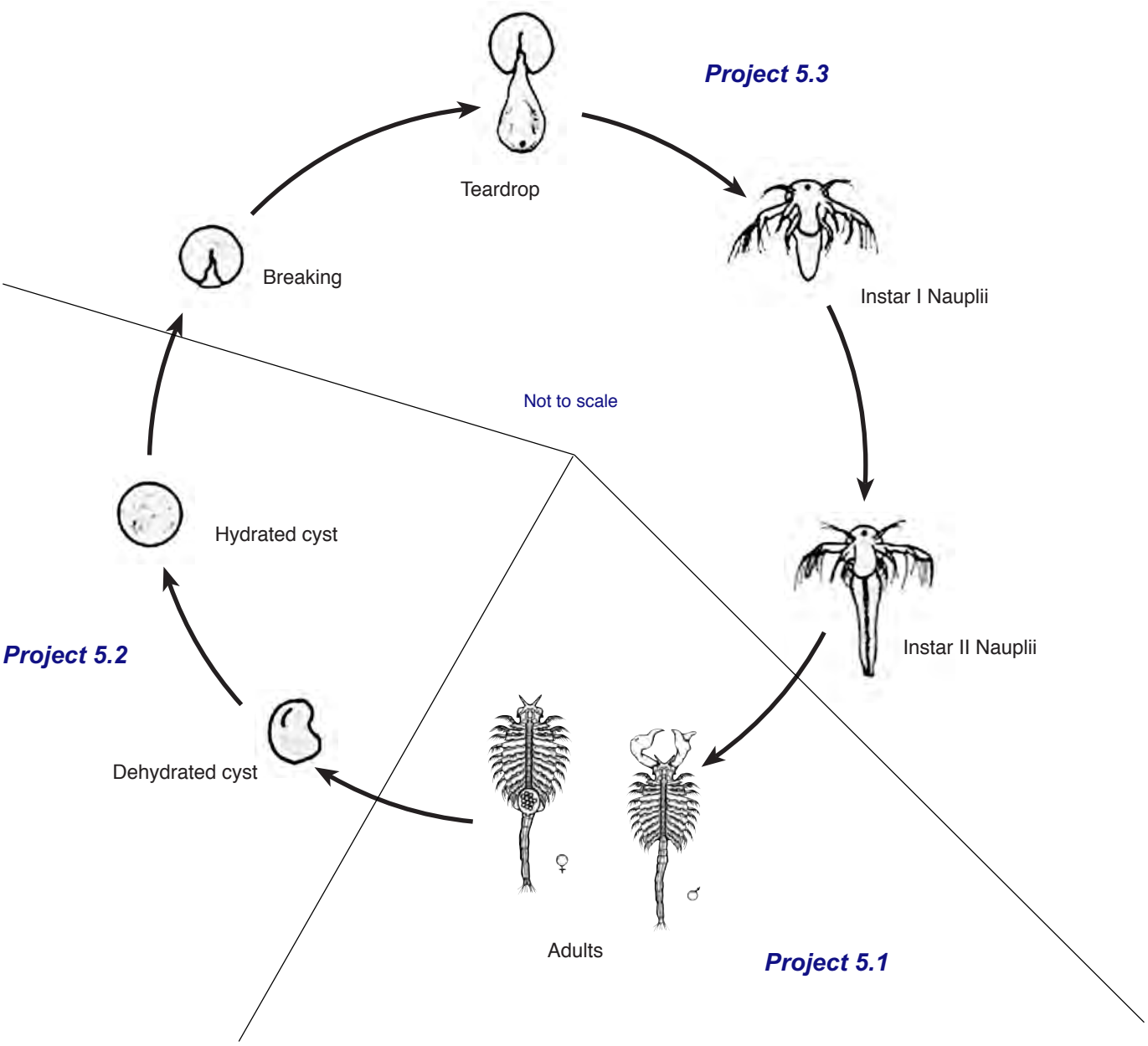


Figure 98.2 Stage 3 Rinsing

(Illustration Bob Moffatt, After National Key Center for Teaching and Research in Aquaculture, Tasmania)



Stage 1 Dehydrated cyst	Stage 2 Hydrated cyst	Stage 3 Breaking
Stage 4 Teardrop	Stage 5 Hatch (Instar I Nauplii)	Stage 6 First moult (Instar II Nauplii)

Figure 99.1 Artemia life cycle and project summaries (Not to scale)
(Illustration Bob Moffatt, After National Key Center for Teaching and Research in Aquaculture, Tasmania)

PROJECT 5.3 HATCHING ARTEMIA

Background

Cryptobiosis is the biological state known as 'suspended animation' that allows organisms to survive harsh environmental conditions.

When the female senses environmental conditions that may kill small artemia, she coats the eggs in a protective cyst.

The development of the embryo stops at about 4000 cells and remains in the environment in that state until conditions improve.

- The embryo senses the change and begins to develop bursting out of the cyst – this is called hatching.

Hatching any egg requires care and the right conditions – artemia are no different careful watch must be kept on temperature, salinity, pH, oxygen levels, cyst density and light levels if hatching is to be successful.

To get the best results use keep your hatching conditions within the following ranges.

1. Temperature in the range of 25-30 °C:
2. Salinity around 20-25 parts per thousand NaCl
3. Oxygen levels above 2 mg/L
4. Cyst density should be less than 5 grams (one and a half level teaspoons) of decapsulated hydrated cysts per litre.

Materials

- Artemia hatchery from Project 9
- Salt water with a salinity of 20-25 grams of salt per litre
- Plastic tea spoon and plant pot
- Strong light
- Dissecting microscope

Procedure

Make sure all water being used is around 25 °C, and the area where hatching will occur is at the same temperature.

- The light cupboard is a great place and the light may aid hatching (See Project 1.4).

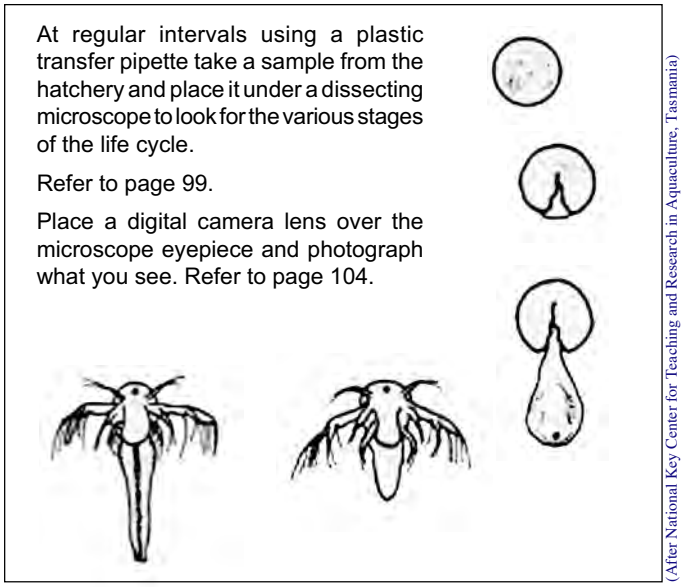


Figure 100.1 Stages of development

Step 1 Fill the hatchery to the 1 litre mark with salt water with a salinity of 20ppt (see Figure 100.2).

If you are using seawater you will have to dilute it 570 mls of pure seawater mixed with 430 mls of distilled water to give a salinity of 20ppt.

Alternative you can dissolve 20 grams of aquarium salt in 800mls of water and then add water to make the total volume up to one litre.

Step 2 Add one and a half teaspoons (5 grams) of decapsulated cysts to the water.

Step 3 Turn on the air and adjust to give regular and small bubbles. If foaming occurs immediately remove the cysts and re-wash them, then repeat step 1.

Step 4 Maintain the temperature around 25-30 °C with gentle aeration until the cysts have hatched – this may take 24 hours.

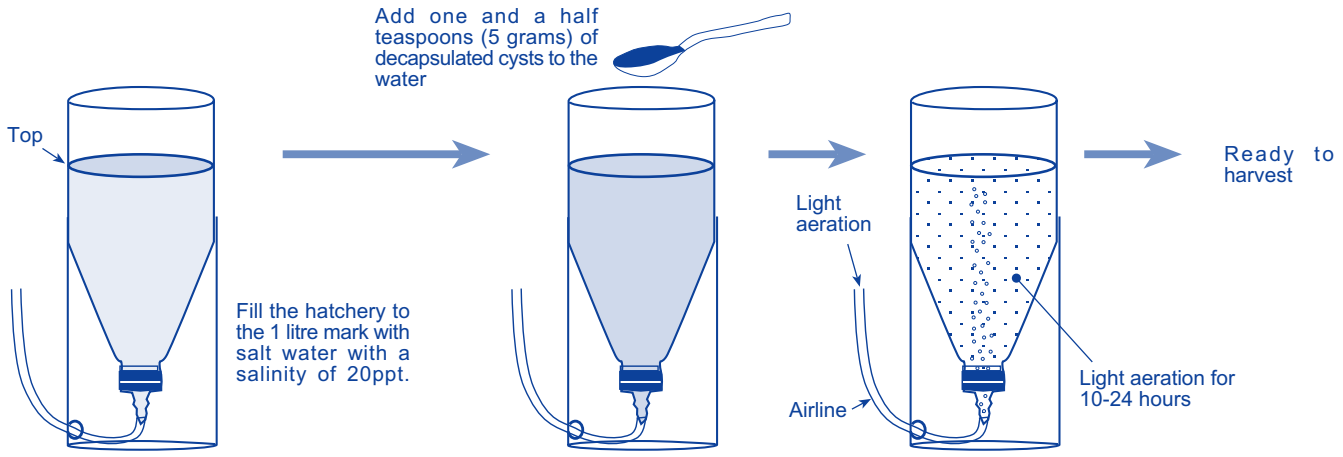


Figure 100.2 Hatching procedure
(Illustration Bob Moffatt, After National Key Center for Teaching and Research in Aquaculture, Tasmania)

Step 5 Harvesting nauplii (See Figure 101.1 and 101.2)

- The good thing about nauplii is that they are phototactic - attracted to light.
- To harvest the Nauplii, remove the air tube from the retainer holes at the top of the hatchery and cover the top of the hatchery with an inverted plastic flower pot (see Figure 101.1).
- Shine a light onto the uncovered section of the bottom of the hatchery.
- After a few minutes lower the airline to drain out the nauplii into a petri dish or beaker (see Figure 101.2).
- Now examine under a dissecting microscope.

Mick O'Connor

Figure 101.1 Havesting naupli

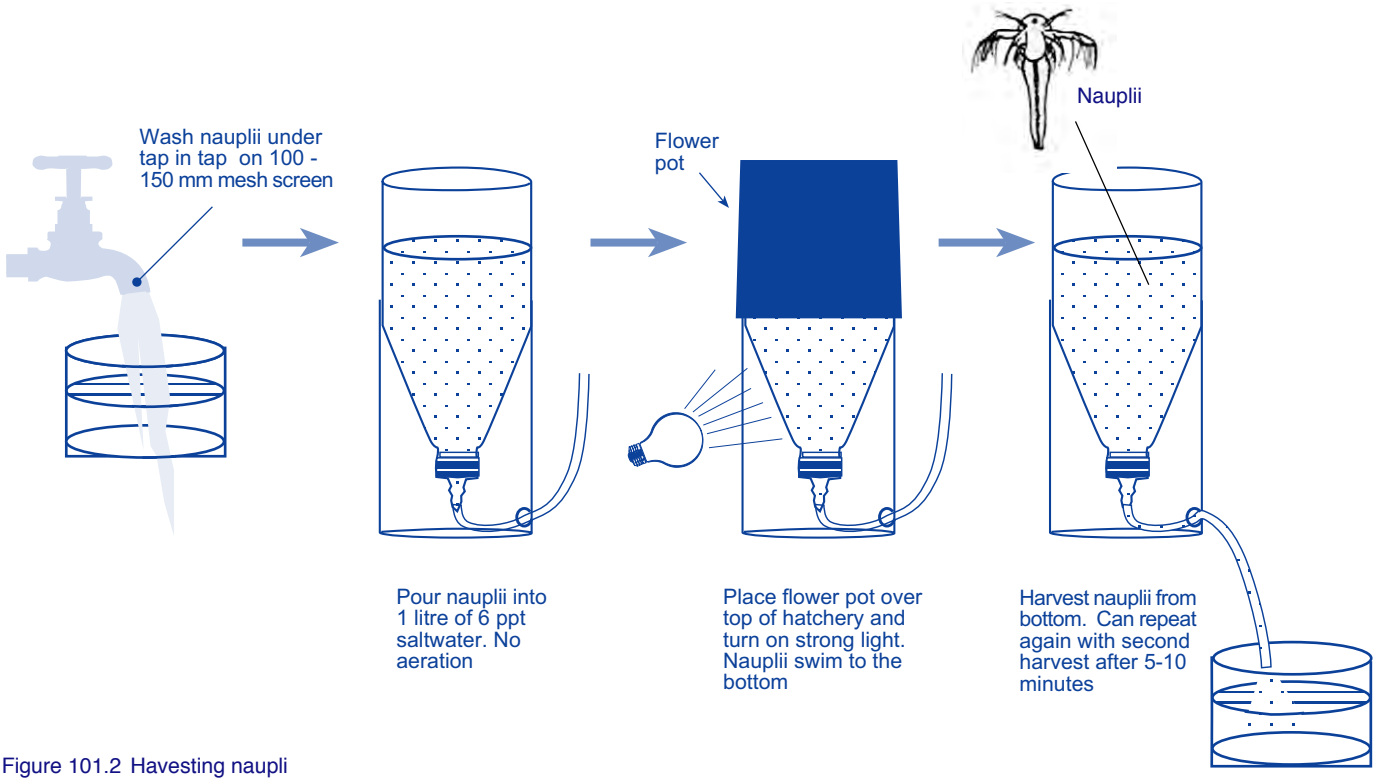


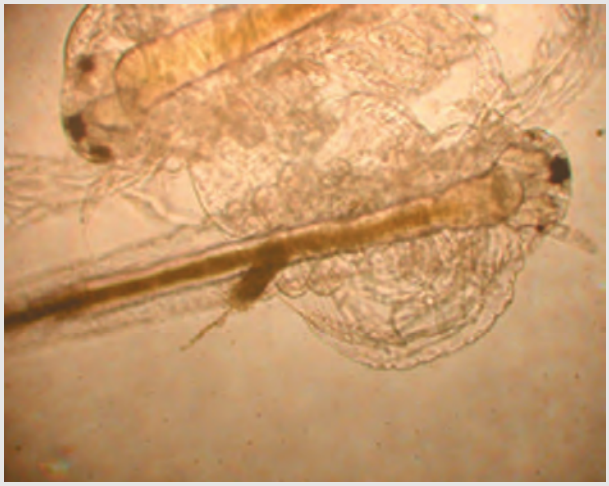
Figure 101.2 Havesting naupli

(Illustration Bob Moffatt, After National Key Center for Teaching and Research in Aquaculture, Tasmania)

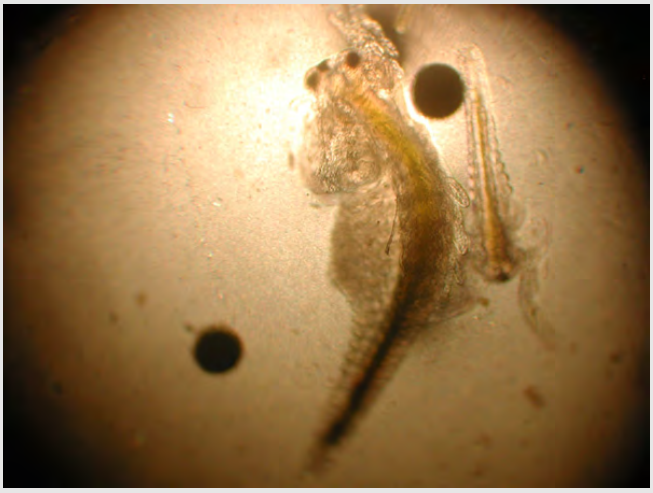
PROJECT 5.4 TAKING DIGITAL PHOTOGRAPHS

At regular intervals using a plastic transfer pipette take a sample from the hatchery and place it under a dissecting microscope to look for the various stages of the life cycle. Some samples are shown below.

Adult and egg

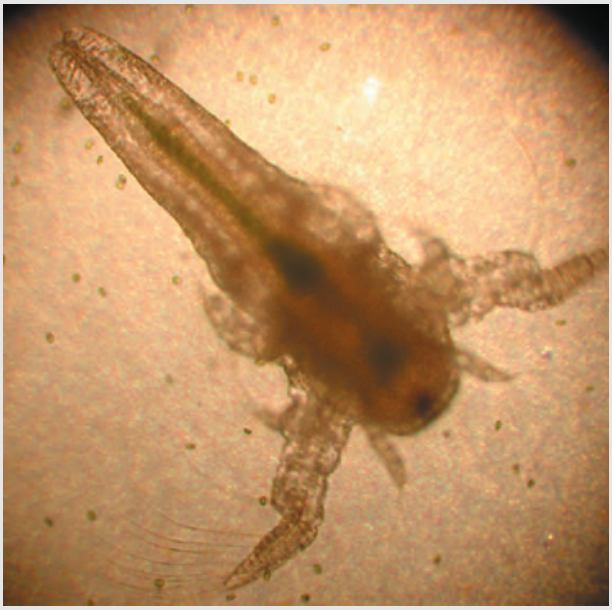


Adults

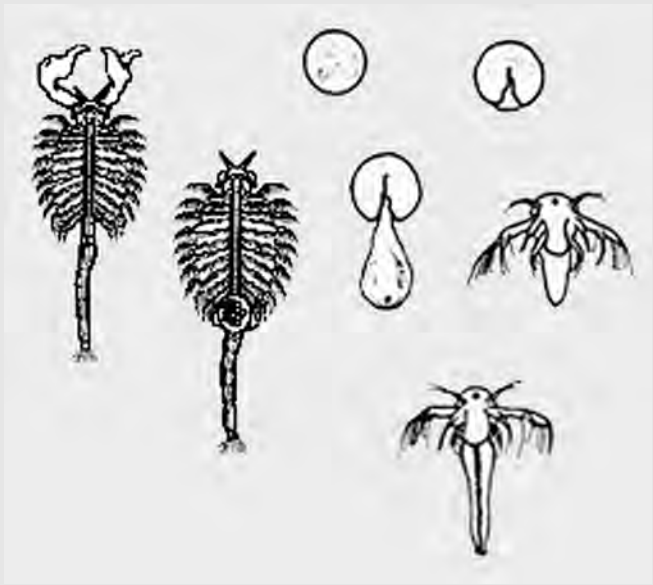


One adult, two eggs and Instar II Nauplii

Preadult



Instar II Nauplii



Various stages

(After National Key Center for Teaching and Research in Aquaculture, Tasmania)

Mick O'Connor

Figure 102.1 Example of digital photographs

PROJECT 5.5 DETERMINING THE SEX OF CRAYFISH

Background

The sex of crayfish can be determined quite easily by looking for the reproductive organs on the underside of the crayfish.

These reproductive organs or genital papillae in the male crayfish are short projections on the bases of the last pair of walking legs.

The female has oval openings on the bases of the third-last pair of legs.

Just to make it interesting and a little confusing....

- It is common (1 in 20) to find individuals with a combination of male and female openings.
- These 'intersexes' usually prove to be of one sex and can function sexually; they are rarely true hermaphrodites able to produce both eggs and sperm.

The female yabby reaches sexual maturity when about 9 to 10 centimetres long

- The male when slightly smaller. (Note: Length is measured from the tip of the rostrum - the spine between the eyes - to the end of tail fan.)
- Nearly all mature females spawn, but the majority of young are produced by 2 year olds, as they outnumber the older age groups.

When freshwater crayfish mate, the male deposits a small packet of sperm gel on the female, near the reproductive openings.

The female then passes the eggs out through the openings and across the sperm packet.

As the eggs pass through the sperm they are fertilised.

- The eggs are guided to the underside of the tail (kept cupped during egg laying), where they are fastened on to the swimmerettes (the small legs on the abdomen) and carried until they hatch.
- Juveniles have special hooks on their legs to allow them to cling to the hairs of the female's swimmerettes; they moult several times before leaving the parent.

Background reading

Chapter 13 of *An Introduction to Marine Studies, 2nd Edition*, gives a good background to crayfish biology and breeding.

Available from:

www.wetpaper.com.au

Materials

- Mature yabby

Procedure

Step 1 Take the yabby holding it around the cephalothorax to avoid being bitten.

Step 2 Turn your hand over so you can see the underside.

Step 3 Look for the reproductive organs.

Step 4 Determine and record the sex using the Figures 103.1 and 103.2.

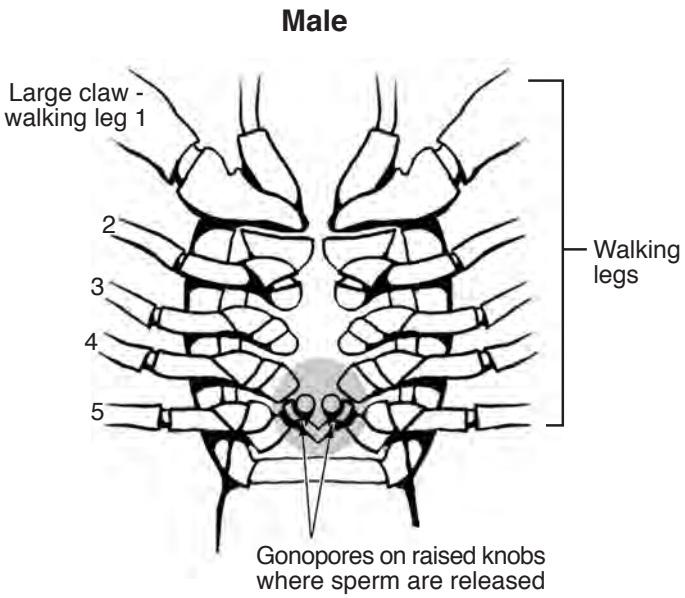


Figure 103.1 Male crayfish
Kerry Kitzelman

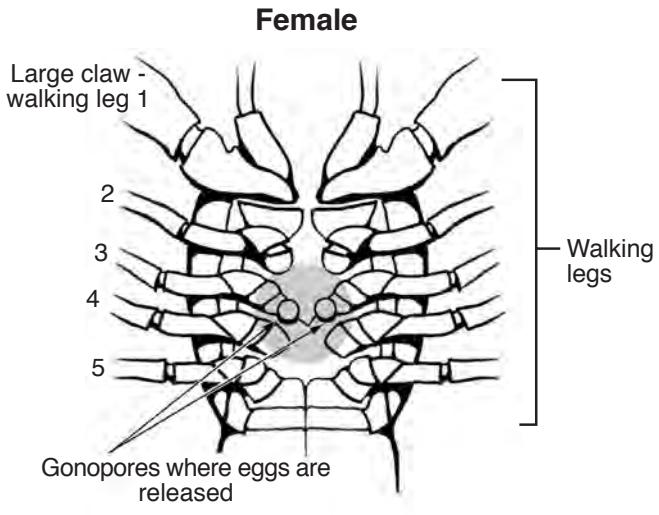


Figure 103.1 Female crayfish
Kerry Kitzelman

PROJECT 5.6 BREEDING CRAYFISH

Background

This project is restricted to the most widespread and commonly used yabby in schools, Blue Claw (*Cherax destructor*), so aptly named given its ability to destroy almost anything in its way, only to be eclipsed by its 'knack' of escaping from any aquarium without curved overhangs.

Yabbies are ideal animals for school aquaculture because:

- They are prolific breeders, easy to grow, hardy, can be kept in glass aquariums, attractive and provide students with hours of amusement if not distraction with their antics in their tanks.
- Their life cycle can be readily observed and studied as the young hatch and stay with the female in the early laval stages

The complete life cycle of the yabby is easily observed and starts with mating male and female.

Life cycle

Project 5.5 details how to determine the sex of crayfish by looking at the underside and counting down from the head,

- Females have oviducts (bumps) at the base of the second pair of walking legs.
- Males have male genital papillae (bumps) at the base of the fourth pair.

Yabbies are prolific breeders and reach sexual maturity before they are one year old.

- They will mate up to five or six times in a summer breeding season with large mature females able to produce up to four hundred eggs at a time.
- When conditions are right the male yabby turns the female on her back and deposits a sperm packet know as a spermatophore near the oviducts where the eggs will come out.
- The female then curls her tail under with her tail almost touching her body to form a protective chamber. She then and picks open the spermatophore to mix sperm with her eggs as she lays them from her oviduct.
- As the green coloured eggs are fertilized, the female attaches them with 'glar' (a powerful glue) onto the fine 'hairs' on her swimmerettes.
- The eggs become black and then as the embryo develops its yellow yolk sac can be see just below the surface of the egg. They will hatch after about three weeks (20-40 days) depending on water temperature.

After hatching the young yabbies will go through three larval stages with a moult in between each stage before swimming away from their mother.

- In these stages they attach themselves to the same 'hairs' as the eggs attached to, but with a special snap hook in their tail rather than glue.
- Immediately after hatching - the first stage baby Yabby doesn't really look like and adult yabby. Its cephalothorax is large and out of proportion – it contains yellow yolk sac for the young yabby to feed on.

Bob Moffatt

Figure 104.1 Aquarium yabby condo

- After a moult, the yolk sac is smaller and the yabby more developed. By the third stage, the young yabbie looks like a small adult – it is still attached but can walk off the female to forage.
- After a third moult, the juveniles become independent of their mother. Once the young yabbies have left the mother she will mate again.

Conditions for mating

- Water Temperature - between 25^0C and 28^0C
- Dissolved Oxygen - above 3 mg/L
- pH Levels - between 7.5 and 8.5
- Alkalinity and Water Hardness - a lack of calcium in the water will also result in soft shelled yabbies. Have some marble chips or dead coral in the tank.
- Male/female ratios - in ponds normal ratio is 1 male to 3 females. In an aquarium 1:1
- Light - 14 hour photoperiod

Materials

- Aquarium and heater (optional)
- Lights and timer
- Male and female crayfish
- Shelter – PVC pipe or extruded bricks

Procedure

In the wild crayfish reproduction is brought on by increases in day-length and in water temperature, corresponding with the onset of spring. Try to mimic these conditions although spawning females do not eat much.

Step 1 Set up the aquarium with timer lights, shelter and water adjusted to just above 20^0C. Set the timer light to give at least 14 hours of light.

Step 2 Select a male and female yabby as close as possible to the same size.

Step 3 Introduce both into the aquarium and begin gradually lifting the water temperature to 27^0C over about a week.

Step 4 Keep an eye on the crays as soon as they are introduced, for fighting – if needs be add more shelter or a temporary glass partition to avoid damage or death. Look to who is the aggressor for future reference and do not breed from him or her.

Step 5 The crays will do the rest!! Observe and photograph.

PROJECT 5.7 HATCHING CRAYFISH AND CARING FOR YOUNG

Bob Moffatt

Figure 105.1 Crayfish with eggs

Background

See Project 5.6

Conditions for Hatching

- Water Temperature - Between 25^0C and 28^0C
- Dissolved Oxygen - above 3 mg/L
- pH Levels - between 7.5 and 8.5
- Alkalinity and Water Hardness. - have some marble chips or dead coral in the tank yabbies to provide calcium needed for their exoskeleton during moulting

Materials

- Aquarium
- Lights and timer
- Male and female cray fish
- Shelter – PVC pipe or extruded bricks and shadecloth

Procedure

Remember that crayfish mate and hatch young in spring – Increasing daylight or in this case a photoperiod of about 14 hours and warm temperatures are required.

As the hatching and subsequent growth occurs oxygen levels should be kept above 5mg/litre

Step 1 Male and female can be left in the aquarium with ample shelter, water adjusted to just above 20oC and set the timer to give at least 14 hours of light.

Step 2 Carefully monitor the female over the three week hatching period.

Step 3 Monitor each moult – photographing provides a good exercise.

Step 4 Cut three shadecloth strips 30 mm wide and 400mm long.

- Double them and bind the folded ends 25mm from the fold with string or rubberband to form a 'jelly fish shelter' for the young crays to hide in.

Step 5 Sterilise the shelter and add to the tank when the first baby swims away from the mother.

Step 6. Canibalism is now a real risk – keep up the feed and introduce more shelter if required.

Step 7 Remove the babies and place in a separate tank with shelter and plenty of food.

Step 8 Regularly size the population segregating the crays according to size.

Bob Moffatt

Figure 105.2 Commercial red claw condos

PROJECT 5.8 FEEDING ALGAE TO ARTEMIA

Background

Project 4.2 outlines the equipment and processes needed to grow atremia.

The algae are fed to the atremia in their growth container, and will survive the shock of going from their algal culture concentration of about 25 g/litre salt to the full strength sea water that the atremia are growing in.

The shock will be lessened each time algae and their water is fed to the atremia if water is taken out of the livestock tank after the algae are added rather than prior, as described in the method below.

Materials

- Jar or large beaker
- Fine mesh aquarium net
- Algal culture (*Nannochloropsis* - often called instant algae)
- Atremia culture

Procedure

Step 1. Place the aquarium net over the open end of the beaker or jar to prevent atremia entering.

Step 2 Use the jar or beaker to take a portion of water out of the livestock tank.

- Discard.

Step 3 Stir the algal culture.

Step 4 Using the same jar/beaker, take the same amount of water containing algae from the algal culture tank and place in the livestock tank.

Step 5 Continue process until the water remains a light green.

- The light will keep excess algae alive in the atremia tank and will ensure a continued food supply for the atremia who will eat when they feel like it.

Step 6. Using a suitable suction tube remove some atremia and place under the dissecting microscope.

- As they are transparent you will be looking inside them and will be able to see the algae that they have eaten.
- By feeding a mixture of different algae studies can be performed to see if one species is preferred as food over another.

Background reading

Chapter 14 of *An Introduction to Marine Studies, 2nd Edition*, pages 166-167 gives a good background to different ways stockfood is used.

Available from:

www.wetpaper.com.au

Instant algae

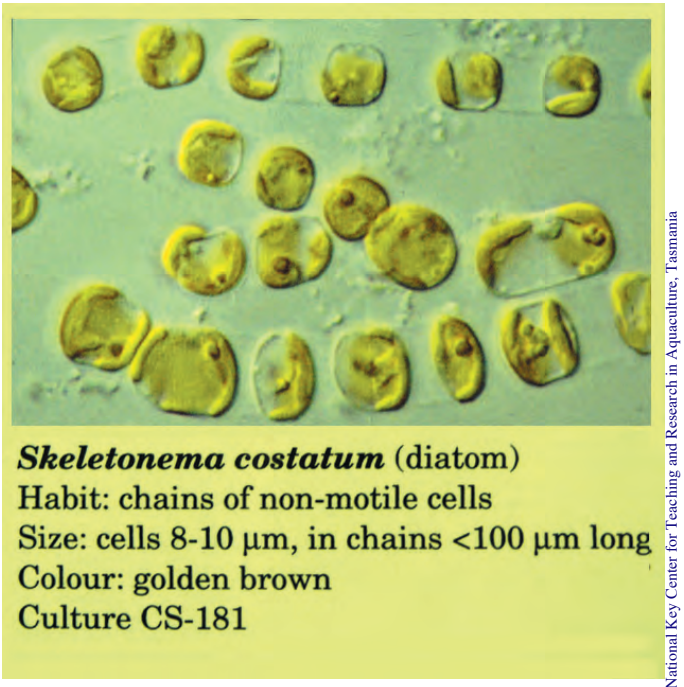
Nannochloropsis can be purchased from

Marine Laboratories Microalgae Research Centre in Hobart,

Phone 03 6232 5316, Fax 03 6232 5471.

MICROALGAE SITE

www.marine.csiro.au/microalgae



National Key Center for Teaching and Research in Aquaculture, Tasmania

Figure 106.1 Algae (*Nannochloropsis*)

Mick O'Connor

Figure 106.2 Artemia adult

PROJECT 5.9 FEEDING DAPHNIA TO LIVESTOCK

Background

Daphnia are an excellent live food for aquarium and aquaculture species being readily accepted by a wide range of vertebrate and invertebrate organisms.

The daphnia are often fed microalgae prior to feeding to other livestock - this practice is called enrichment with the daphnia acting as a carrier and food source in their own right.

When feeding live food there is always the risk of transferring other organisms to the livestock tank from the food culture site. Care should be taken to avoid culturing and transferring pathogenic or parasitic organisms.

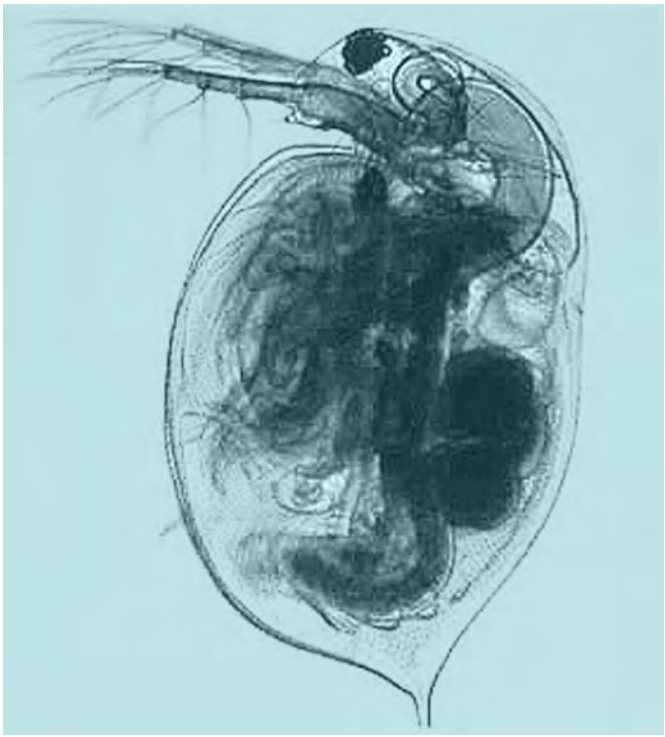


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Figure 107.1 Daphnia

Materials

- Small hand held plankton net
- 100 micrometre or stocking sieve
- Siphon
- Two five litre buckets

Procedure

The simplest procedure is to harvest Daphnia is by dragging a hand held plankton net through the culture and transfer the daphnia directly to the livestock tank - however it may damage some daphnia!

A less shocking method is to harvest and introduce gradually.

Step 1 Take 1 litre of water from the daphnia culture and one litre from the livestock (that you will feed the daphnia to) tank.

Step 2 Mix them in one of the buckets, then filter through the 100 micrometre or stocking filter into the other bucket – keep this filtered water.

Step 3 Return the filtered daphnia and organic matter to the daphnia tank.

Step 4 Wash the original bucket and the sieve in clean water.

Step 5 Half fill this clean bucket with water from the daphnia tank and stand the sieve in it so the water just covers the sieve membrane.

Step 6 Using a siphon suck up daphnia and direct into the sieve.

- Lift the sieve out of the bucket and transfer the daphnia in the sieve to the bucket with the 50:50 filtered mix for the daphnia to acclimatise.

Step 7 After an acclimatisation period tip the daphnia and water into the livestock and return the siphoned water to the culture tank.

- Do not take too many daphnia or your culture may crash – never take more than 20% at any harvest.

Feeding and health check tables

Use the tables over to record feeding rates.

PROJECT 5.10 FEEDING ROTIFERS TO LIVESTOCK

Background

Rotifers are the most common and widely used 'first food' for fin fish larvae and invertebrates in the aquarium and aquaculture industries. In themselves they are not nutritious – they lack the highly unsaturated fatty acids needed by fish larvae for survival and growth.

Rotifers must be enriched by feeding microalgae rich in these essential fatty acids to them, before they in turn are fed to the fish larvae or invertebrates. They act as nutrient carriers for transporting the high-value essential fatty acids (EPA and DHA) and other nutrients from the microalgae to the target species.

- *Nannochloropsis* (Figure 108.2) is the most common enriching algae used in hatcheries and is the most successful.
- It contains high concentrations of almost all essential fatty acids but lacks the omega-3 fatty acid DHA, and is blended with other algae containing DHA.

Harvesting

Rotifers should be harvested every day, taking 20-30% of the culture. Remember that these animals only live for 8-12 days and are most fertile in the first few days of their lives with egg production declining rapidly after that.

Daily harvesting ensures that the culture always contains young and fertile rotifers.

Source unknown

Figure 108.1 Rotifer

Materials

- Large beaker or jar
- Stackable sieves 150 microns and 50 micron (Feeding method 2)
- Syphon and bucket

Procedure

Two basic methods of feeding rotifers to livestock are feeding the rotifers in their culture medium to the livestock or separating the rotifers and feeding them to livestock.

Feeding the rotifers in their culture medium to the livestock

Step 1 Using a jar or beaker take a portion of water out of the livestock tank.

Step 2 Now using a coffee filter, filter this water into the rotifer culture. The filter will take out any material from the livestock water preventing contamination of the rotifers.

Step 3 Stir the rotifer culture.

Step 4 Turn off the filter in the livestock tank.

Step 5 Using the same container, take the same amount of water and rotifers from the culture tank and place in the livestock tank.

- leave the filter off for about an hour while the livestock feed on the rotifers.
- If the filter is left turned on it will take the rotifers out of the water before the livestock can feed on them.

Separating the rotifers and feeding them to livestock

Step 1 Arrange the stackable filters with the 120 micron filter on top of the 50 micron.

Step 2 Now syphon 20-30% of the rotifer culture into the top filter and let it run through the bottom one into the bucket.

- the top 150 micron filter will take out any organic matter and allow the rotifers to go through.
- the 50 micron filter will allow the water to go through but will hold the rotifers.

Step 3 Switch off the filter in the livestock tank and transfer the rotifers from the filter.

Step 4 Measure the water in the bucket, then discard half of it.

Step 5 Add an equivalent amount of clean water as discarded, back to the rotifer culture effecting a partial water change.

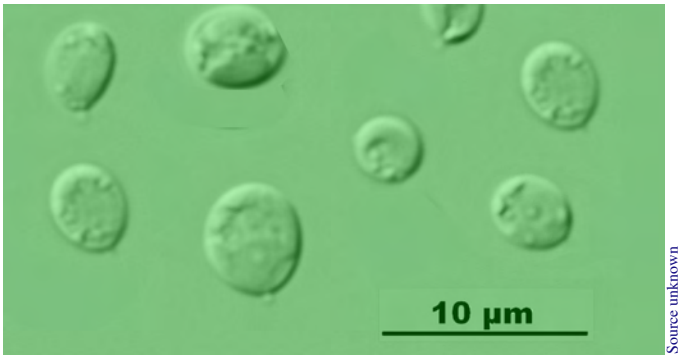

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Figure 108.2 *Nannochloropsis*

PROJECT 5.11 FEEDING AND HEALTH CHECK TABLES

Feeding schedule

Date	Amount fed (g)	Comments	Date	Amount fed (g)	Comments

Health check

Date	Comments	Date	Comments

PROJECT 5.12 WATER QUALITY MONITORING TABLES

1. Nitrate (ppm)

Date	Reading	Comments	Date	Reading	Comments

2. Nitrite (ppm)

Date	Reading	Comments	Date	Reading	Comments

3. pH

Date	Reading	Comments	Date	Reading	Comments

4. Ammonia (ppm)

Date	Reading	Comments	Date	Reading	Comments

5. Calcium hardness (ppm)

Date	Reading	Comments	Date	Reading	Comments

6. Potassium hardness (ppm)

Date	Reading	Comments	Date	Reading	Comments

7. Temperature

Date	Reading	Comments	Date	Reading	Comments

8. Other variables

Syllabus module	Mandatory & additional content	Suitable exercises
Module 22 - Aquarium Design, Construction and Activities Repair This module introduces students to the principles involved in the construction and maintenance of an aquarium. It gives students an appreciation of the different requirements of marine and terrestrial animals and some of the special needs of fish when kept as pets.	1. Construct a working aquarium involving the following principles: filtration of solids, removal of wastes, control of algae, dissolved oxygen supply, balanced community relationships 2. Recognise and rectify problems that may arise in aquarium maintenance construct a biofilter 3. Construct a biofilter Additional Content • Collect aquarium fish • Monitor water quality using remote sensors and digital recording techniques	Making a glass aquarium Making a plastic tub aquarium Making a delux plastic tub aquarium Making a crayfish tank Making a flow through crayfish tank and growth container Making an artemia hatchery Making a larger grow out tank Making a biofilter Making a sponge filter Making an undergravel filter Making an algal scrubber
Module 23 - Underwater Farming This module introduces the concepts in and basic practices involved in aquaculture. The module raises students' awareness of the finite nature of marine resources and the pressure placed on marine species used for human food. It shows aquaculture as a feasible supplementation and alternative to large-scale wild capture.	1. Maintain aquatic organisms in an aquarium 2. Raise aquatic organisms from eggs or juveniles 3. Test the effects of diet on growth rates in aquatic organisms Additional Content • Wild catch a common estuary fish and grow it out • Measure the growth rates of fish • Assess operational procedures on an aquaculture farm	Making a glass aquarium Making a plastic tub aquarium Making a delux plastic tub aquarium Making a crayfish tank Making a flow through crayfish tank and growth container Making an artemia hatchery Making a larger grow out tank Growing rotifers Growing daphnia Growing artemia Growing worms Growing fish Hatching crayfish and caring for young Feeding algae to artemia Feeding algae to oysters Feeding artemia to livestock Growing out fish from fingerlings Examination of commercial fish food Crayfish growth trials
Module 24 - Designing Systems for Aquaculture This module introduces systems used in intensive and extensive aquaculture. They will be required to analyse and evaluate the systems currently used in both systems.	1. Research the economic and environmental costs of flow-through and recirculating aquaculture systems 2. Design an intensive aquaculture system on a sloping site 3. Construct a biofilter 4. Debate the advantages and disadvantages of sea cages 5. Design a crayfish pond	Making a biofilter Making a sponge filter Making an undergravel filter Making an algal scrubber See also the Wet Paper Publications — *Marine Science and Introduction to Marine Studies* for additional information and exercises (www.wetpaper.com.au)
Module 25 - Economics of Aquaculture This module provides the opportunity for students to complete a case study of an existing or hypothetical aquaculture facility to determine its economic viability and profitability.	1. Use first or second-hand investigations to determine the total operating costs of a selected intensive or extensive aquaculture enterprise 2. Discuss the ethics of intensive and extensive aquaculture enterprises 3. Use first or second-hand investigations to determine the gross income of a selected intensive or extensive aquaculture enterprise 4 Use first or second-hand investigations to calculate the level of profit of a selected intensive or extensive aquaculture enterprise	See also the Wet Paper Publications — *Marine Science and Introduction to Marine Studies* for additional information and exercises (www.wetpaper.com.au)

Syllabus module	Mandatory & additional content	Suitable exercises
Module 26 - Growing Stockfeed for Aquaculture This module introduces methods of growing food for animals used in aquaculture concentrating on feeding the early stages of stock adjustment.	1. Grow a variety of microalgal species 2. Grow rotifers 3. Examine microalgae under the microscope 4. Examine rotifers under the microscope Additional Content • Hatch atremia from cysts • Cultivate and grow atremia • Cultivate and grow daphnia • Establish and maintain stock cultures of algae	Using Poly pipe and fittings Making an air filter Making algal growth container Making a light cupboard What's inside daphnia What's inside rotifers What's inside artemia Growing algae Growing rotifers Growing daphnia Growing artemia Decapsulating artemia Hatching artemia Mating artemia Mating crayfish Hatching crayfish and caring for young Determining the sex of crayfish Decapsulating artemia Hatching artemia Mating artemia
Module 27 - Biology of Native Crayfish This module introduces students to the basic anatomy and physiology of native crayfish and their reproduction.	1. Identify crayfish from live or preserved specimens and/or photographs 2. Determine the sex of crayfish from live or preserved specimens and or photographs 3. Research the function of specific organs in a crayfish body Additional Content • Raise crayfish in an aquarium or aquaculture facility • Mate crayfish	Examination of the external features of a crayfish Making a crayfish measurer Crayfish/prawn dissection Mating crayfish Hatching crayfish and caring for young Determining the sex of crayfish Making a crayfish tank Making a flow through crayfish tank
Module 28 - Growing Crustaceans This module introduces the relationship between basic anatomy, physiology and behaviour of crustaceans to growing these animals for human food.	1. Conduct nutritional trials on crayfish 2. Make an aquarium suitable for housing crustaceans 3. Hatch and raise brine shrimp grow one species of crustacean Additional Content • Mate crustaceans • Raise and fatten juvenile crustaceans	Making a crayfish tank Making a flow through crayfish tank Growing artemia Growing crayfish Decapsulating artemia Hatching artemia Mating artemia Mating crayfish Hatching crayfish and caring for young Feeding algae to artemia Crayfish growth trials

Syllabus module	Mandatory & additional content	Suitable exercises
Module 29 - Fish Biology This module introduces the anatomy and physiology of fish.	1. Examine and record the external features of a bony fish in a database 2. Dissect a bony fish and identify its internal organs research the following features of a selected 3. Marine fish: general description, basic anatomy and physiology, life cycle and diet, adaptation, distribution, economic importance 4. Prepare a word-processed, audio or video report of their research project	Examination of the external features of a fish Making a fish measurer Fish Dissection
Module 30 - Managing Fish Production This module introduces the general principles of animal husbandry, specifically those required by fish farmers.	1. Make simple structures to hold fish for prolonged periods 2. Identify common fish diseases 3. Select suitable species of fish to grow out 4. Select food to optimise growth grow out fish from the fingerling stage	Making a glass aquarium Making a plastic tub aquarium Making a delux plastic tub aquarium Making a larger grow out tank Examination of the external features of a fish Making a fish measurer Fish Dissection Growing fish Growing out fish from fingerlings Examination of commercial fish food
Module 31 - Managing Water Quality This module develops an awareness of the importance of water quality, the factors affecting it and the methods used to monitor water quality. Students are made aware of the effects of poor water quality on aquatic and marine plants and animals.	1. Make a Secchi Disc for turbidity study 2. Perform a turbidity test 3. Collect water samples from various sites for analysis 4. Analyse water samples for: temperature, pH, total dissolved solids, ammonia, total phosphorus, total nitrates, total nitrites 5. Identify water sample sites for water analysis Additional Content • Compile water analysis records to monitor • Changes over a period of time	Making a Secchi Disc Making a salinity hydrometer Testing water for Nitrates Testing water for Nitrites Testing water for Phosphates Testing water for Dissolved oxygen Testing water for turbidity Testing water for total dissolved solids Testing water for pH Making a biofilter Making a sponge filter Making an undergravel filter Making an algal scrubber Making a water sampler Making a syphon
Module 32 - Pests and Diseases of Aquatic Organisms This module introduces the common pests and diseases which may limit aquaculture production.	1. Identify the measures needed to protect species from disease 2. Identify from photographs shell disease and whitetail disease in crayfish 3. Identify form photographs white spot and skin flingus disease in fish Additional Content • Treat fish and/or crayfish diseases in an aquarium	

Queensland Applied senior syllabus 2024
Aquatic Practices 2024 v1.1

Aquatic Practices is a vocationally-oriented subject in Queensland that teaches students practical skills and knowledge for marine settings and workplaces:

https://www.qcaa.qld.edu.au/downloads/senior-qce/syllabuses/snr_aquatic_24_app_syll.pdf

Aquaculture

Students could:

- test and record water quality over time
- survey local retail or wholesale outlets for aquaculture animals
- raise aquatic animals and record changes in external features
- evaluate the potential of different species for aquaculture, and complete a research report on an aquaculture subject
- complete a report using video or explore a website of an underwater farm
- write a report on a visit to a local aquaculture farm or an aquaculture website
- investigate the operational procedures of an aquaculture

Aquariums

Students could:

- identify common aquariums and their use
- test aquarium water quality for toxic waste products
- build and/or set up and maintain an aquarium
- demonstrate observation equipment

SUPPY DETAILS

Project	Item	Descrption	Supplier (Lab technican to complete)
1.2	Air filter for algal culture	Airfilter: 10 ml (16 X 97mm) polypropylene flat bottom test tube with screw cap: Product code: TS 9716UU	
1.10	Growout tank	Crayfish Tank - 1000L Oval Water Tub, Specifications. Length: 2.3m Width: 1.1mm Height: 510mm Capacity: 1000L Weight: 40kg Product code (tub only): 001614	
2.5	Salinity hydrometer	Hydrometer - 15 cm Narrow stem 3.5ml draw, non graduated narrow stem polyethylene transfer pipette Product code: A1503	
	Salt water	Ocean Nature Sea Salt – available in 2kg bags Product code: ON320	
4.1	Growing algae	Guillard's F2 solution available in 1000X concentrate in 25ml syringes, 1 litre bottles or 20 L polycubes ready to be diluted with sea water Product code: none	
		Algae	
4.2	Artemia	Brine shrimp eggs with spoon 7g Product code: BSE-7	
4.3	Rotifers	Rotifers	

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