

Observing animal life



Tidal flats—looking for benthic invertebrates

Benthic macroinvertebrates are often found and sampled in intertidal soft-sediment estuarine habitats. They are regularly used to help identify ecological impacts of pollutant discharges or pick up changes in habitat due to hydrological alterations.

What are benthic macroinvertebrates? Benthic means that they live in or on the surface of the estuary floor. Macro means that they are large enough to see, and invertebrates means that they do not have an internal skeleton. Most of the small animals found in and on muddy shores fit this classification. It includes things that live in or on the surface of the mud, from bloodworms to pipis. It does not include small fish, mudskippers or anything else with bones.

The equipment needed to sample benthic macroinvertebrates is relatively simple—gumboots, some small buckets and a sieve—but identification can be difficult and time consuming.

Sieving the samples kills most of the creatures in it, particularly the soft-bodied ones. Do not attempt this activity if you do not have some form of identification material and the time to sit down and identify all the creatures collected. Samples may be preserved if you can't identify them immediately. A permit from PIRSA may be required to complete this activity, depending on the area you wish to study.

What you may need:

- gumboots, wetsuit boots or tight-fitting old shoes
- 500-mL plastic containers and pop on watertight lids, one per sample site
- large plastic bucket to hold tools and samples
- small hand spade
- 25-m measuring tape
- GPS or detailed topographic map
- permanent markers
- assorted sieves with different mesh sizes (1–10 mm)
- access to an outside garden tap and hose
- disposable gloves
- a pair of tweezers or forceps
- methylated spirits or alcohol (optional)
- small plastic or glass jars with lid
- identification materials
- large magnifying glass or dissecting scope.

To ensure good results:

Estuaries may be very variable. Replication assists in obtaining representative data.

Activity:  Extension

Age: High school

Time: 3

Why? Estuarine invertebrates are likely to reflect the overall health of the habitat.

Safety: Avoid areas that contain large amounts of black smelly mud. These contain hydrogen sulphide, which can oxidize into sulphuric acid in contact with air. Wash your hands well in seawater and do not touch your eyes if you have to work in these areas.

Shoes or wetsuit boots are an absolute must in these areas, as razor fish shells are very sharp. They should be tight fitting so they do not get left behind in the mud. Hats are also a good idea, as you should expect to spend at least a couple of hours out in the sun each monitoring event.

When sieving the samples you should wear disposable gloves, as many of the animals can bite or put out irritating substances. South Australia does not have any benthic invertebrates that are highly toxic to touch. Nevertheless, some can be very irritating, particularly to people with sensitive skin.

What to do:

Examine your soft-sediment shoreline carefully. You need to find a location at least 50 m wide that is representative of your estuary, has easy access and can be worked safely by all members of your group. If the area is near a port, on private property or in an aquatic reserve, permission will be needed first.

Gather all your equipment before going out in the field. Everything on the list from the sieves downward can be left back at base. Items above sieves on the list will be needed in the field. Ensure that it will be low tide when the fieldwork is undertaken.

Once in the field identify a suitable location for your first transect. The second and third transects will be 25 metres on either side of the first one. The transects will need to run from the average high-tide mark, down towards the low-water mark.

Starting at the low-water mark on your first transect, take a GPS reading and record it on your benthic invertebrates datasheet. Take a sediment core by tipping one of the plastic containers upside down and pushing it into the sediment. If the sediment is too firm to do this, use the hand shovel to dig the sediment out and fill the plastic container. Put the lid on firmly and mark the lid with transect name and sample number. Take another sediment core half way along the transect and record a GPS reading.

Collect another sediment core and GPS reading at the end of the transect.

Measure 25 m to the left from the original transect and repeat the procedure. Do this on the other side for the third transect. You now have three samples per transect giving you nine samples in total. If your shore is particularly broad, more samples may be taken if you like. Bear in mind that the processing time is considerable, so nine is usually sufficient.

Once you return to your base, take all the sample containers to the garden tap. Stack the sieves, with the coarsest on top and the finest on the bottom. Place a part of the first sample into the top sieve and slowly wash the sediment through the stack. Once most the sediment has passed through, pick out the animals with your forceps or a gloved hand. Put them in a small, labelled jar with a little water. Empty the sediment from the sieves.

Repeat the above procedure until the first sample has been entirely sieved. Screw the lid onto the jar of animals, label it and put it aside for later.

Repeat this procedure until all the samples have been sieved and the resulting animals are contained in nine little jars. These can be identified immediately or preserved, to identify when you have more time.

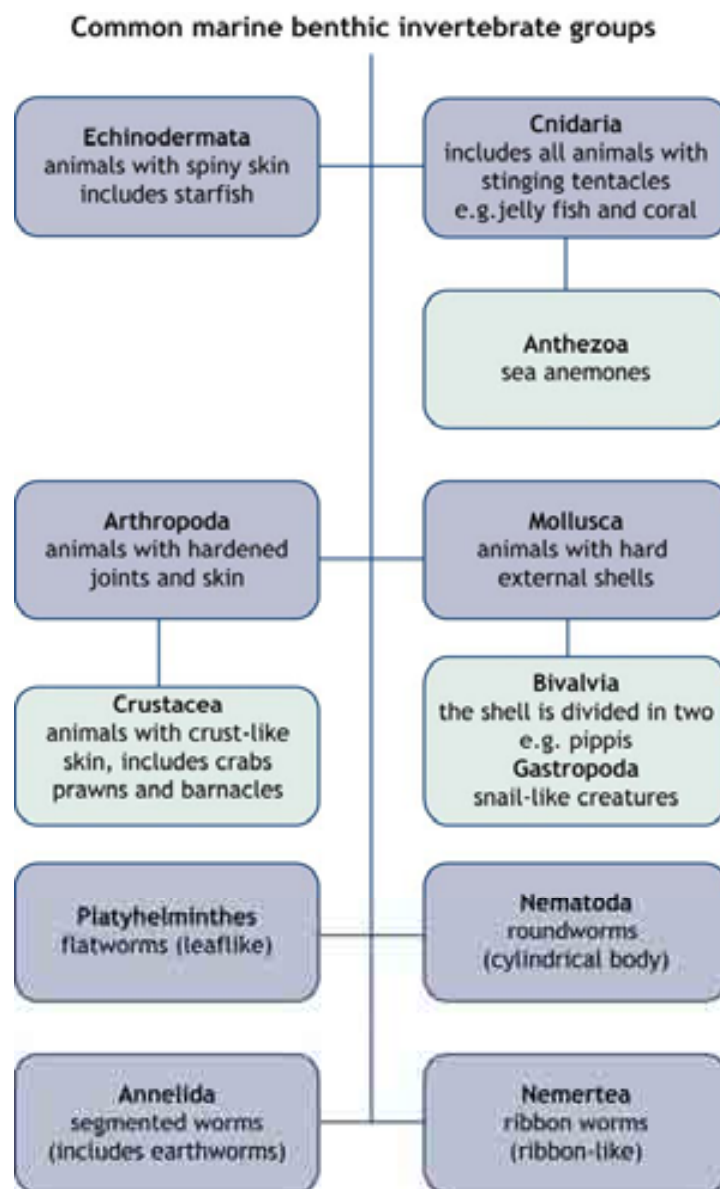


Figure 15—Common invertebrate phyla

Some identifying characteristics are only present when organisms are alive, but remember, many invertebrates will happily eat each other if they are not preserved quickly (eg within a couple of hours). Add some methylated spirits or alcohol to the jars if you wish to preserve the animals.

Identification

Identifying benthic marine macroinvertebrates can be fairly difficult. The taxonomic resolution that is chosen (how detailed an identification you require), depends on the resources and experience of the group. If the group are all beginners, or intermediately skilled, then identifying the macroinvertebrates to phylum or class is probably sufficient.

The common phyla and classes found in soft-sediment shorelines are shown in Figure 15. This chart could be used to find the appropriate section of your other identification resources if you intend to take the identification to a finer level.

Identification that progresses beyond this resolution is probably best undertaken in conjunction with a marine biology or zoology textbook that describes each of the phyla. A few good books are listed at the end of this guideline. Site-specific identification aids may be available for your district. Examples of local identification tools are the resources from Reef Watch SA for the Onkaparinga estuary. Where these are available they are the best resources for this activity.

Data from this type of activity is commonly presented as a species or classification list, possibly with rough counts of the number of each type written next to it. This list should be sent to your Waterwatch coordinator. The list can then be compared with those compiled at other sites, or over time.

It is probably best to revisit the site and do some benthic sampling once a year, although a once-off sampling event can yield valuable data.

Useful references

Dakin, WJ & Bennett, I (1987), *Australian Seashores: A guide to the temperate shores for the beach-lover, the naturalist, the shore-fisherman and the student*, Angus and Robertson Publishers, Australia.

Davey, K (1998), *A Photographic Guide to Seashore Life of Australia*, New Holland Publishers, Sydney.

Haddon, F (1992), *Australia's Seashores: Environmental Field Guide to Flora and Fauna*, Simon and Schuster, East Roseville, New South Wales.

Jones, D & Morgan, G (2002), *A Field Guide to Crustaceans of Australian Waters*, 2nd edition, Reed New Holland, Sydney.

Morgan, D, Best, E, Betjeman, K, Larkum, A, Pitman, M, Thomson, J, Lee, A & Nicholas, J (eds)(1973) *Biological Science: The Web of Life*, Australian Academy of Science, Australia.

Ponder, W, Hutchings, P, Chapman, R (2002), *Overview of the Conservation of Australian Marine Invertebrates: Chapter 2 – The Australian Marine Invertebrate Fauna*, Environment Australia, accessed online 28 May 2003, <www.amonline.net.au/invertebrates/marine_overview/chapt2.html>.

Shepherd, SA & Thomas, IM (1982–1997), *Marine Invertebrates of Southern Australia: Parts I–III*, Government Printer, South Australia.



Tidal flats—counting crab burrows

Burrowing crabs are an important part of estuarine ecosystems. The most common crabs found in South Australian estuaries are shore crabs from the Grapsidae family. Ghost, sentinel, fiddler and semaphore crabs (all from the family Ocypodidae) are also found in muddy estuaries of Southern Australia.

Burrowing crabs are mainly **detritivores**; they will also eat live animals and plants if the opportunity arises. Crabs often leave leftover food and faecal matter in their holes, which quickly breaks down into nutrients for vegetation growing in the surrounding area. They also aerate the soil and provide drainage, which keeps the sediment healthy and productive.

Changes in environmental conditions may result in a decrease in crab populations and therefore a drop in burrow counts. Changes that can cause this are many, but include changes in sediment particle size and hydrology. Decreased crab populations slow nutrient cycling and decrease soil aeration, which may reduce productivity.

In this activity, the number of crab holes present is estimated by counting holes within several 500 mm x 500 mm quadrats. The method does not provide a true estimate of crab populations as some burrows may be home to many crabs or one crab may have many entries into its burrow.

This activity is best repeated every three months, as the number of crab holes present at a site can change very rapidly.

What you may need:

- quadrat count datasheet
- 500 mm x 500 mm quadrat (directions for making a quadrat are included in Appendix 6)
- 10-m (or longer) tape measure
- GPS or topographic map.

To ensure good results:

If crab holes are very numerous you may need to subdivide your quadrat to avoid counting burrows twice.

What to do:

It is recommended that your crab-hole counts be established in an easy-to-access section of homogenous mangal (mangrove forest), a small clearing, the edge of a tidal creek, or in a samphire saltmarsh area.

Establish two parallel eight-metre transects through the site. Transects should be located within 10 metres of each other. Mark the beginning and end of each transect with a visible peg to allow for site relocation.

At the beginning of each transect, place a quadrat to the left of the transect line, and count the number of crab holes in that area. If crab holes are very numerous you may need to subdivide your quadrat to avoid counting burrows twice. In extremely dense concentrations, you may not need to count all the subdivisions of your quadrat. Multiply the number in the portion you have counted by the appropriate factor.

Repeat the quadrat counts every two metres along both transect lines until 10 quadrats have been counted.

Activity:  Core

Age: All

Time: 2

Why? Estuarine macroinvertebrates are likely to reflect the overall health of the habitat. In mudflat and sandflat environments the quantity of crab burrows provides information on the firmness and aeration of the substrate.

Safety: Conduct this activity at low tide.

Data are reported as crab holes per square meter. Results may be variable between sites, so your first monitoring event should be used as a baseline crab hole density for your site.

Crab hole abundance does not equate to absolute crab populations. Large changes in crab hole numbers are needed to indicate changes in crab population.

Identifying the species of crabs present at your site may also be useful. Books covering Crustacea, particularly the Grapsidae and Ocypodidae families are fairly common in technical bookstores. Take photographs of any crabs you find (top and bottom sides) so that you may identify them later.

Useful reference

Jones, D & Morgan, G (2002), *A Field Guide to Crustaceans of Australian Waters*, second edition, Reed New Holland, Sydney, Australia.



Tidal flats—counting surface snails

Tidal mud or sand flats are two of the most common shore types in South Australian estuaries. Although these habitats have a diverse range of fauna, much of the animal life on these flats is hidden during low tides. Only the snails are obvious on the surface, particularly when the tide is just covering the sediment. Surface snails were selected to aid in assessing these habitats due to their ease of detection.

Surface snails usually graze on the thin film of algae and debris that grows or settles on the intertidal sediments. They control the growth of algae and get eaten by a wide range of larger lifeforms, including crabs, birds and fish, particularly the wide variety of wading birds that inhabit tidal flats.

Snails that occur on tidal flats suffer less from humans than snails that live on rocky shores, even though they are still collected for bait, and off-road vehicles create havoc in their fragile habitat. Less direct effects on surface snail populations are many, and include:

- changes in hydrological flows
- increased nutrients
- increased sedimentation
- sulphide-rich mud build-up
- changes to water temperature or salinity
- storm water discharges
- introduction, or changes in numbers, of predatory animals or introduced marine pests.

This method is very similar to the method outlined in the *Rocky shore—counting snails* guideline. This method has been adapted from that activity to make it suitable for use on a soft, flat substrate.

If you wish to record seasonal fluctuations, monthly or quarterly surveys are most useful. Six-monthly or yearly surveys are adequate to detect long-term changes but, as the soft substrate changes quickly, more regular monitoring is recommended.

What you may need:

- quadrat count datasheet
- 50-m tape measure (or greater)
- 500 mm × 500 mm quadrat that may be subdivided with string if needed (instructions for making a quadrat are included in Appendix 6)
- GPS or detailed orthophotographic map
- ruler for measuring snail size
- gastropod identification sheet or an appropriate guide
- camera or drawing pad.

To ensure good results:

If there are many snails, you may need to subdivide your quadrat to avoid counting snails twice.

Activity:  Core

Age: All

Time: 2

Why? Estuarine macroinvertebrates are likely to reflect the overall health of the habitat. Snails may also be affected by overharvesting (bait, souvenirs).

Safety: Always complete this activity at low to mid tide, and start work on your transect from the lowest quadrat, working toward the high-tide mark.

Always wear suitable footwear that will not come off if stuck in soft sediment. An old pair of runners or a pair of wetsuit booties are ideal.

Black mud should be left alone, as it can be very soft and may contain large amounts of sulphide that could oxidize into sulphuric acid if exposed to air. Wash your hands well before eating or touching your eyes if this type of mud is present.

What to do:

Select a site within your estuary that you believe is representative. This is best done at low tide. If this area is in a marine protected area a permit may be required to complete this exercise. You may select an area that is disturbed or one that is pristine.

Establish three transects running between the high- and low-water lines. Select the location of each transect so that the widest range of habitats are sampled. Space them approximately the same distance from each other. Take GPS readings and identify reference points for the start and end of each transect so that they may be located again.

Place quadrat locations evenly along each transect. Three to 10 quadrats are recommended for each transect. Give each quadrat a name depending on which transect it is located on and its position between the high and low water marks. An illustration of this is included in the *Rocky shore – Counting snails* guideline.

If you have enough people, divide your group into one or two persons per transect or quadrat. It is often best to work in pairs with one person counting and one recording.

Starting at the low-water mark, place the quadrats along the left side of the transect. Be careful not to walk on this side at any time, particularly where the quadrats will be placed, as compression damages the sediment structure, particularly when the area is frequently monitored. Always walk, and work from, the right-hand side.

Using a resource such as the SA Museum's *Molluscs of South Australia – Gastropoda* poster, identify the species of snails in your quadrat. A couple of snails from each species may need picking up and sediment rinsing from their shell to identify them. Place them immediately back in the quadrat they were found in. Record the name of each species down the side of your tally sheet for the quadrat. If there are any unknown species measure their width and height, take a photograph or sketch of them, then give them a name, for example 'Snail 1'. These may be identified later from a more detailed guide or by a specialist, or may remain with their temporary name attached to their photograph.

Tally the number of each species occurring in your quadrat. If there are many snails, you may need to subdivide your quadrat to avoid counting errors. Add the numbers from all the subsections together to give a total number of each species for the quadrat.

Once all quadrats have been counted, fasten all the quadrat tally sheets together, ensuring they are each labelled with the date, time, quadrat number and who did the count.

A range of comparisons may be made between each monitoring event, site, transect, quadrat and species. These comparisons include:

- total number of snails per site
- total number of species per site
- total number of snails per species at each site
- changes over time
- detection of harvesting stress or other impacts
- measurement of differences between degraded and pristine sites.



Tidal flats—recording wading birds

Birds are the most conspicuous animals inhabiting saltmarshes and estuaries. Wading birds are also an important part of an estuarine ecosystem. They eat small fish, crustaceans, worms, molluscs and return large amounts of easily accessible nutrients to the ecosystem in the form of faecal matter.

The saltmarshes surrounding an estuary, and the mudflats of the estuary itself, offer a variety of habitats such as mudflats, pools, various types of vegetation and open water. Wading birds have evolved to exploit the resources of the shallow water habitats in estuarine systems.

Human activity affect birds by altering prey abundance and the availability of nesting or hiding sites. For example, herons and egrets need high densities of fish, and excessive pollution might reduce fish populations to the point where herons and egrets cannot get adequate nutrition. Where humans have adversely affected the bird's ecosystem, the population of each bird species will decrease or the affected species will disappear completely.

Birds that need specific habitats or conditions (specialists) may avoid saltmarshes that have been disturbed, while birds that can tolerate a wide range of conditions (generalists) may thrive in these areas.

Birds are amongst the highest predators in the estuarine food web. Other predators include snakes and sharks. Although there are shortcomings to using birds to assess the condition of saltmarshes, birds can yield insight that may be overlooked by studying only plants, invertebrates or fish.

For this activity, monitor only those birds seen on the mud or sand flat from the high-tide mark seaward. This includes any birds flying across the area, if they are close enough to identify. Do not monitor birds in the mangroves or dunes.

What you may need:

- binoculars
- field guides or charts
- pencil
- clipboard and bird observation datasheet.

To ensure good results:

Familiarise yourself with all field guides and other aids at home, rather than in the field. For best results, gather the bird identification materials you are most comfortable with and use a variety of clues (shape, posture, size, coloration, behaviour, habitat and birdcalls) to identify species or groups of closely related species.

What to do:

Point counts are conducted from a single vantage point overlooking the estuary. Observers record all species and individuals seen or heard over a fixed time, so identification needs to be rapid. Arrive between sunrise and 8 am when birds are most active. Bird calling and activity decreases during the day, and you can get a better idea of bird communities by surveying during peak activity.

Locate a vantage point from which you can see a representative portion of the mud or sand flat, including the marsh border. You will want to use the same vantage point for all subsequent visits to the site so make careful note of your locations.

Activity:  Core

Age: All

Time: 2

Why? Waders and migratory birds frequently depend on estuarine habitats. They are sensitive to habitat change, so changes are easily identified.

Safety: Wear solid boots, long pants and a hat. Snakes, prickles, sharp objects and the sun are the main safety concerns.

Look for any birds seaward of the highest tide line and attempt to identify them. If the bird is a wading bird it will have long legs. These include herons and egrets. Duck-like birds and swans, and small birds that run along the mud, such as sandpipers or shorebirds are also to be recorded. Once you identify the general group a bird belongs to, e.g. herons, egrets, swans, sandpipers or a general shorebird, consult a bird book to find the species that the bird most resembles. Sticky notes marking each general section often helps identification. It often helps to obtain a bird list of your area. If you are not sure that you have identified a bird properly and it is not on the list, then it is likely that you have misidentified the bird. See if you can get another look at it. Lists are obtainable for most ornithological groups and from the Birds Australia website at <www.birdsaustralia.com.au>.

It is sometimes helpful to take notes or photographs so you can take the time to identify the bird properly after the survey is finished. If taking notes, ensure that you record details such as size (bigger or smaller than a white-faced heron?), shape (long and thin or plump and round?), coloration (brown? what shade?), and any distinguishing marks or features (any streaking or other noticeable marks?).

This recording is usually done for a fixed amount of time (ideally 20 minutes). If possible record an approximate count of the birds seen, as well as the species. A separate field datasheet should be used for each site and survey date. All datasheets should have the following types of information: observer(s), site location, a rough sketch of the study area with the vantage point location, survey date, time, time of sunrise, weather, and tidal conditions. A good field datasheet will also include a sketch or photographs of the study area. If working in a group, one person should perform all of the data entry so that entries are consistent. If two people are working together, one can observe while the other person records information.

Try to record the activity or habitat of each bird that you observe and check to ensure that you have completely filled out field dataforms.

Surveys should be conducted during different tidal conditions and different times of year. Exposed mudflats at low tides provide important resources for some species, and high tides may force secretive species out of the vegetation where you can see them. Four surveys per year are recommended, although this will depend on the resources and time limitations of the group. Data analysis of bird surveys is possible. However, all that is required for this activity is construction of a species list including rough numbers of each species seen.

From the species list, particularly if counts are included, a range of **metrics** or values may be produced that take into account species tolerances and preferences. The most simple of these are:

- number of species recorded
- number of regionally significant species
- percent abundance of three most common species
- number of species in common between different sites.

Each of these metrics shows different things about the site. Simplified interpretation of these attributes is basically along the lines that the more species found on a site, particularly rare ones, the healthier the site is.

The metric that looks at the percentage abundance of the three dominant species shows whether the site is suitable for a wide range of birds or is a specialist or niche habitat. For example, there may be thousands of birds at a rubbish tip, but they may all be one species—Silver Gulls for example, that are specialised scavengers.

The metric that looks at the number of species in common between sites tells you how much the two sites have in common, habitat-wise. It may also tell you what are the important factors about each site. Two sites that look similar to the human eye frequently attract different birds due to environmental factors not immediately obvious to an observer.

Rocky shore—counting snails

Rocky shores in estuaries are home to a broad range of flora and fauna, depending on the exposure of the site, type of sediment (rocky, sandy or muddy), water flow patterns and water salinity. The life found on these shores varies considerably from site to site. Snails were selected due to their importance in a rocky shore ecosystem and their apparent abundance at most sites worldwide.

Grazing snails feed on the thin film of algae that grows on rocks that remain constantly damp or wet. They control the growth of algae and get eaten by larger lifeforms, including other snails, crabs, birds, fish and reptiles. Without grazing snails a wide range of shore life would need to find alternative food sources, or go elsewhere.

Some snails are carnivorous, such as the little black and white oyster borers.

Common direct human influences on snails include harvesting for bait, food or shells. Less direct things that affect grazing snail populations include changes in hydrological flows, increased nutrients, changes to water temperature or salinity, stormwater discharges, and the introduction of predatory or competitive marine pests.

This activity uses a method that has been simplified and adapted for community use in South Australian estuarine areas. The more detailed Queensland Rocky Reef Watch program is available on the internet for those who are interested:

<www.widebay.net/wmpme/rrw/snails.htm>

The activity records the number and type of grazing snails present in 500 mm × 500 mm quadrats on a regular basis. Quadrats are placed at intervals along transects running between the high- and low-tide marks on rocky embankments or natural rock outcrops along the shores of an estuary. If you wish to record seasonal fluctuations, quarterly surveys are most useful. Six-monthly or yearly surveys are adequate for long-term change detection.

What you may need:

- quadrat count record sheets
- tape measure (30–50 m long)
- 500 mm × 500 mm quadrat, preferably subdivided with string into 100 mm squares (instructions for making a quadrat are included in Appendix 6)
- GPS or detailed orthophotographic map
- ruler for measuring snail size
- gastropod identification sheet or an appropriate guide
- camera or drawing pad
- a tally sheet for each quadrat.

Activity:  Extension

Age: High school

Time: 3

Why? Estuarine macroinvertebrates are likely to reflect the overall health of the habitat. Snail species may also be impacted by overharvesting (food, bait and souvenirs)

Safety: Care is needed when walking in intertidal rocky shore areas. Rocks may be slippery due to algae growth and undercutting can cause them to be unstable. Complete this activity at low tide, and start work on your transect from the lowest quadrat, working toward the high tide mark. If you are in an area where freak waves or strong currents are possible, ensure that a member of the group is selected to keep an eye out for unusually large waves or members of the group that may need help.

Always wear suitable footwear with good grip.

Never reach into places that you cannot see as many animals can cause injury. Gloves may be a good idea in areas where the rocks are rough or where rocks need turning over.

To ensure good results:

In areas with many snails, you may need to subdivide your quadrat to avoid counting errors. As rocky shores can be rather uneven, the surface area covered by a 500-mm quadrat is very variable. Therefore averages of several quadrats may provide more reliable data than information from a single quadrat.

What to do:

Select a site within your estuary that you believe is representative of the estuary. This is best done at low tide. If this area is in a marine protected area a permit may be required. You may select an area that is known to be disturbed or one known to be pristine.

Establish three transects running between the high- and low-water lines. Select the location of each transect so that the widest range of habitats is sampled. Space them approximately the same distance from each other. Take GPS readings and identify reference points for the start and end of each transect so that they may be located again.

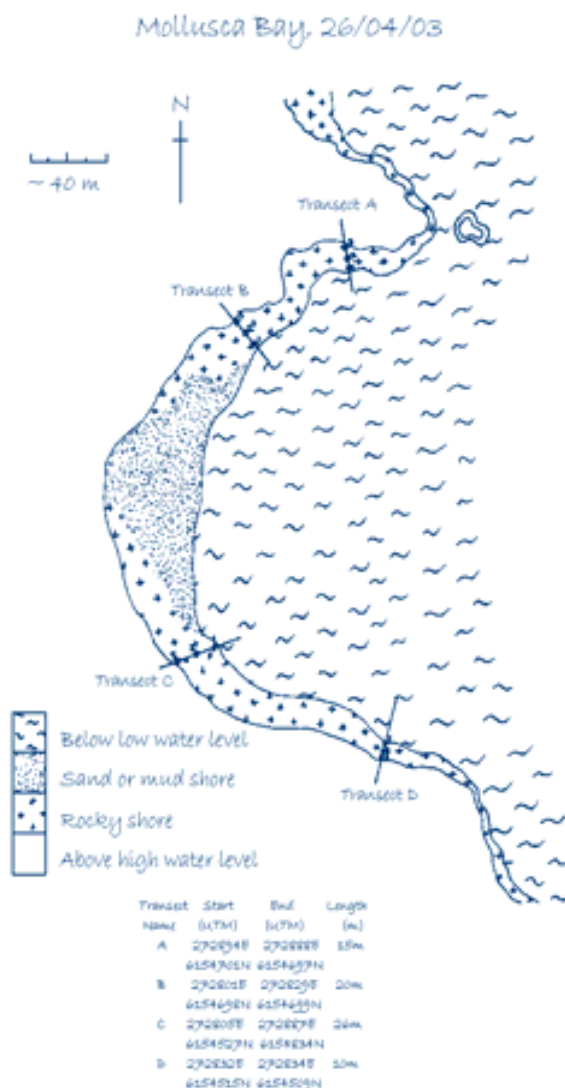


Figure 16—Transect mapping on a site map

Place quadrats evenly along each of the transects. Three to 10 quadrats are recommended for each transect. Give each quadrat a name depending on the transect it is on, and its position between the high and low water marks. Write these on to a site map, as shown in Figure 16. A hand-drawn map is sufficient.

If you have enough people, divide your group into a watcher and one or two persons per transect or quadrat. Small groups may need someone watching for rough water, supervising, and collecting the sheets while the rest of the group works together. It is often best to work in pairs with one person counting and one recording data.

Starting at the low-water mark, flip a coin to decide if the transects are to be placed on the left or the right of the transect line. Using a guide such as the SA Museum’s *Molluscs of South Australia—Gastropods* poster, identify the species of snails in your quadrat. Record these down the side of your tally sheet. If there are any unknown species, measure their width and height, take a photograph or sketch of them, and give them a temporary name, for example ‘Snail 1’. These snails may be identified later from a more detailed guide or by a specialist, or simply remain with a temporary name attached to their photograph.

On rough surfaces, the quadrat area is the area directly below the quadrat when it is held out horizontally and viewed from above.

Tally the number of snails for each species in the quadrat. This includes snails you can find under or around the rocks. Remember to turn your rocks back the way you found them, if you move any or turn them over. If there are many snails, you may need to subdivide your quadrat to avoid counting snails twice. Add the numbers from all the subsections together to give a total number of each species for the quadrat.

Once all quadrats have been counted, fasten all the quadrat tally sheets together, ensuring they are each labelled with the date, time, quadrat number and name of the volunteer who did the count.

A range of comparisons may be made between each monitoring event, site, transect, quadrat and species. These comparisons include:

- total number of snails per site
- total number of species per site
- total number of snails per species at each site
- changes over time
- detection of harvesting stress or other impacts
- measurement of differences between degraded and pristine sites.

A common comparison is to plot counts for each species against each quadrat number (an indication of location relative to the daily tidal cycle) to produce a graph that illustrates how different species are distributed across the shore, as snails often show zonation according to their depth and wetting cycles.

As rocky shores can be rather uneven, the surface area covered by a 500-mm quadrat is very variable. If there are a large number of small rocks in the quadrat, the surface area may be more than double the nominal 0.25 m². Therefore averages of several quadrats may provide more reliable data than information from a single quadrat.



Saltmarsh—looking for invertebrates

Saltmarsh habitats contain many **ecological niches**. To obtain a picture of the biodiversity of your saltmarsh, and to monitor change over a period of several years, you can audit the macroinvertebrates in your marsh.

One type of invertebrate (crabs that occur in the tidal creeks) will be more fully sampled using the techniques outlined in the following guideline, *Saltmarsh—Counting crabs and fish*, whereas this activity will focus on sampling those organisms that occur in the **intertidal** and **supratidal** areas of the marsh.

As some of the activities may be destructive, or you may wish to remove sample organisms for identification, you may need a permit in some areas. If you do not have the time to undertake identification, please do not remove organisms from their habitat.

What you may need:

- site macroinvertebrate record
- equipment listed in the following guidelines:
 - *Tidal flats—Looking for benthic invertebrates*
 - *Tidal flats—Counting crab burrows*
 - *Tidal flats—Counting surface snails*
- D-net for macroinvertebrate sampling in creeklines
- a butterfly net and insect capture box (see Appendix 6)
- muslin bags with tie tops
- camera.

To ensure good results:

Saltmarshes contain areas that are vegetated and areas that are not vegetated (**pannes**). Ensure that you sample your site in a representative manner — place transects and quadrats into areas of high marsh (grasses and saltbush), low marsh (samphires) and pannes.

What to do:

Examine your saltmarsh carefully. Is the vegetation the same right across the site? It probably isn't. Down where the tide comes in every day there may be low growing samphires that look like a lawn. As you move away from the tidal influence there may be bushier samphires, then bluebush and saltbush and maybe even grasses in the driest areas.

Place several 50-m transect lines into areas that represent each of the main **vegetation associations or habitat types**. Record the beginning and ending locations of your transects onto your datasheet. Place your quadrats at the beginning, middle and end of your transects. Try to include some submerged areas in your transect, although not in the quadrats.

In each quadrat, count crab burrows and count the surface snails, following the instructions in the guidelines *Tidal flats—Counting crab burrows* and *Tidal flats—Counting surface snails*. Once you have recorded this data, collect a coring of the surface mud, as detailed in the guideline *Tidal flats—Looking for benthic invertebrates*. This sample will need to be bagged, labelled, and returned to base for processing. Please do not undertake this activity if you will not have the time to identify the organisms.

Activity:  Extension

Age: Adult groups

Time: 4

Why? Macroinvertebrates are likely to reflect the overall health of this mixture of habitats.

Safety: Saltmarshes are home to snakes and other small creatures. They also have large numbers of hidden creeks or overhangs, which are easy to fall down. Look where you are walking and ensure that all members of the group are wearing long pants and covered shoes.

Saltmarsh environments have no shade and can become extremely hot. Wear a hat, take a bottle of water and try to work in the cooler parts of the day.



Figure 17—D-net for sampling macroinvertebrates in aquatic conditions



Figure 18—Butterfly net

Other macroinvertebrates live amongst the vegetation in the saltmarsh. In each of your quadrats, carefully examine any plants to see if there are insects, spiders, millipedes, centipedes, slaters, snails or crabs present. Record the numbers and types of these that you find. If you wish to remove any specimens for later, more detailed identification, place them in small bottles labelled with the quadrat details. Alternatively, take photographs of any animals whose identity you are unsure of.

Record the location of any submerged areas on your transect. Are they pannes or creeklines? Record what type of habitat they are. Use your D-net (see Figure 17) to sweep for macroinvertebrates that may be dwelling amongst the submerged stems of plants edging the water body, and across the submerged sediment surface. Examine your net carefully. Identify any organisms you have caught, or place them into small labelled bottles filled with alcohol for later identification.

Finally, use your butterfly net (see Figure 18) to brush the vegetation along the length of your transect. Capture any insects that fly up and either record them immediately or place them into your insect capture box. At the end of each transect, empty your insect capture box into a labelled muslin bag and tie the top. The bags can be returned to base for identification.

The data you have collected provides useful

baseline information about the site. Because of variables such as the weather, it is difficult to compare the results that you obtain on any two years, but collecting this type of data over five or more years will allow you to detect long-term changes. Even when this data is collected only once for a site, it may be used by later workers to detect species losses from a site many years after the original data was collected.

From the species list, a range of **metrics** or values may be produced that take into account species tolerances and preferences. The most simple of these are:

- a species list
- number of species recorded
- community composition (each species abundance as percentage of total count)
- percent abundance of three most common species.

Useful references

Identifying macroinvertebrates can be a time-consuming task. The following references vary from simple seashore guides through to detailed technical references. Should you not be able to identify an organism, you could take it to an expert (SA Museum or PIRSA) for identification, or you could take a

photograph of the animal, and give it a temporary name or number, which you could use in future studies. If you do this, you need to provide a copy of the photograph to your Waterwatch coordinator or any other person who wishes to use your data.

Dakin, WJ & Bennett, I (1987), *Australian Seashores: A Guide to the Temperate shores for the beach-lover, the naturalist, the shore-fisherman and the student*, Angus and Robertson Publishers, Australia.

Davey, K (1998), *A Photographic Guide to Seashore Life of Australia*, New Holland Publishers, Sydney.

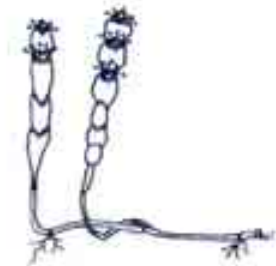
Haddon, F (1992), *Australia's Seashores. Environmental Field Guide to Flora and Fauna*, Simon and Schuster, East Roseville, New South Wales.

Jones, D & Morgan, G (2002), *A Field Guide to Crustaceans of Australian Waters*, second edition, Reed New Holland, Sydney.

Morgan, D, Best, E, Betjeman, K, Larkum, A, Pitman, M, Thomson, J, Lee, A & Nicholas, J (eds)(1973), *Biological Science: The Web of Life*, Australian Academy of Science, Australia.

Ponder, W, Hutchings, P, Chapman, R(2002), *Overview of the Conservation of Australian Marine Invertebrates: Chapter 2 – The Australian Marine Invertebrate Fauna*, Environment Australia, accessed online 28 May 2003, <www.amonline.net.au/invertebrates/marine_overview/chapt2.html>.

Shepherd, SA & Thomas, IM (1982–1997), *Marine Invertebrates of Southern Australia: Parts I–III*, Government Printer, South Australia.



Saltmarsh—counting crabs and fish

Saltmarshes support large populations of creatures that swim—fishes and many types of invertebrates including crabs. Some fish and crabs use saltmarshes at all stages of their lifecycles, while others only visit for spawning or as juveniles.

Sampling swimming animals is not simple, as their presence varies from tide to tide and between different marshes. They are highly mobile and catching them requires patience. However, they are an important indicator of degrading factors such as tidal restrictions and acid sulphate soils.

Submerged saltmarsh habitats that you may sample fish and crabs in include tidal creeks, channels, and inundated **pannes**. As this is a fishing activity you will need a permit from PIRSA.

Sampling fish and crabs is difficult in saltmarshes as the irregular terrain affects netting efficiency. The use of minnow traps is recommended for this activity, as they are easy to deploy, cheap to make, and require few people to operate. Unlike seine netting however, minnow traps do not provide quantitative data, as you cannot tell from what area the fish and crabs were captured.

What you may need:

- saltmarsh fish and crabs datasheet
- several minnow traps.

To ensure good results:

If you are interested in comparing sites, such as a tidally restricted saltmarsh against an unrestricted saltmarsh, you will need at least three traps for each marsh. The traps should be placed in the creek being sampled at each site, spaced equally along the salinity gradient.

What to do:

Place the traps at the edge of the tidal creek, but where they will stay completely underwater at low tide. To do this, it is often easiest to place the traps at low tide. Ensure the trap stays in position by placing a rock or brick on top of it.

Leave the traps in the water from one low tide to the next. Retrieve the traps and empty the contents into individual buckets.

Identify and count the catch from each trap. Measure the size of the organisms (nose to start of tail fin for fish, width of carapace from the base of the largest spines for crabs). Examine any fish for signs of disease such as red spot and fin rot, or parasites. Record the information and return the animals to the water.

From the species list, a range of **metrics** or values may be produced that take into account species tolerances and preferences. The most simple of these are:

- number of species recorded
- community composition (each species abundance as percentage of total count)
- fish condition
- percent abundance of three most common species
- number of species in common between different sites.

Activity:  Extension

Age: Adult groups

Time: 4

Why? Fish and crabs respond rapidly to changes in tidal regime that occur when restrictions are placed in saltmarshes.

Safety: Saltmarshes contain rough terrain and ill-defined waterways. Always work in pairs, wear long sleeves and trousers and enclosed footwear. Hats are advisable as saltmarshes offer little shelter.

Useful references

Jones, D & Morgan, G (2002), *A Field Guide to Crustaceans of Australian Waters*, second edition, Reed New Holland, Sydney, Australia.

Hutchins, B and Swainston, R (1986), *Sea Fishes of Southern Australia*, Swainston Publishing, Perth, Australia.



Across the estuary—recording raptors

Hawks, eagles and kites are raptors. They are an important part of an estuarine ecosystem. They eat fish and smaller birds, removing the weak and infirm from the genetic pool of their prey.

The larger raptors, such as the Osprey, are the top predators in their food chains, so their populations quickly reflect changes to prey distribution, pesticide or heavy metal contamination in their food.

For centuries, scientists and naturalists have studied avian life history, behaviour, environmental requirements, and responses to environmental disturbance and pollution. The concept of using birds as sensitive environmental indicators has long been established. Scientists have studied how nesting, hatching, and fledging success can reflect environmental conditions. The effect of Dichloro-Diphenyl-Trichloroethane (DDT) on raptor shells is a well-known example, discussed by Rachel Carson in her seminal work *Silent Spring* (1962).

Raptors are long-lived and highly mobile, and over the course of their lives can learn to recognise favourable locations for breeding, nesting, feeding and migration. They choose to live in wetlands that have the best conditions to meet their needs. Raptors can usually recognise whether the vegetation is suitable for nesting, or if prey abundance is sufficient for themselves and their young, and if predation risk is low.

This activity monitors the complete visible horizon when the observer is sitting above the high-tide mark, including dunes, spits and mangroves. It only involves identifying raptors (hawks, kites, eagles and ospreys), although notes of other birds can be made.

What you may need:

- binoculars
- field guides or charts
- pencil
- clipboard and bird observation datasheet.

To ensure good results:

Familiarise yourselves with all field guides and other aids at home, not in the field. Point counts are conducted from a single vantage point overlooking the marsh, and require observers to record all species and individuals seen or heard within a fixed amount of time, so identification needs to be rapid. For best results, gather the bird identification materials you are most comfortable with and use a variety of clues (shape, posture, size, coloration, behaviour, habitat and birdcalls) to identify species or groups of closely related species.

What to do:

Arrive between sunrise and 8 am when birds are most active. Bird calling and activity decreases during the day, and you can get a better idea of bird communities by surveying during peak activity.

Locate a vantage point from which you can see a representative portion of the estuary. You will want to use the same vantage point for all subsequent visits to the site so make careful note of your location.

Identify if the bird is a raptor by looking at its body shape and beak. All raptors are shaped like an eagle or hawk. Identification of raptors requires both visual and auditory observations. Although some

Activity:  Core

Age: All

Time: 1

Why? Bird use of estuaries is well known and they are sensitive to change, making them good indicators of habitat health. Raptors are the top of the food chain and require a plentiful supply of prey.

Safety: Wear solid boots, long pants and a hat. Snakes, prickles, sharp objects and the sun are the main safety concerns of this activity.

raptors are very distinctive, many others look similar and often confuse even the most skilled observers. Many raptors have a distinctive call, which makes identification easier. Pay attention to the bird's behaviour, including feeding, resting and flying, and the types of habitat it occupies. Be mindful of the time of year, because in the summer and autumn you are likely to see immature birds or post-breeding adults whose plumage is different from what is illustrated in most books. It often helps to obtain a bird list of your area. If you are not sure that you have identified a bird properly and it is not on the list, then it is likely that you have misidentified the bird. See if you can get another look at it. Lists are obtainable for most ornithological groups and from the Birds Australia website at <www.birdsaustralia.com.au>.

Record all birds meeting the criteria that are observed within your visible horizon when sitting above the high-tide mark, including dunes, spits or mangroves. This is usually done for a fixed amount of time (ideally 20 minutes). Record an approximate count of the birds seen.

Try to record the activity or habitat of each raptor that you observe and check to ensure that you have completely filled out field data forms.

A separate field datasheet should be used for each site and survey date. All datasheets should have the following information: observer(s), site location, a rough sketch of the study area with the vantage point location, survey date, time, time of sunrise, weather and tidal conditions. A good field datasheet will also include a sketch or photographs of the study area.

Data analysis of raptor surveys is possible, and is similar to those methods discussed in the *Tidal Flats – Recording Wading Birds* guideline, however fewer species of raptors are usually seen, so simple species lists and counts are the most common methods of presenting the data.



Across the estuary—recording invasive species

Introduced marine pests are plants or animals that do not naturally occur in an area. These have been introduced through human activity and are usually capable of seriously threatening native species.

Ships and shipping practices are largely responsible for the introduction of marine pests. Marine life grows on the hulls of large ships and ballast water discharge often includes marine life pumped on-board in a different state or country. Ports are the major hot-spots for marine and estuarine pest control.

Recently another source of aquatic pests has been found. With rapid, high-density shoreline development, the number of houses near major waterways has increased exponentially. Many of these have marine aquariums, which usually contain introduced plants and animals. Much aquarium life has been bred to be nutrient, turbidity and pH tolerant, so that it can survive in the less than optimum conditions present in many home aquaria. If these genetically selected life forms are let into our nutrient polluted waterways, they can spread rapidly and cause extensive damage to already stressed ecosystems.

Global warming and El Nino effects have also been blamed for allowing marine pests to rapidly colonise Australian shores.

Eradicating marine pests is very hard once they have established themselves in their new environment. Early detection and rapid action are the only real methods for controlling these species.

Several authorities in Australia deal with invasive marine species and may be able to provide information on identifying any you find. The Australian Quarantine Inspection Service (AQIS) has the primary responsibility of dealing with new introductions.

The Centre for Research on Introduced Marine Pests (CRIMP), is a branch of CSIRO that specialises in controlling marine pests. They are the publishers of Dianne Furlani's *A Guide to the Introduced Marine Species in Australian Waters* (1996).

CRIMP also produce a series of marine pest fact sheets, which may be downloaded from the following website <crimp.marine.csiro.au//Marine_pest_infosheets.html>

PIRSA has a list of introduced marine pests in South Australia. This list can be found at: <www.pir.sa.gov.au/pages/sus_res/mar_hab/pests.htm>.

Reef Watch (online at www.reefwatch.asn.au) has developed a monitoring program called *Feral or In Peril* that provides interested parties with waterproofed identification slates for identifying a number of introduced marine pests and few species of conservation significance that occur in South Australian coastal waters, as well as a monitoring manual that details the type of information Reef Watch requires, to enable any sighting data to be entered into its database.

What you may need:

- site record form
- list of known marine pests in SA
- factsheets with photographs of pests known to be in or near your estuary
- camera or drawing pad and pencil
- topographic map or GPS

Activity:  Core

Age: All

Time: 1

Why? Invasive species may completely alter an area of habitat. Early warning of the presence of an invasive species may allow authorities to take action before a pest becomes impossible to eradicate.

Furlani, DM (1996), *A Guide to the Introduced Marine Species in Australian Waters*, Technical Report Number 5, Centre for Research on Introduced Marine Pests, CSIRO, Hobart. The guide includes more than 70 species descriptions of marine organisms, along with photos and diagrams. It is produced in ring binder form without the binder. Updates are produced occasionally. It costs approximately \$50 and can currently be obtained from CRIMP, CSIRO, GPO Box 1538, Hobart, Tasmania 7001, <www.marine.csiro.au/crimp/nimpis/>.

What to do:

Monitoring for marine pests is often done while completing other monitoring exercises. Keep an eye out for known pest species or anything unusual while working on other methods.

If you find something unusual, take a photograph or sketch it. Attempt to identify it using the resources discussed in the introduction. If it does not match any of the listed species it may be a native that you are unfamiliar with, or an unlisted exotic. Take your photograph or drawing to an expert, for example the Museum of South Australia, Reefwatch or the Marine Habitat section of PIRSA to get the organism identified.

If you are certain, or nearly certain, that your subject is a potential marine pest then follow the procedure below:

- record the name of the species you think it may be
- record as much information about the specimen(s) you observed, such as colour, size, patterning and habitat (including substrate, temperature and salinity)
- assess how bad the infestation is. How many can you see? Have you seen it elsewhere? What area do they cover?
- record the location you found your specimen(s). You could use a topographic map or GPS to do this
- record the date and time of the observation, along with contact details of the people who made the observation
- do not disturb the organism. Some creatures such as brittle stars are very fragile and may reproduce from 'broken bits'. This is especially a problem with algae.

Telling someone

You should contact your Waterwatch coordinator, Reef Watch and/or AQIS to inform them of your discovery. Provide them with all the details and pictures, so that they are able to undertake a complete investigation into the sighting.



Across the estuary—monitoring bacteria

Bacteria are present in all environments and form an essential part of the **detrital cycle**. Not all bacteria are benign. Water that is contaminated with faecal matter from people or warm-blooded animals like cattle can carry bacteria that cause dangerous diseases. Unfortunately, many of these pathogens (disease-causing bacteria and viruses) are very difficult to detect in water samples as they may only be present in small numbers, yet still pose a danger to health.

Fortunately, there are other organisms that live in faecal matter in huge numbers, and we can use the presence of these bacteria as indicators of faecal contamination. Some of these more common bacteria may also cause disease themselves, but their main importance is that they warn us that more harmful pathogens may be present. The main groups of bacteria we use as indicators are described below.

Coliform bacteria are rod-type bacteria that ferment milk sugar, producing acid and gas as a by-product. While many coliform bacteria are found in faecal matter, not all the members of this group are. Some are found in plant material or soil. As a result, the total coliform test is usually used for testing drinking water, but not for testing freshwater swimming areas, as you would expect to find some soil-borne bacteria in swimming waters.

Recent studies (Geissler *et al*, 2000) of traditional and newer methods of testing for total coliforms in seawater have highlighted some interesting aspects to these tests. In marine waters, all types of total coliform tests had some false positive results. On examination of these false positives, it was discovered that the non-coliform bacteria that were reacting with the microbial media belonged to several groups of bacteria—the marine vibrios, the aeromonas group and several different enterobacteria. All of these were capable of causing illness in humans, suggesting that the total coliform test may still have a role to play in monitoring estuarine waters (Balows *et al*, 1991).

More specifically related to faecal matter is the **faecal coliform** group. There is an exception: a bacterium in this group, *Klebsiella pneumoniae*, grows on waste from paper pulp and is not associated with faeces at all.

Another bacterium, *Escherichia coli*, is a single species from the faecal coliform group that is only found in the gut of warm-blooded animals. This makes it an excellent indicator of contamination.

Finally, a group of bacteria called **enterococci** may be used as indicators. They are cocci-type bacteria belonging to the Streptococci group.

There are several tests available for these organisms. Some are presence-absence tests, some have two tests in one, and some produce count data. The following tests (Table 5) are suitable for use by community groups.

Activity:  Extension

Age: High school

Time: 4

Why? Certain bacteria are associated with faecal contamination of waterways. Their presence indicates that other, more harmful organisms may also be present. Such bacteria pose a risk to humans via swimming or through shellfish contamination.

Safety: Cultured bacteria must remain inside their sealed incubation container and should be either bleached or heated prior to disposal.

If the test requires checking for fluorescence, do this in a darkened environment and avoid looking directly at the light.

Table 5—Available bacterial tests

Organism	Presence–Absence test	Count test
Total Coliforms & <i>E. coli</i> (2 in 1)	IDEXX Colilert-18 (colour for TC and fluorescence for <i>E. coli</i>) Merck ReadyCult Coliforms 100 (colour for TC and fluorescence for <i>E. coli</i>)	Merck Envirocheck Contact C Slides Total Coliforms/ <i>E. coli</i> (all colonies develop specific colours)
Enterococci	IDEXX Enterolert (fluorescence) Merck ReadyCult Enterococci (colour)	

What you may need:

- record forms
- sterile container for collecting samples
- sterile disposable gloves
- sterile distilled water (boiled & cooled) for diluting samples
- sterile 10-mL disposable plastic syringe body for each sample
- sterile IDEXX 100-mL containers for incubating any presence–absence tests and doing dilutions
- incubator (bought or home-made, instructions for making may be found in Appendix 6)
- a 4–6 W UV lamp with a wavelength of 366 nm if a presence–absence test that uses fluorescence is selected.

To ensure good results:

Ensure you do not contaminate your samples—wash your hands prior to sampling, use sterile gloves and follow the sampling guidelines provided earlier in this manual.

All microbiological media have use-by dates. Check the dates of all media prior to going out into the field.

Timing of incubation is important. You cannot rely on positive results that are read after the appropriate time has been exceeded. See media package inserts for details.

What to do:

Collect a sample of water in a sterile container being careful not to contaminate it.

All seawater tests require dilution, so use a sterile syringe to withdraw 10-mL of the sample, and place this sub-sample directly into a 100-mL sterile IDEXX container. Top the container up to the 100-mL mark with sterile distilled water. Screw on the lid and shake lightly to mix. You are now ready to test the sample.

Presence–Absence tests

Take a sealed dose of the selected reagent and empty it into the 100-mL IDEXX bottle containing the diluted sample. Reseal the bottle, swirl until media is dissolved and place in the incubator at the correct temperature. Incubate for the specified time. The temperature and time vary depending on which media you select—read the package inserts to obtain these details.

Once the time is up, examine the colour of the liquid, as described on the package insert. If the test requires checking for fluorescence, do this in a darkened environment and avoid looking directly at the light.

Count tests

These slides are coated with agar and sealed in a sterile tube. Unscrew the lid of the tube and withdraw the slide. Dip the slide into the diluted sample water in the IDEXX 100-mL bottle for 5–10 seconds. Withdraw the slide from the sample, and allow to drain in air for a few seconds, then reinsert into the tube and close the lid tightly. Incubate the tube at the required temperature for the requisite time.

The contact slides have numbers on each side. Side 1 is a plate count agar, which grows a wide range of colonies. Side 2 is 'Chromocult' agar, and colonies of coliform bacteria grow red, while colonies of *E. coli* grow blue on this agar. Visually compare (do not count) the colonies on Side 2 with the pictures on the chart provided in the Envirocheck package insert. Record the number (0, 10^3 , 10^4 , 10^5 , 10^6 , 10^7) then multiply it by 10 to correct for the dilution factor and record the final result as 'colony forming units per mL'.



References

- Armstrong, P (1987), *Reading and Interpretation of Australian and New Zealand Maps*, 2nd edition, Longman Cheshire, Melbourne.
- Balows, A, Hausler, WJ, Herrmann, KL, Isenberg, HD & Shadomy, HJ (1991), *Manual of Clinical Microbiology*, 5th edition, American Society for Microbiology, Washington.
- Cameron, WM & Pritchard, DW (1963), 'Estuaries' in Hill, MN (ed), *The Sea*, Volume 2, John Wiley and Sons, New York, 306–324.
- Chapman, V (1960), *Saltmarshes and Salt Deserts of the World*, Interscience Publishers, New York.
- Christianson, IG, Clayton, MN & Allender, BM (1981), *Seaweeds of Australia*, Reed Books, Sydney.
- Carson, R (1962), *Silent Spring*, Houghton Mifflin, New York.
- Coultas, CL and Hsieh, Y (ed) (1997) *Ecology and Management of Tidal Marshes—A Model from the Gulf of Mexico*, St Lucie Press, Delray Beach, Florida.
- Dakin, WJ & Bennett, I (1987), *Australian Seashores: A Guide to the Temperate shores for the beach-lover, the naturalist, the shore-fisherman and the student*, Angus and Robertson Publishers, Australia.
- Dashorst, G & Jessop, J (1998), *Plants of the Adelaide Plains & Hills*, 2nd Edition, South Australia State Herbarium, Finsbury Press, South Australia.
- Datson, B (2002), *Samphires in Western Australia: A Field Guide to Chenopodiaceae Tribe Salicorniae*, Department of Conservation and Land Management, Perth, Western Australia.
- Davey, K (1998), *A Photographic Guide to Seashore Life of Australia*, New Holland Publishers, Sydney.
- Environment Protection Authority (2003), *Environment Protection (Water Quality) Policy*, EPA, Adelaide.
- Furlani, DM (1996), *A Guide to the Introduced Marine Species in Australian Waters*, Technical Report Number 5, Centre for Research on Introduced Marine Pests, CSIRO, Hobart.
- Geddes, M & Hall, D (1990), 'The Murray Mouth and Coorong' in Murray-Darling Basin Commission (ed), *The Murray*, Murray-Darling Basin Commission, Canberra.
- Geissler, K, Manafi, M, Amoros, I & Alonso, JL (2000), 'Quantitative determination of total coliforms and *Escherichia coli* in marine waters with chromogenic and fluorogenic media', *Journal of Applied Microbiology*, 88:280–285.
- George, A (ed)(1984), *Flora of Australia: Volume 4—Phytolaccaceae to Chenopodiaceae*, Commonwealth of Australia, Printed by Griffin Press, South Australia.
- Geoscience Australia (2002), *Australian Estuaries and Coastal Waterways*, a CD ROM produced for the National Land and Water Resources Audit, AGSO GeoScience Australia, Canberra.
- Geoscience Australia (2002), *Ozestuaries Database*, accessed 19 February 2007, <www.ozestuaries.org>.
- Haddon, F (1992), *Australia's Seashore: Environmental Field Guide to Flora and Fauna*, Simon and Schuster, East Roseville, New South Wales.
- Heap, A, Bryce, S, Ryan, D, Radke, L, Smith, C, Smith, R, Harris, P & Heggie, D (2001), *Australian Estuaries and Coastal Waterways: A Geoscience Perspective for Improved and Integrated Resource Management*, a report for the National Land and Water Resources Audit, AGSO GeoScience Australia, Canberra.
- Jessop, J & Toelken, H (1986), *Flora of South Australia. Parts I–IV*, 4th edition, South Australian State Herbarium, South Australian Government Press, Adelaide.
- Jones, D & Morgan, G (2002), *A Field Guide to Crustaceans of Australian Waters*, 2nd edition, Reed New Holland, Sydney.



- May, JD (1999), 'Spatial variation in litter production by the mangrove *Avicennia marina* var. *australasica* in Rangaunu Harbour, Northland, New Zealand' in *New Zealand Journal of Marine and Freshwater Research*, 33:2.
- Mitrovic, S (1995), *What Scum is That?* Water Quality Services Unit, Department of Land & Water Conservation, Government of NSW.
- Morgan, D, Best, E, Betjeman, K, Larkum, A, Pitman, M, Thomson, J, Lee, A & Nicholas, J (eds) (1973), *Biological Science: The Web of Life*, Australian Academy of Science.
- National Land and Water Audit (2002), *Australian Catchment, River and Estuary Assessment 2002*, Volumes 1 and 2, Commonwealth of Australia, Canberra.
- Nicholson, E, Ryan, J & Hodgkins, D (2002), 'Community data—where does the value lie? Assessing confidence limits of community collected water quality data', *Water Science and Technology*, 45:11, 193–200, IWA Publishing.
- Otley, H (2000), *Huon Valley Water Quality Report 1996—2000: an aid to catchment management decisions*, A report based on water quality data collected by the Huon Healthy Rivers Waterwatch Groups, Huon District of Forestry Tasmania and the Department of Primary Industries, Water and Environment, Hobart.
- Packham, JR & Willis, AJ (1997), *Ecology of Dunes, Saltmarsh and Shingle*, Chapman and Hall, London.
- Ponder, W, Hutchings, P & Chapman, R (2002), *Overview of the Conservation of Australian Marine Invertebrates: Chapter 2—The Australian Marine Invertebrate Fauna*, Environment Australia, accessed 21 January 2006, <www.amonline.net.au/invertebrates/marine_overview/chapt2.html>.
- Pritchard, DW (1952), 'Estuarine hydrography', *Advances in Geophysics*, 1:243–280.
- Purinton, TA & Mountain, DC (1997), *Tidal Crossing Handbook—A Volunteer Guide to Assessing Tidal Restrictions*, Parker River Clean Water Association, Massachusetts.
- Shepherd, SA & Thomas, IM (1982–1997), *Marine Invertebrates of Southern Australia: Parts I–III*, Government Printer, South Australia.
- State Emergency Service (1991), *Map Reading Handbook*, 2nd edition, TASMAR, Environment and Planning, Tasmania.
- Tchobanoglous, G & Burton, FL (eds) (1991), *Metcalf & Eddy Wastewater Engineering: Treatment, Disposal, Reuse*, McGraw-Hill International Editions, Singapore.
- Waterwatch Australia Steering Committee (2002), *Waterwatch Australia National Technical Manual, Volume 4—Physical and Chemical Parameters*, Environment Australia, Canberra.
- Wilton, K & Saintilan, N (2000), *Protocols for Mangrove and Saltmarsh Habitat Mapping*, Australian Catholic University.
- Zedler, JB (ed) (2001), *Handbook for Restoring Tidal Wetlands*, CRC Press, Boca Raton, Florida.










Appendix 1—Monitoring Framework

Desk-based activity		Core activity	
Activity with a field component		Extension activity	









1 Designing your monitoring program





Activity	Type	Age group	Time req.	Why is this monitored?
Selecting activities for your program		All	3	
Selecting your sampling location/s		All	3	

2 Sampling site observations










Activity	Type	Age group	Time req.	Why is this monitored?
Recording weather details		All	1	The weather affects many other parameters being measured in a monitoring program, and should be included to aid interpretation of the collected data.
Recording time and tide height		All	1	The state of the tide affects many other parameters being measured in a monitoring program, and should be included to aid interpretation of the collected data.
Recording any odours		All	1	Algal blooms, some effluents and anoxic conditions all produce various odours.
Recording water depth		All	1	The depth of water affects many other parameters being measured in a monitoring program, and should be included to aid interpretation of the collected data.
Recording water surface conditions		All	1	The water surface condition affects parameters such as Secchi depth as well as the amount of mixing occurring in the water, so should be included to aid interpretation of the collected data.
Recording water colour		All	1	Water colour may indicate dissolved matter, the colour of the substrate, the presence of muddy sediments, floating oils or the presence of an algal bloom.
Measuring flow rate		All	2	The speed that water moves provides an indicator of the volume of stormwater or riverine flows into an estuary, or the strength of the tides. Calmer areas are more likely to host certain types of submerged aquatic vegetation.

3 Understanding estuarine processes










Activity	Type	Age group	Time req.	Why is this monitored?
Measuring the water temperature		All	1	Temperature plays an important role in determining the amount of oxygen that can remain in the water, as well as affecting the reproductive status of many organisms. Warmer or colder water than usual for an area may allow invasive species to gain a foothold. Temperature also controls the rate of many chemical processes. Useful for comparing drains/outfalls with the ambient temperature.
Measuring pH		All	2	pH controls the solubility of many metals such as iron and copper in water. Low pH may indicate acid rain events or acid soil drainage; values outside 5–10 indicate industrial pollution or cataclysmic events. Estuaries are normally between 6.5 and 8.5 (lower in the fresh end, higher in the marine end).
Measuring turbidity/water clarity		All	2	Measures water clarity, which controls the depths at which submerged vegetation can grow. Turbidity may indicate erosion or algal blooms and is a very useful measure for mapping plumes from stormwater outlets. SA EPA considers <1 = good and >10 = poor in estuarine conditions.
Measuring salinity/conductivity		All	2	Salinity is the dissolved salt content of water. Salinity and temperature are the primary causes of stratification in estuaries. Salinity affects the amount of oxygen that can be dissolved in the water. Flocculation of suspended solids is also affected by salinity. Plants and animals are distributed in estuaries according to their salinity preferences.
Obtaining a topographic map or aerial photograph of your estuary		High school	na	The width-to-depth ratio of an estuary has an impact on the mixing of the fresh and saline waters within it. Mapping shoreline changes over time, with aerial photography, allows any changes in the quantity of flow and the effect of riparian clearing to be noted.
Determining whether your estuary is positive or negative		High school	3	This factor determines the salinity patterns in the estuary.
Classifying your estuary		High school	2	Different types of estuaries are very different in their mixing regime and in how pollution is distributed. Understanding what type of estuary you are studying is an essential step in understanding why things happen the way they do in your estuary.
Mapping the salinity gradient of your estuary		High school	4	Estuaries vary in salinity from the head to the mouth.





Activity	Type	Age group	Time req.	Why is this monitored?
Mapping the salinity profile of your estuary		High school	3	Stratified estuaries have quite different water quality at different depths. To determine how a pollutant will be distributed in an estuary it is necessary to understand its stratification pattern.
Mapping the turbidity plume from a stormwater outlet		High school	4	This activity measures the impact of catchment erosion.
Determining where the 'turbidity maximum' is for your estuary		High school	4	The turbidity maximum is an important location, marking where maximum sedimentation may be expected in an estuary.
Studying the effect of tidal restrictions on wetlands that fringe your estuary		High school	4 (initial) 1 (usual)	Estuarine wetlands develop their character (saltmarsh, melaleuca wetland, freshwater–tidal wetland) from the hydrological regime. Monitoring the ground and surface water quantity and quality can provide insight into the health of, and reasons for long-term change in, these wetlands.

4 Chemical assessments











Activity	Type	Age group	Time req.	Why is this monitored?
Measuring dissolved oxygen		All	3 (test) 2 (meter)	Most aquatic organisms require at least 3–5 mg/L dissolved oxygen to survive. The concentration in water varies depending on salinity, temperature and time of day. Below 4 mg/L indicates hypoxia, below 0.5 mg/L indicates anoxia.
Measuring sulphide		All	3	This is measured as a surrogate for dissolved oxygen. There should be no sulphide present in surface waters of an estuary.
Measuring orthophosphate		All	2	Soluble ortho-P is rapidly taken up by algae. It is frequently an indicator of fertiliser runoff. Total ortho-P includes the more easily dissolved particulate orthophosphates (those loosely bound to clay particles, or incorporated into planktonic algae, for example).
Measuring ammonium		All	2	SA EPA considers <0.05 is good and >0.5 is poor in estuarine conditions. High concentrations of ammonium are toxic to fish.
Measuring chlorophyll-a		Adult groups	4	Chlorophyll is the green pigment in plants and is a good measure of the plankton present in water as well as the trophic status of the estuary. This is a surrogate test for nutrients.
Measuring alkalinity		All	2	Measures the water's ability to neutralise acidity. Fresh varies between 30–90 mg/L, seawater is around 116 mg/L.
Measuring total suspended solids		High school	4	Related to turbidity (and from the same sources), suspended solids measure the amount of solid particles in the water. These fine particles can clog fish gills and foul benthic filter feeders.
Using mass balances to estimate point-source pollutants		High school	4	Many contaminants are toxic to marine life.
Measuring soil salinity and soil pH		High school	2 or 3	The soil salinity and pH of saltmarsh areas determines the types of vegetation that will develop. These parameters can change rapidly after tidal gates or other restrictions are put into an estuary, causing large changes in the vegetation and sometimes causing a release of acid into the main estuary.

5 Looking at the vegetation and habitat

Activity	Type	Age group	Time req.	Why is this monitored?
Keeping a visual record of plankton blooms		All	1	Plankton blooms are often the result of high nutrient concentrations.
Getting to know phytoplankton with a net tow		High school	3	Different types of plankton are always present in the water. The dominant types vary depending on the conditions. For example, diatoms require a source of silica (usually present in riverine water entering the estuary) as well as nutrients, while dinoflagellates only require nutrients (such as may be found in sewage outfalls). Plankton blooms are often the result of high nutrient concentrations.
Counting and identifying phytoplankton in a water sample		Adult groups	4	The number of plankton cells present in a water sample, as well as their type, provides information on the health of the estuary.
Monitoring a seaweed bed		High school	3	The more complex types of brown seaweed (macroalgae) may respond to disturbance by disappearing from seaweed beds.
Monitoring an intertidal SAM bed		High school	3	SAM (submerged aquatic macrophyte) beds are an essential habitat in estuaries, and are major fish nursery areas. Most SAM beds require high light levels to survive, and appropriate inundation, temperature and salinity regimes.
Monitoring submerged SAM beds		Adult groups	4	SAM beds are an essential habitat in estuaries, and are major fish nursery areas. Most SAM beds require high light levels to survive, and appropriate inundation, temperature and salinity regimes.
Assessing the fringing vegetation of the estuary for continuity		High school	2	Width and continuity of riparian vegetation is related to water quality, particularly water clarity. Other important indicators are weediness and erosion.
Monitoring mangrove leaf/twig litter production		High school	3	Leaf and flower production is an indicator of mangrove health. The litter from mangroves is an important part of the detrital cycle.
Monitoring mangrove expansion		High school	2	Regular examination of the seaward and landward edges of a forest can determine if the forest is expanding or retreating landward. This can indicate local relative sea-level change or sediment availability.

Activity	Type	Age group	Time req.	Why is this monitored?
Studying saltmarsh plants		High school	3	Saltmarsh areas are valuable wader habitat and fish nurseries. Saltmarshes are very vulnerable to hydrological change, which may be revealed by a change in vegetation.
Monitoring leaf/twig litter production in a <i>Melaleuca</i> wetland		High school	3	In SA, <i>Melaleuca halmaturorum</i> fringing wetlands are often present in the estuarine parts of swampy watercourses. They may be seasonally inundated, and frequently have stands of <i>Phragmites</i> . These freshwater-intertidal wetlands are an important filter system for floodwaters, and also attenuate the progression of floods to the more marine parts of the estuary.
Using aerial photographs to map habitat change		High school	3	This measures the changes in coastal habitats over a long period of time.
Measuring beach litter (cleanups!)		All	2 or 3	Many non-biodegradable articles cause harm to marine life (entanglements and ingestion), people (diver entanglements, cuts to beach-users) and to marine vessels (propeller snags, etc). The estuarine benthic habitat may also become smothered with non-biodegradable wastes.

6 Observing animal life

Activity	Type	Age group	Time req.	Why is this monitored?
Tidal flats—Looking for benthic invertebrates		High school	3	Estuarine macroinvertebrates are likely to reflect the overall healthiness of the habitat.
Tidal flats—Counting crab burrows		All	2	Estuarine macroinvertebrates are likely to reflect the overall healthiness of the habitat. In mudflat and sandflat environments the quantity of burrows provides information on the firmness of the substrate.
Tidal flats—Counting surface snails		All	2	Estuarine macroinvertebrates are likely to reflect the overall healthiness of the habitat. Snails may also be impacted by over-harvesting (eg food, bait and souvenirs).
Tidal flats—Recording wading birds		All	2	Estuarine organisms are likely to reflect the overall healthiness of the habitat. Migratory and resident wader use of estuaries is well known and they are sensitive to habitat change, so changes are easier to identify.
Rocky shore—Counting grazing snails		High school	3	Estuarine macroinvertebrates are likely to reflect the overall healthiness of the habitat. Snails may also be impacted by over-harvesting (eg food, bait and souvenirs).
Saltmarsh—Looking for invertebrates in saltmarsh habitats		Adult groups	4	Estuarine macroinvertebrates are likely to reflect the overall healthiness of this mixture of habitats.
Saltmarsh—Counting crabs and fish in saltmarsh creeks		Adult groups	4	Estuarine organisms are likely to reflect the overall healthiness of the habitat. Fish and crabs respond rapidly to changes in tidal regime that occur when restrictions are placed in saltmarshes.
Across the estuary—Recording raptors		All	1	Estuarine organisms are likely to reflect the overall healthiness of the habitat. Bird use of estuaries is well known and they are sensitive to habitat change, so changes are easier to identify.
Across the estuary—Recording invasive species		All	1	Invasive species may completely alter an area of habitat. Early warning of the presence of an invasive species can allow authorities to take action before the pest becomes impossible to eradicate.
Across the estuary—Monitoring bacteria		High school	4	Certain bacteria are associated with faecal matter contamination of waterways. Their presence indicates that other, more harmful organisms may also be present. Such bacteria may pose a risk to humans via swimming or through shellfish contamination.

Appendix 2—Beaufort Scale

Beaufort Number	Description	Wind Speed (kph)	Wind Speed (knots)	Observations
0	calm	0	0	Tree leaves don't move; smoke rises vertically; sea is calm, mirror like.
1	light air	1–5	1–3	Tree leaves don't move; smoke drifts slowly; direction of wind shown by smoke, not by vane; sea is lightly rippled.
2	slight breeze	6–11	4–6	Tree leaves rustle; flags wave slightly; vanes show wind direction; small wavelets or scale waves.
3	gentle breeze	12–19	7–10	Leaves and twigs in constant motion; small flags extended; long unbreaking waves.
4	moderate breeze	20–29	11–16	Small branches move; flags flap; waves with whitecaps.
5	fresh breeze	30–38	17–21	Small trees sway; flags flap and ripple; moderate waves with many whitecaps.
6	strong breeze	39–50	22–27	Large branches sway; umbrellas used with difficulty; flags beat and pop; larger waves with regular whitecaps.
7	moderate gale	51–61	28–33	Sea heaps up, white foam streaks; whole trees sway; difficult to walk; large waves.
8	fresh gale	75–86	34–40	Twigs break off trees; moderately high sea with blowing foam.
9	strong gale	75–86	41–47	Branches break off trees; tiles blown from roofs; high crested waves.
10	whole gale	87–101	48–55	Some trees blown down; damage to buildings; high churning white seas and exceptionally high waves hiding ships from view.
11	storm	102–120	56–63	Widespread damage to trees and buildings; mountainous waves; sea covered in white foam.
12	hurricane	120+	64+	Severe and extensive damage.

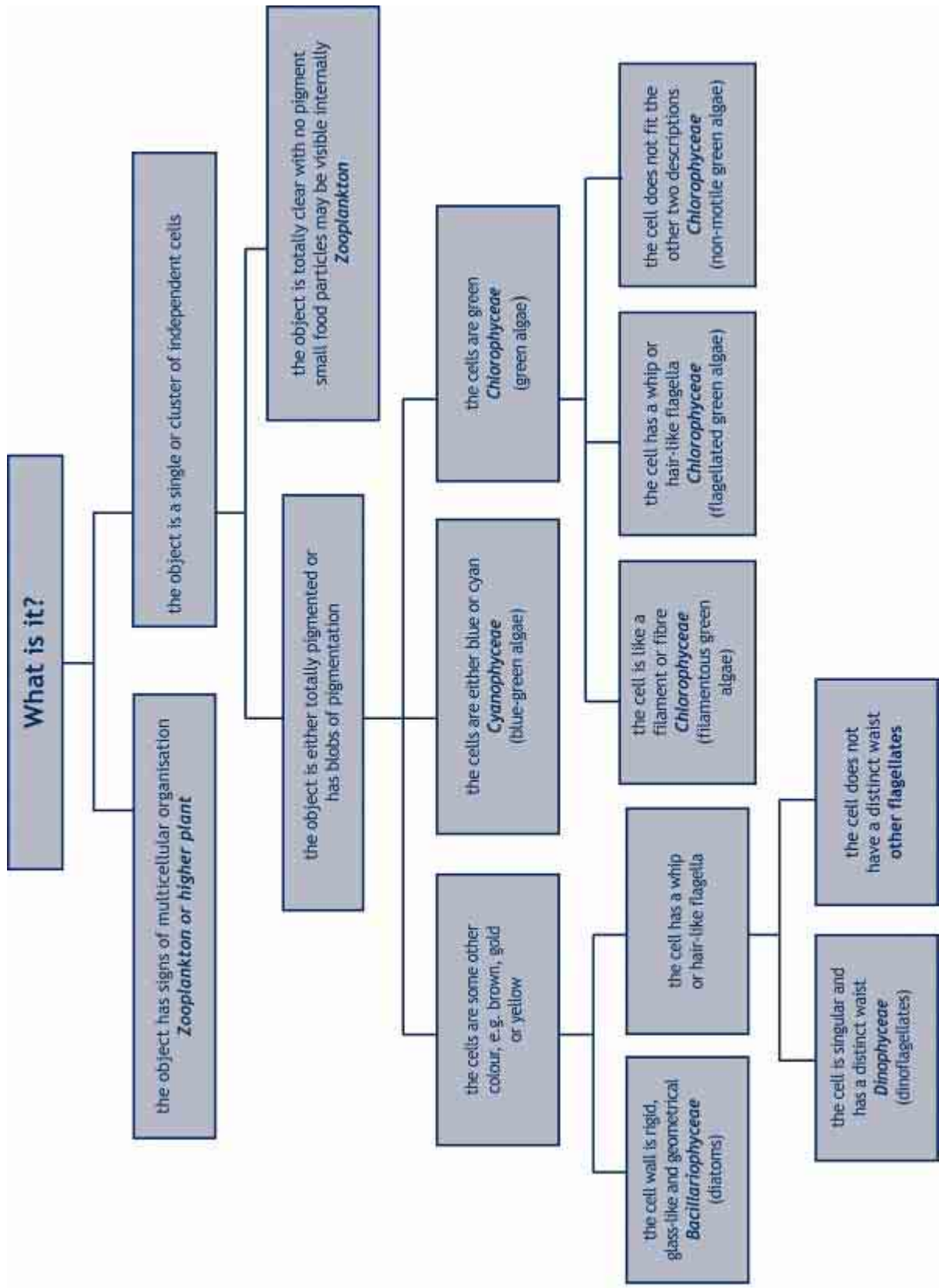
Appendix 3—Water colour chart

255,255,255	255,255,204	255,255,153	255,255,102	
204,255,255	204,255,204	204,255,153	204,255,102	204,204,153
153,255,255	153,255,204	153,255,153	153,204,0	153,153,51
102,255,255	102,255,204	102,255,153	102,153,0	102,102,51
102,204,255	0,204,204	0,204,102	51,102,0	102,51,0

This chart is based on the HTML-RGB (red, green, blue) colour scheme used for describing colours used on PC and TV monitors. The particular colours selected are 'non-dithering' colours that are likely to reproduce well on most printers, and on monitors that display at least 256 colours.

The range of colours is similar to the range found in the Forel-Ule colour scale that is commonly used in estuarine studies.

Appendix 4–Plankton identification flow chart



Appendix 5—Forms

Site record

Group:	Tide data:	
Site name:	Stage of tide:	
Samplers:	Last tide—height: time:	
Date and time:	Next tide— height: time:	
Weather observations:	Odours and observations: (include dumping, algal blooms and invasive species)	
Air temperature:		
Wind:		
Cloud cover:		
Rain:		
Surface water conditions:		
About the water body:		
What depth is the water you are sampling?	<input style="width: 50px; height: 20px;" type="text"/>	m
What colour is the water?	<input style="width: 50px; height: 20px;" type="text"/>
What temperature is the water?	<input style="width: 50px; height: 20px;" type="text"/>	°C
What is the clarity of the water?	<input style="width: 50px; height: 20px;" type="text"/>	m
Is the water flowing in one direction?	<input style="width: 50px; height: 20px;" type="text"/>	
If so, what is its flow rate?	<input style="width: 50px; height: 20px;" type="text"/>	m/sec
Measuring salinity:	Water quality parameters:	
Method: Hydrometer <input type="checkbox"/>	Method: Conductivity <input type="checkbox"/>	
Raw SG:	EC: mS/cm	pH <input style="width: 50px; height: 20px;" type="text"/>
Total Dissolved Salts (TDS)		Dissolved oxygen <input style="width: 50px; height: 20px;" type="text"/> mg/L
..... g/L		DO saturation <input style="width: 50px; height: 20px;" type="text"/> % sat
		Orthophosphate (as) <input style="width: 50px; height: 20px;" type="text"/> mg/L
		Ammonium (as) <input style="width: 50px; height: 20px;" type="text"/> mg/L
		Alkalinity (as.....) <input style="width: 50px; height: 20px;" type="text"/> mg/L
Extra chemical parameters: (record any other test results here, along with the units measured and chemical species being reported)		

Hydrology field data record

Group:	Location numbers and locations: (eastings and northings or longitude and latitude)
Estuary name:	
Samplers:	
Date and time:	

Map of estuary showing sampling locations:

Hydrology data – Surface water:

Location number	Time	Surface water height (m AHD)	Location number	Time	Surface water height (m AHD)

Hydrology data – Ground water:

Location number	Time	SWL	pH	EC (mS/cm)	Temp	Raw SG	TDS (g/L)

Beach litter record

Group:	Location numbers and locations: (eastings and northings or longitude and latitude)
Estuary name:	
Samplers:	
Date and time:	

Map of estuary showing collection location:

Record rubbish collected as: Number in category (Weight of category)

.....

Comments:

Plankton count record

Group:	Location numbers and locations: (eastings and northings or longitude and latitude)
Estuary name:	
Samplers:	
Date and time:	

Map of estuary showing collection location:

Dilution factor: 10

	Tally	Cells counted	Cells/mL	Surety
Blue-greens				
Filamentous greens				
Flagellated greens				
Non-motile greens				
Diatoms				
Dinoflagellates				
Other flagellates				

Total plankton per mL

Saltmarsh vegetation record

Group:	Location numbers and locations: (eastings and northings or longitude and latitude)
Estuary name:	
Samplers:	
Date and time:	

Maps

Map of estuary showing location	Comments about the site (weeds, etc)
---------------------------------	--------------------------------------

Drawings of vegetation transects with measurements:

Riparian vegetation record

Group:	Location numbers and locations: (eastings and northings or longitude and latitude)
Estuary name:	
Samplers:	
Date and time:	

Maps

Map of estuary showing location	Comments about the site (weeds etc)
Drawing of riparian vegetation transect with measurements	

Other details:

Litter production record

Group:	Location numbers and locations: (eastings and northings or longitude and latitude)
Estuary name:	
Samplers:	
Date and time:	
Map of estuary showing sampling locations:	

Record trap contents as: Number (Dry weight)

	Leaves	Flowers	Bark	Seeds or propagules	Other items
Trap 1					
Trap 2					
Trap 3					

Quadrat counting record (macroinvertebrates)

Group:	Location numbers and locations: (eastings and northings or longitude and latitude)
Estuary name:	
Samplers:	
Date and time:	
Map of estuary showing sampling locations:	

Transect 1	Species and counts
Quadrat 1	
Q2	
Q3	

Transect 2	Species and counts
Quadrat 1	
Q2	
Q3	

Transect 3	Species and counts
Quadrat 1	
Q2	
Q3	

Transect 4	Species and counts
Quadrat 1	
Q2	
Q3	

Saltmarsh invertebrates record

Group:	Location numbers and locations: (eastings and northings or longitude and latitude)
Estuary name:	
Samplers:	
Date and time:	
Map of estuary showing sampling locations:	

Transect 1	Grazing snails	Crab burrows	Benthic core invertebrates	Macro-invertebrates on plants in quadrat	Aerial sweep of transect	D-net findings for submerged areas of transect
Quadrat 1						
Q 2						
Q 3						

Transect 2	Grazing snails	Crab burrows	Benthic core invertebrates	Macro-invertebrates on plants in quadrat	Aerial sweep of transect	D-net findings for submerged areas of transect
Quadrat 1						
Q 2						
Q 3						

Tidal flat benthic macroinvertebrate record

Group:	Location numbers and locations: (eastings and northings or longitude and latitude)
Estuary name:	
Samplers:	
Date and time:	

Map of estuary showing sampling locations:

Transect 1	Species and counts
Core 1	
Core 2	
Core 3	

Transect 2	Species and counts
Core 1	
Core 2	
Core 3	

Transect 3	Species and counts
Core 1	
Core 2	
Core 3	

Transect 4	Species and counts
Core 1	
Core 2	
Core 3	

Saltmarsh fish and crabs record

Group:	Location numbers and locations: (eastings and northings or longitude and latitude)
Estuary name:	
Samplers:	
Date and time:	

Map of estuary showing sampling locations:

Record trap contents as: Number of each species (Total weight of each species)

	Fish species	Crab species
Trap 1		
Trap 2		
Trap 3		

Appendix 6—Equipment

Making a Secchi disk

Secchi disks (Figure 19) are used for a number of measurements in estuaries. Their main function is to measure water clarity. However, they may also be used to test water depth and water colour. More details are given on how these disks are used in the guidelines.

Below we explain how to make a 300-mm Secchi disk, even though a disk is included in Waterwatch core kits for estuarine monitoring. This is because they are easy to lose in fast-flowing water. They also make a good fundraiser if you wish to sell them to other groups or organisations, as commercially available Secchi disks are relatively expensive.

Materials

- a sheet of aluminium or high density plastic at least 300 mm square
- large compass or pen attached by its nib to a piece of string
- jigsaw with an appropriate sized blade
- large eye-bolt with three large nuts and two washers
- drill with a bit slightly larger than the shank of the eye-bolt
- non-stretch corrosion resistant cord or plastic coated stainless steel wire
- something to mark the cord with (permanent marker or bands of heat-shrink plastic)
- primer in a spray can
- black and white paint in spray cans
- paper and sticky tape for masking
- newspaper to protect work surfaces
- a couple of clamps
- a solid workbench
- a hammer.



Figure 19—Secchi disk

Method

Mark a 300-mm circle on your sheet of aluminium or plastic.

Clamp the sheet of material $\frac{1}{2}$ or $\frac{2}{3}$ onto the workbench.

Cut the section of the circle hanging off the workbench, then move the sheet around, clamp it down and cut the remainder of the circle.

Drill a hole through the centre of the circle.

Spray the disk with primer on both sides.

Spray the disk with white on both sides.

Mask two diagonally opposite quarters off, on either side of the disk.

Spray the unmasked quarters on each side with black paint.

Fit the eye-bolt through the hole with at least one washer and a nut on either side. More nuts will give more weight, so that the disk will sink better.

Cut the end of the bolt off and hammer it so the nuts cannot undo.

Attach the cord or wire to the eye ensuring that it is secure.

Mark the cord at known intervals, possibly every 100–500 mm.

Making a depth beeper

Depth beepers (Figure 20) are the most accurate and reliable method of determining water depth in a groundwater well. They are also easy to carry and economical to use. This is the method used by most well monitoring specialists.

The beepers are constructed using a small battery-powered electronic circuit, a waterproof or water resistant box and a length of electrical cable. This cable may be of any length, but the activities in this manual do not require a length of more than 2–3 m.

The unit described in this method is relatively economical. Purchasing all the materials required to construct the unit should cost \$20–\$25, as long as you have a soldering iron, a drill and some screws or something else to mount the circuit board into the box. The beeper uses battery power and is suitable for construction by a hobbyist.

Materials

- 9-volt battery
- battery clip
- electronic buzzer (4–15V DC) (Dick Smith code L7009)
- light emitting diode (LED) of any type or colour
- one wire of a 2–3 m long piece of figure-8 cable (Dick Smith W2020)
- BC557 transistor
- BC547 NPN transistor
- 1 m Ω ¼ W carbon film resistor
- 2 x 100 k Ω ¼ W carbon film resistors
- 10 k Ω ¼ W carbon film resistor
- soldering iron and solder
- 'Varoboard' or matrix board with 2.5 mm hole spaces
- clear plastic box
- drill with assorted bits
- something to seal any holes made in the box (silica, rubber seals or waterproof tape)
- screws to mount the circuit board in the box
- a wine cork or rubber eraser
- waterproof glue or silicone sealant

Method

Cut the matrix board to fit the area required for the circuit, ensuring that it will fit into the box.

Using the circuit diagram (Figure 21), construct the circuit on the matrix board. Instructions on how to do this are available from most electronic hobby stores.

Attach the battery clip onto the board at the place indicated in the diagram, and attach the battery.

Attach the electrical cable to the circuit board.

Check to make sure the unit works by dipping the end of the cable into a glass of water and pulling it out. The unit should make a noise and the LED should turn on.

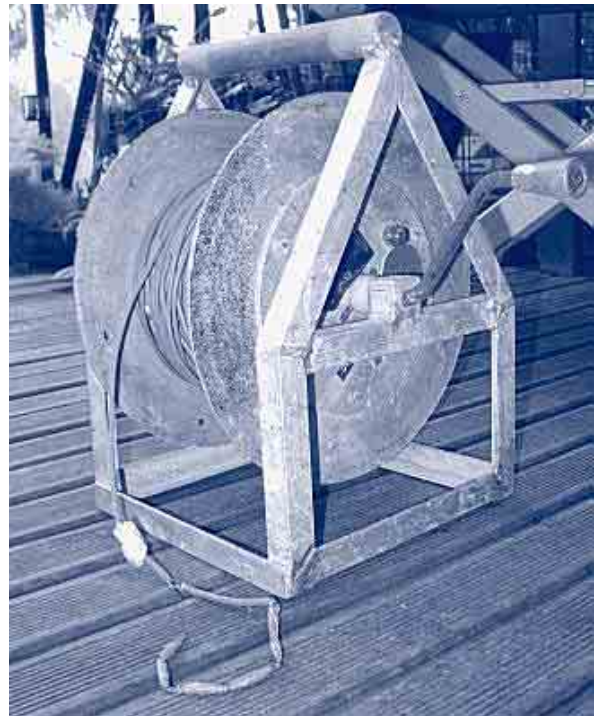


Figure 20 – Depth beeper for deep wells

Drill a hole into the side of the box, and thread the cable through.

Mount the circuit board in the box, ensuring that the battery is secure and accessible.

Screw the lid onto the box and ensure that the unit still works and that you can still easily see and hear the buzzer and the LED. If you can't, drill a number of small holes into the lid of the box and cover them with a thin water resistant material, e.g. tape, cling wrap or fine fabric.

Once you are happy with how the box works, cut the covering of the wire at the end so that at least 0.50 mm of each wire is showing.

Drill two small holes or force a large needle into the cork or eraser, forming two holes at least 10 mm apart.

Separate the two wires of the cable at the end and push them through the cork or eraser so that 10 mm or so is protruding out of the lower side.

Fasten the cork or rubber to the cable using the waterproof glue or silica sealant.

You are now ready to go!

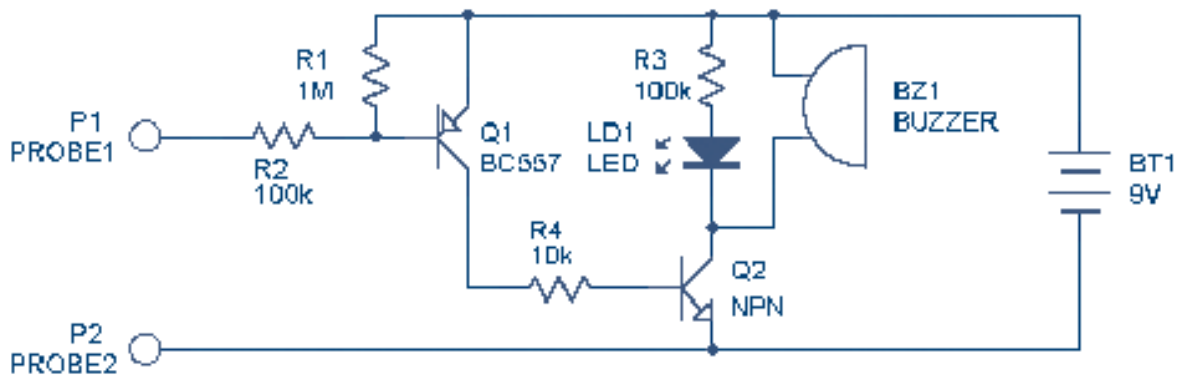


Figure 21—Circuit diagram for depth beeper

BT1 = battery and battery clip

BZ1 = electronic buzzer

LD1 = light Emitting Diode (LED)

P1 = first wire of the cable

P2 = second wire of the cable

Q1 = BC557 transistor

Q2 = BC547 NPN transistor

R1 = 1 M Ω ¼ W carbon film resistor

R2 = 100 k Ω ¼ W carbon film resistor

R3 = 100 k Ω ¼ W carbon film resistor

R4 = 10 k Ω ¼ W carbon film resistor

Making an incubator

An incubator or environmental chamber is a box that is maintained at a specified temperature. They are used to keep samples for bacterial testing warm. These chambers are quite expensive to purchase, but they are relatively easy to construct. A hand-made incubator may cost as little as \$35, although the sky is the limit.

Any sort of insulated box may be used to make the incubator—a broccoli box, an esky or even an old fridge is appropriate, depending on the size and robustness required.

A competent (licensed) person should do the electrical wiring of the incubator as it uses mains power. Instructions on how to wire the bulb mount and the thermostat are usually provided with these items, so they are not discussed here.

The heat source for the incubator could be an incandescent globe, a ceramic globe, a heat mat or a heating cord. The heat source should not put out large quantities of UV light. Large amounts of UV may kill the bacteria you are trying to monitor.

Electricity is dangerous, particularly near water or if the wiring is done incorrectly. Get a licensed person (your local electrician) to install the wiring. It is possible that you could make an incubator with 12-volt power, which would make it safer, more portable and would allow a hobbyist to do the wiring, but it would require a specialised thermostat and globe.

Materials

- insulated box
- heat source and fitting if required
- two small pieces of craft wood (MDF) (optional)
- > 2-m electrical extension cord
- thermostat (available from a hardware store or pet shop)
- sharp knife, drill or jigsaw
- selection of self-tapping wood or plastic screws.

Method

Ensure that the insulated box is intact and can be sealed with no large gaps or holes.

Cut a small hole in the top large enough to pass an electrical cable through.

Cut the extension cable in half.

Cut the female plug (the one with the slots for the pins) off the electrical cable.

Thread the plain half of the extension cable through the hole in the box.

Wire the fitting for the heat source to the wire inserted into the box.

Mount the fitting onto the inside of the box. Placing a piece of craftwood on the outside of box and mounting the fitting through the box onto the wood may be the most stable way, if the box is lightly made.

Cut another two holes next to each other near the middle of the box. Thread the lead from the heat source through one, and the other half of the extension lead through the other.

Wire the thermostat so that the light is controlled by it and the male plug is available to plug into the mains power supply.

Ensure that all exposed wires and holes in the box are covered.

Plug the box in and ensure it works, eg turn the thermostat to above room temperature and check that the heat source turns on.

Monitor the temperature to ensure it remains fairly constant, before using the incubator.

Making a depth water sampler

Depth samplers vary in complexity. A search on the internet reveals a wide range of samplers varying considerably in complexity and price. This appendix details how to make a basic sampler for shallow (< 4 m) depths. Deeper sampling requires a more complicated device. Strong currents may also make sampling difficult using this device.

The pole of the sampler may be made from anything, as long as it is rigid and is long enough to reach the required depth, with a bit left out of the water to hang on to. Telescopic poles made of different widths of pipe could be used if the depth is great; broom handles or bits of dowel are fine for shallow water.

Another method is to use a strong cord instead of the pole and a well-weighted bottle. The weights are often fishing weights or a concrete block attached to the base of the bottle.

Materials

- a pole
- a sample bottle with a narrow neck
- a small piece of wood
- a couple of nails and a hammer
- two large worm-drive hose clamps
- something to mark the pole with, eg paint or a permanent marker
- cork or rubber bung that fits the opening of the bottle
- a length of string as long as the pole.

Method

Nail the small piece of wood onto the end of the pole, making an 'L' shaped foot plate.

Place the sample bottle neck-upwards onto the foot plate.

Use the hose clamps to fasten the sampling bottle to the pole, resting the base of the bottle on the wooden foot-plate.

Mark the pole with the distance (in centimetres) from the bottle opening.

Fasten the string through the cork or bung.

Insert the cork into the mouth of the bottle.

Attach the other end of the string onto the opposite end of the pole. The string is easier to handle if it runs through some eyelets that are mounted on the pole.

To use the sampler, lower the bottle into the water until it reaches the appropriate depth. Pull the cork out using the string and allow the bottle to fill completely. Once the bottle is full, lift it directly out of the water.

Making a filtration unit

Filtering devices (Figure 22) are available in many shapes and prices. Each group using a filter will probably have a different unit, depending on the resources of the group. This section discusses what is available and their merits.

The units are discussed in two parts: filter holders and pumps. Items from each of these categories may generally be mixed and matched to suit your group's requirements, but be aware that there are some that will not go together. Ask your laboratory supplier about this before purchasing equipment. Assorted bits of clear silicone piping and adaptors may solve the problem.



Figure 22 – Assorted filtration equipment

Filter holders and vessels

Sartorius filter These are the most luxurious combination of filter holder, sample holder and vacuum vessel, but they are expensive (several hundred dollars). They attach simply to most pumps, and a few bits of clear pipe and/or a couple of adaptors allow you to use them with the cheaper forms of pump such as the syringe.

47 mm filter holder



This is a two-piece filter holder that screws together with a filter paper inserted in between the two halves. They are a step down from the filtering unit, as they have no vacuum vessel. The lack of vacuum vessel means that they are difficult to attach to a standard pump, but a large syringe works particularly well.

Disposable filter

These are also designed for use on a large syringe. They are a sealed disposable unit containing the filter paper. They are single use, as removing the paper destroys the unit. They are not suitable for suspended solids testing, as you can't weigh the filter first, but are fine for chlorophyll analysis. At \$10–\$20 each, they are not cheap. They do not make good financial sense if you plan to use a lot of them. If you only intend to use them once or twice a year for chlorophyll testing they may be the cheapest option, given that you can buy them singly, whereas filter papers must be purchased in lots of 100.

Pumps

Laboratory vacuum pump

A laboratory pump is ideal for suspended solids testing, and for chlorophyll testing if the sample site is nearby. They have a strong pumping action, come with a gauge and require very little time to filter a large volume of water. Laboratory pumps are very expensive, often costing several thousands of dollars, so are only recommended if your school or group already has one. A local laboratory may be happy to let you use theirs, as long as the turnaround time is fast enough for the tests being performed.

Hand held pump	Hand held pumps are often used out in the field, particularly those that have a gauge attached. They may operate from a range of energy sources: muscle, petrol, car or battery power. They have similar drawbacks to the laboratory pumps as they are fairly expensive, ranging from several hundred dollars to several thousands. Most of these pump styles require a vacuum vessel, so a Sartorius filter unit is usually recommended.
Water powered vacuum pump	Water (or venturi) pumps are an economical (\$50–\$150) method of creating a weak to moderate vacuum without expending large amounts of effort. These pumps work best off mains water, but will work perfectly adequately off rainwater tanks or other non-pressurised water sources. One of their main advantages is that they do not necessarily require a vacuum vessel; water being sucked into the pump does not harm it at all.
Large syringe	Large syringes (>100 mL) are a great way of filtering small volumes (up to two litres) of water. A 140-mL syringe can cost anywhere between \$15 and \$50 depending on the retailer and manufacturer. They are not common in laboratory supply houses, but the Tyco Healthcare group makes one called a Kendall Monoject Syringe, reference number 1100-514031, which is available in South Australia.
Pipette bulb	A pipette bulb is absolutely the cheapest pumping method, apart from simple gravity. A bulb will cost you between \$3 and \$10. They usually only generate a low level vacuum and it takes considerable pumping to maintain the pressure. Bulbs are therefore only recommended for small volumes. A pipette bulb requires a vacuum vessel, so this may make a large syringe look cheaper in the long term, unless your school or group already has a vacuum vessel of some sort.

Making a quadrat

Quadrats (Figure 23) are used for counting animals or plants in an area. Each quadrat has a known area, so that the total number of a species can be quantified by multiplying the results from several quadrat samples.

The instructions below tell you how to make a 500-mm square quadrat from PVC pipe, although they may be made from anything and be of any size or shape.

Materials

- 2-m of ≤ 15 -mm diameter white PVC pipe
- four elbows to fit the PVC pipe
- a hacksaw with an appropriate blade
- sandpaper
- drill with an approximately 3-mm bit (optional)
- 4.5-m cooking twine

Method

Cut the PVC pipe into four equal lengths approximately 500-mm long.

Fit the pipes into the elbows to form a square.

Measure the inside width of the square to ensure that measures $500\text{-mm} \pm 2\text{-mm}$. If the quadrat measures more than this use the sandpaper to shorten each of the pieces of pipe.

Remove any writing from the pipe by buffing it off with the sandpaper.

If wish to be able to subdivide the quadrat, drill a hole through the pipes every 100 mm.

You can then thread the twine through the holes to divide the quadrat into 100-mm squares as required.



Figure 23—Quadrat

Making a leaf trap

A leaf trap is a 500 mm × 500 mm net, mounted on four star droppers or hung from a tree. Traps may be made larger than this, but this is an easily managed size. The traps are used to collect leaves, seeds and flowers that fall from trees, so that the plant litter production may be monitored.

The porosity of the fabric used in the net depends entirely on the plant and parts of the plant you would like to measure. The smallest part of a mangrove that is likely to fall into the net is a flower, so the netting may be fairly coarse, eg tulle or 50–80% shade cloth. The smallest part of a *Melaleuca* tree that is likely to fall into a net is its seeds, which are only tenths of a millimetre across. To measure the amount of these falling, you will need a very fine cloth, eg mulching fabric or heavy calico.

Materials

- a 500-mm wooden frame or a PVC quadrat
- > 1.05 m of 1.15 m wide fabric
- sewing machine or needle and thread
- cable ties (optional)

Method

Cut four triangular pieces of fabric, approximately 520–550 mm along each edge.

Sew them into a pyramid shaped net, well sealed at the bottom.

Turn the net seam-side out.

Attach the top, or open end, of the net onto all four sides of the wooden frame or quadrat. Cable ties or sewing may be used for this step.

Making a plankton net

Plankton nets (Figure 24) are long cone-shaped nets, with the wide end held open using a circular frame and the narrow end finished with an opening large enough to attach a sampling bottle to it. The circular frame is often attached to a rope harness at three or four points, then attached to a long tow-rope to allow it to be towed behind boats or alongside jetties.



Figure 24—Plankton net

The material for making some grades of plankton net can be almost as expensive as buying the net ready-made. Marine plankton nets are a special nylon netting, which has a known mesh size of between 50 and 300 microns, depending on the organisms you wish to collect. If you can find a cheap, or reasonably economical source of fabric, making the net is a simple process.

Materials

- a 300–500 mm circle (depending on the width of the fabric) made of heavy-duty wire or bent metal
- > 1 m of netting fabric of the appropriate mesh size
- sewing machine and appropriate (nylon monofilament) sewing thread
- nylon rope for making harness and tow-rope
- polyethylene wide-mouthed sample bottle and worm-drive hose clamp

Method

Sew the mesh into a long cone shape using a double seam to enclose the edges of the material. The large opening is to have a diameter slightly larger than the metal circle, and the small opening should have a diameter of approximately 70 mm, or slightly larger than the neck of your sample bottle.

Hem the openings so that the net will not fray and sew the big opening onto the circular frame.



Figure 25—Sample bottle and attachment

The sample bottle (Figure 25) is attached to the net by a worm-drive hose clamp. If you are only doing qualitative plankton tows, then a simple polyethylene sample bottle will suffice.

If you wish to do quantitative tows, then you will need to ensure that the sample bottle has two little windows cut into it near its neck, with glued-in mesh covering them. This reduces back-pressure in the sample bottle, ensuring a more representative collection is made.

Making a water dipper

A water dipper (Figure 26) has two main uses — it enables you to obtain a sample from below the water surface, and it is a usefully sized container for measuring salinity with a hydrometer.

Making one of these is straightforward. By using a longer length of dowel you can increase both your reach and the depth it is possible to sample from.

Materials

- length of dowel
- small block of wood, at least 90 mm long
- several large headed nails
- hand saw
- a spade bit approximately the size of the dowel
- a plastic 250-mL measuring cylinder

Method

Cut the end of the wooden block off at an angle, generally 45°–60°.

Use the spade bit to drill a hole directly into the angled face, but not all the way through.

Insert the dowel into the hole and nail it into place from underneath the wooden block.

Cut two sides of the measuring cylinder's base away, so that there are two tags left on either side.

Nail the cylinder onto the wooden block using these tags.



Figure 26—Water dipper

Glossary

abundance—presence of an organism, recorded in numbers

acid—a pH of less than 7

alkaline—a pH of more than 7, historically called 'basic' by chemists

alkalinity—the buffer capacity or capacity of the water to neutralise acids; it is not a reference to the pH of the water, and does not mean that the water is alkaline

ambient—surrounding or natural level or concentration

anaerobic—living or occurring in absence of oxygen

anoxic—conditions in which oxygen is absent

anthropogenic—man-made or caused by man

apical shoot—new leafy growth coming from the apex, end, top or point of a branch or plant

area—the area of an estuary is the area below the mean high-tide mark, between the mouth of the estuary and its head

assemblage—collection of individuals, usually of different types

assessment—an evaluation of selected ecosystem attributes

autotrophic—able to produce its own food, for example, by photosynthesis

baseline study—first assessment of an ecosystem against which future changes are measured

beeper—small electronic device attached to a cable that produces a beeping sound when the end of the cable is immersed in water—regularly used for groundwater monitoring. Also called a pinger

benthic—the bottom layer of a water body, the surface of the sediment

biomass—amount of living material (plant or animal) in an area

bloom—the discoloration of water caused by a large population of floating or suspended algae

bottom—the bottom of an estuary or a meniscus is the lowest point of that object

brackish—water that is salty, but less so than seawater

capillary action—capillary action pulls water against gravity due to adhesion between the water and the surface

chlorophyll—green pigment in plants that enables them to use the energy of the sun for photosynthesis

community—group of organisms belonging to a number of different species that co-inhabit and interact in the same area

Coriolis forces—force due to the rotation of the earth that may cause a large fluid body to assume a circular motion when it flows (eg weather systems)

data quality plan—document describing the quality of data you plan to produce, and the methods you plan to use to ensure this quality

datasheet—paper form used to record field data in a set format

depth—the depth of an estuary is the depth of water covering an area at the time the measurement is taken—if tidal conditions are recorded, the depth can be corrected for tide at a later date

detrital cycle—the cycle of decomposition, where small organisms break down dead materials, returning these materials to the rest of the food chain

detritivores—organisms that break down dead materials

dinoflagellate—a phylum of plankton

diurnal variation—variation of a parameter eg dissolved oxygen, between night and day—*di* means two and *-urnal* means per day

diversity—the number of species of plants, animals, mammals, birds or reptiles that occur in an area

DO saturation—the amount of oxygen that water of a given salinity and temperature can hold

ecological niche—location and role for which a species is well suited within its community, including its habitat, what it eats, its activities, and its interaction with other living things

ecosystem—dynamic complex of plant, animal, fungal and micro organisms and associated nonliving environments interacting as an ecological unit

enterococci—round bacteria that occurs only in the guts of mammals

epiphyte—non-parasitic plants growing on the surface of other plants or other object, eg brown slime or staghorn ferns

epiphytic growth—a brown slimy growth of epiphytes, often found on seagrass

***Escherichia coli* (*E. coli*)**—faecal coliform that occurs only in faecal matter from mammals, including humans

eusaline—salinity similar to that of seawater

evaporation volume—the volume of water that evaporates from a given area

faecal coliform—rod-shaped bacteria that occur only in faeces or faecal matter

flowchart—a chart showing the flow of materials through a process

gauging data—water height and flow data, often obtained from a gauging weir in a river or a staff gauge in a pond or tidal area

georectification—the correction of an aerial or satellite photograph to reflect true geographic locations

groundtruthing—the process of making sure data collected remotely is correct on the ground

groundwater—underground water

groundwater table—the upper surface of the underground saturated zone

habitat type—the habitat type of a site define what grows or lives in that area—the habitat type of a site depends on flooding regimes, availability of shelter or water, climate and elevation

head—the head of an estuary is the landward limit of tidal variation

hydrology—the science of water movement and flow

hypersaline—more that twice the salinity of seawater

hypoxic—this is water that is not quite anoxic, but heading that way—generally less than 4 mg/L DO is considered hypoxic

infauna—fauna living below the surface

input volume—the input volume in an estuary is the volume of water entering it

inputs—substances that are put into or used in a process

intertidal—the area between the high- and low-tide marks

invertebrates—animals without an internal skeleton

landward margin—the landward edge of an intertidal habitat

long—when dealing with estuaries the length of an estuary is the distance from the head to the mouth, flowing the main water body

macro—large enough to see with the naked eye

macro algae—macro algae are multiple-celled algae that are large enough to see with the naked eye

macrophyte—a plant that has leaves, stems, roots and flowers—aquatic macrophytes may be submerged (seagrasses) or emergent (reeds)

mangal—forest containing mangrove species and associated plants or animals

mass balance—chart or table that shows all the inputs and outputs to a factory, ecosystem or estuary—theoretically the inputs should equal the outputs

meniscus—the curved surface of a liquid where it comes in contact with a solid object, particularly the walls of a tube—this is caused by capillary action pulling up the edges of the liquid and the middle of the liquid being pulled down by gravity

methodology—collection of methods used in an activity

metric—invented measure or index assessing a condition, eg environmental health, that is normally difficult to quantify

micron (μm)—thousandth of a millimetre.

monitoring—repeated observation or assessment of a system on a regular scale—this is often done to detect change

monitoring program—plan of what is going to be monitored, including where and how

mouth—the mouth of an estuary is where the salinity is indistinguishable from that of seawater, this is often where a sand-bar crosses part of the estuary or some other landmark that can change the hydrology of the area is present

National Pollutant Inventory (NPI)—the federal register of known polluting activities

neap tide—the tides where there is the smallest difference between low tide and high tide each lunar month

negative estuary—an estuary where the input of freshwater is exceeded by evaporation

net primary productivity—the total production of biomass from primary sources, eg water, nutrients and solar power by an individual ecosystem

net evaporation—the change in water level that occurs in a water-filled pan exposed to the elements

orthophoto—a georectified aerial photograph, often overlaid with property details

outputs—the products, by-products and waste from a system

pan evaporation—the same as net evaporation.

pannes—small low areas in a saltmarsh where the mud has no plants growing and water gathers—often called 'rotten spots'

paleoriver—an ancient and no longer flowing river often only detectable by remote sensing technology

parameter—measure used to describe some characteristics of a system or population

phytoplankton—microscopic organisms that use photosynthesis to obtain their energy from the sun and float around in freely waterways

piezometers—small groundwater wells used for monitoring the height of the water table

plopper—brass or metal object with a concave bottom, which when lowered into a piezometer makes a distinct sound when the water table is struck

positive estuary—an estuary where freshwater inputs exceed evaporation losses

productivity—accumulation of biomass over a period

propagules—seeds or other organs (eg spores) of propagation

quadrat—an object that has fixed unit of area, which is used for sampling—may be square, rectangular or circular

qualitative—descriptive, non numerical, assessment

quality plan—document describing the quality of data you plan to produce, and the methods you plan to use to ensure this quality

quantitative—numerical; based on counts, measurements or other values

raptor—bird of prey (eg hawk, eagle or kite)

riparian zone—the edge of a river or waterway, where the aquatic habitat meets the terrestrial habitat

riparian vegetation—vegetation that grows beside fresh or brackish streams or rivers, in the zone between low- and high-water mark

runoff coefficient—a runoff coefficient is multiplied by the rainfall to determine the amount of water that runs off a parcel of land—there are different coefficients for each type of land use

rural area—areas that do not contain large amounts of buildings or other development

sabkha—large saltpan and mudpan area edging gulfs—such as the Gulf St Vincent and the Persian Gulf

seral—seral communities are plant or animal communities that occur in a specific part of a disturbance succession or zonation pattern—the communities are not static, the area and edges change constantly due to environmental impacts or variation

sodic—soil that is high in sodium which causes the soil to lose its structure—commonly found in areas that contained saline soils, but have been leached of chloride by rain or freshwater inundation

spatial—pertaining to space and area

Specific Gravity (SG)—the weight or mass of a liquid in kg/L at 15.6°C

spring tide—either the highest high tide or lowest low tide for a lunar month

staff gauge—a fixed gauge used to measure changes in water height

Standing Water Level (SWL)—the height of the groundwater table before any pumping or testing is done

standpipe—the part of a piezometer extending above the ground

stratification—development or formation of distinct layers within a system

supratidal—area above the average high tide, which floods on spring high tides

survey—an organised inspection

tidal exchange—tidal exchange is the exchange of water when water flows in and out of an estuary under the influence of the tide

tidal restriction—a bridge, headland, embankment, ford or earthworks that restricts water movement and reduces the speed, time or height of tidal movement

tide—movement of a large water body due to the gravitational pull of the moon and the sun

transect—line or narrow belt used to survey the distributions of organisms across a given area

transfers—transferring the output of one system to the input of another, which then outputs it, eg water from your sink going down the drain and into a sewerage works, which is then discharged into a water body

turbidity maximum—the turbidity maximum in an estuary is the zone where much of the sediment from the river is settled out

urban area—urban areas are areas that have a high number of houses and other developments

variable—any measurable aspect of a sample that is not constant

vegetation associations—groups of plants that regularly occur together—examples of this would be salt bush and pig-face, or sea rocket and coastal spinifex

volume—the volume of an estuary is the amount of water it holds on an average tide