module 7

WATERWATCH AUSTRALIA NATIONAL TECHNICAL MANUAL

Estuarine Monitoring

Module 7 – Estuarine Monitoring

Waterwatch Australia National Technical Manual
by the Waterwatch Australia Steering Committee

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Preface

The *Waterwatch Australia National Technical Manual* was prepared by the Waterwatch Australia Steering Committee to provide guidance and technical support to the Waterwatch community monitoring network throughout Australia.

The guidelines and information reproduced in this Manual have been agreed by the national Waterwatch and Coastcare networks based on their knowledge and experience in coordinating community monitoring programs in Australia with advice from the scientific community.

The Manual has been published as a series of modules. Each module is a stand-alone document addressing an important aspect of community waterway monitoring. The following modules are available in the Manual:

1. Background
2. Getting Started: the team, monitoring plan and site
3. Biological Parameters
4. Physical and Chemical Parameters
5. Data ... Information ... Action!
6. Groundwater Monitoring
7. Estuarine Monitoring (this module)

Much of the material included in this module was drawn from State estuarine technical manuals particularly those published or in draft form from South Australia, Queensland and New South Wales. A workshop was held in Sydney on 27–28 July 2005 that reached agreement on the parameters to include in the module and where possible methods of measurement.

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Before you begin

This section contains a definition of an estuary and the Geoscience Australia classification of estuaries. It discusses an estuary monitoring plan, including the common catchment and estuary pressures and management issues and the parameters you can consider measuring to better understand estuary health and change. This section also discusses how to map your estuary, and the importance of historical information, including oral histories, in understanding your estuary.

Please read this section before proceeding and before you go into the field.

What is an estuary?

An estuary is a 'semi-enclosed coastal body of water where salt water from the open sea mixes with freshwater draining from the land'. A more complete list of terms important in understanding and monitoring estuaries is contained in the Glossary.

Estuary monitoring plan

Estuarine monitoring is a means of gathering information about the condition or 'health' of an estuary and to understand changes that may be occurring. It consists of observations or measurements of the characteristics of the water itself, of the shoreline and bed of the estuary, and also of the aquatic life and vegetation in and around the water. Monitoring can also be used to initiate action in the catchment to benefit the estuary, or to take action in the estuary itself.

How and what you monitor depends on the issues affecting your estuary and why you are gathering information about it. You need a reason to monitor. Sometimes, a single issue will trigger the interest of individuals and groups, or a number of issues in the catchment or estuary will provide the reasons for monitoring. One way to identify issues is to ask questions about uses, values and threats. Once you have agreed on the reason for monitoring it is possible to form a specific objective and to design a detailed monitoring plan.

It is important to prepare a monitoring plan before you begin. Module 2 provides help with preparing a monitoring plan. It builds the monitoring plan around eleven questions:

Q1 Why are you monitoring?

Q2 Who will use your data?

Q3 How will the data be used?

Q4 What will you monitor?

Q5 What data quality do you want?

Q6 What methods will you use?

Q7 Where will you monitor?

Q8 When and how often will you monitor?

Q9 Who will be involved and how?

Q10 How will the data be managed and reported?

Q11 How will you ensure your data are credible?

What you choose to monitor will depend on the management issues or pressures occurring in your estuary or catchment, often caused by human activities. These pressures change the state or condition of the environment and by carefully selecting the right indicator or parameter for measurement, the cause of environmental change may be determined and a management or policy action taken.

Table 7.1 provides a summary of common catchment and estuary pressures and management issues, the possible impact on your estuary and guidance on the parameters you may wish to measure to better understand estuary health and change.

Case Study - Black Bream Estuary

Black bream estuary is adjacent to a caravan park and is popular for swimming and fishing. Local residents are concerned that sewage from holiday-makers is having a negative impact because every summer algae wash up on the shore causing bad smells. They also want to know whether it is safe to swim in the estuary.

First the residents formed a group and mapped the estuary and found out some of its history. An old fisherman reckons fish numbers have declined since the estuary's heyday. Next, the group looked in the *Waterwatch Manual* and talked with the coordinator about which measures are most useful to track sewage. They discovered that another possible cause of algae might be reduced summer inflow of fresh water from the catchment.

The group decided that measuring for nitrogen, phosphorous and pH was well within its ability and decides to measure them on a regular basis. They also organised for bacteria measurements to be taken and to assist the environmental protection agency with its sampling. At the same time group members collected background information on the estuary and the catchment, such as the time, tide height, water depth and catchment inflows that is relevant to interpreting their results. They found that sewage levels were a little high but there was good tidal exchange of water with the ocean.

The Waterwatch group is also working with the local shire and caravan park to reduce the stress on the estuary. The group is going well, enjoying an occasional barbeque and making new social connections.

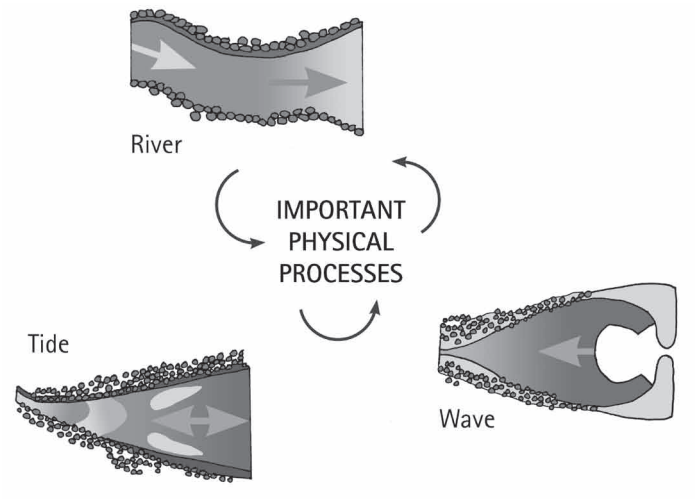
Table 7.1: Management issues and parameters

Management issue/pressure	Possible impact	Suggested parameters	
Coastal development (urban and agriculture)	Loss of habitat – mangroves, saltmarsh, seagrass Disturbance of acid–sulphate soils Altered hydrology	Algal blooms Invasive species pH Turbidity Salinity Dissolved oxygen Phosphorus	Nitrogen Chlorophyll-a Seagrass Macroalgae Mangroves Saltmarsh Crab burrows
Sewage	Eutrophication – which leads to loss of flora and fauna Increased algal growth Human health Shellfish health	Odours Algal blooms Fish kills and animal deaths Dissolved oxygen Phosphorus	Nitrogen Chlorophyll-a Crab burrows Bacteria
Sedimentation	Increased turbidity Loss of seagrass beds Loss of macroalgae Estuary mouth and channel changes	Flow rate Invasive species Turbidity Phosphorus Nitrogen	Seagrass Macroalgae Mangroves Saltmarsh Crab burrows
Agricultural and urban run-off	Eutrophication Sedimentation Pesticides Excessive rubbish	Algal blooms Fish kills and animal deaths Beach litter pH Turbidity Dissolved oxygen Phosphorus Nitrogen	Chlorophyll-a Seagrass Macroalgae Mangroves Saltmarsh Crab burrows Bacteria
Upstream dams	Sedimentation of estuary Greater saline influence further up estuary Barriers to fish migration	Flow rate Turbidity Salinity	Seagrass Macroalgae Mangroves
Entrance modification	Changes to internal tidal regime Removal of habitat Reduces fish stocks at local levels	Flow rate Fish kills and animal deaths Salinity Dissolved oxygen	Seagrass Macroalgae Saltmarsh
Over-fishing	Reduced fish stocks at local levels Changed rocky shore flora and fauna	Invasive species Rocky shores	

Estuary classification

Geoscience Australia has classified Australia's estuaries by condition and key ecological processes for the National Land and Water Resources Audit. Classifications for 974 of Australia's estuaries and coastal waterways are provided on the OZESTUARIES database at <www.agso.gov.au/ozestuaries>. You should see whether your estuary has been classified and what condition it is considered to be in on a national scale.

Figure 7.1: Geoscience Australia estuary classification system



Source: National Land and Water Resources Audit.

The Geoscience Australia classification is based on physical forces (wave, tide and river energies) driving the form and function of Australian estuaries and coastal waterways. Physical processes influence estuary shape and ecology and the classification is based on the relative dominance of these physical processes.

Table 7.2 provides a summary of the classes of estuaries and a summary of distinctive characteristics, susceptibility to impacts and management requirements.

Mapping your estuary

What is it and why does it matter?

Understanding the mixing processes in your estuary requires an understanding of the geography of the estuary. Salt water and fresh water mix differently depending on the shape of the 'container' they are in.

To demonstrate this, mixing patterns in a tall 250 ml measuring cylinder may be compared with mixing patterns in a wide shallow bowl. Simply pour 100 mL of thick oil into the two containers, then 100 mL of very thin oil on top. Gentle swirling of the shallow bowl will cause considerable mixing, while gentle swirling of the deep, narrow measuring cylinder will have little effect.

Suggested methods, equipment and reporting

Examination of maps of your estuary may help to explain some of the readings you obtain in the field. Mapping also allows you to measure shoreline changes, such as erosion or development of sandbanks that may change flow patterns within the estuary.

A wide variety of maps are available. Standard references for your estuary should include at least one large-scale topographic map. As your understanding of your estuary deepens, you may wish to add bathymetric maps (that map the underwater features), aerial photography, orthophoto maps (aerial photographs that have been georectified and have topographic details superimposed on them), and satellite imagery. Maps may be purchased on paper, or digitally on CD ROM.

Equipment

The equipment you will need for mapping your estuary includes:

- large-scale maps or aerial photos of your estuary – try to get a map that includes your estuary on one sheet (if you have a really large estuary, choose the scale that shows you the most detail, and paste as many maps together as you need)
- tide books
- measuring tools, such as callipers, rulers and compasses
- plastic overlays with 2 mm x 2 mm or 5 mm x 5 mm dot grids
- fine tipped permanent markers

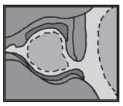
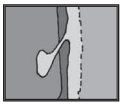
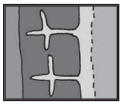
Procedure

Using the scale on your map as a guide, work out the **length** of your estuary. It may be tricky determining where the head of the estuary is – it is usually considered to be the point farthest upstream that still has a rise and fall in tide. This may not be marked on your map and you may need to make some field observations.

Take several measurements across the **width** of your estuary. What is the average width of your estuary? Does it vary much?

Now try to calculate the **area** of your estuary. Place a plastic dot grid over the map and trace around the estuary, using a fine tip permanent marker. Calculate the number of dots located within the boundaries of the traced area, converting the area to square centimetres (cm^2). For example on a 2 mm dot grid, 25 dots = 1 cm^2 and on a 5 mm dot grid, 4 dots = 1 cm^2. Dots located on the boundary line are only counted if over 50 per cent of the dot covers the subject area.

Table 7.2: Classification of Australian estuaries

Class	Subclass		Sediment trapping efficiency	Turbidity	Circulation	Sedimentation	Eutrophication
Wave-dominated		Wave-dominated estuary	High	Naturally low	Salt wedge/ partially mixed	High risk	High risk
		Strandplains	Low	Naturally low	Negative/ salt wedge/ partially mixed	Low risk	High risk
Tide-dominated		Tide-dominated estuary	Moderate	Naturally high	Well mixed	Some risk	Low risk
		Tidal flats/ creeks	Low	Naturally high	Well mixed	Low risk	Low risk
River-dominated		Tide-dominated delta	Low	Naturally high	Well mixed	Low risk	Low risk
		Wave-dominated delta	Low	Naturally low	Salt wedge/ partially mixed	Low risk	Moderate risk

shallow water terrestrial land intertidal habitat deep water

Source: Heap et al. 2001.

Depth is an important measure to estimate, and may be available from bathymetric maps, or from your harbour master. Is your estuary deep with steep sides, or wide and shallow with deeper channels?

Once you know the average depth, you can calculate the volume of water in your estuary by multiplying the square metres of area by the metres of depth (for a rough estimate). Each resulting cubic metre is a kilolitre of water. To get a more accurate measure, divide your estuary up into different zones, and calculate the area and volume for each zone.

If your group becomes very interested in mapping, you may be prepared to spend the time to learn how to use a computerised geographic information system (GIS). These packages simplify the georectification and scaling of aerial photographs, and allow you to overlay your field measurements onto topographic and other base maps. They provide a sophisticated analysis.

Interpreting your results

The information you gather in this activity will be useful in preparing a monitoring plan and undertaking the other activities contained in this module.

Each day the tide may come in and out of your estuary. When the moon is full, the spring tides bring in large amounts of water, while the neap tides of half-moon have the smallest tidal exchange. By looking at the difference in height between high and low tide at neaps and springs, and then looking at your topographic and bathymetric maps, you may be able to gain a rough idea of the volume of water that comes in and goes out with the tide. This water, the tidal exchange, has a major influence over how long it takes for pollutants to disperse in your estuary. An estuary with a small tidal exchange holds the pollutants for considerably longer than a similar estuary with a large exchange.

You may gain quite a lot of information relating to shoreline deposits, erosion areas and sand bars from your maps. Besides using that information in your fieldwork, note any changes you observe. Large changes in the shoreline or in deposition areas such as sandbanks may reflect land management practices in the catchment or changing coastal tide and wave patterns.

Historical change

What is it and why does it matter?

Estuaries are often naturally dynamic areas that are constantly undergoing change because of the interaction between rivers, tides and sand and sediment movement. Many estuarine areas have been changing over the last 100 years, with large areas of land cleared for agricultural, industrial and urban development, thus reducing key habitat for estuarine fauna, and changed river flows due to river regulation. Alternatively, in some regions mangrove habitat has expanded due to changes in the environment.

Suggested methods, equipment and reporting

It is possible to monitor historical change in vegetation community types, by comparing the community extent in the past to that of the present. You can do this by using aerial photos, where the boundaries and expanse of vegetation types are clearly defined, thus an increase or loss in community extent can be seen and measured. The amount of change that has occurred over time in vegetation communities can be assessed by placing a plastic grid overlay over the map and calculating the area covered in the past and present.

Equipment

The equipment you will need for recording the history of your estuary includes:

- aerial photos of the area, both historical and recent
- plastic overlays with 2 mm x 2 mm or 5 mm x 5 mm dot grids
- stereoscope (optional)
- fine tipped permanent markers

Procedure

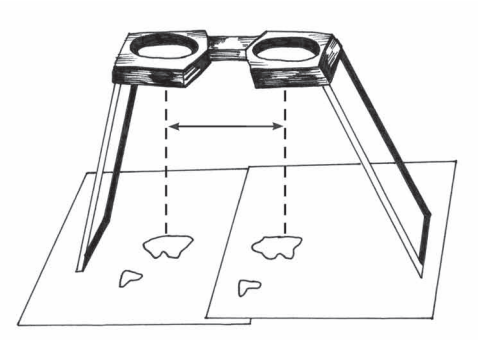
To aid in later interpretation, familiarise yourself with the study area, identify habitat types, and note the shape, size, patterns, and tone of the features.

Lay your photographs and maps out in front of you. Locate landmarks you have physically visited. Can you find any? Do they look different to how you thought they would look?

Secure a plastic dot grid on top of the aerial photo using a paper clip. Trace around the area of interest, using a fine tip permanent marker. Ensure the outside of the line being drawn matches the outside boundary of the area on the photo.

If you have a stereoscope, place two aerial photos under the stereoscope. Both photos should show the same area with approximately 30 per cent overlap. Looking through the stereoscope, move the photos until both images overlap and a 3D image is created (stereoscope is not essential but aids interpretation by creating a 3D image).

Figure 7.2: Using a stereoscope



Mark the landmarks with a permanent marker. Trace around any holes and/or gaps within the subject area and mark clearly to avoid confusion when calculating the area. Do the landmarks on the transparency line up with those marked on the map? How far out are they? Aerial photos are distorted due to the curve of the earth and the angle the plane was at when the photo was taken. How distorted are your photos?

Once you have finished tracing all the objects on the photographs, reduce or enlarge them on a photocopier until they are the same scale as your map. Once you are happy with them, photocopy them onto film that has graph paper markings on it, or overlay two bits of film and stick them together.

Determine the area of each square on the film. Count the number of squares that fall completely within each of the traced objects and estimate the number of partial squares. Calculate the total area (on the film) of each traced object. Multiply this by the appropriate scale to get the area of each object on the land.

Interpreting your results

Compare the results from historical photos to the recent photos to determine any change in habitat area. Data is interpreted as a loss or expansion of habitat area or as an amount of change over time. This exercise allows you to assess the impacts of human activities on remnant habitat and change in ground surface type, for example, soft surface such as grass or trees to hard surface such as roads, paving or concrete. Has any object or land cover type changed significantly in its total area? Are the individual areas of vegetation smaller or larger?

Has any object or land cover type changed significantly in location, for example has it moved inland or seaward? By how much has it moved?

It is beneficial to test the results obtained from this mapping exercise, to ensure their accuracy. The way to test your results is to visit the study site and compare on-ground habitat and features with the aerial photos you used.

Oral histories

What is it and why does it matter?

Storytelling is the time-honoured practice where culture and tradition are passed on from person to person, generation to generation, within communities and beyond, to enrich and inform future generations. Oral histories are authentic historical documents that form part of our cultural heritage. Personal knowledge and life experience are integral to the collective memory of our families, communities and organisations.

Suggested methods, equipment and reporting

Older residents of the area around your estuary have knowledge and personal reminiscences that are an important historical resource. They may include:

- information on the dates of the opening and closing of estuary mouths, forming of breakwaters or sandbars
- changes to vegetation, loss of habitat or expansion of mangroves
- changes to the depth of the estuary or the pattern of tidal flows
- interesting tales of fishing, swimming and living in the estuary
- information on previous generations, who lived and worked in the estuary
- historical information on structures, wrecks and places.

The Oral History Association of Australia was formed in 1978 and there are branches in every state and territory. The objectives of the Oral History Association are to promote the practice and methods of oral history, to educate in the use of oral history methods, to encourage discussion in all aspects of oral history and to foster the preservation of oral history records. You can visit their web site at <http://www.ohaa.net.au>.

Equipment

The equipment you may need for recording the oral history of your estuary includes:

- note pad and pen or pencil
- tape recorder (if your subject agrees to be recorded)
- release form (see below)

Procedure

Firstly, you need to work out what your topic is about.

In order to gather primary information you will need to interview someone. This is part of the process of recording oral history. This means finding the right person to get the best information possible. You may already have someone in mind who could tell you lots of things about the area.

Contact your prospective interviewee and ask permission to do your interview. Reach agreement as to how the information they are going to provide may be used or accessed. Legal releases are an essential part of oral history. They confirm to the interviewer and the interviewee that the information will be used in an agreed way. A simple release might look like this:

I, [*interviewee's name*] give my permission to [*name of interviewer or project*] to use this interview, or part of it, for research, publication and/or broadcasting (delete one of these if required) and for copies to be lodged in [*name of library or archive*] for the use of other bona fide researchers.

Signed [*interviewee*]

Date _____

Interviewer [*name and signature*]

If the person you wish to interview agrees to participate, set a date and time for the interview that is suitable for both of you.

Before the interview, you will have to make a list of relevant questions to ask the interviewee. You will need between 10 and 20 questions. Your questions need to be in order (that is, they must be organised into sub-topics) so your interview flows in a logical sequence.

Some tips:

- Make sure you introduce yourself and explain the reason for the interview.
- Be polite and respectful.
- Be prepared with serious questions. You don't want to waste people's time.
- Ask permission to tape the interview. If they say no, be prepared to write out (transcribe) the responses.
- Ask your interviewee if they have any photographs or literature they can show you, if this is relevant. This often leads to some good stories and descriptions.
- Give a copy of your work to the interviewee and make sure it reaches them by the promised date. This allows them to check that what you have recorded and transcribed is as they remember it.
- As a researcher you must be flexible and willing to take direction from the interviewee as to how they want their information used or accessed.
- Do not use any material in a way that goes against the wishes of your interviewee.
- Do not ask questions that could result in a yes/no answer – you need detailed information and description.
- Practice asking politely for more details.
- If they run out of things to say, ask more questions.

Interpreting your results

Write a report from the oral histories collected that describe the most important changes in your estuary. Compare this information with changes you have mapped from old aerial photographs, and use it to help you prepare your monitoring plan. Be aware that oral histories are people's perspectives and that information can become exaggerated or distorted over time. Lodge a record of your oral history with a local library, or contact the Oral History Association to find out where and how to best store your oral history.

Site observations and events

This section discusses use of the site record sheet (at Appendix 4) – how to complete details on the weather, the time and height of the tide, water depth, the flow rate of things like stormwater, riverine or tidal flows in an estuary, and any odours at the site. It also covers event-based monitoring, algal blooms, fish kills and animal deaths, invasive species and beach litter.

Site record sheet

The site record sheet (see Appendix 4) provides a record of information about the conditions you encountered on the day of monitoring. It includes:

- weather
- time and tide height
- water depth
- flow rate
- odours.

Measurements should, where possible, be taken at the same time in the tide cycle each time you monitor, that is, on the ebbing or flooding tide at the same time after the change in tide.

Recording weather details

The weather affects many other parameters being measured in a monitoring program, so should be included to aid interpretation of collected data.

Hot weather may reduce the ability of estuarine waters to hold dissolved oxygen, windy conditions may stir up sediments, and heavy rain may provide plumes of turbid runoff. Additionally, it may be difficult to obtain accurate Secchi disk measurements in strong glare or when the surface of the water is choppy.

In general, it is best to monitor in calm conditions. Avoid monitoring in strong storms, as there are safety issues to consider.

Equipment

The equipment you will need for recording weather conditions in your estuary includes:

- site record form
- Beaufort scale
- thermometer
- weather records for the previous few days from the Bureau of Meteorology web site at <www.bom.gov.au>.

Procedure

On your site record form, record the weather at the time you are visiting your sampling site.

You may record whether it is sunny or cloudy, and estimate how much of the sky is covered by cloud. If it is raining, record this, and also record whether it has rained earlier that day.

You may use the same thermometer you use for measuring water temperature to measure air temperature. Place the thermometer in a shaded dry place and allow it to equilibrate with the surrounding air temperature for five minutes, and then read the temperature. Wet thermometers or those lying in the sun will give wrong readings.

Estimate the speed of wind that may be blowing using the Beaufort wind speed scale (see Table 7.3).

If you can obtain the weather records for the previous few days from the Bureau of Meteorology web site, attach these behind your site record sheet. The weather leading up to your sampling event may have a bearing on what you discover on site. For example, a series of hot, humid nights can result in very low oxygen levels just before sunrise and can sometimes cause fish deaths.

Recording time and tide height

The state of the tide affects many other parameters being measured in a monitoring program, so should be included to aid interpretation of the collected data.

Equipment

The equipment you will need for recording time and tide height in your estuary includes:

- site record form
- timepiece
- tide timetable.

Procedure

On your site record form, record the time you are visiting your sampling site. Look up the tide timetable for the nearest tide station to your monitoring site and decide what stage the tide is at. Is it:

- high tide
- low tide
- ebb (falling) tide
- flood (rising) tide?

Also record the time and height of the tide that occurred immediately before your visit, and the time and height of the next tide that will occur.

Recording water depth

The depth of water may affect other parameters being measured, such as temperature and dissolved oxygen.

Equipment

The equipment you will need for recording water depth in your estuary includes:

- Secchi disk or depth staff
- site record form.

Procedure

Gently lower the Secchi disk into the water until it just touches the bottom of the waterway. Read the water depth from the cord.

Table 7.3: Beaufort Wind Scale

Beaufort scale number	Descriptive term	Units in km/h	Units in knots	Description on land	Description at sea
0	Calm	0	0	Smoke rises vertically	Sea like a mirror
1–3	Light winds	19 km/h or less	10 knots or less	Wind felt on face; leaves rustle; ordinary vanes moved by wind	Small wavelets, ripples formed but do not break; a glassy appearance maintained
4	Moderate winds	20–29 km/h	11–16 knots	Raises dust and loose paper; small branches are moved	Small waves – becoming longer; fairly frequent white horses
5	Fresh winds	30–39 km/h	17–21 knots	Small trees in leaf begin to sway; crested wavelets form on inland waters	Moderate waves, taking a more pronounced long form; many white horses are formed – a chance of some spray
6	Strong winds	40–50 km/h	22–27 knots	Large branches in motion; whistling heard in telephone wires; umbrellas used with difficulty	Large waves begin to form; the white foam crests are more extensive with probably some spray
7	Near gale	51–62 km/h	28–33 knots	Whole trees in motion; inconvenience felt when walking against wind	Sea heaps up and white foam from breaking waves begins to be blown in streaks along direction of wind
8	Gale	63–75 km/h	34–40 knots	Twigs break off trees; progress generally impeded	Moderately high waves of greater length; edges of crests begin to break into spindrift; foam is blown in well-marked streaks along the direction of the wind
9	Strong gale	76–87 km/h	41–47 knots	Slight structural damage occurs – roofing dislodged; larger branches break off	High waves; dense streaks of foam; crests of waves begin to topple, tumble and roll over; spray may affect visibility
10	Storm	88–102 km/h	48–55 knots	Seldom experienced inland; trees uprooted; considerable structural damage	Very high waves with long overhanging crests; the resulting foam in great patches is blown in dense white streaks; the surface of the sea takes on a white appearance; the tumbling of the sea becomes heavy with visibility affected
11	Violent storm	103–117 km/h	56–63 knots	Very rarely experienced – widespread damage	Exceptionally high waves; small and medium sized ships occasionally lost from view behind waves; the sea is completely covered with long white patches of foam; the edges of wave crests are blown into froth
12+	Hurricane	118 km/h or more	64 knots or more		The air is filled with foam and spray. Sea completely white with driving spray; visibility very seriously affected

In shallow water, a measuring staff may be used instead of a Secchi disk. To prevent the staff digging into any soft sediment, the staff should have a footplate fitted.

As measuring the water depth may disturb the sediment, this measurement should occur after all other measurements have been taken.

The marked cord on a Secchi disk may stretch or shrink over time, so the markings on the cord should be checked against a metal rule before using the disk in the field.

Measuring flow rate

The speed at which water moves provides an indicator of the volume of stormwater, riverine or tidal flows in an estuary. Flows in estuaries are complex; however, recording flow rate can help you determine whether erosion or accretion of the shoreline is likely to occur, whether an area is likely to host certain types of aquatic vegetation, and (if the flow rate is taken in a stormwater drain, effluent outfall or creek that is joining the estuary) the load of pollutants entering the estuary.

Flow rate may impact on turbidity, dissolved oxygen concentrations and stratification. When examining fringing wetlands it is important to record flow rate as part of determining whether tidal restrictions are occurring in these vulnerable areas.

The rate of flow may be multiplied by the cross sectional area of the waterway being measured, to determine the volume of water passing a given point over a period of time. Details for calculating this information are contained in Module 4 of this manual – Physical and Chemical Parameters.

Equipment

The equipment you will need for measuring flow rate in your estuary includes:

- activity record form
- several oranges
- net on a pole
- 50-metre measuring tape
- stopwatch.

Procedure

Use your tape to mark the distance over which you will measure the flow rate. A person should stand at the beginning and at the end of the distance to be measured. The person at the beginning should have a stopwatch; the person at the end should have a net on a pole, for catching the oranges.

Place an orange in the water upstream of the first person. As the orange passes the first person, they should start timing. As the orange passes the second person they should call out loudly to the first person, then net the orange out of the water. The first person should stop the watch as soon as they hear the second person call out. Repeat this at least three times – try to use the same orange. However, as the orange is biodegradable, losing the odd one (if the flow is fast, for example) is not significant.

Take an average of the three readings, and use this as the time taken in the following calculation:

> **Flow rate (m/sec) = distance travelled ÷ time taken × 0.9 (correction factor because velocity varies between the middle and edge of a channel)**

The longer the distance over which you time your orange, the more likely you are to obtain an accurate result, so use a minimum distance of 10 metres, but preferably use a 50-metre distance. Choose an area where there are no structures or rocky outcrops jutting into the flow, as they will modify the flow around them.

Recording odours

If your site smells, record this on your site record, along with a description of the smell. Does it smell like fish, sewage, new-cut grass, rotten eggs, over-ripe fruit, fuel? Algal blooms, some effluents and anoxic conditions all produce various odours.

As well as smell, take a note of anything else unusual – are new pipes discharging near the site; is there an oil slick on the water? Oil slicks from pollution are usually brightly coloured, but sometimes in summer, the faintest slick of rainbow colours on the water surface may be from the oils in mangrove leaves.

The presence of dead fish and crabs is an indicator that should be recorded, as well as the presence of scum or foam on the water, rust-coloured ooze from the soil next to the water, the presence of lots of floating rubbish, or unusually turbid (muddy) water.

Event-based monitoring

What is it and why does it matter?

The purpose of event-based monitoring is to capture the impacts of significant catchment run-off events on estuarine or coastal receiving waters. It involves fairly frequent monitoring in the days, and probably up to two weeks, after a rain event.

Event-based monitoring is a way of capturing changes in turbidity, pH, dissolved oxygen during, and chlorophyll-a (Chl-a) after, heavy rainfall events.

Increased **turbidity** is an immediate impact of run-off. In undisturbed catchments, the increases may be relatively small, but in highly disturbed situations they may be greater and longer lasting.

The fine particles entering estuaries during storm events can have immediate impacts on light levels in the water. They can also have long-term effects on light penetration as fine particles, once settled, may be intermittently re-suspended by wind or tide.

Monitoring **pH** levels during and/or after rain events may help determine the extent and source of acid and/or turbid water. The first flush of floodwaters, particularly after long dry spells, can carry acid waters from disturbed acid sulphate soils into an estuary. This can have serious impacts on local biota – if acid sulphate soils are extensive, enough acid can be generated to decrease the pH from 7–8 to as low as 1–2. The fact that these levels may persist for only a few days indicates the need for event-based monitoring.

The extent of **dissolved oxygen** depletion and its effect on fish is normally greatest after a heavy rain event. Organic matter sourced from the land and carried by the floodwaters starts to decompose naturally. The resulting bacterial activity consumes oxygen from the water. If the flooding is severe enough, organic matter lying in anoxic or oxygen-depleted water on the bottom of the estuary can be re-suspended in the water column. This organic matter becomes exposed to oxygenated water and also begins consuming oxygen.

Monitoring of **Chl–a** after a flood event, in addition to regular monitoring, can indicate the nutrient input from the event. Decomposing organic matter also releases nutrients. Together with the nutrients washed into estuaries after rain events, this can lead to short-term increases in Chl-a levels for up to 4 to 7 days after a significant rain event.

Event-based monitoring is demanding as it requires a response at short notice; it will be suitable for only a few groups. However, data collected is extremely valuable for identifying the extent of changes taking place during a rain event and shortly afterwards.

Data on the severity of acid run-off and oxygen depletion can be very useful for agencies and scientists. Due to the unpredictable nature and short notice of rain events, many government and scientific agencies are often unable to respond quickly enough to gather useful data. Local groups can often collect vital data in these events.

Estuary mouth opening/closing

Some estuaries build up sand bars across their entrance and periodically open and close to the sea. When such an estuary mouth is open, water exchanges freely between the estuary and the sea with the tidal cycle. Many estuaries along the coastline of temperate Australia, particularly coastal lagoons, naturally close at times of small freshwater input when sandbars form across their mouth. Closure of estuaries causes ponding of water behind the sandbar and reduced or no flushing of the system occurs.

Closure becomes particularly important when nutrient inputs are increased above natural levels. The lack of exchange with the sea means nutrient levels continue to increase within the closed system, often resulting in eutrophication.

Closure of an estuary mouth also affects the movement of animals to and from the sea. This is particularly important for fish species that migrate into and out of estuaries as part of their life cycle.

Visual and/or photographic reports from visits to the estuary mouth at similar tidal states (for example, lowest water spring tides) will show whether the estuary is opened or closed to the sea. Tide gauges in estuaries can also be used to monitor mouth opening and closing as a loss of tidal signal indicates that the mouth is closed.

Equipment

The equipment you will need for measuring estuary opening/closing includes:

- camera
- GPS (if available)

Procedure

Record details of the estuary opening/closing including:

- location, using GPS or a topographic map or geographical features
- date and time
- weather and wind details
- extent and severity
- sketch map
- photographs.

Interpreting your results

Estuary mouths can open or close suddenly as a result of catchment rainfall and storm events. Landholders or fishermen may also open estuary mouths to reduce flooding or for other benefits. It is important to build up a long-term record of estuary mouth opening and closing, as there are likely to be correlations with estuary health measures you are concerned about. If you observe a sudden change in your estuary mouth notify the relevant management agency (such as local government or the parks service).

Suggested methods, equipment and reporting

With careful **site selection**, event-based monitoring programs can identify the extent, severity and possible source of oxygen depletion, sediments, nutrients and/or acid run-off from a rain event.

One approach could be to select:

- a site at, or just downstream of, the pollution source or area of concern
- a control site upstream of the pollution source or area of concern
- a site well downstream of the pollution source, where the pollution can mix with the water body.

If possible, try to use some sites that are used during regular monthly monitoring programs so event-based data can be compared with monthly monitoring data.

Equipment

In addition to the standard equipment required for each test you will need:

- an extension pole with sample bottle
- a boat (optional) – the spatial coverage of the event-based monitoring program can be greatly improved if you have access to a boat; although there can be safety concerns (see safety).

Safety

- During floods, never stand in fast-flowing water or water above knee height.
- Always use an extension pole when collecting samples.
- Select sites that can be safely accessed during rain and flood events – bridges and other structures can be ideal.
- Do not use a boat in a flooded waterway above an estuary – this can be extremely dangerous.
- Watch out for large woody debris (such as snags, logs and branches) in the water.

Procedure

When monitoring, always try to sample the first flush. This is critical for monitoring pH as the first flush will normally carry the greatest quantities of acid. If possible, it is also useful to collect samples at regular intervals (the more frequent the better – every hour is ideal) to identify the rate at which a parameter is changing.

Dissolved oxygen can be monitored at each site during, and every 24 hours after, a rain event to capture the rate of oxygen decline. Chl-a samples can be collected about three days after an event. Samples should be collected every 24 hours for 7 days.

Using an extension pole and sample bottle, collect a sample of water from 20 cm below the surface, as close as possible (keeping safety in mind) to the middle of the stream, estuary or current.

Conduct the water quality tests following the relevant procedures. Record the time of day and the time lag between the sampling time and the start of the rain event. Also record any unusual occurrences such as fish deaths.

Interpreting your results

Data from event-based monitoring can be compared with data collected during normal monthly sampling. This will identify the extent of change between event and non-event conditions.

Monitoring of pH can help to identify the source of acid. If pH values at a drainage or creek outlet are significantly lower than those at reference sites upstream and downstream of the main water body, there may be acid sulphate soil disturbance upstream of the outlet.

Dissolved oxygen data can be used to determine the severity of oxygen depletion. If one site is monitored at regular intervals, the rate of oxygen depletion can also be determined. However, the source of the organic matter causing the depletion may be difficult to identify, as there are many potential sources in a catchment.

Chl-a levels naturally increase after floods, but the occurrence of unusually large post-flood blooms could indicate excessive nutrient load.

Algal blooms

What is it and why does it matter?

While algal blooms occur naturally in many estuaries in response to environmental conditions and changes, they can also be the result of increases in nutrient levels (eutrophication) or increased light availability. When a bloom occurs, the algae form into a dense mat, appearing to float on the water surface.

The detrimental effects of this include production of toxins from blue-green algae, unpleasant odours, and a reduction in light penetration and photosynthesis rates, resulting in a decline in dissolved oxygen levels. Benthic blooms of algae also smother underlying vegetation, causing reductions in dissolved oxygen in shallow waters. This affects both aquatic flora and fauna communities.

Monitoring algal blooms occurrences may provide information on the cause of fish mortality and other impacts, indicating aspects in need of future monitoring and management actions.

Detecting algal blooms, and identifying the involved species, is important due to the potential impacts they have on human and animal health.

Suggested methods, equipment and reporting

Algal blooms can be detected initially by visual assessment and in some cases the cause of some algal blooms is not well understood. It is important to report their type, size, location and frequency and to notify the relevant authority to assess the severity of the bloom and whether the bloom is toxic.

Equipment

The equipment you will need for measuring algal blooms includes:

- camera
- GPS (if available)
- sealable bottle
- esky with ice
- rubber gloves.

Procedure

As algal blooms are random occurrences site selection is dependent on the location of the bloom. During normal monitoring activities, details of algal blooms and other unusual occurrences can be recorded and management agencies (such as local council or environmental protection agency) notified immediately.

Record details of the algal bloom or unusual event, including:

- location, using GPS or a topographic map or geographical features (for example, 'under bridge')
- date and time
- algal species (if known)
- extent and severity
- weather and wind details.

Sketch a map of the affected area indicating any landmarks and industry or agricultural uses. Take photos of the bloom as it appears in the water, and in a neutral or light coloured (but not white) bucket, away from sunlight.

To avoid contamination risks, all sample containers should be thoroughly cleaned with hot water and detergent, and rinsed several times in the water to be collected, before a sample is taken.

Wearing protective gloves, fill the sealable bottle with a sample of the water containing the algal bloom. Be careful to avoid direct contact with the bloom as it may cause skin irritations.

Dilute the sample with water adjacent to the bloom to allow light penetration, as dense concentrations of algae in the bottle will speed up oxygen depletion, and cause mortality of the algal species. Identification becomes more difficult once the algal samples have died.

Store the sample bottle on ice, away from light. Do not freeze samples as, upon expansion, the cell walls of the algae samples may burst, preventing accurate identification. If possible take several replicate samples (three recommended) for analysis. You need to take the samples to a laboratory for identification.

Interpreting your results

Monitoring algal blooms and unusual occurrences may indicate changes in environmental conditions. By reporting these events to management agencies, understanding of the cause of the events may be detected and managed, decreasing the chance of repeat events.

Fish kills and animal deaths

What is it and why does it matter?

Fish kills and animal deaths may occur naturally or be caused by human impacts.

The frequency and magnitude of fish kills is an indicator of biological condition and are generally believed to reflect the integrity of an estuarine, coastal or marine system.

Fish kills are unexpected and generally short-lived events conspicuous for the death of large numbers of animals. Kills are also aesthetically unpleasant because they litter coastal waters with rotten smelly carcasses.

Causes include anoxic and hypoxic events, infectious diseases, toxic algae, and uncommon weather patterns. Fish kills can result in:

- depletion of valuable stocks
- increased susceptibility to overfishing
- disruption of food chain dynamics and the interdependencies between species
- promotion of colonisation by noxious species and elimination of species essential to the healthy functioning of communities
- flow-on effects (such as illness and/or death in other species) if birds and other predators consume contaminated fish.

Disease-causing bacteria and pathogens are naturally present in estuarine, coastal and marine systems. Healthy animals generally show no ill effects from their presence unless there is a change in a predisposing environmental factor such as overcrowding, nutrition or water quality.

Poor environmental conditions will stress animals, resulting in a decline in the ability of their immune systems to protect them from disease. For example, exposure of fish to acidic water and toxic heavy metals associated with disturbed acid

sulphate soils damages their skin and gills, increasing their susceptibility to fungal infections such as red spot disease (epizootic ulcerative syndrome).

The presence of litter in estuarine, coastal and marine systems can harm animals that eat, become entangled in, or are suffocated by, the litter. Toxic substances can also leach from litter which then bioaccumulate up the food chain. Species of endangered or threatened marine mammals, turtles and seabirds are particularly at risk from litter.

Shark nets and drum lines offer some protection to swimmers by 'fishing' for potentially dangerous sharks and reducing their numbers around protected beaches. Unfortunately, they also indiscriminately capture other marine life, including harmless sharks, rays, dolphins, dugong, whales and turtles, some of which are endangered or threatened.

Boat strike is another human-related cause of death for marine mammals and reptiles. These animals are air breathers and must surface regularly, therefore, putting them at risk from human boating activities.

Suggested methods, equipment and reporting

Fish kills and animal deaths are unexpected occurrences and in some cases the cause of the deaths is not well understood. It is important to record as much information at the site of the kill and also in the surrounding area. As with algal blooms it is best to record this information and notify the relevant authority for further action.

Equipment

The equipment you will need for monitoring fish kills and animal deaths includes:

- a camera
- GPS (if available)
- sealable bags, plastic bottles, jars
- esky with ice
- rubber gloves.

Procedure

Notify a management agency (such as your local council, fisheries, parks service, environmental protection agency or environmental department) as soon as you find fish kills or animal deaths.

You need to photograph dead animals and the area affected and record details relating to the fish kill or animal death including:

- location and estimate of the size of the affected area
- date and time of discovery
- number, size and identification of affected species, including other animals such as crabs
- presence of other sick or dying fish or animals
- presence of skin lesions, wounds or injury
- presence of entanglement or litter
- presence of unaffected fish or animals
- the state of decay of deceased fish or animals
- the presence of any unusual materials, such as oil slicks, discoloured water, rubbish
- industries or agricultural uses in the fish kill vicinity.

You can collect samples of the dead fish, animal, water, sediment and unusual materials to send away for analysis. When collecting samples:

- wear protective gloves, take samples of the dead fish, animal, invertebrates, sediment and water
- store dead fish in plastic bags
- store sediment samples in glass jars
- store water samples in plastic bottles
- keep samples on ice or in a deep freeze if the relevant agency cannot collect the samples for more than 24 hours.

Interpreting your results

You should report the number of kills, species involved, as well as the number of animals killed. Once sufficient information on animal kills is available for a location, it will be possible to produce tables or graphs showing trends and their statistical significance. These trends can then be reported as an estimate of change from previous baseline data.

Invasive species

What is it and why does it matter?

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 a population or the diversity of native species.

Ships and shipping practices are largely responsible for introducing 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 was identified. With rapid, high-density shoreline development, the number of houses with marine aquariums near major waterways has increased exponentially. These aquariums usually contain introduced plants (such as Japanese Seaweed) and animals (such as the Northern Pacific Seastar). Much aquarium life has been bred to be nutrient, turbidity and pH tolerant, so it can survive in the less than optimal conditions present in many home aquariums. If this aquarium life is released into our

waterways, it can spread rapidly and cause extensive damage to already stressed ecosystems.

Once marine pests have established themselves in their new environment it is very hard to eradicate them. Early detection and rapid action are the only real methods for controlling these introduced species.

Japanese Seaweed

Suggested methods, equipment and reporting

A number of authorities in Australia deal with invasive marine species and may be able to provide information to help you identify any you may find. The Australian Quarantine Inspection Service (AQIS) has primary responsibility for dealing with new introductions.

The Consultative Committee on Introduced Marine Pest Emergencies is composed of state and Commonwealth representatives and is set up to deal with marine pest incursions. Contacts exist in each state and further information can be obtained from the Marine Pest Mapping and Information System web site at <http://data.brs.gov.au/mapserv/marinepest/index.phtml>.

The Centre for Research on Introduced Marine Pests (CRIMP) is a branch of CSIRO that specialises in controlling marine pests. CRIMP has published Dianne Fulani's *A Guide to the Introduced Marine Species in Australian Waters* that includes descriptions, photos and diagrams of more than 70 species of marine organisms.

CRIMP also produces a series of marine pest fact sheets, which may be downloaded from <http://crimp.marine.csiro.au//Marine_pest_infosheets.html>.

Equipment

The equipment you will need for detecting and recording invasive marine species includes:

- site record form
- camera
- drawing pad and pencil
- topographic map and/ or GPS
- fact sheets and field guide books.

Procedure

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 tasks.

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 listed species it may be a native with which you are unfamiliar, or an unlisted exotic. Take your photograph or drawing to an expert to get the organism identified.

If you are certain, or nearly certain, that your subject is a potential marine pest you need to:

- 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 does it cover?
- record the location at which you found the specimen(s) – you could use a topographic map or GPS
- record the date and time of the observation, along with contact details of the person or people who made the observation
- leave the organism undisturbed – some creatures, such as brittle stars, are very fragile and may reproduce from 'broken bits'. This is especially a problem with algae.

Interpreting your results

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

Beach litter

What is it and why does it matter?

The amount of litter accumulating on beaches and within estuarine environments is increasing throughout Australia. Litter can easily be collected and assessed by community groups; its removal enhances the amenity of estuarine areas.

Some effects discarded litter can have on the environment are:

- suffocation and toxic reactions in marine mammals, fish, reptiles and birds caused by ingesting various types of litter
- drowning and tissue damage in fauna caused by entanglement in discarded fishing nets, lines and ropes
- degradation of habitat

- increase in safety hazards due to broken bottles, sharps, cans etc.
- decrease in aesthetic quality of an area.

Suggested methods, equipment and reporting

Litter should be collected regularly, ideally every month and no longer than every three months. You should dispose of all collected litter at an appropriate facility. Where possible, items should be recycled, and all dangerous goods should be disposed of safely. It may be beneficial to make a photographic record of the condition of the study area for later comparison.

Equipment

The equipment you will need for recording and cleaning up beach litter includes:

- beach litter record form (Appendix 5)
- heavy-duty plastic, hessian or 'Clean-up Australia Day' bags
- scales accurate to 0.2 kg
- trundle wheel or 50 meter measuring tape
- clipboard and datasheet
- topographic map or GPS
- special container for sharps
- gloves and protective shoes for everyone.

Procedure

Sites selected for litter monitoring should be:

- readily accessible at all times to allow regular monitoring programs
- not subject to regular cleaning operations by other agencies
- known to be a litter accumulation site.

Scientific field procedure

Set up 3 x 100 m to 500 m transects (depending on site and amount of litter) parallel to the shoreline, using the trundle wheel or tape measure, recording and marking start and end points.

Collect all visible litter within 5 m either side of transect, as found along the length of the transect, ensuring the whole area is covered.

Sort all the litter collected into categories, as listed on the datasheet. Count the number of items in each category. Remove sand from all items and weigh each category to the nearest 0.25 kg (see Figure 7.3).

Repeat this procedure for each replicate transect established. This method should usually be conducted monthly if possible although less frequent monitoring is still useful.

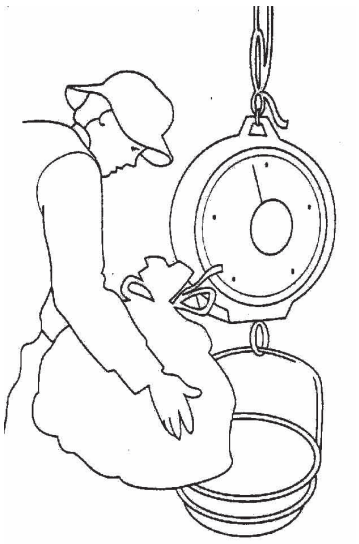
Random field procedure

Fan out over the monitoring site. Randomly search the area for debris, collecting all visible litter with a certain number of people for a designated amount of time.

Sort all the litter collected into categories, as listed on the datasheet. Count the number of items in each category. Remove sand from all items and weigh each category to the nearest 0.25 kg.

This method provides a snapshot of the type and quantity of litter but will not reveal trend information in the data.

Figure 7.3: Weighing beach litter



Interpreting your results

Litter collection allows documentation of the type and quantity of litter and the possible sources of this pollution. Collection also lets you assess whether education campaigns about litter reduction are working.

The origin of litter may be determined by sorting the litter into categories. Origins of litter include:

- boats – offshore and within estuaries
- stormwater
- direct dumping
- windblown.

Physical and chemical parameters

The physical and chemical parameters discussed in this section are:

- water temperature
- pH
- turbidity
- salinity and electrical conductivity
- dissolved oxygen
- phosphorus
- nitrogen
- chlorophyll-a
- bacteria.

Water temperature

What is it and why does it matter?

The temperature of a waterbody directly affects many physical, biological and chemical characteristics, thus playing a vital role in the health and functioning of an estuary. Water temperature greatly influences estuarine processes such as plant photosynthesis, metabolic rates of aquatic animals, rates of development and reproduction, mobility, migration and distribution patterns, and the response of organisms to toxins, parasites and disease. Water temperature can also have major effects on water chemistry.

Long-term temperature changes may have a considerable impact on estuarine ecosystems. Warmer water temperatures encourage rapid growth of heat-tolerant organisms, which often include introduced species brought in on ships from tropical ports. As an example, Pacific Oysters, introduced for aquaculture, will often reproduce in the warm sheltered waters of estuaries, where they settle on rocky shores and reduce the space available for native species.

Water temperature is a key factor controlling the rate of biological processes, such as algal growth. For every 10 degrees Celsius (°C) increase in temperature, the rate of biological processes almost doubles. This is why algal blooms and deoxygenation are much more likely to occur in summer than in winter.

Water temperature influences dissolved oxygen levels. As water temperature increases, its capacity to hold oxygen decreases, which can in turn affect aquatic organisms.

Rapid temperature shifts can stress many aquatic fauna, which are normally accustomed to more gradual changes. Temperature readings are required to convert dissolved oxygen readings from milligrams per litre (mg/L), to per cent saturation, and to convert conductivity readings to salinity readings. Therefore, always record temperature readings when monitoring dissolved oxygen and salinity.

What factors affect water temperature?

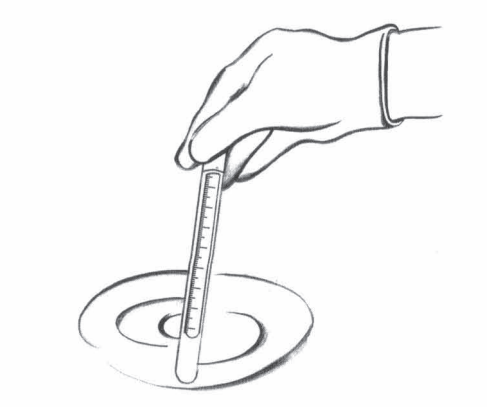
In estuaries, water temperature fluctuates in response to a range of factors including:

- depth
- vegetation
- exposure to sunlight and amount of shade
- latitude
- turbidity
- seasons
- tides
- time of day
- location
- amount of mixing or stratification
- level of inflows from tributaries
- human pressures and impacts.

Suggested methods, equipment and reporting

Water temperature can be measured using a thermometer or a water quality meter. Detailed methods, equipment and reporting for monitoring temperature are contained in Module 4, Physical and Chemical Parameters.

Figure 7.4: Measuring temperature



pH

What is it and why does it matter?

pH is a measure of the acidity or alkalinity of a substance. To measure pH, a pH scale with values ranging from 0–14 is used. Where a pH of 0 is most acidic, 7 is neutral (in water at 25°C), and 14 is the most alkaline (see Figure 7.5). pH varies in different environments as is evident in freshwater, which ranges between 6.5 and 8.0 (sometimes higher), and seawater which has a range of between 8.0 and 8.5.

Estuaries, however, have pH values that lie between those of freshwater and seawater.

Due to humic acids leached from sandy soils, freshwater coastal lagoons and creeks can have pH values lower than 5. The pH of water controls the solubility of many metals such as iron and copper. Low pH may indicate acid rain events or acid soil drainage.

The pH of water bodies needs to be kept within the natural range to protect species and maintain healthy biodiversity. Though they may occur naturally, for example in shallow water overlying seagrass areas, higher than expected pH values may indicate blooms of phytoplankton or macroalgae.

What factors affect pH?

Pollution generated from rain events, such as acid runoff from exposed acid sulphate soils on land, can drain into an estuary and reduce pH values dramatically. Alternatively, biological processes of photosynthesis associated with plants and algae can raise pH levels well above the normal marine range through removal of dissolved carbon dioxide in the waterway. Other factors that may have an affect on pH include:

- source of the water
- time of day
- water temperature
- geology and soils
- discharge of industrial wastes
- atmospheric deposition
- salinity.

Suggested methods, equipment and reporting

You can measure pH with strips (with a resolution of 0.5 pH units), pH liquid test kits (with a resolution of 0.2 units) or with a probe/multi purpose meter (with a resolution of less than 0.1 pH units). The pH strips are the cheapest and most rugged option but are less precise than the other two methods and some types of strips are sensitive to salinity changes.

Detailed methods, equipment and reporting for monitoring pH are contained in Module 4, Physical and Chemical Parameters.

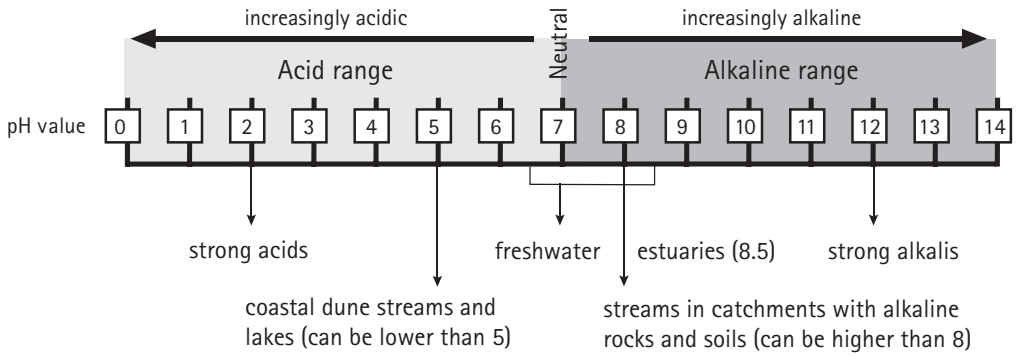
Interpreting your results

Before data can be interpreted, it is necessary to have some knowledge of the natural pH range of the estuary. Some estuaries or coastal creeks may have a naturally low pH, while the pH of sites with strong seawater influence is likely to be high. Therefore, use reference sites in natural or low impact areas to provide a baseline against which to compare data from other sites.

In most estuaries, pH values vary between 6.5 and 8.4 depending on the level of salinity and freshwater influence. Values outside this range would indicate some disturbance of the system due to one or other of the causes discussed above.

Fluctuations of more than 0.5 pH units from the natural seasonal maximum or minimum in freshwater should be examined, as well as in marine waters where the pH varies by more than 0.2 units from the normal values.

Figure 7.5: pH scale



Turbidity

What is it and why does it matter?

Turbidity is an indirect measure of the suspended solids in the water. As the amount of suspended solids in the water increases, the depth that light can penetrate is reduced. The turbidity (cloudiness) controls the depths at which submerged plants can grow. Increased turbidity may indicate erosion or algal blooms and is a useful measure for mapping stormwater plumes.

High turbidity in some estuaries may be indicative of poor land-use practices within the catchment. Elevated levels of turbidity over time can reduce the health of estuaries and impact on estuarine processes by:

- reducing the depth, range and extent of seagrass and macrophyte beds
- absorbing heat and making water temperature rise faster in turbid waters
- decreasing the concentration of dissolved oxygen
- stopping light reaching submerged aquatic plants and reducing their photosynthetic productivity rates
- deterring some fish species or clogging the feeding apparatus of filter feeding animals
- settling into spaces on the bedrock, and decreasing the amount and type of habitat available for creatures that live in crevices or are cryptic in nature
- carrying harmful bacteria and attached contaminants such as pesticide and herbicide residues, heavy metals and nutrients
- changing the substrate from sand to silt/mud depending on depositional zone.

What factors affect turbidity?

Turbidity is affected by:

- rainfall and catchment runoff
- soil erosion
- bed and bank erosion
- waste discharge
- stormwater
- riparian vegetation
- salinity
- flow
- re-suspension
- algal growth
- aquatic macrophyte growth.

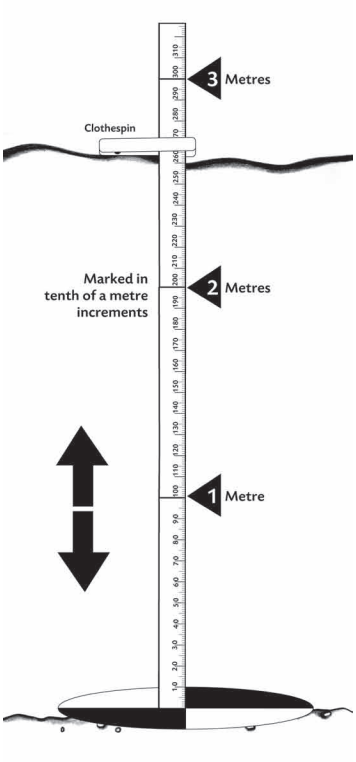
Suggested methods, equipment and reporting

There are generally three ways to measure turbidity; by using a:

- Secchi disk (see Figure 7.6)
- turbidity tube
- turbidity meter procedure.

Detailed methods, equipment and reporting for monitoring turbidity with these methods are contained in Module 4, Physical and Chemical Parameters.

Figure 7.6: Using a Secchi disk to measure turbidity



Interpreting your results

Interpreting turbidity readings requires measurements to be made over a long time to determine the natural turbidity in your estuary. Large variations occur in Australian estuaries depending on chemical, physical and biological factors so it is important to be aware of the range of your estuary's turbidity readings before making assumptions.

Turbidity can also vary considerably depending on recent run-off history. Once such information is available, you can use turbidity data and carefully selected sites to identify potential hotspots or sources of sediment and/or suspended particles.

To distinguish between natural and human-induced impacts on turbidity, it is essential to have good background knowledge of the physical characteristics of the estuary. Different estuaries are likely to have different natural ranges for turbidity levels, which are influenced by rainfall, soil type, tidal range and estuary type. For example, turbidity in wave-dominated estuaries with small tidal ranges tends to be much lower than that in estuaries dominated by strong tidal currents, which keep particles in suspension for longer periods.

Salinity and electrical conductivity

What is it and why does it matter?

Salinity is a measure of the quantity of dissolved salts per unit mass of water. For example, seawater is approximately 35 parts per thousand (ppt), while that of freshwater is less than 5 ppt. In estuaries, salinity largely depends on the interaction between marine and freshwater inflows; thus the salinity range may vary greatly between 35 ppt when the tide is high to less than 5 ppt in areas of low tide and freshwater inflow. Salinity regimes within an estuary are greatly influenced by the ebb and flow of tides as well as variations in freshwater input. Therefore, salinity distributions are in a constant state of change daily, seasonally and annually.

Salinity is normally highest at the mouth and decreases gradually as the water moves upstream to the freshwater reaches. However, in some estuaries, particularly those in dry coastal regions, when freshwater flows are low or non-existent, evaporation in the mid to upper reaches can cause water to become even more saline than seawater (hypersaline) by as much as 60 per cent.

Due to the close interaction between estuaries and the sea, salinity plays a major role in shaping estuarine ecology. As much of the flora and fauna has adapted to a particular salinity regime, there can be very distinct distributions of communities.

Electrical conductivity (EC) is a measure of a substance's ability to conduct an electric current. In relation to water, the greater the amount of dissolved salts the greater the ability of the water to pass a current, hence the greater the EC. Seawater conducts electricity more easily than freshwater; thus EC is routinely used to measure salinity. The basic unit of measurement for conductivity is microsiemens per centimetre ((S/cm) or EC units. Salinity measurements along an estuary are a good indicator of whether it has experienced recent freshwater inflows or whether it is in the middle of a long, dry period. This information is important when interpreting other water quality information gathered in the estuary. Salinity levels have great impacts on estuarine flora and fauna and profiles of salinity can indicate stratification within an estuary.

Conductivity readings are necessary to convert concentrations of dissolved oxygen to percentage saturation.

Positive and negative estuaries

Positive estuaries are those where the amount of freshwater that naturally enters the estuary exceeds the amount of freshwater lost to the air as evaporation. Such an estuary will have a salinity gradient that is freshwater at its head and will gradually increase to seawater at its mouth.

In contrast, a negative estuary loses more freshwater to evaporation each day than enters the estuary. This type of estuary will have hypersaline water at its head, and the salinity decreases to that of seawater at the mouth.

Negative estuaries are very slow at dispersing pollutants – and this may be even more noticeable if tidal exchange is low. Positive estuaries are common in areas where there is high rainfall and low evaporation, but can exist in low rainfall areas where a small river is fed from an extremely large catchment.

Mapping salinity gradient

Estuaries have a gradient of salinity along them, starting with either fresh water (in positive estuaries) or hypersaline water (in negative estuaries) at the head of the estuary, and either increasing or decreasing in salinity until at the mouth of the estuary the salinity is that of seawater.

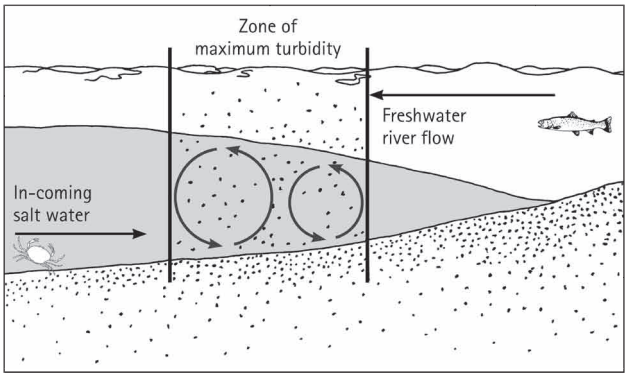
To look at the salinity gradient in your estuary, multiple samples need to be taken along the estuary – along the edges of the estuary and as close to midstream as you can get (usually from a bridge, or from a jetty or boat).

Mapping salinity profile

Stratification, or layering, of different salinity waters often occurs in estuaries. Highly saline water is dense. It is heavier than seawater, so when the tide comes in or a stormwater discharge occurs, the fresher water flows over the top of the more saline water (see Figure 7.7). Stratification can result in reduced dissolved oxygen levels in the deeper layers of water.

To get a representative sample in a stratified estuary, multiple samples need to be taken at a range of depths as close to midstream as you can get, usually from a bridge, or from a jetty or boat. This method is called profiling.

Figure 7.7: Estuary stratification



A common range of sampling depths would include: 20 cm below the surface, 1 m, 2 m and then every 2 m after that, until the bottom of the water body is reached. If your estuary is particularly shallow, you may like to make the sampling depths closer together. Ideally you would take at least three samples (bottom, middle and near surface) from each location. Salinity, dissolved oxygen (DO), pH and temperature readings are taken for each sample.

What factors affect salinity?

Salinity can be affected by many factors including:

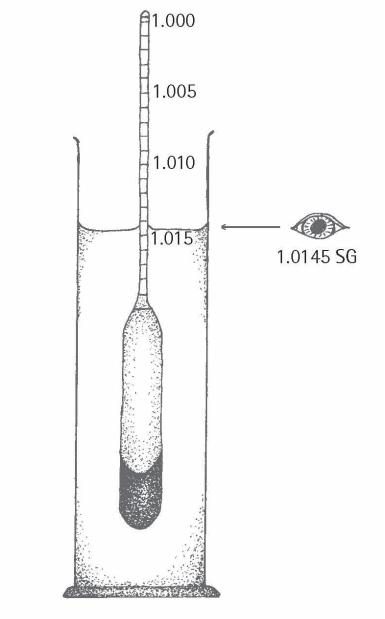
- run-off
- geology and soils
- land-uses
- flow
- temperature
- evaporation and dilution
- discharge of contaminants
- ingress of marine waters.

Suggested methods, equipment and reporting

Salinity can be measured directly using an electrical conductivity meter. However, some estuaries have salinities of up to three or four times that of seawater. Conductivity meters that measure these sorts of salinities are expensive and require regular calibration. In such a situations a hydrometer can be used to measure specific gravity (which correlates to Total Dissolved Solids as g/L) and then converted to a measure of salinity (see Figure 7.8).

Detailed methods, equipment and reporting for monitoring salinity with an electrical conductivity meter are contained in Module 4, Physical and Chemical Parameters. The Waterwatch South Australia *Estuarine Monitoring: Guidance Manual* provides a detailed description of the use of a hydrometer, and includes tables for converting specific gravity to total dissolved solids, to salinity.

Figure 7.8: Reading a hydrometer



Interpreting your results

Salinity measurements will change seasonally. During the wetter months more freshwater will enter the estuary, so salinity levels will tend to be lower, particularly in the mid to upper reaches. Salinity will increase during the drier months when the tides push the saline seawater further back into the estuary.

Each estuary has a relatively consistent range of salinity and electrical conductivity values that, once known, can act as baseline data against which to compare regular measurements of these variables. Any changes outside these normal ranges may indicate altered river discharge or tidal flow into the estuary.

Dissolved oxygen

What is it and why does it matter?

Dissolved oxygen (DO) measures the amount of oxygen dissolved into the water, and available to aquatic plants and animals for breathing. When dissolved oxygen levels drop, aquatic life starts to die. Some organisms need higher or lower oxygen concentrations, so the sensitive ones die first. Fish coming to the surface for air is one of the first signs of low DO levels.

High nutrients increase the growth of algae in the water. Algae are plants that breathe oxygen at night via respiration, and create oxygen during the day via photosynthesis. In clear waters with low nutrients, the night to day changes in DO content are minimal. In nutrient-rich waters the amount of algae in the water increases, and the night–day DO change may become extreme.

Sources of oxygen in the water include:

- atmospheric oxygen that is constantly exchanged between air and water through interactions at the water surface including waves, ripples and tumbling water
- photosynthesis by aquatic plants and algae expelling oxygen into the water as a waste product.

The concentration of dissolved oxygen in a waterbody is an indicator of the health of the ecosystem. Low levels (< 3 mg/L) of DO will harm most aquatic life through lack of oxygen, whereas high levels can lead to supersaturation. DO levels will generally be low where there are high levels of organic waste entering an estuary, due to the increased rates of respiration from bacteria feeding on the organic matter.

What factors affect dissolved oxygen?

Factors affecting dissolved oxygen concentrations include:

- water temperature
- photosynthesis by aquatic plants and respiration by aquatic flora and fauna
- breakdown of organic matter in the water by bacteria
- water movement and mixing
- flow
- daily, seasonal and long-term cycles of biological and chemical characteristics
- removal of vegetation
- presence of nutrients and chemicals in the water
- altitude
- depth – thermal and salinity stratification.

Suggested methods, equipment and reporting

Dissolved oxygen may be measured using either a dissolved oxygen test kit or a dissolved oxygen meter. Detailed methods, equipment and reporting for monitoring DO with these methods are contained in Module 4, Physical and Chemical Parameters.

Interpreting your results

When reporting dissolved oxygen values, percentage saturation values should be used because the solubility of oxygen is affected by both the temperature and salinity of the water. For example, fully saturated, cold freshwater contains nearly twice as much oxygen as fully saturated, warm seawater (that is, 11.3 mg/L compared to 6.2 mg/L).

Most waters also exhibit a varying daily cycle of oxygen concentration, with minimum values before dawn and maximum values in the afternoon.

Measurement of the daily fluctuations in dissolved oxygen concentrations can enable assessment of the impact of nutrients on plant growth in an estuary. Dissolved oxygen concentrations can be used to indicate the presence of organic pollution from discharges and catchments.

When sourcing reasons for fish kills, dissolved oxygen levels are needed, to determine whether low DO was a factor.

Phosphorus

What is it and why does it matter?

Phosphorus (P) is a mineral nutrient that is essential for all forms of life. Phosphorus occurs naturally in estuaries at low concentrations and generally comes from the weathering of rocks and decomposition of organic matter, however it can also enter your estuary through runoff or discharges (see Figure 7.9).

Phosphorus limits and controls the rate and abundance of plant growth through impairing their ability to take up dissolved orthophosphate. A sudden increase in orthophosphate can stimulate algal growth leading to algal blooms. Algal blooms potentially produce toxins and may cause decreased levels of dissolved oxygen, whilst blue-green algae blooms can have the potential to be extremely toxic to humans and animals. Generally, phosphates do not pose a danger to human or animal health and as such are not regulated in our drinking water.

What factors affect phosphorus?

Various factors affect phosphorus concentrations in an estuary. They include:

- soil type
- geology and rock type
- animal and human wastes
- seasonal conditions
- urban runoff
- phosphate containing fertilisers.

Suggested methods, equipment and reporting

The two methods for measuring phosphorus are:

- dissolved reactive test
- total phosphorus test.

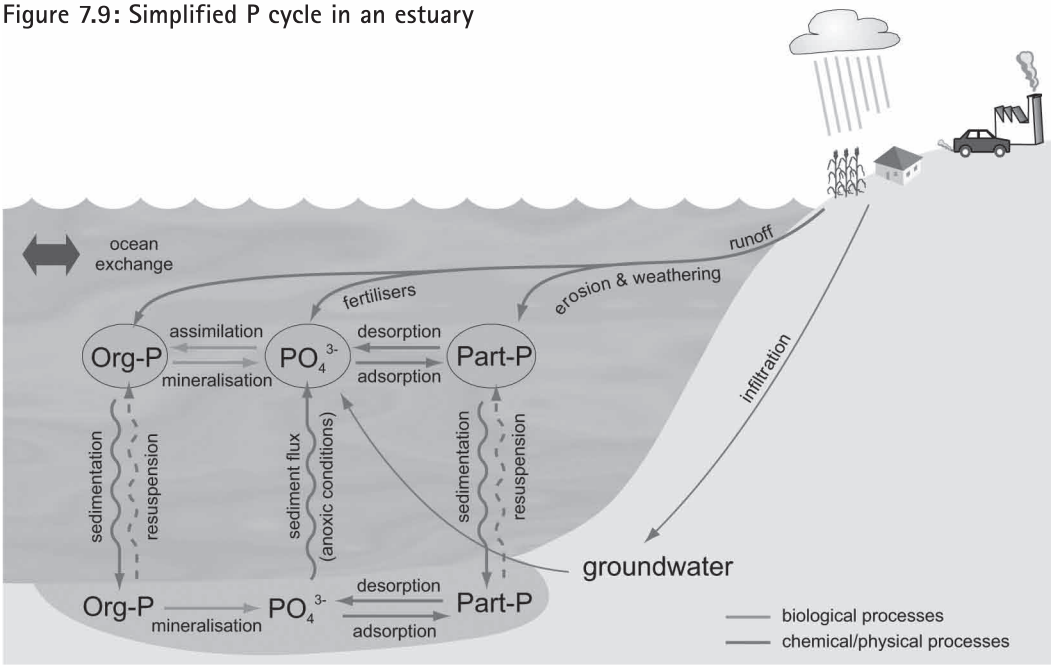
Because phosphorus levels often vary with depth, especially during the summer when the estuary may be stratified, you may need to collect samples at different depths.

Detailed methods, equipment and reporting for measuring phosphorus with these methods are contained in Module 4, Physical and Chemical Parameters.

Interpreting your results

Over time your set of measurements for phosphorous concentrations will enable you to identify when a particular phosphate value does not fit the usual pattern for your estuary. In doing so, you may be able to identify potential algal blooms or nutrient problems and advise the appropriate authority for further management action.

Figure 7.9: Simplified P cycle in an estuary



Source: RiverScience, The Science behind the Swan–Canning Cleanup Program.

Nitrogen

What is it and why does it matter?

Nitrogen is an essential component of most biological processes. Nitrogen is derived from the atmosphere where nitrogen gas (N_2) is the main constituent. Inorganic nitrogen exists in a free state as a gas, as nitrates (NO_3), nitrites (NO_2) or ammonia (NH_3), whilst organic nitrogen is found in proteins and other compounds. Nitrate is a soluble form of nitrogen and as such can be taken up easily by aquatic organisms thereby making it the most meaningful compound of nitrogen to measure (see Figure 7.10).

If nitrogen concentrations increase, the resultant problems can include algal blooms, loss of species diversity and excessive growth of aquatic weeds. Elevated levels of nitrogen can be attributed to diffuse or point source pollution; continued monitoring may identify these pollution sources.

Ammonia is a product of decomposing organic waste and as such can be used as an indicator of the amount of organic matter in the estuary.

Australian estuaries are predominantly nitrogen limited because of efficient de-nitrification of the nitrogen loads. However, there are situations where estuaries can be phosphorus limiting and where the limitation fluctuates between nitrogen and phosphorus on a seasonal basis.

What factors affect nitrogen?

Nitrogen is measured by the concentrations of nitrates in the water, and in general nitrates can be affected by:

- rock type and geology
- soil types
- runoff
- vegetation
- decomposing flora and fauna
- industrial discharges
- animal and human wastes.

Suggested methods, equipment and reporting

The two methods for measuring nitrate concentrations are:

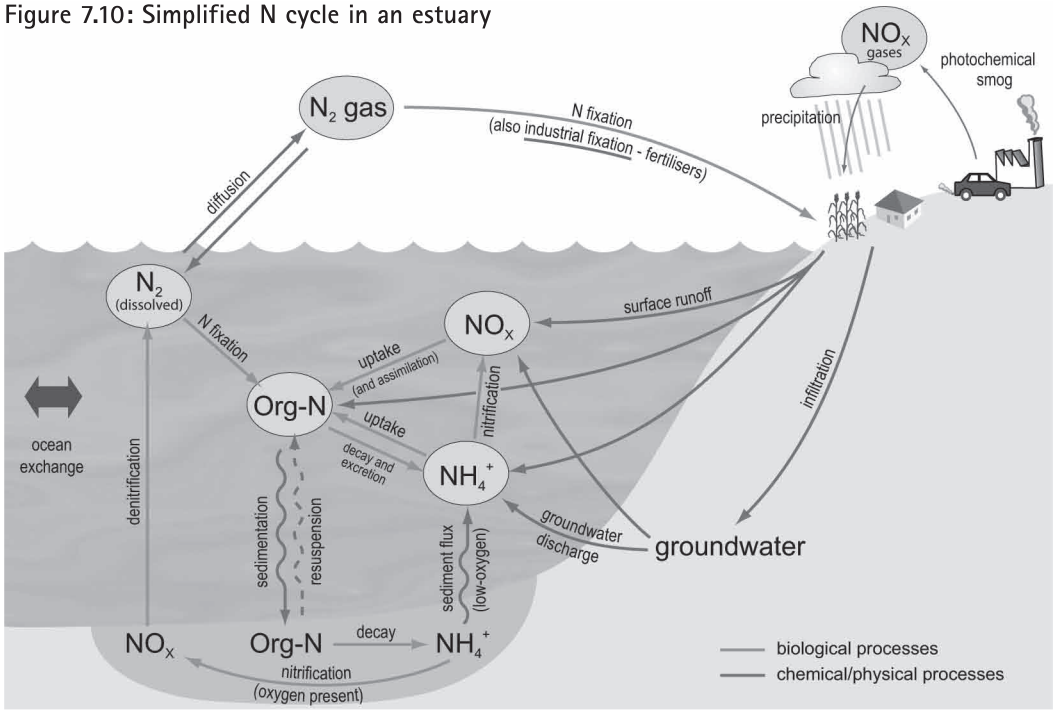
- colour comparator
- Colorimeter/Spectrophotometer.

Detailed methods, equipment and reporting for measuring nitrogen with these methods are contained in Module 4, Physical and Chemical Parameters.

Interpreting your results

As many factors influence nitrogen concentrations within a waterbody, it is not possible to define an upper limit for nitrate levels that will ensure protection of all ecosystems. Your Waterwatch coordinator will be able to provide you with information about the relevant trigger values for your estuary as defined in the ANZECC water quality guidelines.

Figure 7.10: Simplified N cycle in an estuary



Source: RiverScience, The Science behind the Swan–Canning Cleanup Program.

Chlorophyll-a

What is it and why does it matter?

Chlorophyll-a (Chl-a) is the green, photosynthetic pigment found in plants, macroalgae, and phytoplankton growing in estuaries. Chl-a is essential to plants by being active in the capture of light energy for photosynthesis. Most commonly concentrations of Chl-a can be used as an indirect measure of the concentration of phytoplankton cells in the water.

Phytoplankton thrives on nutrients from the surrounding water, and subsequently transforms light and nutrients into plant matter. As such it is the main contributor to primary productivity in the estuarine environment.

Increasing the nutrient load in estuaries usually leads to increased growth of phytoplankton; thus estimation of phytoplankton concentrations is one measure of the primary production in an estuary. Phytoplankton concentrations are generally low in nutrient-poor estuaries whilst high concentrations can be found in nutrient-rich estuaries. Therefore measuring Chl-a can provide a test for nutrients.

High concentrations of phytoplankton are detrimental to an estuary's functioning as it has an adverse effect on dissolved oxygen and water clarity whilst some blooms may be directly toxic to other aquatic organisms.

Suggested methods, equipment and reporting

There is no field test for Chl-a. Instead, samples are collected by passing a known volume of water through filter paper to trap the chlorophyll. The filter paper is then stored in an airtight plastic vial, and kept on ice for transport to a laboratory for analysis. Though samples can be frozen for up to a month before analysis, storage and transportation may make this method logistically difficult, particularly in remote areas.

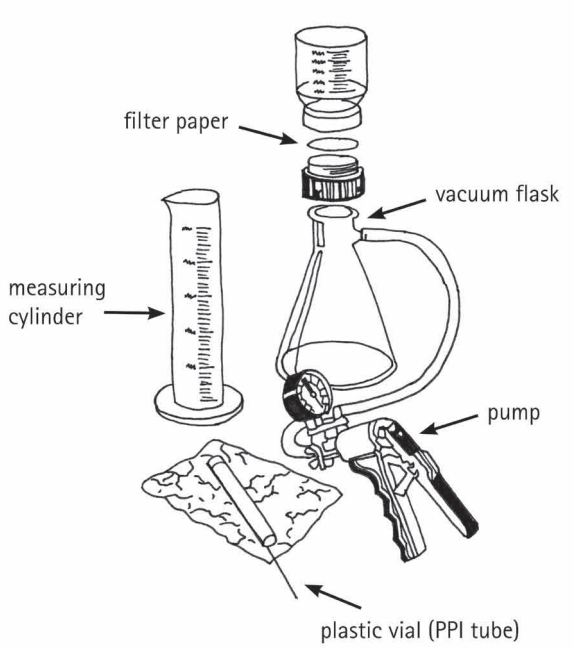
In the laboratory, specific chemicals are used to destroy the algal cells and dissolve the Chl-a into a solution. This is then put into a spectrophotometer to determine the concentration of the pigment.

Equipment

The equipment you will need for recording chlorophyll-a includes:

- site record form
- 2 L sampling bottle with lid
- 500 ml measuring container
- a vacuum pump with gauge for use in the field, or a sample location close to a laboratory with a suitable pump
- 500 ml vacuum flask to fit gauge
- 47 mm glass fibre (GF/C) filter paper
- forceps
- sealable airtight, labelled small plastic containers – each container needs to be labelled, with space to record the site location, date and time of collection and amount of water filtered
- alfoil to wrap small containers in
- small plastic snap lock bags to put small containers and alfoil in
- a small esky with ice or rapid access to a freezer.

Figure 7.11: Chlorophyl–a sample kit



Source: Environmental Protection Agency, Queensland.

Procedure

Collect a two-litre sample from the water body 20 cm below the surface. Pour 500 mL of the sample into the measuring flask, ready for filtering.

Place the filter paper in the filter, ensuring that the wrinkled side of the paper faces upwards. Place 250 mL from the measuring flask into the top of the filter device and use the pump to create a vacuum in the flask. Keep the water level above the filter device semi-full using water from the 500 mL flask, but be careful not to block the filter.

Once the 500 mL flask has been emptied refill to the 500 mL line and continue with the filtering process. After all water has been filtered, or the filter has started to block up, and the filtering device is empty, leave the filter in place for several seconds to ensure the filter paper has been sucked dry.

Record the volume of water that was filtered. Remove the filter paper from the filter screen gently using the forceps, ensuring that the upper surface of the filter paper (which contains the chlorophyll-a) remains untouched. Fold the paper (upper surface on the inside) over twice then place into a small, labelled container.

Record the site location, date, time sampled, and volume of water filtered on the label and on your datasheet.

Wrap the containers completely with aluminium foil to stop light from degrading the chlorophyll-a then place into a plastic bag in the esky or freezer. Freeze the samples immediately.

Arrange to transfer the samples from the storage area to a laboratory that will complete the analysis. Frozen samples may be kept in storage for up to a month before being transported to a laboratory for analysis, although the temperature must not exceed 0°C.

Interpreting your results

By monitoring Chl-a in an estuary, it is possible to gain an understanding of the health of the system in terms of nutrient availability to organisms and pollution. High phytoplankton concentrations may be used as an indicator of declining health in a system, and measurements of Chl-a in conjunction with other water quality parameters may provide useful information on the status of your estuary.

Data is expressed in micrograms per litre (ug/L). Chl-a concentrations vary seasonally, and tend to be at their highest during the warmer months when light and temperature levels are higher. As phytoplankton needs light, its growth may be inhibited in turbid waters, resulting in low Chl-a readings. It is therefore recommended that turbidity/water clarity levels or Secchi depth be monitored in association with Chl-a sampling.

The amount of flushing a site receives will also influence results, as regular flushing will wash algal cells away. As the more confined upper reaches of an estuary tend to be flushed less than the lower reaches, they generally have higher concentrations of Chl-a.

Bacteria

What is it and why does it matter?

Bacteria are present in all environments, and form an essential part of the detrital cycle. However, not all bacteria are benign. Water that is contaminated with faecal matter from people or warm-blooded animals like cattle can carry bacteria that pose a risk to humans through swimming or through eating contaminated shellfish. 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 human health.

Fortunately, other organisms live in faecal matter in huge numbers, and we can use the presence of these bacteria as 'indicators' of faecal contamination. Some of the more common bacteria may also cause disease themselves, but their main importance in waterway monitoring is that they warn us of the presence of more harmful pathogens. 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 by-products. While many coliform bacteria are found in faecal matter, 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.

More specifically related to faecal matter is the faecal coliform group. One specific bacterium, *Escherichia coli (E. 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.

Faecal coliforms enter streams from a variety of sources including sewer and septic systems; stormwater carrying animal droppings; runoff from intensive farming and broadacre farming; and waterfowl and livestock defecating directly into the water. Counts of these colonies can rise dramatically following wet weather events as sewer and septic systems overflow and when stormwater enters the estuary.

Monitoring faecal coliform provides a measurement of excrement in the estuary (from humans, domestic animals, livestock and wildlife) increasing the disease risk and nutrient inputs. Increasing concentrations generally indicate rising pollution levels in the waterbody.

Presence of faecal coliform is an indication of the presence of other more potentially dangerous pathogens in the water, such as Cryptosporidium, Giardia and Campylobacter.

What factors affect faecal coliforms?

A number of key factors influence the survival of faecal coliforms in waterways. These include light, turbidity, temperature and pH, however temperature and pH play a lesser role in determining faecal coliform survival.

Sunlight is probably the most important variable affecting faecal coliform die-off in waterways; a 90 per cent reduction in the population of faecal coliform might be expected in a few hours of bright sunlight, whilst in darkness the organisms may persist for many days. In sewage-contaminated or oxygen-stressed waters faecal coliform survival is extended.

Suggested methods, equipment and reporting

When monitoring for bacteria ensure that safety considerations are taken for both the safety of the person monitoring and also to ensure you don't contaminate the sample

Equipment

The equipment you will need for measuring and recording bacteria includes:

- record forms
- sterile container for collecting samples
- sterile disposable gloves
- sterile distilled water (boiled and cooled) for diluting samples, or faecal coliform water sample bottle
- sterile 10 mL disposable plastic syringe body for each sample
- sterile 100 mL containers for incubating any presence–absence tests and doing dilutions
- commercial test kit.

Procedure

Several types of commercially available tests are available for bacteria measuring. Some are presence–absence tests, some have two tests in one, and some produce count data. Speak to your Waterwatch coordinator about whether you wish to undertake your own tests, or whether it is possible to have samples tested in a laboratory.

Ensure you do not contaminate your samples – wash your hands before sampling, use sterile gloves and follow the sampling guidelines provided on the test kit.

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 container. Top the container up to the 100 mL mark with sterile distilled water. Screw on the lid and shake lightly to mix the waters. You are now ready to test the sample.

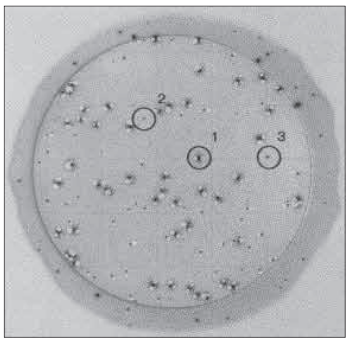
Presence–absence tests require use of a sealed dose of reagent to be placed into a 100 mL bottle containing the diluted sample. The bottle is sealed and swirled until the media is dissolved and then placed into an incubator at the correct temperature.

Once the time is up, examine the colour of the liquid, as described on the package insert. The test may also require use of a special fluorescent light in a darkened environment.

Count tests involve use of pre-prepared slides coated with agar and sealed in a sterile tube. The slide is inserted into the diluted water sample in the 100 mL bottle for 5–10 seconds. The slide is allowed to drain in air for a few seconds, and then resealed and incubated.

The contact slides have numbers on each side. Numbers (0, 103, 104, 105, 106, 107) are recorded and multiplied by 10 to correct for the dilution factor and the final results are recorded as 'colony forming units per mL' (see Figure 7.12).

Figure 7.12: E coli count plate



Data Interpretation

These tests may give highly variable results when not conducted by water authority laboratories with suitable equipment. The presence of faecal coliforms within the estuarine environment may act as an indicator of pollution, including more harmful sources of bacteria such as Giardia and Cryptosporidium. Thus, monitoring this parameter can help determine whether further action is necessary in order to provide estuaries that are safe for economic, ecological and recreational use.

Safety when testing for bacteria

- Dispose of the nutrient plates safely after incubation. The supplier of the nutrient plates may collect and dispose of them, or you need to have access to an autoclave to do this yourself. Check with your supplier.
- Always use forceps to hold objects coming into contact with the sample – never use bare fingers.
- Do not breathe into or out of sample contents to avoid contamination and prevent any risks to your health.
- Wash hands with soap before and after carrying out the analysis.
- Always sterilise any forceps or filtration units by dipping them into 'alcohol' such as methylated spirits and ethanol before and after use.
- Use one filtration unit per sample and re-sterilise before using again.
- Avoid sampling surface water or water from the bottom with sediments, since these areas contain greater numbers of coliform bacteria than the main water body, and will not indicate a representative sample.

Vegetation and habitat

The vegetation and habitat discussed in this section is seagrass, macroalgae (that is, seaweed), mangroves and saltmarsh.

Seagrass

What is it and why does it matter?

Seagrasses are flowering plants that live in marine and estuarine habitats. They are rooted in the sediments with leaves appearing above ground and, like terrestrial grasses, reproduce via seeds and flowers. They can usually be recognised by their green colour and are generally found in dense beds in areas of low wave action, predominantly within estuaries and bays.

There are only 58 described species of seagrasses worldwide, with one-third of these restricted to Australia, however this small number of species does not reflect the importance of seagrass ecosystems.

Seagrasses play a number of roles considered beneficial to the health of estuaries, including:

- providing nursery habitat for fish, crustaceans and invertebrates
- stabilising the sediment, thereby decreasing turbidity
- absorbing dissolved nutrients and converting them to plant material, thereby reducing the occurrence of toxic algal blooms
- increasing plant production of inshore waters, thus expanding the base of the food chain
- helping to reduce wave action and current energy.

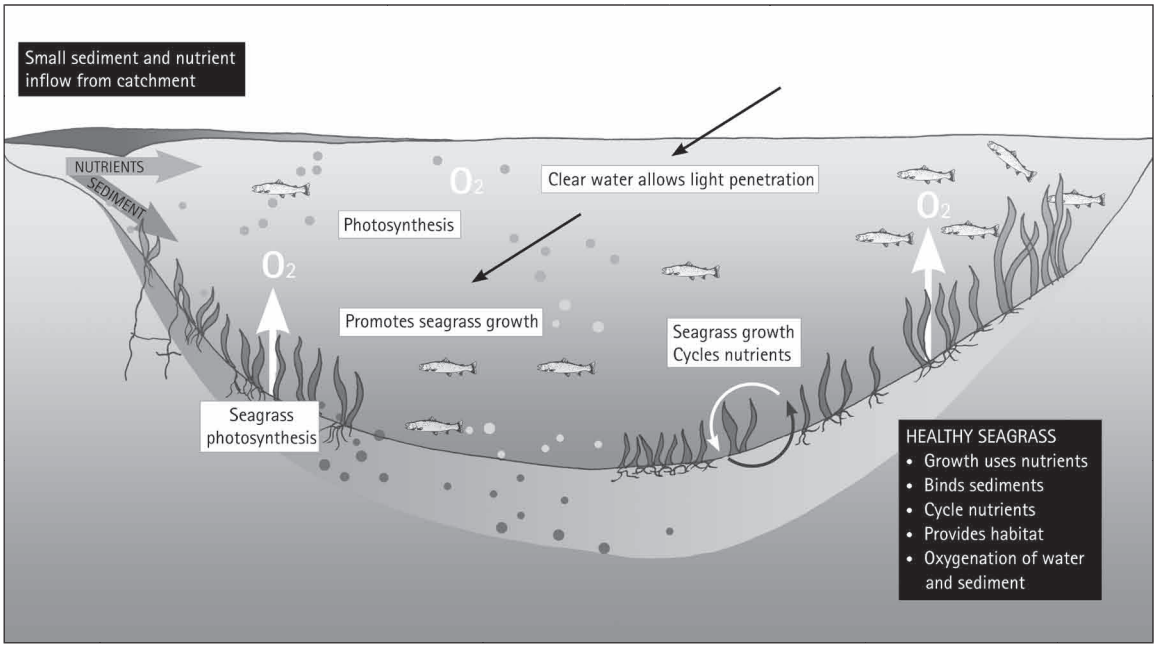
In comparison with unvegetated habitats there is a large abundance and diversity of animals associated with seagrass beds. These include:

- infauna – animals living in the sediment, among the rhizomes of the seagrass
- mobile epifauna – smaller, mobile animals associated with the surface of the sediment
- sessile epifauna – animals permanently attached to the seagrass stems or leaves
- epibenthic fauna – larger, mobile animals that are associated with seagrass beds rather than individual shoots
- periphyton – a thin layer of microscopic bacteria, which quickly colonise any exposed area of seagrass.

Natural processes and human activities often cause frequent change in seagrass meadows. Productivity occurs mostly in the warmer months, with the timing varying among species; however, overall growth rate depends on the turbidity of the water, temperature and nutrient concentrations.

Seagrass communities are susceptible to changes in water and environmental quality, which makes them a useful indicator of environmental health. The degradation or loss of seagrass beds can be an indication that the health of an estuary has been compromised.

Figure 7.13: Healthy seagrass environment



Source: Concept diagram by Department of Land and Water Conservation, graphic by Frank Lopez.

What factors affect seagrass?

Successional changes are related to natural events such as severe wave action, and human activities such as dredging, discharge of nutrients into estuaries, poor boating practices, land clearing and habitat destruction.

Seagrass decline can also be attributed to a reduction in light penetration. Algal blooms, suspended particles and sediment in the water column, excess nutrients and runoff, and industrial pollutants all cause decreased light penetration.

Suggested methods, equipment and reporting

Seagrasses can be measured in a number of ways such as:

- mapping seagrass beds
- measuring and monitoring the percentage cover
- measuring the density and condition.

Mapping seagrass beds

In response to changing environmental conditions, seagrass beds can shift in location and increase or decrease in density and condition. If this is the case, the resultant beds will need to be mapped in order to show new patterns of distribution and abundance.

If you have a GPS use it to establish the boundaries of the bed, otherwise shoreline features, buoys, shoals and landmarks will help you mark the location. You can do the mapping from a boat, by snorkelling, or by using published maps that may be available from relevant management agencies.

When mapping the bed, note any differences such as changes in species composition, epiphytic growth, density and general condition of the beds. It is a good idea to seasonally map the beds to ascertain shifting patterns.

Older maps and aerial photos are useful for comparing changes over time and, coupled with a review of historical events, can lead to identification of factors that may have affected distribution of seagrass beds in the estuary.

Percentage cover

Percentage cover indicates the status and stage of development of a seagrass meadow. Measuring and monitoring percentage cover over time provides access to an early warning indicator of seagrass decline. Appendix 3 provides a guide to estimating the percentage cover of seagrass.

In general, if percentage cover is high and has varied little over time, this is indicative of a stable, well-developed community. However, a reduction in cover and failure to recover from disturbances may indicate natural or human induced stress.

Seagrass watch

In Queensland, the need for information on long-term monitoring of seagrass meadows and increased concern over their decline led to establishment of the community-driven Seagrass Watch program. This program, which operates in several locations from Cairns to Moreton Bay, is supervised by the DPI Marine Plant Ecology Group, based in the Northern Fisheries Centre, Cairns. Its aim is to provide a reliable early warning of changes in the status of Queensland's seagrasses, and to measure these changes. More information on Seagrass Watch is available on the web site <http://www.seagrasswatch.org/>.

In New South Wales, the Community Environment Network is implementing a Community Seagrass Monitoring Project. More information is available at <http://www.cccen.org.au/Projects/seagrass/index.htm>.

Density and condition

Seagrass communities are highly productive environments, and an above ground biomass estimate can be taken as a measurement of their productivity. By using a visual technique of estimating above ground biomass, you can identify increases and decreases in seagrass density over time. Condition can also be measured by using a visual technique.

A coating of sediment on seagrasses can indicate an increase in suspended sediment in the water column, whilst large amounts of algal epiphytes can be indicative of increased nutrient concentrations within the estuary. Any increases in these factors could be considered an indication of environmental decline.

Equipment

The equipment you will need for measuring seagrass includes:

- 50 x 50 cm quadrat
- 2 plastic star pickets (do not use steel star pickets as they may corrode or pose potential hazard to others)
- mallet
- subsurface buoy with stainless steel trace
- site labels (if photographing on site)
- 50 m measuring tape
- 4 x 50 cm plastic pegs
- compass and GPS
- seagrass percentage cover or density and condition illustration sheet and record form.

Procedure

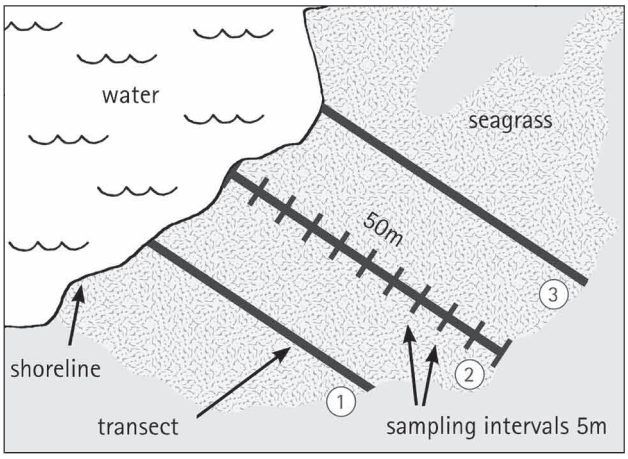
Try to select a site in the seagrass community (50 m x 50 m) that is representative, has low variability and is not difficult to revisit for future monitoring. Be mindful to minimise any disturbance to the site.

For future reference, record the position of the selected site using a GPS or compass bearings. This will make relocating it easier at a later date.

Establish three transects (see Appendix 1) at each site. Permanently mark out the middle transect at each site, by driving a star picket into the ground at the landward edge until the top is only 10 cm above the water surface. Attach a subsurface buoy and record the position using a GPS. You will need permission from the management agency if you intend to establish permanently marked plots.

Make sure the transect runs at right angles (90°) to the shore, by taking a compass bearing. Holding the 50 m tape, follow the compass bearing for 50 m. To mark the end of the transect, drive a picket into the ground at this point. Run transects 1 and 3 directly parallel to the left and right of the middle transect (transect 2).

Figure 7.14: Establishing transects in seagrass habitats



Place the 50 cm x 50 cm quadrat (see Appendix 2) to the right of the transect at 0 m. Estimate the percentage cover of seagrass in the quadrat and/or seagrass density and condition. Record the results onto the datasheet, along with the species of seagrass found in the quadrat and an estimate of the percentage of total cover represented by each.

Measure the percentage cover of epiphytes on the seagrass blades within the quadrat. This figure should be the percentage of seagrass leaf area that is covered by epiphytes. Estimate the percentage cover of both filamentous and macroalgal cover in the quadrat. Record the presence of fauna species (including worm and yabbie holes) and any other features of interest.

Repeat quadrats at 5 m intervals along the transect up to and including the 50 m mark. Repeat this procedure for each transect. You should take measurements seasonally and always record your information accurately.

Interpreting your results

Data collected using these methods primarily show trends over time at and between sites. The best way of depicting the results is in the form of graphs and/or histograms. Seagrass meadows exhibit natural changes in density and condition, for example percentage cover is generally higher in the warmer months, and as such it is necessary to collect data for a minimum of two years to account for this natural variability.

Rates of change in percentage cover should be compared with those of nearby sites to determine if the event is local or more widespread. Reasons for decline could include environmental stress whilst those beds experiencing gains could indicate recovery after a flood event.

It is important to remember that different sites naturally have different rates of cover and beds with low percentage cover are not always indicative of a degraded bed. High epiphyte and algal cover may indicate nutrient enrichment at the site, and should be monitored whilst the source of the problem is being investigated.

Macroalgae (seaweed)

What is it and why does it matter?

Macroalgae are large algae that are visible to the naked eye. They are commonly referred to as seaweed, and are found attached to rocky shorelines, washed up on or growing on sandy substrates in estuaries. Species of macroalgae range from large leaf-like structures, to encrusting mats or filaments that have detached from algal beds, and are classified into three main divisions:

- *Chlorophyta* or green algae
- *Phaeophyta* or brown algae
- *Rhodophyta* or red algae.

Regardless of these names, green algae are not always green, brown algae are not always brown and red algae are not always red.

Some species of algae may appear in response to environmental changes, such as increased nutrient concentration or availability, or become dominant if already present in the area. These species are known as 'nuisance' species and can be used as indicators of the environmental health of the area.

Suggested methods, equipment and reporting

Monitoring macroalgal species can provide baseline data on the types and distribution of species within an estuary. The appearance, loss or change in domination of macroalgal species may indicate changes in environmental conditions or nutrient availability.

Equipment

The equipment you will need for measuring macroalgal species includes:

- large sealable sample containers
- esky with ice
- gloves
- species identification chart/book.

Procedure

Wearing gloves for protection, collect samples of macroalgal species from the water or shore. Place in a sealable container.

Place sample on ice away from sunlight, do not freeze; refer to identification list for macroalgal species; and record details of sample location.

If identification cannot be made, samples may be sent to a laboratory for identification purposes. If nuisance macroalgae are present, you need to record the species type, appearance, location, site and (if necessary) a description of the macroalgae.

Waterwatch Queensland

Waterwatch Queensland has produced a handbook, *Common Macroalgae of Coastal Southern Queensland*, which accompanies the Waterwatch Queensland Community Estuarine Monitoring Manual. It provides information on macrophytic algal species in coastal estuarine areas of southern Queensland, and is designed to be used in field observations.

You can visit their web site at <http://www.qld.waterwatch.org.au>.

Interpreting your results

By identifying and monitoring macroalgae in estuarine environments, it is possible to detect the effects that environmental conditions, such as nutrient and light availability, are having on the ecosystem. By understanding the distribution of macroalgal species under normal conditions, it is possible to use macroalgal species as indicators of environmental change.

Mangroves

What is it and why does it matter?

Mangroves are the trees and shrubs that grow in areas of muddy silts – river mouths, or other areas within the intertidal zone sheltered from strong wave action and tidal currents. Mangrove systems, which can tolerate conditions that most other plant species cannot, often grow in soft, muddy soils devoid of oxygen, and can also tolerate frequent inundation from sea water.

The northern areas of Australia have the greatest diversity of mangrove species. Of the 38 species in Queensland, 30 occur north of the Daintree River. Diversity steadily decreases from north to south, with six species present in southern Moreton Bay, two in the Sydney area, and one, *Avicennia marina*, in Victoria. There are none in Tasmania.

Mangrove communities exhibit a number of adaptations that make it possible for them to grow in intertidal environments. For example, mangroves can store excess salt in leaves, which are then dropped from the tree to the ground, or excrete salt through the leaves, and can compensate for low oxygen levels by using pneumatophores (peg-like roots that protrude from the sediment around the tree, and have pores through which oxygen can be absorbed).

The zonation exhibited in mangrove communities is dependent, in large part, on the salinity levels present. Some species are more tolerant to salt concentrations, and thus can survive in the landward zonation where tidal inundation is less common and evaporation leaves high salt concentrations.

Mangrove communities are internationally recognised for the important economic and ecological roles they play. Mangroves can act as buffers against storm activity, thus protecting coastlines and other assets, as well as filtering water as it enters the estuary, removing nutrients, pollutants and other materials.

Ecologically, mangroves are highly productive and provide nursery, breeding and spawning grounds for a range of species, including commercially important species of fish and crustaceans. Other species, such as molluscs and gastropods, are often dependent on this habitat type at some stage in their life. Further, mangroves support high faunal densities as they provide a structural refuge for organisms, harbouring high densities of prey species, low predation pressure, high sediment organic content and small particle size, making this a favourable habitat for fish and invertebrates. Mangrove forests also provide important roosting areas for a range of bird species.

Mangrove communities are increasingly under pressure from human-induced changes. Many have been cleared to allow urban development, canal estates and land reclamation, and distributions have changed due to factors such as increased sedimentation. Further pressures include land and stormwater run-off, agricultural pollution, and increased sediment and nutrient loads.

Suggested methods, equipment and reporting

Mangroves can be monitored in a number of ways including:

- mapping extent
- regeneration
- mangrove structure
- mangrove canopy cover.

Mapping extent and change

It is beneficial to map the mangroves within your estuary, in order to assess any changes in distribution throughout the monitoring program and over longer time frames. This will indicate where conditions have changed, declines in mangrove area, or encroachment on other habitat types, such as saltmarsh.

Where possible, a GPS should be used to record the boundaries of the mangrove forest, otherwise mark distributions using a topographic map. When mapping the area note any changes in species composition, density and general condition of the mangroves.

Regular examination of the seaward and landward edge of a mangrove forest can determine whether the forest is expanding, or retreating landward. For the development of mangroves, sand or mud flats must accumulate until their level is high enough to allow colonisation by plants. This can only occur where there is sufficient sediment supply (either from land or offshore) and where the water flows are of low energy. The presence of different species of mangrove seedlings than the predominant canopy species may indicate future changes.

Construction of embankments, death of offshore benthic flora, such as seagrass beds, and channelling of stormwater flows may all have an impact on the process of bank formation by changing tidal regime, water flow rates or wave action.

Changes in these communities occur slowly, and simple photographic monitoring may detect changes within a few years. One particularly useful way of assessing this change is to establish a photo point at, for example, a boat ramp, that traverses the width of a mangrove forest.

Equipment

The equipment you will need for monitoring mangroves includes:

- camera
- fixed locations for taking photographs (may be paint marks on a boat ramp, above which you stand your camera tripod, a marked jetty rail or something similar)
- a metre rule or other measuring stick with the date marked on a sheet attached to it.

Procedure

Choose a ramp, wharf or other location where you can see along both the seaward edge of a mangrove forest, and along the landward edge. Make sure you can access both areas to place your metre rule against a tree.

This activity is best conducted annually, to record the expansion or retreat of the edges of the mangrove forest.

Establish a markable location close to the seaward edge of the mangroves from which to take your photographs. Place your metre rule against a mangrove tree that will be in the foreground of the photograph. Take a shot that looks along the seaward edge and that includes your metre rule as a scale and date.

Move inland until you reach a point where you can see along the landward edge of the forest. Repeat the procedure outlined above.

Once you have collected several years worth of photographs you may wish to scan the prints, and enlarge or reduce them so the metre rule is the same size in all of them – most image packages will allow you do this – and to overlay the photographs so you can see the difference. Alternatively, you could scale the prints using a photocopier, and then place a grid over the scaled photographs to estimate the change.

Mangrove regeneration

Due to their intolerance to shade, seedlings of most mangrove species are absent, or in low densities, under a mature forest canopy. However, the death of mature trees leaves a gap in the canopy, allowing increased light to reach the forest floor, and triggering establishment of seedlings. These seedlings rapidly colonise the light gap, beginning the process of regeneration and eventually refilling the gap.

Light gap regeneration rate can be an indicator of mangrove system health. Seedlings respond to environmental changes more rapidly than do mature stands, and monitoring can produce useful data in as little as three months. A heavy deposition of sediment from a flood event or major changes to site hydrology can induce stress, resulting in decreased growth rates and increased mortality.

Long-term hydrological changes, such as sediment deposition or greater tidal and freshwater influence, may have occurred at the site over the past 10–30 years, resulting in colonisation by a different species from the original. For example, long-term sediment deposition may raise the elevation of a site, resulting in conditions more favourable for species normally found higher up the tidal gradient. Other hydrological changes, resulting in more tidal or freshwater influence, could create conditions favouring species normally found closer to the seaward or landward margin.

Identifying what species of mangrove is replacing the previous forest can indicate whether climatic or hydrological changes have occurred.

Photographic techniques

Photographs are used to provide a permanent record of the condition of a site, and can allow comparisons of a site over time. A few hints on taking photographs that will ensure consistency between sampling events follow:

- Use the same camera each time a photograph is taken.
- Take the photograph from the same location at each sample event to show changes at the site. This is achieved by using the same reference point and photographic angle each time.
- Take the photo at the same time of day on each sampling occasion.
- When photographing quadrats, take the photo at two angles:
  - from directly above
  - from 45 to 60 degrees (approximately navel height).
- Record the photo and site details on the data sheet so any photos taken can be matched with the site information later.

Equipment

The equipment you will need for monitoring mangrove regeneration includes:

- 50 m fibreglass tape measure
- 2 m tape measure or measuring stick
- 2 PVC plastic stakes
- plastic calipers
- compass
- permanent marker
- flagging tape.

Procedure

In a light gap or a recovering mangrove forest, establish a transect (see Appendix 1) containing a minimum of 25 mangrove seedlings. Monitor the height and stem diameter of each seedling within the transect every three months and calculate the approximate trunk volume of the seedlings.

When selecting a site, use recent aerial photographs or local knowledge to find canopy or light gaps in a mangrove forest, and confirm that there are seedlings present by personal inspection. At least four light gaps from one homogenous mangrove community are needed for data interpretation.

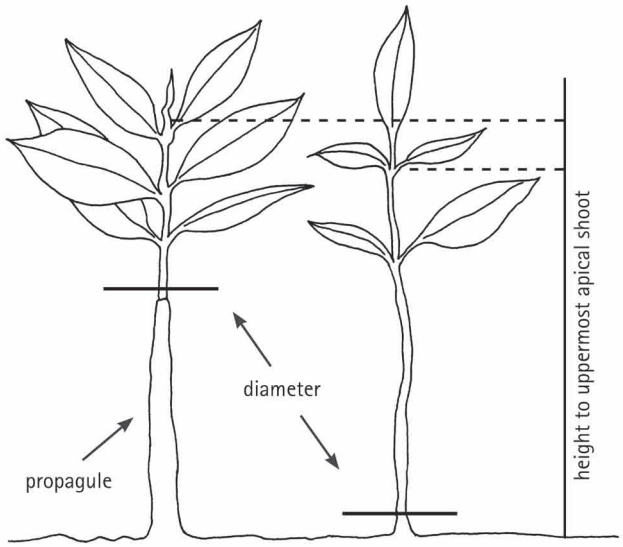
Establish a transect running north–south through the middle of a light gap. Walk to the middle of the gap and take bearings directly to the north and to the south. Mark with stakes the spots where the north–south bearings intercept the boundary of the gap. These will be the start and end points of the transect.

Draw a mud map of the gap showing its dimensions, an arrow representing north, the position of the transect and the surrounding forest type.

Starting at the northern point, lay out the 50 m tape measure through the middle of the gap, to the southern boundary. The transect needs to be wide enough to include at least 25 seedlings within its boundaries. Start with one that is 2 m wide (1 m either side of the tape measure), and count the number of seedlings.

Increase or decrease the distance until a suitable width, containing an adequate number of seedlings, is established. Record this width on the datasheet. If seedlings are extremely dense, establish four quadrats (see Appendix 2), each with 25 or less seedlings at regular intervals along the transect. If seedlings are sparse (less than 50 in the entire gap), or if the light gap is small, sample the entire gap.

Figure 7.15: Measuring the height of a seedling



Using flagging tape, tag each seedling along the transect with a unique number. Beside the corresponding number on the datasheet, record the distance of each seedling along the transect and its position (distance to the left or right) from the middle of the transect (for example, 12 m, left 0.7 m). This allows for easy location of the seedlings during repeat surveys.

Using the measuring stick, record the height of the seedling by measuring from the ground to the base of the uppermost apical shoot. If the seedling is growing from a propagule, take the measurement from just above (see Figure 7.16). Record the result on the datasheet.

Using the plastic calipers, measure the stem diameter of the seedling at the base of the cotyledon, which is just above the original propagule. If there is no propagule, take the measurement at the base of the stem, just above the swelling.

Count the number of leaves on each seedling. If there are more than 25, record the result as > 25 leaves.

When the site is resurveyed, the death of each seedling should be recorded beside its unique number – that number should not be reassigned. New seedlings should be assigned new numbers and their height, stem diameter and leaf number recorded.

Monitor every three months if possible. If you want to conduct longer-term studies, you will eventually need to modify the monitoring methods as seedlings increase in size and decrease in density.

Mangrove regeneration is not suitable in locations with deep mud, as visits will damage the site, potentially damage plants, and reduce growth rates. To prevent site damage, limit the number of volunteers participating in the survey.

Interpreting your results

The rate of increase in seedling biomass indicates the rate of regeneration of a site. To calculate this, measure the relative volume (as opposed to biomass) of seedlings per square metre within the transect, and its rate of increase over time.

Slow or no increase in relative seedling volume and/or high seedling mortality rates may indicate environmental stress. Use the formulas below to calculate the relative volume of seedlings within a plot.

Relative volume of seedling (cm^3) = 1/3*phi*D/2*H

Where:

phi = 3.14 (approx.)
D = diameter of trunk (cm)
H = height of plant (cm)

Seedlings differ widely in shape, due to their leaves and branches, making true volume difficult to measure. Based on the two key indicators of plant size (stem diameter and height), and the 1/3 multiplication factor, this formula actually measures the volume of the plant stem as if it were pyramid-shaped. This is a relative measurement, allowing the growth rate of seedlings to be monitored and compared with that of other seedlings.

Calculate the total volume of seedlings in the transect by summing the volumes of all seedlings measured. Divide this figure by the area of the transect to give volume per square metre (cm^3/m^2).

Calculate the density of seedlings by summing the number of seedlings and dividing by the area of transect (number of stems per m^2). As the light gap matures, density is likely to decrease naturally, as many of the seedlings die due to competition.

Leaf counts also provide an indicator of seedling progress in the early stages of development. However, after 25 leaves have established counting leaves becomes very time consuming.

Mangrove structure

Mangrove structure refers to the composition of a mangrove community in terms of canopy height, stem density, age, tree diameter and species represented. It varies considerably between different forest types, and between the same forest types in different locations. It is influenced by many natural factors including climate, tidal inundation, soil pH and salinity, sediment particle size and amount of freshwater.

Measuring mangrove structure provides baseline data on the diversity and structure of a mangrove community at a particular site. This information can be useful for interpreting other parameters.

Formal measurements provide quantitative data on the structure or level of ecological development of a mangrove community. Data is expressed as:

- stems (living and dead) per hectare
- basal area (square metres per hectare)
- tree height.

Mangrove canopy cover

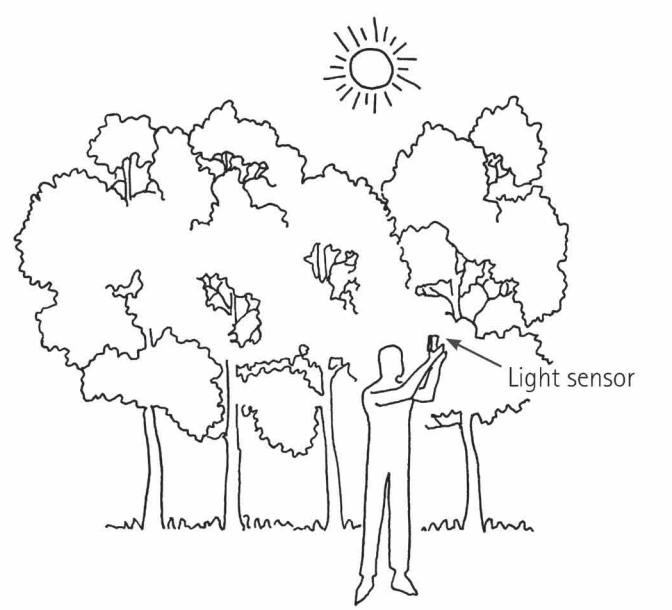
Canopy cover is the area of vegetation covering a mangrove forest. It is dependent on the density of the trees and the amount of foliage present and is often calculated as the amount of sky blocked by the canopy. The leaf area index (LAI) is an index score relating the total area of leaf surface within a plant community to the ground area of that community, and provides a measure of canopy cover.

Plants under natural and human induced stress tend to shed leaves, thus reducing their leaf area. Leaf area index data can be used to monitor both short- and long-term foliage changes in a community and can be compared to data from other mangrove forests. Changes in foliage characteristics may be caused by insect attack, defoliation caused by storm damage, or increased rates of primary productivity. By monitoring leaf area index it is possible to detect changes that may be reflective of stress or disturbance in the environment.

When calculating leaf area index a series of light readings are taken under the canopy and outside the mangrove community in the sun (see Figure 7.16). Leaf area index is determined by calculating the ratio of light under the canopy, to the light in the adjacent open space. Methods for undertaking leaf area index are contained in the Waterwatch Queensland *Community Estuarine Monitoring Manual*.

It is important to distinguish between natural and human-induced changes when interpreting data. As leaf area in canopies will naturally vary slightly from season to season, with a peak during the summer months, leaf area index can also vary naturally between sites and between different communities.

Figure 7.16: Taking light measurements with a light meter



Saltmarsh

What is it and why does it matter?

Saltmarshes are intertidal wetland communities of low growing herbs, grasses and shrubs growing in sheltered areas, normally in the upper intertidal zone between mangroves and the land. They tolerate poor aeration and high levels of salinity common in the soils of this zone, which are only infrequently flushed by larger spring tides. Once these subside, the dampened soils dry out, leaving behind the sea salts. Soils can become so saline and/or anaerobic that areas of bare ground, known as saltpans, occur.

Saltmarsh communities occur in all Australian states, with the largest areas in the dry tropical north. However, diversity increases steadily from north to south, with the greatest diversity in southern Australia, where there are about 26 species.

Migratory wader birds use salt marsh and saltpan communities as roosting and feeding sites, while other birds use salt marshes as breeding sites. Juvenile fish use them during spring tides, and retreat to the estuary as the tide recedes.

What factors affect saltmarsh?

Saltmarsh vegetation patterns can change in response to changing soil salinities and tidal flushing regimes. Land reclamation and development, such as canal estates, marinas and industrial development, have replaced saltmarsh communities in coastal areas. Drainage has been used to convert marsh areas into agricultural lands, and embankments have changed hydrology significantly.

Long-term rises in sea level and in stormwater may increase flushing. This could lead to reduced levels of soil salinity, and the creation of conditions more favourable to the growth of mangrove communities. Construction of tidal floodgates and other structures in some areas can remove tidal influence, resulting in replacement of saltmarsh by terrestrial species.

Other threats include introduced species (for example, *Spartina* spp.), drainage for mosquito and sandfly control, illegal dumping of rubbish, use of recreational vehicles, and cattle grazing.

Suggested methods, equipment and reporting

Monitoring is useful in assessing changes to saltmarsh communities over time, or to monitor the progress of saltmarsh rehabilitation projects that involve changed tidal regimes (that is, after floodgates have been removed or modified).

The suggested method is to record percent cover and species dominance of saltmarsh along a transect (see Appendix 1). The transect line can run either perpendicular to (from land to the mangrove fringe), or parallel to, the tidal gradient.

Equipment

The equipment you will need for monitoring saltmarshes includes:

- 50–100 m tape measure
- 50 cm x 50 cm quadrat
- saltmarsh identification guides
- compass and GPS
- topographic or site map
- plastic or painted wooden stakes
- mallet or hammer
- saltmarsh vegetation record form.

Procedure

It is important to select a monitoring site that is easy to access. Walking across saltmarshes damages them, and they may take a long time to recover. The harder the site is to access, the more damage monitoring it will do. Using the edge of a boardwalk or walking path as the transect line is probably the best site in terms of minimising damage and making this activity easier on participants.

The next most important point is to ensure that the site appears to be representative of the state of saltmarshes in the area. Closely examine aerial photographs of the marsh you wish to monitor. Observe the species composition, cover and canopy height of the saltmarsh and the extent of each habitat.

Establish a transect line. The direction and length of the intersect line in the transect depends on the objectives of the monitoring program, site characteristics and access issues.

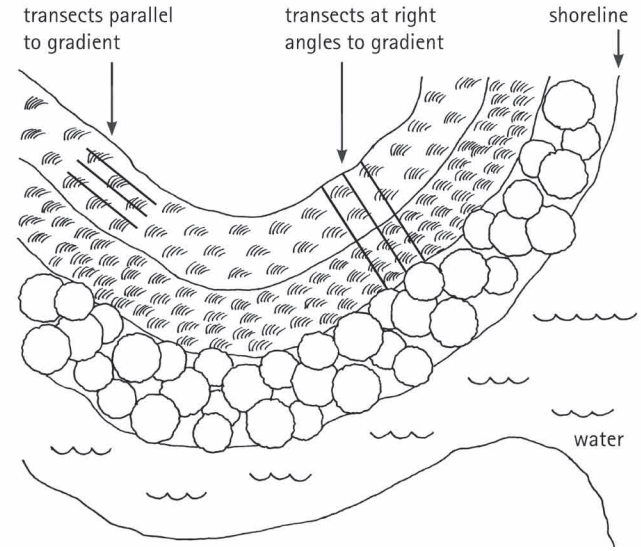
Intersect lines that run from the mangrove edge to the highest point in the marsh will generally capture the fullest range of habitat gradients, however lines running parallel to the tide will give you the largest sample of one habitat type.

Each transect should be 50 m to 100 m long. If you are running the transect line across the marsh, measure this out and lay the measuring tape along the ground. At the start point of the line hammer a small stake in (you will need permission from the land manager if you want to leave a permanent marker). Record the location using a GPS or your site map. Hammer another stake in at the other end and record its location. Attach the measuring tape to each stake so it is relatively straight and level.

If you are using a boardwalk as a transect, simply mark the starting and ending points with a dab of paint on the boards, and record their locations using your GPS or map.

It is recommended that you establish 100 m to 300 m of transect lines within each area of saltmarsh you wish to monitor, although this will depend on your resources and time availability.

Figure 7.17: Diagram of transect layout



Start at the beginning of your transect and look at the plants that intersect the transect. What is growing directly below the measuring tape, or right next to the boardwalk? Use your identification guides to identify it to either genus or species level. If you can't identify it, take a picture or describe it. How much of the first metre of transect line does each species cover (see Appendix 3)?

Record the distance along the transect that this species covers. For example, you may have a 2 metre wide swathe of *Sarcocornia quinqueflora*. It is not necessary to record bare ground if the patches are less than 20 cm across, but bare areas greater than 20 cm should be recorded.

Look at each metre along the transect line and make a similar assessment. If you get large lengths of the same thing, just record this as one entry and draw brackets enclosing the area it covers.

When you get to the end of the first transect, work out the total amount of each species in lineal metres or percentage. Record the unit that you use. Repeat this procedure for each transect.

Do not monitor immediately after a very high storm tide. The saltmarshes are likely to be flattened, which will bias results.

Interpreting your results

The information you obtain will tell you about individual species dominance, the total percentage cover of saltmarsh species and the percentage cover of weed species.

The presence and distribution of species is a simple but useful indicator of ecosystem function over time, with their loss or decline indicating potential environmental stress.

A useful purpose of saltmarsh monitoring is to perform 'before and after impact' assessment for regeneration projects or hydrological changes on the site. If the monitoring is being used for this purpose, control sites should be established to determine if the detected changes are site specific or are the result of more widespread variations, such as the weather.

This activity detects changes that occur slowly over a long period and that may not be noticeable to the eye. An annual measurement, in the same season may be needed for three years or more, before you can detect slow degradation. Two measurements, a year apart, may pick up any rapid improvement that occurs on remediated sites. A once-only measurement will tell you quite a lot about the type of saltmarsh you are monitoring, but will not tell you much about any environmental impacts that may be occurring, apart from the presence and quantity of weeds.

If composition is being used as an indicator of salinity, it is important to know how salt-tolerant local saltmarsh species are. It may also be possible to identify key indicator species of saltmarsh health or structure in some areas where the presence or absence, growth or decline of such species may indicate ecosystem change.

Animal life

The animal life discussed in this section is crabs (and their burrows), the various animal species that inhabit the rocky shores (snails have been selected due to their importance in a rocky shore ecosystem), and wading birds.

Crab burrows

What is it and why does it matter?

Estuarine crabs break down much of the leaf and other organic matter produced by mangrove forests. Their burrows also increase the ratio of soil surface area to air, resulting in some aeration and oxidation of the mostly anoxic mangrove soils. This oxidation can be important for the growth of mangrove plants.

Changes to the crab population can affect the nutrient cycling and oxidation of intertidal soils, which in turn can affect the productivity of mangroves.

Crabs can be sensitive to pollution. Their absence from a mangrove forest may indicate that the site is experiencing human-induced stress.

Suggested methods, equipment and reporting

Data on crab burrow density may complement other mangrove monitoring exercises.

Equipment

The equipment you will need for monitoring crab burrows includes:

- 50 cm x 50 cm quadrat
- 10 m fibreglass tape measure

Procedure

The method is to determine the number of crab burrows in a survey area, estimated by counting burrows within 50 cm x 50 cm quadrats (see Appendix 2).

This method is normally used in association with other methods, but if establishing a new site, you need to ensure it is in a homogenous mangrove forest, in an area representative of the surrounding forest.

Establish three parallel 10 m transects (see Appendix 1) 5 m apart, through the site. Mark the beginning and end of each with a peg to help locate the site again later (you will need permission from the management agency if you want to leave permanent markers).

Starting at 0 m, place a quadrat to the left of the transect and count the number of crab burrows within it. Burrows on the edge of the quadrat should be counted only if the centre of the hole is within the quadrat.

If crab holes are numerous, use a 25 cm x 25 cm area of the quadrat and multiply the results by four. Repeat every 2 m along the length of the transect.

Repeat monitoring every three months.

Interpreting your results

Data is interpreted as crab holes per square metre (holes m^2). Results may be highly variable between sites, so establish a baseline burrow density for each site.

Long-term trends showing a significant decline in burrow numbers may indicate declining crab numbers and/or that the site is experiencing stress. As crabs can have multiple burrow entrances and some species have been known to share burrows, the relationship is not linear.

As crab hole abundance does not equate to absolute crab populations, significant changes in burrow counts would need to be recorded to indicate changes in population.

Plugs can also cover crab holes at low tide and there are holes in the mud that may not be crab holes. Care needs to be taken in drawing conclusions from your results.

Rocky shores

What is it and why does it matter?

Rocky shores in estuaries are home to a broad range of animals and plants that live in the area between high and low tide (see Figure 7.18). These organisms must be able to cope with extremes of temperature and salinity, inundation by seawater, exposure to drying air twice every 24 hours, and pounding waves that can dislodge and crush some species. They also have to avoid being eaten by birds, molluscs and crabs at low tide and by fish and other marine life at high tide.

Several distinct habitats exist on rocky shores; they include:

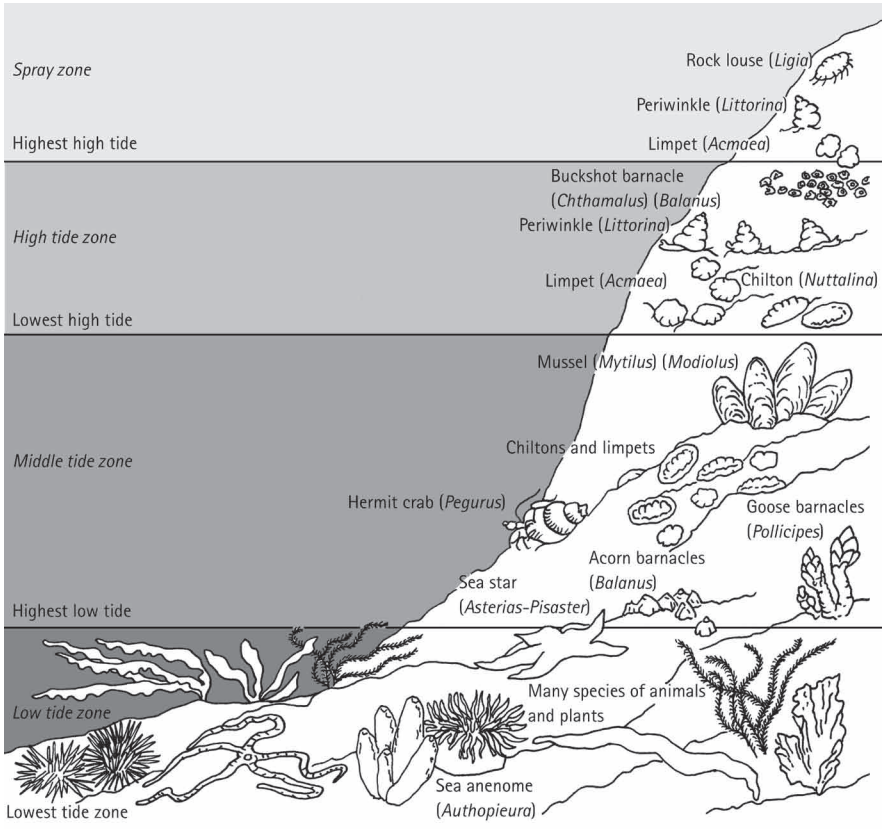
- **Rock platforms** are formed when waves, wind and rain carve rock into flat platforms. Often, the back of the rock forms a cliff, while the ocean edge of the platform steps down into the water.
- **Rockpools** are formed by the grinding action of large boulders and hold enough water at low tide to allow some flora and fauna to survive.
- **Boulder fields** are areas with large boulders and gentle slopes.

Rocky shores are not only subject to extremes in physical conditions they are also at times heavily impacted by human-induced events. Collection of intertidal species for food, bait or display is becoming a widespread and intense activity, as is pollution from stormwater, sewage and chemicals in the marine environment. Such impacts are having a negative affect on species distribution and abundance on rocky shores; monitoring needs to be undertaken to identify when and where adverse effects are occurring.

Snails, for example, have been selected for monitoring due to their importance in a rocky shore ecosystem and their apparent abundance at most sites nationally. Baseline inventories of snail species abundance and distribution, allows for comparisons to be made over time.

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 are, in turn, 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, and the shore may become dominated by plants.

Figure 7.18: Relationship between tide levels and the lifeforms that inhabit rocky shores



What factors affect grazing snails?

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

Snail surveys can quantify the effects of human-induced or natural disturbance, such as over harvesting, trampling or pollution. Continuous monitoring provides resource managers with early warning signs of abnormal conditions.

In many parts of Australia, the highly urbanised nature of the coast has led to rocky shore species being targeted for snail collection, food and bait. As is often the case, larger specimens are taken, thus leading to a decrease in snail size and abundance over time.

Suggested methods, equipment and reporting

Comparison of snail size and abundance within the same shore or comparisons between shores can provide evidence that over-harvesting is occurring or that other impacts may be occurring at a site.

Equipment

The equipment you will need for monitoring snail communities on the rocky shores includes:

- 3 tape measures (50 m long)
- 50 cm x 50 cm quadrat (preferably one for each transect)
- collection tray
- ruler
- grazing snail identification guide
- camera (for later identification of unknown species)
- wet boots or a pair of old sneakers with good grip
- quadrat count record sheets
- GPS or map.

Procedure

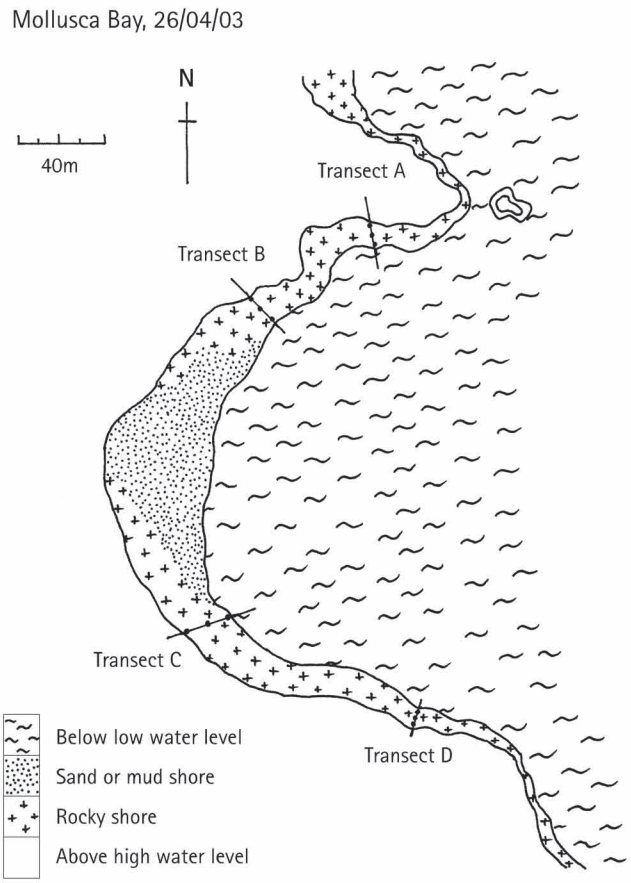
Select a site you believe is representative of the type of rocky shores found in the estuary. This is best done at low tide. If this area is in a marine protected area you may need to obtain a permit to complete this exercise. You may select an area that is known to be disturbed or one known to be protected.

All monitoring should be carried out at low tide, therefore aim to arrive at the shore at least one hour before low tide.

Establish three transects (generally between 20 m and 50 m long and between 5 m and 20 m apart, depending on the size of the site) running between the high and low water lines. Select the location of each transect (see Appendix 1) 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 they may be located again.

Place quadrat (see Appendix 2) locations evenly along each transect. Three to ten 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 (see Figure 7.19), a hand-drawn map is sufficient.

Figure 7.19: Transect mapping on a site map



Transect name	Start (UTM)	End (UTM)	Length (m)
A	272894E 6154701N	272888E 6154697N	15m
B	272801E 6154698N	272829E 6154699N	20m
C	272805E 6154527N	272887E 6154834N	26m
D	272832E 6154515N	272834E 6154509N	10m

Starting at the low water mark, place a quadrat on one side of the transect line. Using a guide, identify the species of snails that exist in your quadrat. Record these down the side of your tally sheet. Measure each snail and record the length, grouping same size range species together. 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.

Tally the number of each species occurring in the quadrat. This includes any snails you can find under or around the rocks in that section. Remember, if you move or turn any rocks over turn them back the way you found them. 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.

Repeat this procedure until all quadrats along the transect have been sampled and recorded.

Monitoring monthly or quarterly is most useful for recording seasonal fluctuations, whereas half-yearly or yearly surveys are adequate for longer-term monitoring projects. Monitoring should take place over consecutive years.

Distribution and abundance comparisons of grazing snails can be made at the same site over time, or for more robust results, comparisons can be made between protected (for example, in a Marine Park) and unprotected areas or between accessible and inaccessible sites.

Safety on a rocky shore

Always work in pairs with one person counting and one recording.

Always exercise extreme caution and beware of waves and rising tides. When working close to the shoreline, take extra care and only work at the lowest of tides.

Always wear suitable footwear with good grip.

Never reach into places you cannot see into. Some species have spines, prickles or can sting or bite you.

Do not touch live *Conus* shells as some species have poisonous spikes. Also be wary of blue-ringed octopuses in rockpools.

Interpreting your results

By monitoring a rocky shore it may be possible to quantify the extent of human impact on both flora and fauna. Human impacts include trampling, removal of organisms and pollution. Trampling can have long-term effects on flora and fauna with recovery being dependent on their sensitivity to disturbance. Removal of organisms alters size ratios, which can impact on population growth and survival rates, and pollution can have devastating effects.

By establishing a baseline inventory and monitoring habitats over time, it is possible to gain an idea of the species present and their abundance. This gives a fair indication of site disturbance and predicts the need for future management.

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
- changes over time
- detection of harvesting stress or other impacts
- measurement of differences between impacted and non-impacted 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 50 cm quadrant is very variable. If there are a large number of small rocks in the quadrat, the actual surface area may be more than double the nominal 0.25 m^2. Therefore averages of several quadrats may provide more reliable data than information from a single quadrat.

Wading birds

Birds are the most conspicuous life form inhabiting estuaries. Wading birds are an important part of an estuarine ecosystem having evolved to exploit the resources of the shallow water habitats present in estuarine systems. They eat small fish, crustaceans, worms, and molluscs; and supply large amounts of easily accessible nutrients back to the ecosystem in their faecal matter.

Many wading birds are migratory and are listed under international migratory bird agreements, which aim to protect critical habitats. However, as loss of habitat in Australia, South–East Asia and important northern hemisphere breeding grounds continues, we need to better understand long-term trends in wader populations.

Information on the populations of wading birds, both migratory and resident, can determine the importance or significance of an estuary and provide an early warning of population decline.

The *Atlas of Australia's Birds* allows volunteers to regularly record birds and to submit their information to Birds Australia. Birds Australia compiles this information and produces regular reports, including reports on water birds and migratory wading birds. More information is available from the Birds Australia web site at <www.birdsaustralia.com.au>.

The Australasian Wader Studies Group is a special interest group of Birds Australia. The group is a non-government organisation dedicated to studying waders (otherwise known as shorebirds) throughout the East Asian–Australasian Flyway.

If you want to include bird monitoring in your estuary monitoring plan, or want to know whether any bird monitoring is already happening, contact Birds Australia.

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Glossary

This glossary contains selected estuarine terms; additional definitions of terms can be found in Module 1 Background.

Aeolian – the erosion, transport, and deposition of material by wind; works best when vegetation cover is sparse, or absent.

Benthic micro-algae (BMA) – microscopic plants that inhabit the sediment surface (or substrate), including diatoms and dinoflagellates.

Bio-clastic – sediments made up of broken fragments of organic skeletal material, for example, shells.

Bioturbation – organisms, mainly worms or crustaceans that disturb the sediment by burrowing or during feeding; their activities mix the sediment layers and may cause substantial sediment resuspension.

Coastal waterway – a body of water situated on or near the ocean coast, with some association with the ocean; includes embayments, wave- and tide-dominated estuaries, wave- and tide-dominated deltas, coastal lagoons, and tidal creeks.

Coastal lagoon – coastal waterways in which waves are the principal factor that shapes the overall geomorphology; characterised by a sandy barrier that can partially or totally constrict the entrance, backed by a mud basin, and typically have negligible river input.

Coastal protuberance – a prominence or bulging out of the coastline, typically formed from deltaic sediments.

Conceptual model – a depiction or representation of the most current understanding of the major ecosystem features and processes (including biological, physical, chemical and geomorphic components) of a particular environment (for example, estuaries).

Cut-off embayment – typically, small basins within wave-dominated estuaries or wave-dominated deltas that have been bypassed by the principal fluvial current flow, and therefore have restricted exchange with the main body of the coastal waterway.

Deposition – the dropping of material that has been picked up and transported by wind, water, or other processes.

Drowned river valley – a bedrock valley that has been submerged by rising sea level, and has not been significantly infilled by sediment. See also: Embayment.

Ebb tide – a falling tide; the phase of the tide between high water and the succeeding low water.

Embayment – a coastal indentation (or bedrock valley) that has been submerged by rising sea level, and has not been significantly infilled by sediment. See also: Drowned River Valley.

Epiphyte – an epiphyte is a plant that grows upon or attached to another living plant using it for support; many aquatic species of algae, including seaweeds, are epiphytic.

Epifauna – animals that live on the sediment but do not burrow into it.

Euryhaline – organisms able to tolerate a wide range of salinity.

Facies – sum total of features that reflect the specific environmental conditions under which a given sediment was formed or deposited; the features may be lithologic, sedimentological, or faunal.

Flood tide – a rising tide; the phase of the tide between low water and the next high tide.

Flushing – exchange of water between an estuary or coastal waterway and the ocean.

Freshwater – water, typically derived from inland or rainfall, with less than 0.03 per cent ionic content.

Function – the function of an estuary is how it acquires the materials and energy needed, processes its waste products, and interacts with adjacent waters and the surrounding landscape.

Halite – mineral formed by evaporation, composed of NaCl.

Halophytic – salt-tolerant vegetation.

Headward – the landward or upstream section of an estuary or coastal waterway.

Hypersaline – water with a high concentration of salt, for example, greater than the ionic content of seawater.

Intermittently closed and open lakes and lagoons (ICOLLs) – referring to coastal lagoons and some wave-dominated estuaries under low runoff conditions.

Intertidal – the environment between the level of high tide and low tide.

Macroalgae – large algae including red, green and brown algae.

Macrotidal – coastal ocean or waterway with a high mean tidal range, for example, greater than 4 metres.

Mesotidal – coastal ocean or waterway with a moderate mean tidal range, for example, between 2 and 4 metres.

Microtidal – coastal ocean or waterway with a low mean tidal range, for example, less than 2 metres.

Mouth – the entrance of the coastal waterway, or the place where the sea meets or enters the coastal waterway.

Neap tide – tide smaller than the mean tidal range; occurs about every two weeks, during half moons.

Negative estuary – an estuary in which evaporation exceeds freshwater inflow and therefore hypersaline conditions exist.

Phytoplankton – microscopic, planktonic plants that exist within the water column.

Prograde – the outward building of a sedimentary deposit, such as the seaward advance of a delta or shoreline.

Residence time – the average time a hypothetical particle of water spends in solution between the time it first enters and the time it is removed from a coastal waterway.

Resuspension – a process in which sediment particles on the substrate are brought back into water column suspension by waves, tides, or wind.

Salt-wedge – an intrusion of seawater into a coastal waterway in the form of a wedge along the seabed; the lighter fresh water from riverine sources overrides the denser salt water.

Seagrass – marine flowering plants that generally attach to the substrate with roots.

Spring tide/king tide – tide greater than the mean tidal range; occurs about every two weeks, when the moon is full or new.

Strand plain – a series of dunes, typically associated with, and parallel to, a beach and sometimes containing one or more small creeks or lakes.

Sub-tidal – permanently below the level of low tide; an underwater environment.

Supra-tidal – above the level of high tide; a terrestrial environment.

Tidal creek – coastal waterways in which tides are the principal factor that shapes the overall geomorphology; typically occur on prograding, muddy coasts and contain a narrow channel that drains the immediate hinterland that is fringed by intertidal habitats.

Tidal current – an alternating, horizontal movement of water associated with the rise and fall of the tide, these movements being caused by gravitational forces due to the relative motions of the moon, sun and earth.

Tidal prism – volume of water moving into and out of an estuary or coastal waterway during the tidal cycle.

Tide-dominated delta – coastal waterway in which tides are the principal factor that shapes the overall geomorphology, and river input is sufficient to have filled the basin. Typically funnel-shaped, and the wide entrance may form a coastal protuberance that contains elongated tidal sand banks fringed by inter- and supra-tidal habitats.

Tide-dominated estuary – coastal waterway in which tides are the principal factor shaping the overall geomorphology. Typically funnel shaped with a wide entrance containing elongated tidal sand banks. The margins are fringed by extensive intertidal habitats, separated by tidal channels.

Upwelling – the rise of seawater from depths to the surface, typically bringing nutrients to the surface.

Vertical accretion – accumulation of sediments or other material resulting in the building-up or infilling of an area in a vertical direction.

Washover/back barrier deposit – deposit of marine-derived sediment landward of a barrier system, often formed during large storm events.

Wave-dominated delta – coastal waterway in which waves are the principal factor that shapes the overall geomorphology, and river input is sufficient to have filled in the basin so there is limited space for continued sediment accumulation. Such areas are characterised by a sandy barrier and a river channel that has a direct connection with the sea

Wave-dominated estuary – coastal waterway in which waves are the principal factor in shaping the overall geomorphology. Characterised by a sandy barrier (partially constricting the entrance) that is backed by a broad central basin and a fluvial delta, where the river enters the basin.

Zooplankton – non-photosynthetic, heterotrophic planktonic organisms, including protists, small animals, and larvae that exist within the water column.

Appendixes

These appendixes contain:

- a description of transects and how to establish them
- a description of quadrats and how the make them
- a saltmarsh and seagrass percentage cover diagram
- a site record sheet
- a beach litter record sheet

Appendix 1

Transects

What is a transect?

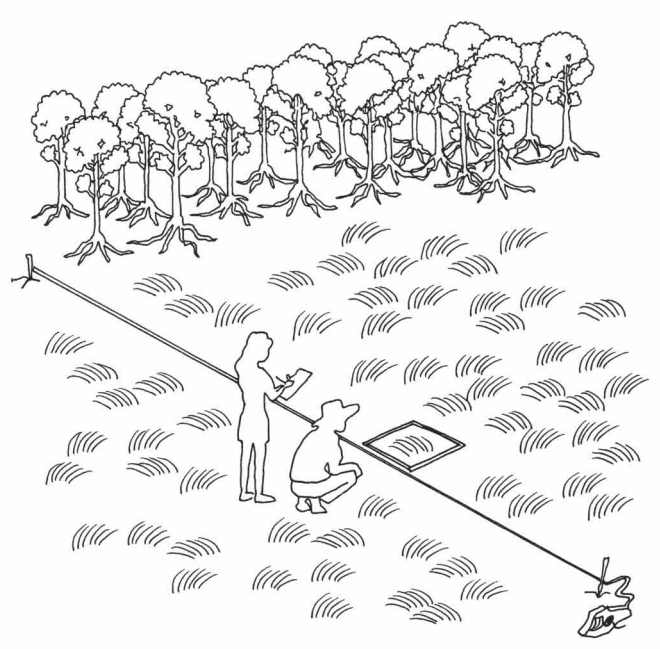
A transect is a line or narrow belt within a given area, used when surveying the distribution of organisms in that area. Transects are normally marked out with tape measures that can be from a few metres to several kilometers long. There are two types of transect – line intercept and belt.

A line intercept transect is essentially a straight line through a survey area. Of the population being studied, only those members that touch, underlie or overlie the transect line are recorded.

A belt transect has a defined width and internal area (length x width). All individuals within these boundaries are recorded. Due to their defined internal area, belt transects can be used to determine density.

Quadrats (see Appendix 2) can also be placed along transects at regular intervals.

Figure 7.20: Using a transect



How to establish a transect

Mark the start of the transect (at 0 m) with a peg, pole or other object that can be fixed into the ground. Record the intended direction of the transect as a compass bearing. This is useful when establishing long transects and allows for easy location of the transect at a later date.

To lay out the transect, set the compass to the bearing and note a feature or distant landmark on the same bearing. Tie the end of the tape measure to the transect marker and walk in a straight line following the bearing, or walk directly towards the feature you have noted. Stop when the length of the transect or the boundary of the study area has been reached. Turn to face the start point and take a compass bearing – it should be 1800 +/- the original bearing.

As visibility is likely to be restricted in a mangrove forest, note a distant tree in the direction of the bearing and walk towards it, laying out the tape measure in the process. Once at the tree, note another tree in the bearing direction and continue.

Appendix 2

Making a quadrat

What is a quadrat?

A quadrat is a square, rectangular or circular frame with a defined internal area, used to measure attributes of a population or subject.

How to make a quadrat

To make a quadrat you will need:

- for a 50 cm x 50 cm square quadrat:
- four x 50 cm lengths of PVC plastic conduit, 20 mm or 25 mm in diameter
- four PVC elbow joints
- tape measure
- hacksaw.

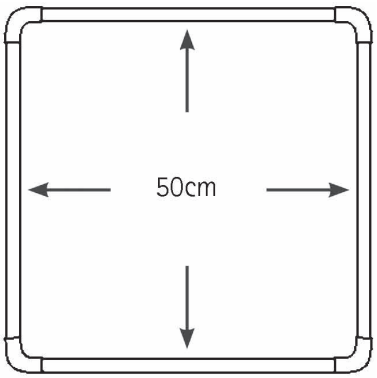
Note: Vary the length of conduit sections according to the size of quadrat required.

Each side of a 50 cm x 50 cm quadrat measures 50 cm from one inside edge to the inside edge of the opposite side. Join the four 50 cm lengths of conduit with the elbows to make a square. Due to the elbows, the quadrat will be larger than the desired 50 cm x 50 cm.

Measure its internal length and breadth and subtract 50 cm from each measurement. The figure obtained is the length of PVC piping that needs to be cut from each 50 cm length in order to finish with a quadrat that has a 50 cm x 50 cm internal dimension.

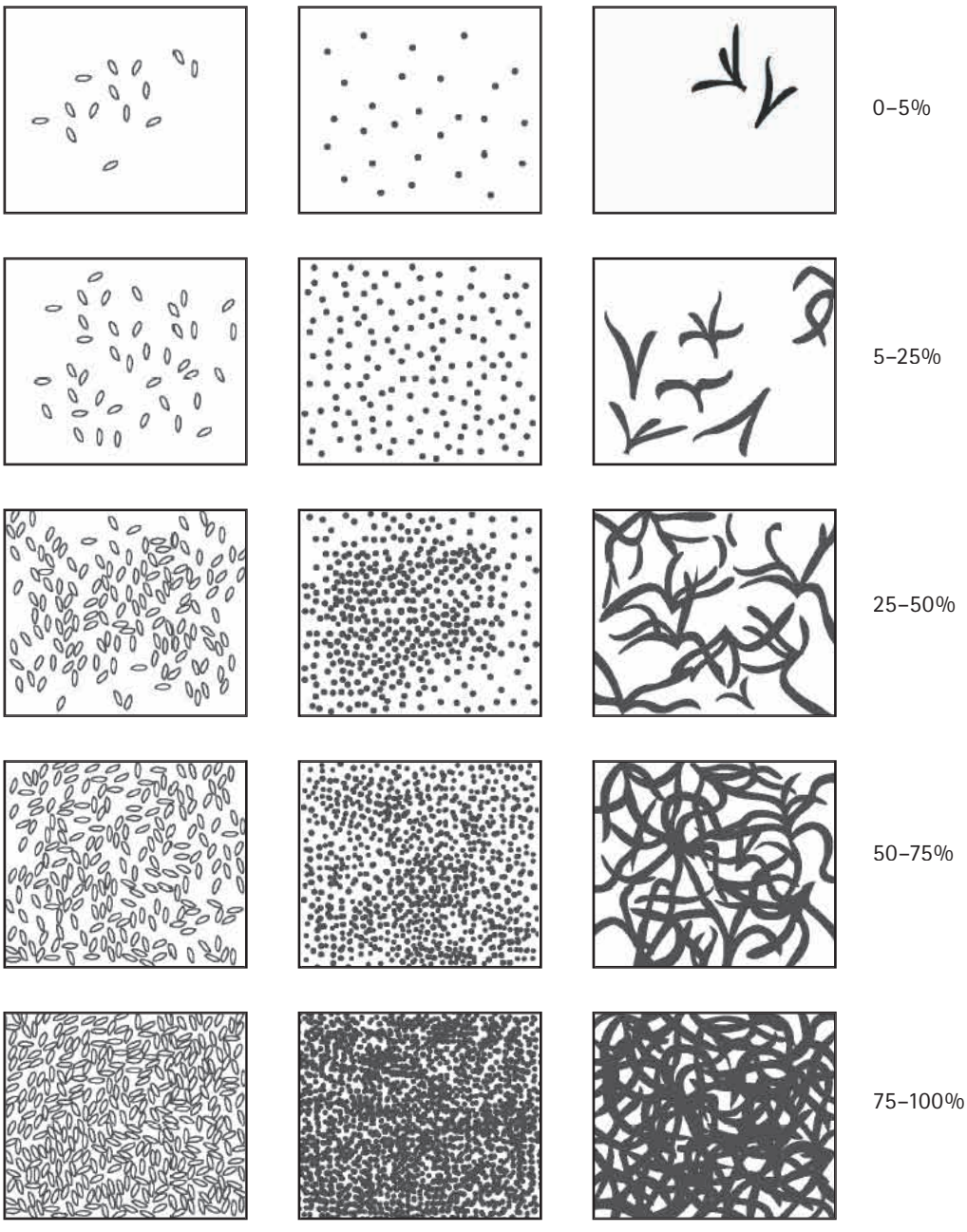
The quadrat can be glued together. However, if you roughen the ends of the PVC the elbows will grip it and it can be left unglued, making it possible to dismantle for easy transportation.

Figure 7.21: Quadrat construction



Appendix 3

Saltmarsh and seagrass percent cover



Appendix 4

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?			M
What temperature is the water?			°C
Is the water flowing in one direction?			
If so, what is its flow rate?			m/sec

Measuring salinity:		**Water quality parameters:**		
Method: Hydrometer	Method: Conductivity			
Raw SG:	EC: ..... mS/cm	pH		
Total dissolved salts (TDS) ..... g/L		Dissolved oxygen		mg/L
		Turbidity		
		Orthophosphate (as ...)		mg/L
		Nitrogen (as ...)		mg/L
		Chlorophyll-a		

Appendix 5

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: