Marine Science

Suggested and Mandatory Practicals

Source: Active wild

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Contents

Unit 1

Oceanography

Source: Scientific American

Suggested practical:

- A convection experiment

◊ Mantle convection currents with golden syrup

Aim: To observe convection currents.

Materials:

- Golden Syrup
- Large glass beaker
- Packet of thin biscuits
- Bunsen burner, tripod and heatproof mat
- Access to a freezer

Method:

1. Pour the syrup into the beaker and leave in the freezer for 1 hour. Do not allow it to freeze, simply reduce the temperature to gain maximum viscosity.
2. Break a biscuit in half carefully and place the two halves, touching, on the surface of the syrup. The biscuit halves need room to move apart, so ensure that the two halves are not too big.
3. Place the beaker and contents on a tripod with a gauze in place to ensure localised heating.
4. Light the Bunsen burner and heat the beaker using a low blue flame directed at the centre of the beaker's base for around 10 minutes (depending on the speed of heating and quantity of syrup used). Slowly the central cone section of the syrup will change in colour as its temperature rises and its viscosity falls.
5. Observe what is happening. After around 10 minutes (depending on the speed of heating and quantity of syrup used) the biscuit halves will have been driven apart by the convection currents.

Sourced from: Institute of Physics, https://core.ac.uk/download/pdf/5213901.pdf
Done by: Chris Pacey, St Patrick's College Townsville & Nika Langham, Pioneer SHS Mackay

Note:
It is now thought likely that there are three possible driving mechanisms for plate tectonics:

- Movement of mantle convection currents as above,
- The mass of the subducted plate (the sinking slab) at the subduction zone dragging the surface part of the plate across the surface,
- At constructive margins, the new plate material sliding off the higher oceanic ridges.

More on this from *A "mantle plume" in a beaker – but not driving plates Mantle plumes 'yes' – but convection currents driving plates, probably 'No'* from
https://www.earthlearningidea.com/PDF/86_Mantle_plume.pdf

◊ Convection currents

Aim: To observe convection currents.

Materials:

- 1 tank
- 2 food dye (blue & red)
- Frozen coloured water
- 1 round tank
- 1 conical flask & stopper
- 2 pins
- 2 paper cups
- Water
- Hot and cold water
- Fresh and salt water

Method:

Sourced from:
Done by: Chris Pacey, St Patrick's College Townsville

◊ Convection currents

Aim: To observe convection currents.

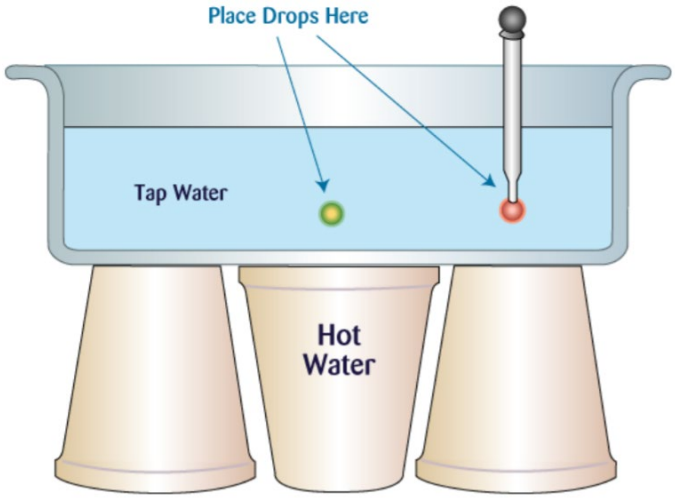
Materials:

- 20 x 30 cm Pyrex pan
- 3 Styrofoam cups
- Tap water
- Hot water
- Ice or chemical cold pack
- Food colouring
- Droppers
- Paper and pencil for logging observations

Method:

This activity can be rather wet.

1. Fill the Pyrex pan 2/3 full with water.
2. Turn two of the cups upside down (to use as a bridge) and set the pan of water on top of the two cups. Fill the third cup with hot water and place it under the centre of the pan of water. Leave the pan for several minutes until the water has stopped moving.
3. Put one drop of food colouring on the bottom of the pan in the centre (directly over the heat source), and put one drop on the bottom halfway to the edge (see picture below).



4. Take the temperature of the water directly over the heat source and off to the side of the pan.
5. Set up the experiment again. This time put hot water on the edges instead of the centre of the pan. What happens if you use ice and make the water colder? How does cold water affect the currents?

Discuss:

- Is there a difference in temperature above the heat/cold source and on the side?
- Describe the movement of water for both parts of the experiment. Which way does the water move over the heat source and over the cold source?
- Summarize the results of water movement in the context of temperature changes.

Sourced from: https://sealevel.jpl.nasa.gov/files/archive/activities/ts1enac3.pdf
Done by: Nika Langham, St Patrick's College Mackay

Suggested practical:

♦ Thermoclines

◊ Dissolved oxygen & temperature

Aim: To test the effect of increasing temperature on the concentration of dissolved oxygen found in water.

Materials: (materials needed for one group)

- One LaMott Dissolved Oxygen Kit (or similar kit)
- One Celsius thermometer
- One 1000 mL Beaker
- 1000 mL of distilled water
- One aquarium air pump/aerator
- One heating pad
- One wall clock with a second hand
- One wooden or plastic spoon

Method:

1. Place the 1000 mL beaker on top of the heating pad(s). The heating pads should be in the "off" position.
2. Fill the beaker with 1000 mL of distilled water, and properly setup an air pump/aerator (turned "on").
3. Allow one day to permit the surface agitation produced by the air pump/aerator to allow oxygen to dissolve into the distilled water establishing a baseline equilibrium concentration.
4. Begin the experiment. To ensure mixing, gently (but, thoroughly) stir the water with the spoon for one minute. Stirring will disrupt any thermocline (and thermocline-driven DO disparities) that might have occurred.
5. Record the temperature of the water in the beaker at a mid-depth, and cantered in the beaker.
6. Then, using the directions accompanying the LaMott's (or other) dissolved oxygen (DO), kit test and record the dissolved oxygen content of the water.
7. Turn on the heating pad(s) for ten minutes.
8. After ten minutes, gently stir the water, and then record the temperature and DO of the water (mid-depth, and laterally cantered).
9. Continue to record the temperature and DO content of the water at regular intervals as time permits.
10. Create a graph of their results.

Discuss:

- What effect could thermal pollution, and subsequent decrease in DO, have on an aquatic ecosystem such as a local stream, lake, pond, etc.?
- What are some sources of thermal pollution?
- What might be some methods or techniques to mitigate or eliminate sources of thermal pollution?

Sourced from: https://nmsgraysreef.blob.core.windows.net/graysreef-prod/media/archive/education/activities/pdfs/gr_dissolved_oxygen_vs_temperature.pdf

◊ The Influence of Water Temperature & Salinity on Dissolved Oxygen Saturation

Aim: To determine water temperature, salinity, and dissolved oxygen levels.

Materials:

- 9x 1 L beakers
- Three shallow containers
- Water
- Aquarium air pump and tubing
- Thermometer
- Salinometer
- Dissolved oxygen meter
- Graph paper

Method:

1. Add 500 ml of water to each 1 litre beaker as follows and label accordingly:
   a) 3 with freshwater (0 ‰)
   b) 3 with seawater (@ 30-35 ‰)
   c) 3 with brackish water (@ 15 ‰).
2. Set up three water baths using shallow containers:
   a) 1 containing ice water (@ 0°C)
   b) 1 containing ambient room temperature water (@ 10°C)
   c) 1 containing heated water (@30°C).
3. In each water bath place a beaker representing each salinity and aerate each beaker for 10 minutes (which should be sufficient to reach oxygen saturation in each beaker)
4. Determine the dissolved oxygen (DO) concentration for each sample.
5. Graph your data as salinity vs. DO and temperature vs. DO.

Discussion:

- Summarize the relationship between temperature and salinity with respect to dissolved oxygen saturation.
- Discuss how these factors could play an important role in the management of aquaculture systems.

Sourced from: https://irrec.ifas.ufl.edu/media/irrecifasufledu/teach-aquaculture-/module-5/How-DO-Changes-With-Salinity.pdf

◊ Thermocline

Aim: To investigate stratification of the ocean due to changes in temperature and salinity.

Materials:

- Rectangular tank with a divider (can be ordered from sciencekit.com)
- 2 Beakers (preferably with pour spouts)
- 2 contrasting colours of food colouring
- Tap water (warm and room temperature)
- Ice
- Bottle containing pre-made salty water solution: Combine approximately 75g of salt dissolved in 1L of water
    - o Note: Kosher salt yields a clearer solution than table salt, but either can be used

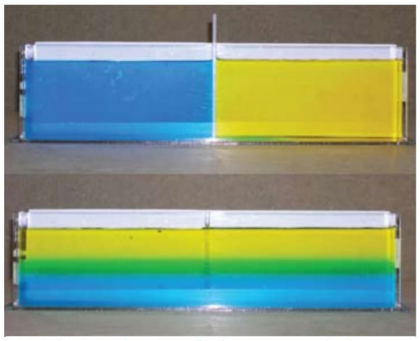
Method:

Part 1 - Salinity:

1. With the divider placed in the tank, fill one side of the container with the fresh water.
2. Fill the other side of the tank with the prepared salty water.
   Note: You may need to shake the salt-water container to make sure the solution remains mixed before adding it to the tank.
3. Add a few drops of food colouring to each side of the tank.
4. Calculate the approximate density of both water masses.
5. Remove the tank divider (try not to disturb the tank as you do this).

Part 2 - Temperature:

1. Empty the tank chamber and put the divider back between the two sections.
2. Fill one side of the tank with warm tap water.
3. Fill the other side of the tank with water cooled by ice (do not pour any ice into the tank).
4. Add a few drops of food colouring to each side of the tank.
5. Remove the tank divider (try not to disturb the tank as you do this).



Tank before (top) and after removal of divider (bottom).

Discuss:

- How might the effects of climate change, such as warming and melting of sea ice, affect the vertical structure of the water column?

Sourced from: https://aquarius.oceansciences.org/docs/hands_on_temp_salinity_stratification.pdf

Suggested practical:

- A wave tank experiment

◊ Shoreline erosion

Aim: To investigate the effect of waves on the shoreline (erosion).

Materials:

- 9x13
- Sand
- Water
- Water bottle

Method:

1. Place sand on one side of the pan. Make it a steep slope
2. Pour water into the other side, until it's halfway up your sand slope.
3. Place your water bottle on the end with the water. Push the bottle down to create small waves. Do this, consistently and evenly for 1 whole minute. Write down what you observe.
4. Push again with your water bottle for another minute. You may make your waves bigger, or keep them the same size. Make observations.

Done by: Chris Pacey, St Patrick's College Townsville

Suggested practical:

- ♦ A beach profile/ dune transect (using sand sifts)

◊ Beach profile

Aim: To investigate the shape of the beach using distance and angle measurements.

Materials:

- 2 ranging poles
- Clinometer
- Tape measure

Method:

1. Follow a straight transect line from the edge of the sea to the end of the active beach. Split the line into segments where the slope angle changes. Each reading is taken from break of slope to break of slope.

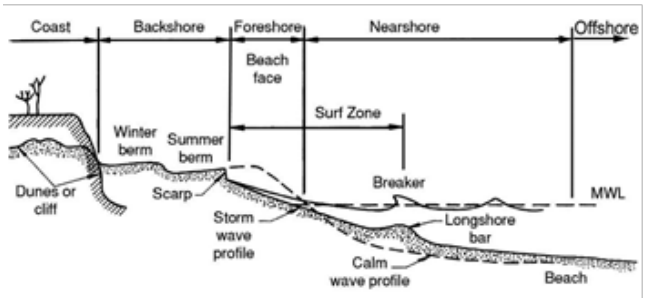
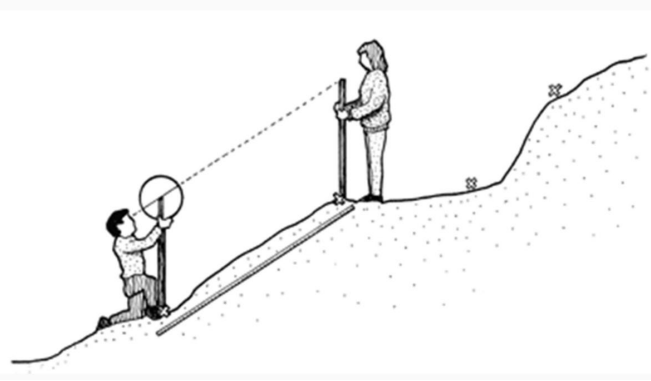


Figure 8 Schematic typical beach profile, terminology and zonation (after Sorensen, 2006)

2. Person A stands at a safe distance from the edge of the sea holding a ranging pole
3. Person B stands holding a second ranging pole further up the beach where there is a break of slope
4. The distance between the two ranging poles is measured using a tape measure
5. The angle between matching markers on each ranging pole is measured using a clinometer



6. Repeat this process at each break of slope until the top of the beach is reached.

Sourced from: https://www.geography-fieldwork.org/a-level/coasts/coastal-management/method/#:~:text=5.-,Groyne%20measurements,one%20side%20of%20the%20groyne
Done by: Nika Langham, Pioneer SHS Mackay

◊ Coarse sediment analysis (pebbles & cobbles)

Aim: To determine the size and shape of course sediments on the beach.

calipers

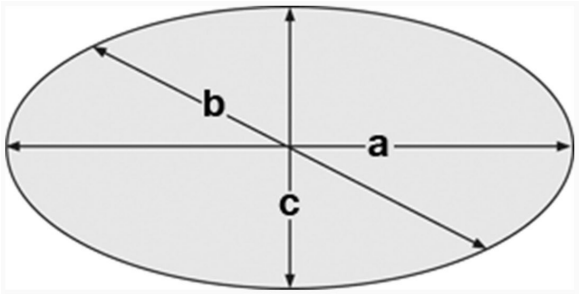
Materials:

- Rule or calipers
- Protractor or concentric circle card

Method:

Sampling can be random, systematic and/or stratified.

1. To measure the size of the coarse sediment- use calipers to measure *a*, *b* and *c* axes of each pebble.



a-axis is the longest axis,

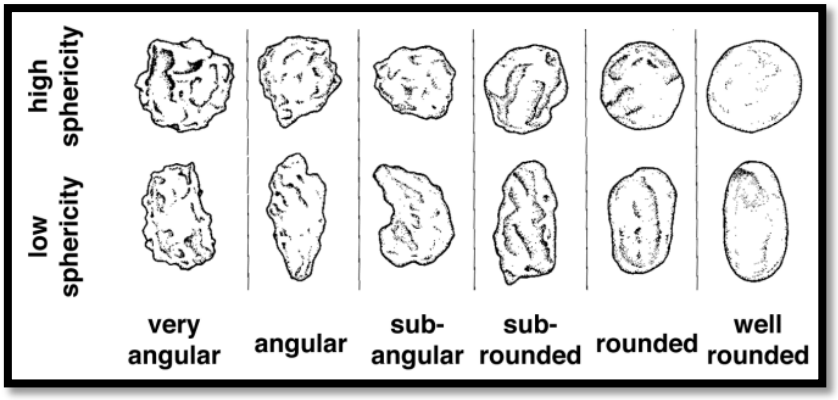
b-axis is the widest axis at right angles to the a-axis,

c-axis is the shortest axis

2. Calculate mean size of coarse sediment for each sample site on the beach:

$$\text{Mean size} = \frac{a+b+c}{3} \text{ for each pebble}$$

3. Measure the minimum radius of curvature (*r*). This is the sharpest corner on the a-axis. This will be needed for the calculation of the Cailleux's Roundness Index.
4. To determine the shape of the coarse sediment by the eye. The simplest way to record pebble shape is to classify the stone as very angular, angular, sub-angular, sub-rounded, rounded or very rounded using a Power's Scale of Roundness.



Power's Scale of Roundness

5. To determine the shape of the course sediment using measurements & calculations.
   The analysis of the shape of coarse sediments can be divided into 4 categories: **shape, sphericity, flatness and roundness.**
   The raw data needed for each pebble are the lengths of the *a*, *b* and *c* axes.

   Zingg's shape classification
   Calculate the ratio *b*÷*a* for each pebble.
   Calculate the ratio *c*÷*b* for each pebble.

   Now classify each pebble into one of the four groups shown in the table

Type of pebble	$b \div a$	$c \div b$
Sphere	> 0.67	> 0.67
Disc	> 0.67	< 0.67
Rod	< 0.67	> 0.67
Blade	< 0.67	< 0.67

   Krumbein's Index of Sphericity
   To calculate Krumbein's Index for each stone/pebble:

$$\text{Krumbein's Index} = \left(\frac{bc}{a^2}\right)^{\frac{1}{3}}$$

   Krumbein's Index (K) K must be between 0 and 1. K = 1 for a perfectly spherical pebble. The lower that K is, the less spherical the pebble.

   Cailleux's Flatness Index
   TO calculate Cailleux's Flatness Index for each stone/pebble:

$$\text{Flatness Index} = \frac{a+b}{2c} \times 100$$

   A perfectly equidimensional particle will have a Flatness Index of 100 and will increase infinitely as it become flatter.

   Cailleux's Roundness Index
   The raw data needed for each pebble are:
   the length of the longest axis (*l*) (previously defined as *a*)
   the radius of curvature of the sharpest angle (r)
   To calculate Cailleux's Index for each stone/pebble:

$$\text{Roundness Index} = \frac{2r}{l} \times 1000$$

   Roundness Index =1000 for a perfectly spherical pebble. The lower the Roundness Index is, the more angular the pebble.
   Cailleux's Roundness Index may be presented using box and whisker plots.

Sourced from:
https://www.geography-fieldwork.org/a-level/coasts/coastal-management/method/#primary-nav
https://www.geography-fieldwork.org/a-level/coasts/low-energy-coasts/data-analysis/#primary-nav

◊ Fine sediment analysis (clay, silt & sand)

Aim: To determine the size and shape of course sediments on the beach.

Materials:

- Graduated sieves
- Protractor or concentric circle card
- Scales

Method:

Samples of fine sediments (clay, silt and sand) will need to be taken from the beach to the lab. Each sample should be placed in a sealed plastic bag and accurately labelled.

Use a set of graduated sieves can be used to sort sediment samples into different size categories (in millimetres or as phi sizes).

1. The sieves are arranged in decreasing mesh diameter with the largest at the top.
2. Place the sediment sample in the top sieve then shake the sieves to sort the sediment into the various sieves.
3. Measure the mass of sediment in each sieve using scales.
4. Calculate phi sizes using conversion table

Sediment size	
mm	**phi**
1.00	0
0.50	1
0.25	2
0.13	3
0.06	4
0.03	5
0.01	6

5. Calculate the percentage mass of sediment in each phi size category.
   For example, if total mass=100g and the mass of material at 5-10mm = 20g, then 20% of the total mass of sediment is 5-10mm in diameter.
6. Present the data in a graphical form.
   This can be done in a number of ways:
   - a histogram with % mass on the y axis and sediment size on the x-axis,
   - a pie charts to show changes along the transect, which might be overlaid on a map or aerial photograph,
   - a scatter graph to show how mean sediment size varies with distance along the beach (see below).

7. Alternatively, use semi-logarithmic graph paper to plot a cumulative frequency graph of phi against mass. Plot phi size on the linear x-axis. Plot the cumulative mass of sediment on the logarithmic y-axis. On your finished graph, find the phi size values at 16% and 84% cumulative mass.
   Use these figures in the following formula

$$\frac{\text{phi at } 84\% \text{ mass} - \text{phi at } 16\% \text{ mass}}{2}$$

   Use the following table to interpret the result

result	interpretation
0.35	very well sorted
0.35 - 0.5	well sorted
0.5 - 0.7	moderately well sorted
0.7 - 1.0	moderately sorted
1.0 - 2.0	poorly sorted
2.0 - 4.0	very poorly sorted
4.0	extremely poorly sorted

Sourced from:
https://www.geography-fieldwork.org/a-level/coasts/coastal-management/method/#primary-nav
https://www.geography-fieldwork.org/a-level/coasts/low-energy-coasts/data-analysis/#primary-nav

Mandatory practical:

♦ Water quality tests

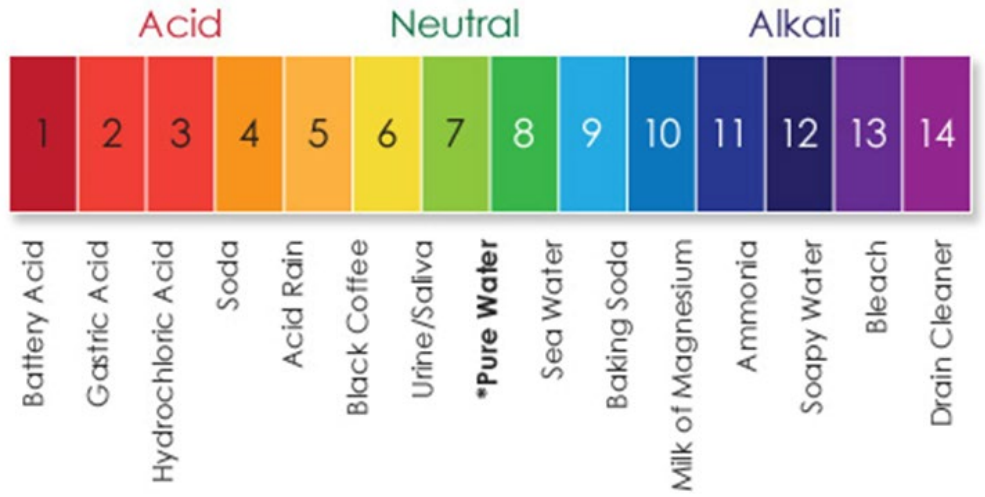
For each test, you will determine water quality index manually. You can access online calculator where you can input your measurement and your water quality score will be calculated.

Online calculator for the water quality score can be found at
http://home.eng.iastate.edu/~dslutz/dmrwqn/water_quality_index_calc.htm

◊ pH

Aim: To determine the quality of water by measuring pH.

Reason for testing:
pH measures hydrogen ions in the water and indicates if the water is alkaline (>7), neutral (7) or acidic (<7). This level of solubility tells what nutrients are available to aquatic life. Metals tend to become more toxic at a lower pH because they are more soluble.
Natural waters will have a pH of approximately 7. Sea water has an average pH of 6.5-8.5. At extreme high or low pH ranges, organism diversity decreases.



Materials:

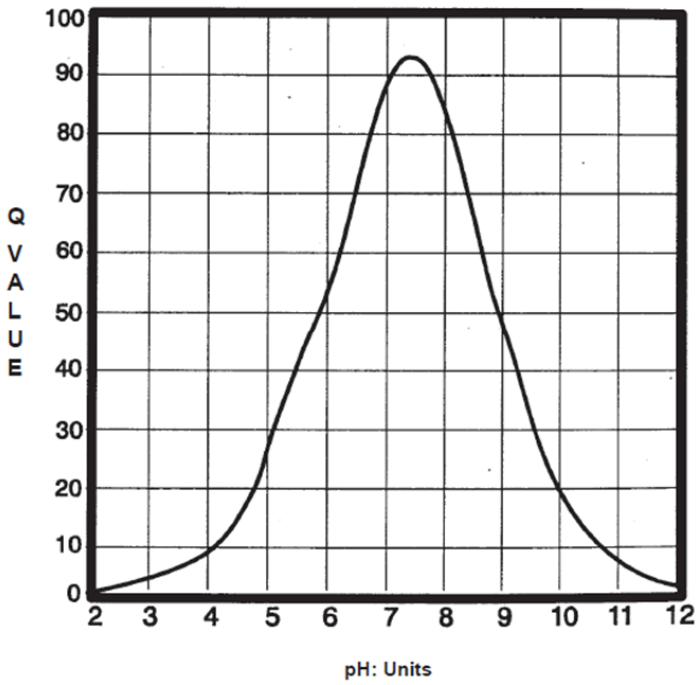
- Xylem YSI multimeter

Method:

Ensure the Xylem YSI multimeter probe has been calibrated.

1. Place the probe below the surface of the water.
2. Slowly swirl the probe under the surface of the water, being careful to ensure it is fully submerged at all times. Record the pH for your site.
3. Compare the result to the pH scale.
4. Record this into results table.
5. Use the Q-Value chart below to determine the Q-Value. Record.
6. Multiply Q-Value by the weighted factor and record your weighted Q-value in results table.

Unit	pH	
Quality	< 5.5	poor
	5.5 – 6.5	average
	6.5 – 8.0	good
	8.1 – 8.5	average
	>8 .6	poor



pH Q-Values

pH (units)	Q-Value
<2	0
2	2
3	4
4	8
5	24
6	55
7	90
7.2	92
7.5	93 (max)
7.7	90
8	82
8.5	67
9	47
10	19
11	7
12	2
>12	0

Results table:

pH	**Q-value**	**Weight factor**	**Weighted Q-value (Q-value x Weight factor)**	**Water quality**
		0.11		

Source: original?

https://www.flinnsci.com/api/library/Download/9e6902d6199442498fd167e4a386588b

Done by: Joline Lalime, Mackay North SHS

◊ Temperature Change

Aim: To determine the quality of water by measuring temperature change.

Reason for testing:
Water temperature has a direct link with toxic absorption, salinity and dissolved oxygen. Some organisms cannot reproduce if water is not at the correct temperature, for example, coral polyps reproduce well in a narrow temperature range above 26° C. Altitude and proximity to coasts will influence natural water temperature e.g.: water in the Snowy Mountains may be 3° C, but this is its natural state, not necessarily poor quality. Keep this in mind when measuring. The ranges provided below are a guide.
The temperature of water affects the way people use water and the abilities of aquatic organism to live, grow, and reproduce. Temperature is a catalyst, a depressant, an activator, a restrictor, a stimulator, a controller, and a killer, and it is one of the most important and influential water quality characteristics for life in water.
Since nearly all aquatic organisms are "cold-blooded," the temperature of the water regulates their metabolism and ability to survive and reproduce effectively. Temperature affects the ability of water to hold dissolved oxygen (see the section on dissolved oxygen for more information). The temperature of the water regulates the species composition and activity of aquatic life in any location.
Each species has a favoured range of water temperatures for its various life processes. Although aquatic organisms can become acclimated to slow, natural changes in water temperature, rapid unnatural changes, such as those caused by industrial thermal pollution or large cool water releases from dams, can be lethal for them, even in the temperature range that organisms can tolerate under natural conditions. Thus, for aquatic life, the rate and amount of change in temperature is as important as the present temperature of the water.

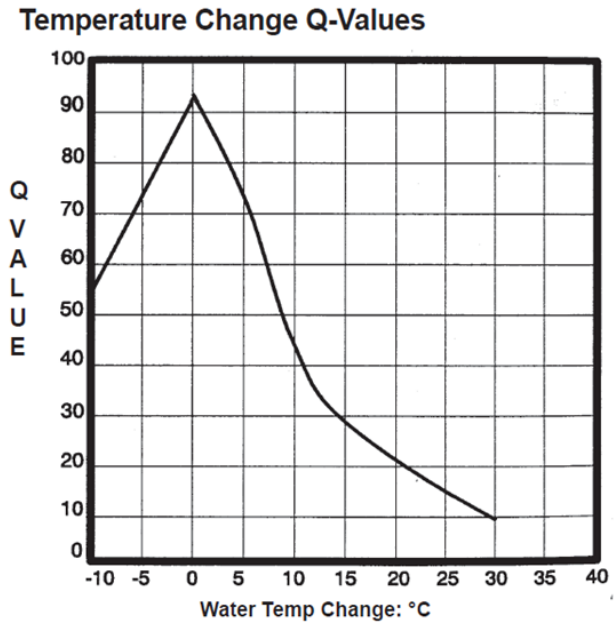
Materials:

- Xylem YSI multimeter

Method:
Ensure the Xylem YSI multimeter probe has been calibrated.

1. Place the probe below the surface of the water (approx. 200mm). Slowly swirl the probe under the surface of the water, being careful to ensure it is fully submerged at all times. Record the temperature for your site (T_1). Record.
2. To find the temperature difference; take a second temperature reading at 1 meter below the surface (T_2). Record. Calculate the temperature difference and record in the results table.
3. Now use the Q-Value chart below to determine the Q-Value. Record this on your data table and multiply by the weighted factor and record your weighted Q-value. Record.

Unit	Degrees Celsius (°C)	
Quality	0 – 9 °C	poor
	10 – 14 °C	fair
	15 – 25 °C	good
	26 – 36 °C	fair
	>37 °C	poor



Change in Temp. (°C)	Q-Value
-10	56
-7.5	63
-5	73
-2.5	85
-1	90
0	93 (max)
1	89
2.5	85
5	72
7.5	57
10	44
12.5	36
15	28
17.5	23
20	21
22.5	18
25	15
27.5	12
30	10

Results table:

T_1	T_2	Temperature difference	Q-value	Weight factor	Weighted Q-value (Q-value x Weight factor)	Water quality
				0.10		

Source: original?
https://www.flinnsci.com/api/library/Download/9e6902d6199442498fd167e4a386588b
Done by: Joline Lalime, Mackay North SHS

◊ Nitrates (NO_3^{2-})

Aim: To determine the quality of water by measuring nitrates.

Reason for testing:
All living things need nitrogen to build proteins. High levels in water lead to plant growth. Some die and sink to the bottom, decompose and use up oxygen in the process, eventually killing fish and insects. Ammonia is a form of nitrogen, a waste product of humans and other animals, which can be toxic to fish.
Nitrate (NO_3^{2-}) is an ion that is formed by the complete oxidation of ammonium ions (NH_4^+) or nitrite ions (NO_2^-) by microorganisms in the soil or water; the nitrite ion is an intermediate product of this process. Nitrate is one of the seven molecular forms that are important in the nitrogen cycle. These seven forms are: molecular nitrogen gas (N_2), nitrous oxide (NO_2), organic nitrogen in amino and amine groups, ammonia or the ammonium ion (NO_3^{2-} or NH_4^+), nitrite, and nitrate. Nitrogen is essential to life on earth, as it is essential for plant growth and development. Nitrogen gas makes up 78% of the air we breathe, although our source for useful nitrogen comes from the food we eat. Nitrogen, in the various forms listed above, passes from the air to the soil, to all living things, and then back into the air through the process called the nitrogen cycle.
While nitrate itself is normally not toxic to humans, and fish can tolerate low levels of nitrate, nitrite is quite toxic to most aquatic animals and to humans. Where there are high levels of nitrate, there are probably high levels of nitrite. Nitrate is easier to measure, so it is used as a marker that may indicate the need for additional testing for nitrite. A high nitrate concentration may also signal the presence of pathogenic bacterial contamination.

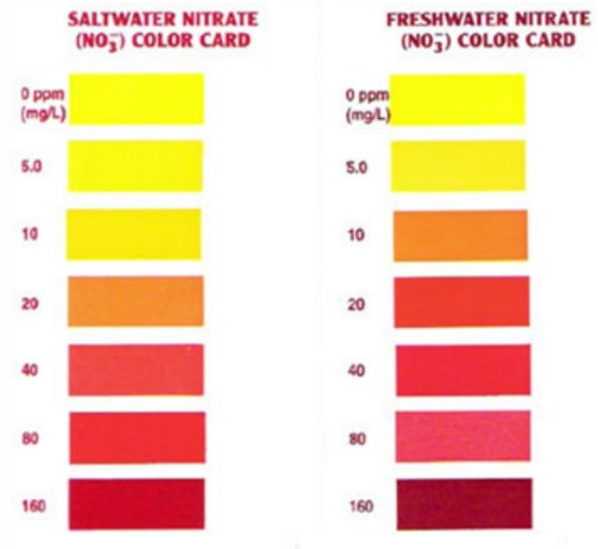
Materials:

- API Nitrate test kit

Method:
Using an API Nitrate Test Kit. Directions:

1. Fill a clean test tube with 5 ml of water to be tested (to the line on the tube).
2. Add 10 drops from Nitrate Test Solution Bottle #1, holding dropper bottle upside down in a completely vertical position to assure uniformity of drops.
3. Cap the test tube and invert tube several times to mix solution.
4. Vigorously shake the Nitrate Test Solution Bottle #2 for at least 30 seconds. This step is extremely important to insure accuracy of test results.
5. Now add 10 drops from Nitrate Test Solution Bottle #2, holding dropper bottle upside down in a completely vertical position to assure uniformity of drops.
6. Cap the test tube and shake vigorously for 1 minute. This step is extremely important to insure accuracy of test results.
7. Wait 5 minutes for the colour to develop.

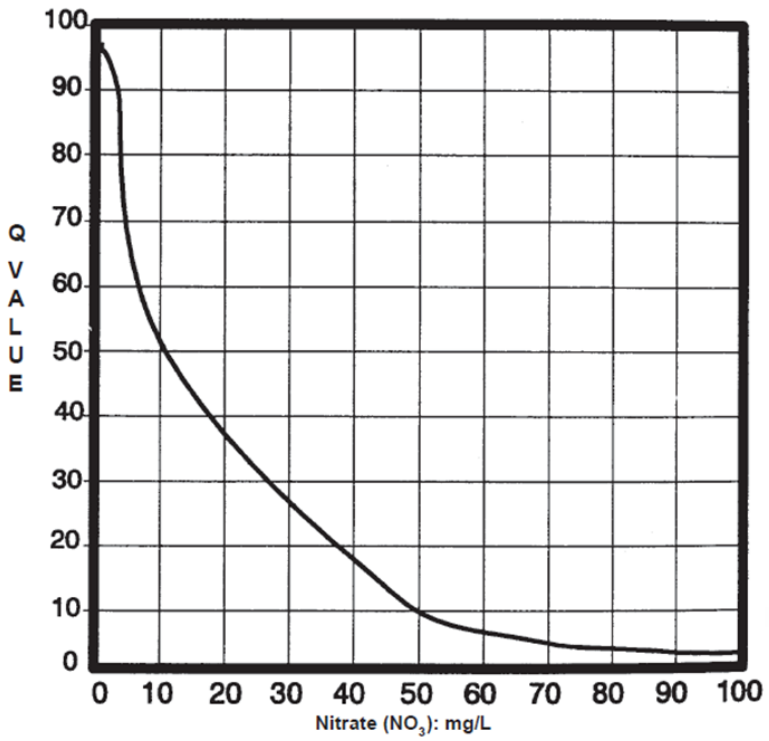
8. Read the test results by comparing the colour of the solution to the appropriate Nitrate Colour Card (choose either Fresh water or Salt water). The tube should be viewed in a well-lit area against the white area of the card. The closest match indicates the ppm (mg/L) of nitrate in the water sample.



Rinse the test tube with clean water after use.

Unit	mg/L	
Quality	< 0.4	Poor
	< 0.2	Fair
	< 0.1	Good
	< 0.05	Excellent

Nitrate (NO_3): Q-Values



Nitrate-N (mg/L NO_3-N)	Q-Value
0	98
0.25	97
0.5	96
0.75	95
1	94
1.5	92
2	90
3	85
4	70
5	65
10	51
15	43
20	37
30	24
40	17
50	7
60	5
70	4
80	3
90	2
100	1
>100	1

Results table:

Nitrates (ppm)	Q-value	Weight factor	Weighted Q-value (Q-value x Weight factor)	Water quality
		0.10		

Source: original?
https://www.flinnsci.com/api/library/Download/9e6902d6199442498fd167e4a386588b
Done by: Joline Lalime, Mackay North SHS

◊ Phosphates (PO_4^{3-})

Aim: To determine the quality of water by measuring phosphates.

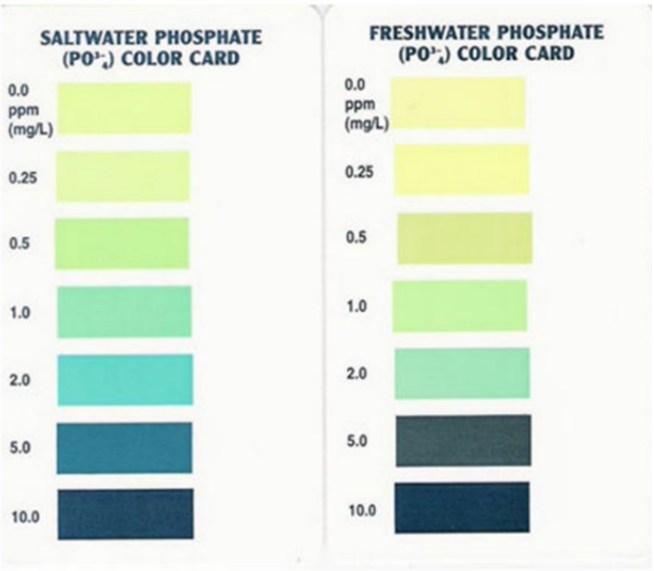
Reason for testing:
All plants and animals need phosphate but too much can cause eutrophication, e.g. algal blooms. It occurs naturally in minerals and rocks. The concentration reflects particular land uses and human activities. Soil erosion and fertilisers increase the amount of phosphate in the water.
Phosphate is generally the rate-limiting eutrophication reagent. That is, in most circumstances, of all the nutrients required for freshwater plant growth, phosphate is the one that is used up first. Therefore, when there is no more phosphate, plant growth stops. Conversely, when phosphate is present, plant growth is stimulated. The presence of excessive amounts of phosphate stimulates excessive plant growth.
Excessive plant growth in aquatic environments stimulated by excessive nutrient content, called accelerated or cultural eutrophication, interferes with water use and is considered a nuisance. Algal growths impart undesirable tastes and odours to the water, interfere with water treatment, become aesthetically unpleasant, alter the chemistry of the water supply, and contribute to the phenomenon of accelerated or cultural eutrophication. Eutrophication often leads to anoxic conditions that occur when plant material is broken down by bacteria, and the dissolved oxygen is consumed during bacterial respiration. Most of the eutrophication problems are associated with lakes or reservoirs, rather than streams and rivers.

Materials:

- API Phosphate test kit

Method:
Using an API Phosphate Test Kit. Directions:

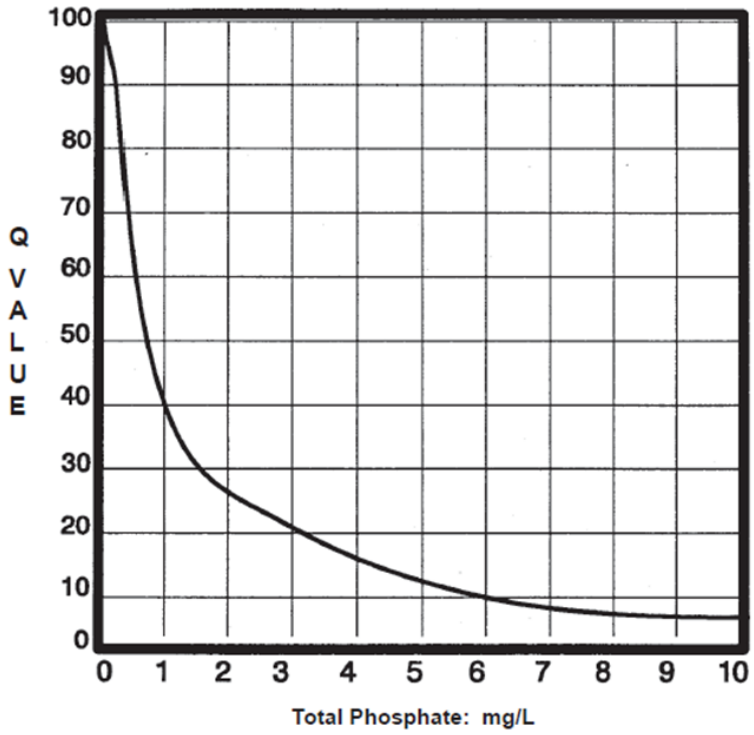
1. Rinse a clean test tube with water being tested.
2. Fill the test tube with 5 ml of water to be tested (to the line on the tube).
3. Holding the bottle vertically add 6 drops from Phosphate Test Solution Bottle # 1. Cap the test tube and shake vigorously for 5 seconds.
4. Now holding the bottle vertically add 6 drops from Phosphate Test Solution Bottle #2. Note: Bottle # 2 contains a very thick solution and may require increased pressure to release drops.
5. Cap and shake the test tube vigorously for 5 seconds.
6. Wait 3 minutes for colour to develop.
7. Read the test results by comparing the colour of the solution to the appropriate Phosphate Colour Card (choose either Fresh water or Salt water). The tube should be viewed in a well-lit area against the white area of the card. The closest match indicates the ppm (mg/L) of total phosphate (PO_4^{3-}) in the water sample.



Rinse the test tube with clean water after use.

Unit	mg/L	
Quality	< 0.15	Poor
	0.05 – 0.15	Fair
	0.02 – 0.05	Good
	< 0.02	Excellent

The Q-value curve for phosphate that is used in the calculation of a water quality index illustrates that water quality declines dramatically as the concentration of phosphate increases.



Total Phosphate (mg/L P)	Q-Value
0	99
0.05	98
0.1	97
0.2	95
0.3	90
0.4	78
0.5	60
0.75	50
1	39
1.5	30
2	26
3	21
4	16
5	12
6	10
7	8
8	7
9	6
10	5
>10	2

Results table:

Total phosphates (ppm)	Q-value	Weight factor	Weighted Q-value (Q-value x Weight factor)	Water quality
		0.10		

Source: original?
https://www.flinnsci.com/api/library/Download/9e6902d6199442498fd167e4a386588b
Done by: Joline Lalime, Mackay North SHS

◊ Total dissolved solids (salinity)

Aim: To determine the quality of water by measuring salinity.

Reason for testing:
Salinity measurement is one of the most important factors in determining the types of plants or animals that can live in or near a body of water. The effects of salinity are felt strongly in the agricultural industry, where the proper landscape irrigation is dependent upon water quality. If the salinity content of irrigation water is too high, salts build up in the soil and starve root systems of water. Plants affected by highly saline irrigation waters will exhibit dwarfing and yellowing of leaves or may lose leaves altogether.
The salt content of bodies of water dictates the organisms that live in that body of water. Many aquatic plants and animals can tolerate a wide range of salinities but often prefer a specific salinity concentration in which to reproduce.

Materials:

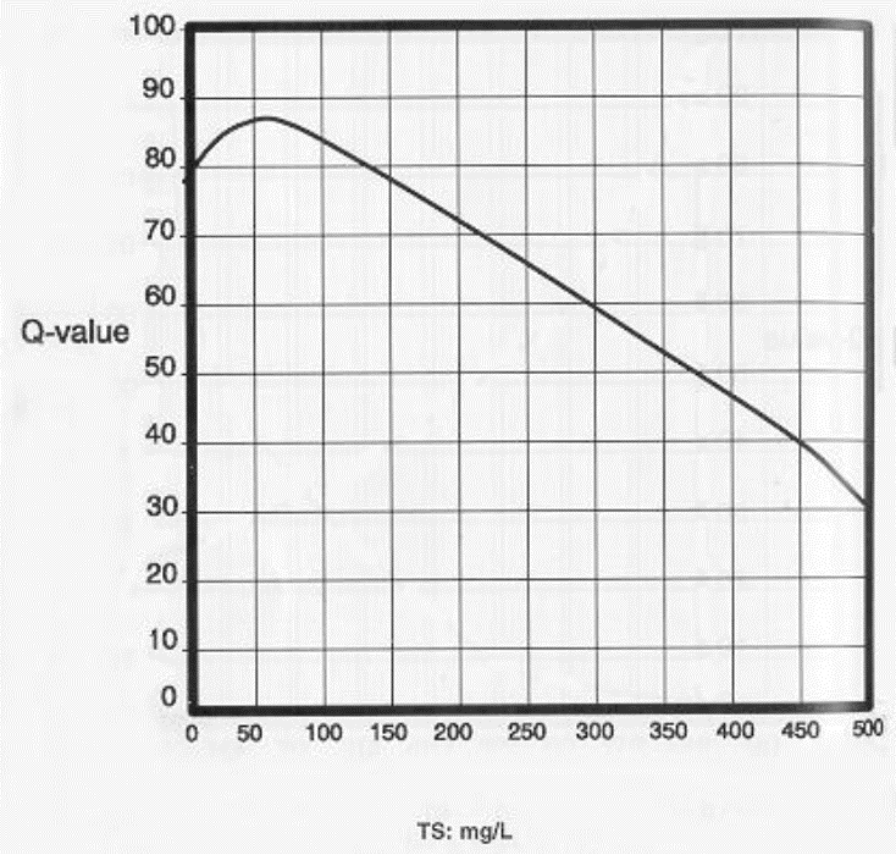
- Xylem YSI multimeter

Method:
Ensure the Xylem YSI multimeter probe has been calibrated.

1. Place the probe below the surface of the water.
2. Slowly swirl the probe under the surface of the water, being careful to ensure it is fully submerged at all times. Record the TDS for your site.
3. Now use the Q-Value chart below to determine the Q-Value. Record this in your data table.
4. Multiply Q-value by the weighted factor and record your weighted Q-value.

Unit	Micro siemens (μS) abbreviated to electrical conductivity (EC)	
Quality	0 – 100	Excellent (30 = rainfall)
	500	Fair
	>750	Poor (840 = sewage effluent)
	1600	Upper limit for drinking
	5000	Upper limit for crops
	8000	Upper limit for livestock
	50 000	Seawater

Generally, as the level of total dissolved solids increases, the Q-value declines and water quality diminishes. The only exception to this curve is when TDS is very low (less than 50 mg/L). A small amount of dissolved nutrients in a water body is healthy for cell development and function in organisms.



Note: 1 ppm = approximately 1 mg/L (also written as mg/L) in water

Results table:

TDS	Q-value	Weight factor	Weighted Q-value (Q-value x Weight factor)	Water quality
		0.10		

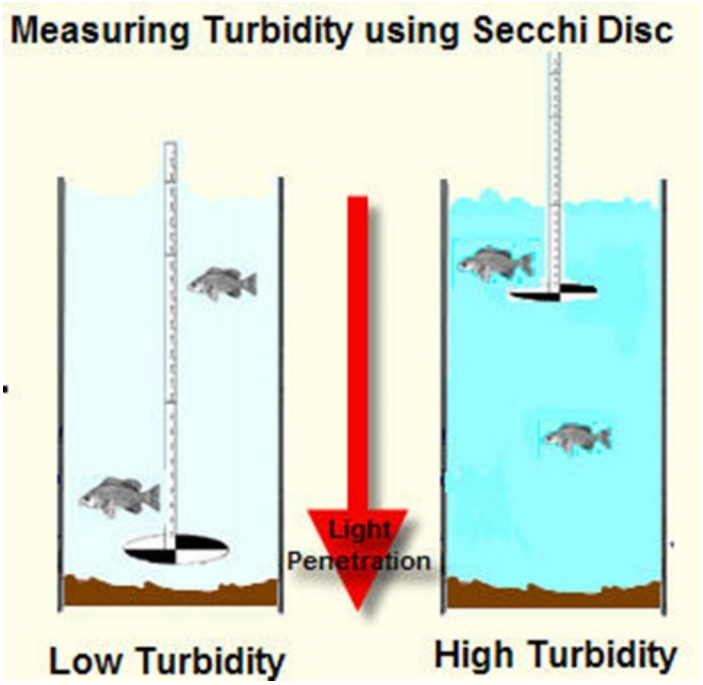
Source: original?
https://www.flinnsci.com/api/library/Download/9e6902d6199442498fd167e4a386588b

Done by: Joline Lalime, Mackay North SHS

◊ Turbidity

Aim: To determine the quality of water by measuring turbidity

Reason for testing:
Turbidity is one of the most common water quality problems. It causes significant problems, no matter what the designated use. Turbidity is regulated under the EPA's Primary Drinking Water Standards, and it is of such concern that daily measurement is required for treated drinking water. Turbidity indicates a possible presence of contamination with pathogenic organisms. Turbidity resulting from eutrophication, or the presence of high levels of nutrients such as nitrates, may progress to anoxic water that supports the growth of anaerobic bacteria, with the resultant release of noxious or malodorous gases. Turbidity blocks the penetration of sunlight through the water, resulting in reduced plant and algae growth and reduced dissolved oxygen concentrations and food sources for fish and other organisms. The large amounts of silt and clay in some turbid water settle on the bottom, blocking bottom-dwelling organisms, eggs, and developing embryos and larvae from essential dissolved oxygen, causing their death. The suspended solids can clog or damage fish gills, suffocating them.
Turbid water absorbs solar radiation, causing increased temperatures in the surface water and the formation of a thermal layer, with the reduced mixing in the water column. This causes lower levels of dissolved oxygen at the bottom of the water column.



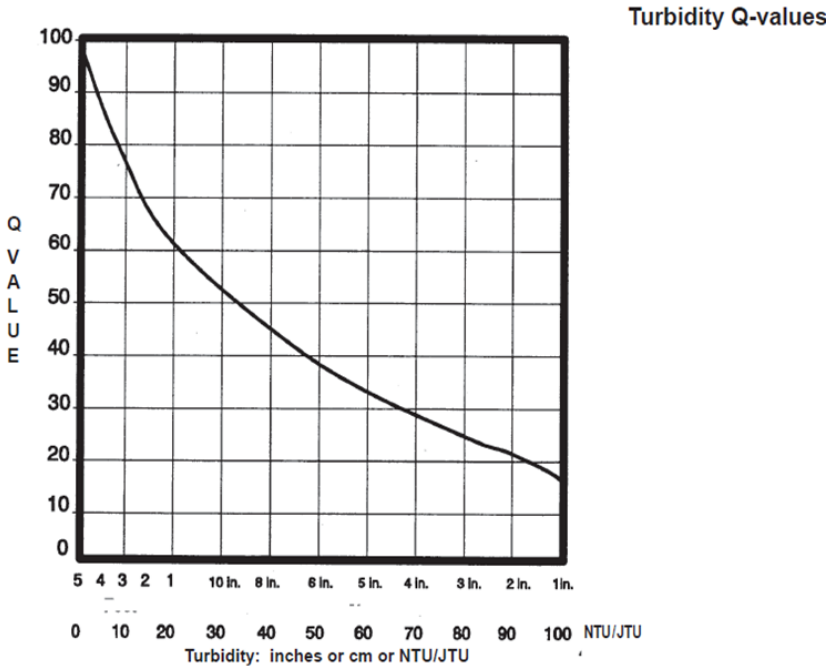
Materials:

- Secchi disc

Method:

1. Lower the turbidity disk into the water until it disappears. Take care with the disc so that it does not bend out with current (add weight if current is a problem).
2. Note the depth (D_1). Lower the disc even further, and raise it upwards. Note the depth when you first see the disk (D_2).
3. Take an average of D_1&D_2 ($\frac{D_1+D_2}{2}$). Record.
4. Use the Q-Value chart below to determine the Q-Value. Record this on your data table in Column B and multiply by the weighted factor and record your weighted Q-value in results table.

Unit	Nephelometric Turbidity Units (NTU)	
Quality	< 10	good
	11 - 29	fair
	> 30	poor



Results table:

D_1	D_2	Average depth ($\frac{D_1+D_2}{2}$)	Q-value	Weight factor	Weighted Q-value (Q-value x Weight factor)	Water quality
				0.08		

Source: original?
https://www.flinnsci.com/api/library/Download/9e6902d6199442498fd167e4a386588b
Done by: Joline Lalime, Mackay North SHS

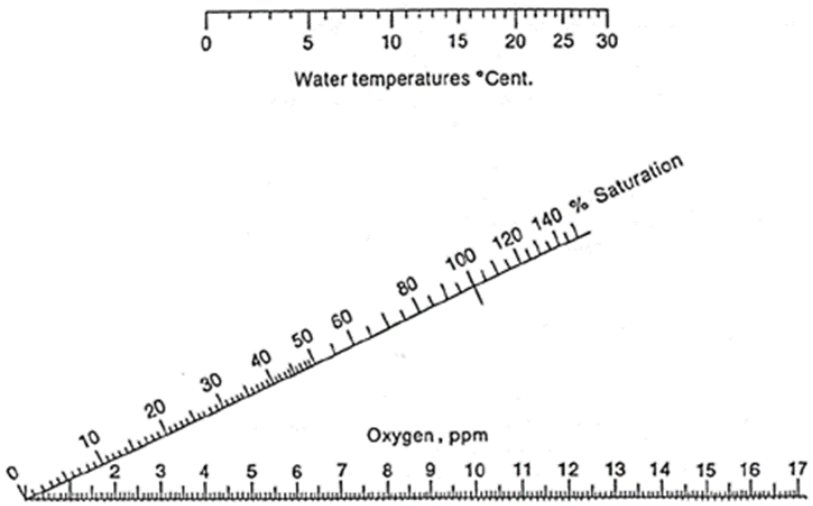
◊ Dissolved Oxygen (DO)

Aim: To determine the quality of water by measuring dissolved oxygen in water.

Reason for testing:
Dissolved oxygen (DO) is oxygen gas (O_2) that is dissolved in water. Dissolved oxygen is a critical factor determining water quality, affecting the growth and health of aquatic communities and the adequacy of water to be used for drinking and municipal purposes.
Aquatic plants and animals need oxygen, except that it is dissolved in water. Fish 'breathe' in dissolved oxygen through their gills. Organic waste and higher water temperatures cause dissolved oxygen levels to fall. High levels of dissolved oxygen mean a greater variety of living things can survive. Most aquatic organisms require dissolved oxygen to live, with some organisms requiring higher concentrations than others.
The presence of DO prevents the occurrence of anaerobic conditions that support the growth of anaerobic bacteria, which produce noxious or malodorous gases, such as methane (CH_4) or hydrogen sulphide (H_2S).

Materials:
- DO probe (Xylem YSI multimeter)
- Thermometer

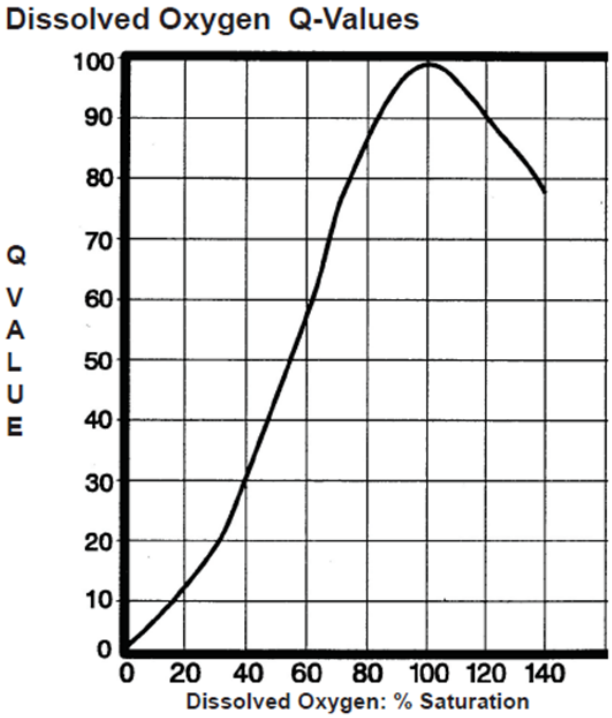
Method:
1. Ensure the Xylem YSI multimeter probe has been calibrated.
2. Place the probe below the surface of the water (approx. 200mm). Slowly swirl the probe under the surface of the water, being careful to ensure it is fully submerged at all times. Record the DO for your site.
3. When you have established the DO concentration in mg/L, measure the water temperature and use the "level of saturation" chart below to determine the percentage saturation of dissolved oxygen. Record this into Column A of your results table.



4. Use the Q-Value chart below to determine the Q-Value. Record this on your data table
5. Now multiply Q-Value by the weighted factor and record your weighted Q-value in results table.

Unit	% saturation	
Quality	0.0 – 4.0	Good
	4.1 – 7.9	Fair
	8.0 – 12.0	Poor
	>12	Check results (too high)

The Q-value curve for DO that was established by the National Sanitation Foundation for their water quality index determination procedure illustrates that water that is significantly supersaturated with DO is as undesirable as water that low in DO concentration. The higher the Q-value, the better the water quality.



DO (% Saturation)	Q-Value
0	0
10	8
20	13
30	20
40	30
50	43
60	56
70	77
80	88
85	92
90	95
95	97.5
100	99
105	98
110	95
120	90
130	85
140	78
>140	50

Results table:

% saturation	Q-value	Weight factor	Weighted Q-value (Q-value x Weight factor)	Water quality
		0.17		

Source: original?
https://www.flinnsci.com/api/library/Download/9e6902d6199442498fd167e4a386588b
Done by: Joline Lalime, Mackay North SHS

◊ Biological oxygen demand (BOD)

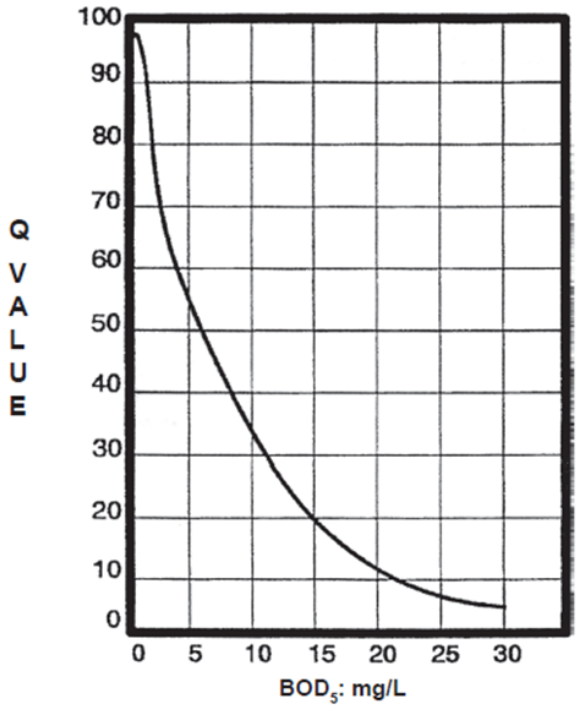
Aim: To determine biological oxygen demand (BOD).

Reason for testing:
Biological oxygen demand (BOD), also known as biochemical oxygen demand, is a bioassay procedure that measures the dissolved oxygen (DO) consumed by bacteria from the decomposition of organic matter. The BOD analysis is an attempt to simulate by a laboratory test the effect that organic material in a water body will have on the DO in that water body.
Biochemical oxygen demand values are a measure of food for naturally occurring microorganisms or, in other words, a measure of the concentration of biodegradable organic material. When nutrients are introduced, naturally occurring microorganisms begin to multiply at an exponential rate, resulting in the reduction of DO in the water. The test does not determine the total amount of oxygen demand present, since many compounds are not oxidized by microorganisms under conditions of the test.
Biological oxygen demand is an important water quality parameter because it greatly influences the concentration of DO that will be in the water. The Q-value curve for BOD used in calculating a water quality index 2 illustrates the relationship between BOD and water quality—the higher the BOD, the lower the water quality, with 0 to 2 mg/L being associated with high water quality and values greater than 10 mg/L being associated with low water quality.
The BOD test is used to measure the potential of wastewater and other waters to deplete the oxygen level of receiving waters. The test is also used to examine influents and effluents from wastewater processing facilities to compute the efficiency of operation of the treatment units.

Materials:
- Bottle to collect water sample
- Dissolved oxygen probe (Xylem YSI multimeter)
- Glass flask with lid
- Aluminium foil
- Marker pen

Method:
1. Collect a water sample in a bottle.
2. Obtain a dissolved oxygen reading for your water sample using the Xylem YSI multimeter probe. Record this reading.
3. Fill glass flask with your water sample to over flowing.
4. Place lid on flask.
5. Wrap flask in aluminium foil.
6. Label flask with your name and area.
7. Place flask in cupboard for five days.
8. After five days remove flask and obtain dissolve oxygen reading using the Xylem YSI multimeter probe. Record this reading.
9. Determine the BOD by subtracting final dissolved oxygen reading from the initial reading.

Unit	ppm or mg/L	
Quality	<2.5	Very good
	<4	Good
	<6	Fairly good
	<8	Fair
	>15	Poor



BOD5 Q-Values

BOD 5 (mg/L DO)	Q-Value
0	96
1	92
2	80
2.5	73
3	66
4	58
5	55
7.5	44
8	40
10	33
12.5	26
15	20
17.5	16
20	14
22.5	10
25	8
27.5	6
30	5
>30	2

Results table:

BOD5 (ppm)	Q-value	Weight factor	Weighted Q-value (Q-value x Weight factor)	Water quality
		0.11		

Source: original?
https://www.flinnsci.com/api/library/Download/9e6902d6199442498fd167e4a386588b
Done by: Joline Lalime, Mackay North SHS

◊ Faecal bacteria

Aim: To determine

Reason for testing:
Faecal coliforms are bacteria derived from the faeces of humans and other warm-blooded animals. These bacteria can enter rivers through direct discharge from mammals or birds, from agricultural or storm runoff carrying bird or mammal wastes or sewage discharge into the water. These are microscopic bacterial which in themselves are harmless, but signify that there may be other harmful bacterial present such as those causing gastroenteritis, dysentery, typhoid fever, hepatitis and/or outer ear infections. The standards for these bacteria are summarised below. The faecal coliform count is also used as an indicator of water quality as shown.

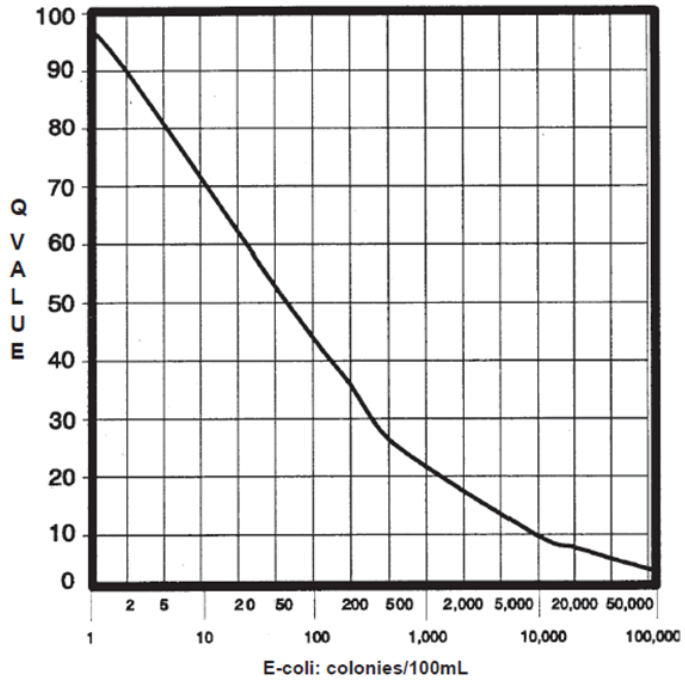
Faecal Coliforms	Safe Colony Numbers
Drinking Water	1/100mL
Swimming/Surfing	200/100mL
Boating/Fishing	1000/100mL
Treated Sewage effluent	No more than 200/100mL

Materials:
- Sterile water bottles for water samples
- Pre-prepared Petri dishes
- Incubator

Method:
1. It is a good idea to wear gloves and avoid interfering with the sample.
2. Water samples will be taken on site in sterile bottles. Remember to label your bottle with the site number.
3. When you get back to the lab, add 1mL of your water sample to specially prepared petri dishes which facilitate the growth of the bacteria. Full instructions below.
4. Once the water has been added to the petri dish, tape the dish closed.
5. Place the petri dishes into an incubator at 35°C.

Unit	Colony forming units (CFU) / 100mL	
Quality	< 1	Very good – i.e.: for drinking
	1 – 35	Good - Primary contact e.g.: swimming
	35 – 230	Fair - Secondary contact e.g.: fishing, boating
	< 700	Poor – no contact

E. Coli Q-Values



E.Coli (colonies/100mL)	Q-Value
0-1	98
2	89
5	80
10	71
20	63
50	53
100	45
200	37
500	27
1,000	22
2,000	18
5,000	13
10,000	10
20,000	8
50,000	5
100,000	3
>100,000	2

Results table:

E-Coli (col/100mL)	Q-value	Weight factor	Weighted Q-value (Q-value x Weight factor)	Water quality
		0.16		

Source: original?
https://www.flinnsci.com/api/library/Download/9e6902d6199442498fd167e4a386588b
Done by: Joline Lalime, Mackay North SHS

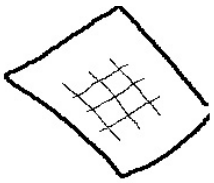
Faecal Coliform Guide

Easygel kits are an easy way to test for bacteria in water. They can be used by folks at home, students, teachers, businesses and professionals. There are two types of kits; Coliscan Easygel for E coli and coliforms and, Total count Easygel for total aerobic bacteria. These test the 3 groups used by the Australian Drinking Water Guidelines to assess water quality.

Not only do these kits detect the bacteria most critical for water quality but they show actual colonies. So, by adjusting for sample size, the actual numbers of colonies for a given volume can be calculated so you know exactly what levels of bacteria are present in your water. Kits come with background information describing the technology, full printed instructions and colour guide. Here is the basic step by step process for using the kits.

1. Thaw the media bottle in a little warm water. Remember bacteria are very small so it's a good idea to wash your hands before getting a sample. Work in a clean spot. Don't touch anywhere on the kits where it might contaminate the sample.
2. Collect a sample in the sterile bottle. To avoid getting any unwanted bacteria keep fingers away from the neck of the bottle and hold the cap in one hand. You can also gently dip the bottle into a dam or spring.
3. Use the sterile dropper to transfer a sample into the thawed media bottle. Use up to 5 ml for fairly clean e.g. town water, but use less, say 2.5 ml for a rainwater tank or protected creek or spring and 1 ml for an unprotected water supply e.g. a dam or creek.
4. Recap the media bottle and gently swirl the bottle to mix the sample. Take a Petri dish that comes in the kit. Lift the lid (the wider part) a little then pour in the media. Put the lid on and leave the dish undisturbed for 45 - 60 mins.
5. Carefully tilt the dish to check if the media has set. If so then tip it upside down (wider part on the bottom). This is to stop condensation from dripping onto the growth media surface.
6. Place the dish somewhere warm and dark such as a kitchen cupboard. Around 25 to 28 deg C will give good growth. In winter or in a cool climate look for a warm spot in the house. A heated seedling raiser makes a good cheap incubator. Keep the dishes covered or away from strong light.
7. Some colonies may appear from 20 - 24 hrs. Count the colonies before 48 hrs. Use the colour guide to identify colonies. To help with the count slowly move a ruler behind the dish and count from side to side. The instructions will tell you how to work out how many bacteria per 100 ml of sample water. If there are more colonies that can be easily counted then you may have to use the grid method. See Appendix 2. Colonies on the Total count media are pale or colourless.

How to count bacteria colonies



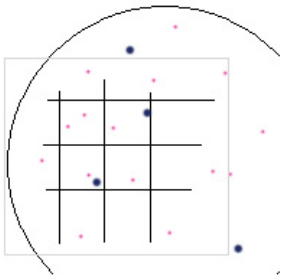
The Easygel method makes estimating bacteria in water or other sources very easy. It is especially good for students and people without special facilities because the method is easy to use and doesn't require any medium preparation or autoclaving etc. The kits are a big advance on the days when testing for bacteria involved the tedious and hazardous process of heating and storing agar medium.

Now its dead easy because with Easygel all that's needed is a bottle of medium, a pre-treated dish (called a plate in Australia), a sterile dropper and a sterile sample bottle. All this comes in the Easygel kits. The method is very cost effective because there is no preparation time and no expensive equipment required.

If there are only a few colonies then all of them can be counted. If you used 2.5 ml of the original sample to add to the Easygel bottle then that is a 2.5/100 fraction of 100mLs, or 1/40th of 100mLs. Colonies are usually reported as Colony Forming Units per 100mLs (CFU's/100 ml). Therefore, to convert your count to CFU's per 100 ml multiply the count by 40.

But what if there are too many colonies to count? Then you have to resort to a little trick. Find a piece of clear acetate sheet, the type that is used for the clear cover of a booklet such as on a bound report. Scratch out a small square cm x cm grid using a ruler and a sharp point. You only need one square but a 2 x 2 grid is better.

Now place the grid over the culture plate, usually from the underside closest to the colonies. Count the number of colonies in each grid. Only include colonies on the edges of the squares if they are more than half way inside the square. Now move the grid to another position. Just move it without looking and leave it there for the next count. Don't deliberately choose areas where there are more colonies - or you will get an artificially high result. Write down each count then average them to get an average per square cm. There are close to 57.4 cm^2 in an Easygel dish so then multiply that average by 57.4. This will give an estimate of the number of colonies per dish. Now, how many mL of sample did you use? Again, if it was 2.5 mL then the multiplying factor is 40. Here is an example. The dark colonies are E coli and the pink colonies are coliforms.



	Grid	E coli	coliforms	
	1	0	2	
	2	1	1	
	3	1	1	
	4	0	1	
	Average	0.5	1.25	per squ. cm
Number per plate		28.7	71.75	
Sample size		2.5	ml	
Multiplying factor		40	(100/2.5)	
Estimate		1148	2870	CFU's / 100ml
		(40*28.7)	(40*71.75)	

The more counts you can make the better. Move the grid randomly each time. In the Lab if we are getting fairly consistent results and there are lots of colonies then we stop at around 10 counts otherwise we go to 15 to 20 counts.

The owner of this water supply probably should not drink the water without treatment!

Notes: You can use this same method for Aerobic plate count, just count the clear to colourless colonies on the Easygel plate after using the APC media. Also, if there are large numbers of colonies on a plate then you may have to draw 1/4 sq. cm grids on the clear acetate sheet and count colonies in each smaller grid. Just multiply by 4 to get one sq. cm counts.

Unit 2

Marine Biology

Source: artPrintbeauty

Suggested practical:

- ♦ Estimate populations, e.g. survey counts, quadrats, species density, percentage coverage, indirect or direct observation, catch and release

Scientists use two common methods to complete a biodiversity study of organisms in one local habitat: plot studies and line transects. A transect is a long, narrow sampling area, while a plot study is a square area that can be divided into subplots. Transects extend over a longer portion of a study area than a plot does, resulting in a larger sample of different plant species. However, plot studies are useful for small areas that have a great variety of plant life.

◊ Estimate populations using line transect technique

Aim: To use a line transect as a method for obtaining a representational survey of a community.

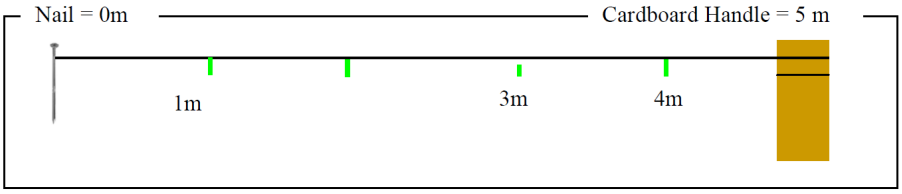
Materials:

- Line transect: 5m of heavy string, large nail, thick cardboard
- Pen/pencil
- Plain paper

Method:

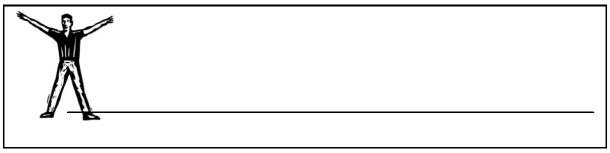
Constructing a line transect:

1. A handle for the line transect can be made from scrap wood or cardboard. Use heavy string for the line and a large nail for the anchor.
2. Measure 5 meters of heavy string, tie it to the handle and wind up.
3. Tie the nail to the other end of the string. Marking the string at every meter with coloured yarn or a marker is helpful for quick measuring outside.



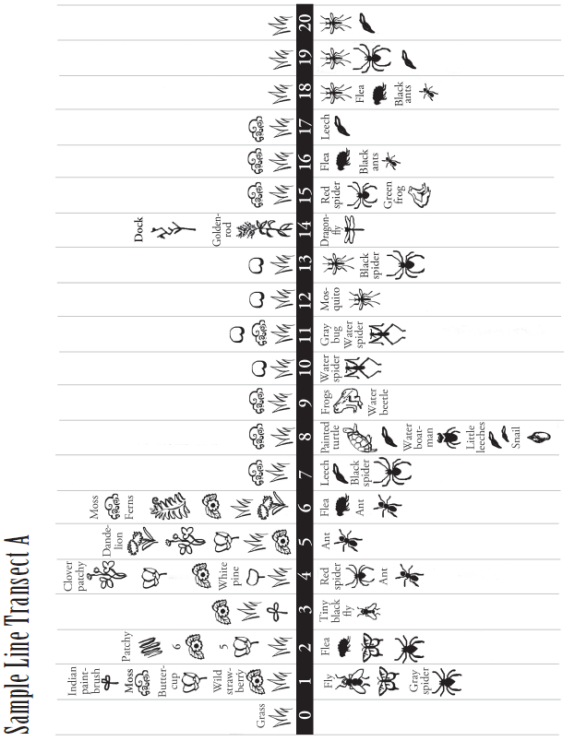
Obtaining data:

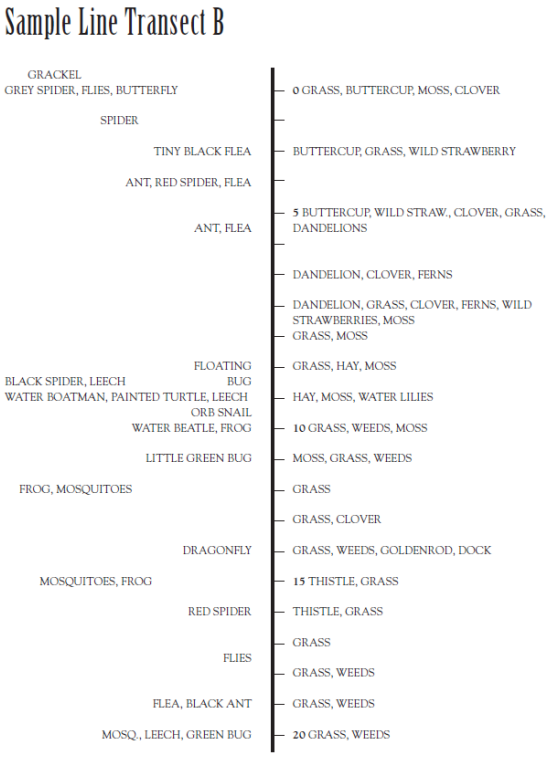
1. The nail is the zero-meter mark of the line transect.
2. Unroll the string and place on the ground. The handle end is the 5-meter mark.
3. Stand at the zero mark, on the line. You can do the transect in either of the following ways:
    a. **Point intercept method**. Record what is directly under each meter or half meter mark on the line. This technique is good for sampling a very large area relatively quickly, but it can miss a lot of information if the area is complex.
    b. **Record all** organisms found within arm's length on either side of the line from the zero-meter mark to the 1-meter mark (then 1-2m, 2-3m, etc).



All items along the line transect should be identified, counted and then mapped. If the species' name is not known for a plant or animal, learners may use more generalized descriptions (i.e. shrub, yellow flower, black beetle, etc.).

Examples of line transect:





Adapted from: A line transect, https://gen.uga.edu/documents/biodiversity/activities/A%20Line%20Transect.pdf
Examples from: Janet Galle and Patricia Warren; Exploring Ecology: 49 Ready-to-Use Activities for Grades 4–8
Extra resource: https://www.hawaii.edu/gk-12/opihi/classroom/measuring.pdf

Done by: Joline Lalime, Mackay North SHS

◊ Estimate populations using line transect technique in intertidal zone

Aim: To use a line transect as a method for obtaining a representational survey of a community.

Materials:

- 50m tape measure
- Pen/pencil
- Plain paper

Method:

1. Lay a 50 metre transect line perpendicular to the shoreline from the low tide mark to the mid tide zone.
2. Record the organism closest (under) the line and the next two closest organisms.
3. Move 5 metres along the line and record three closest organisms.
4. Repeat every 5 metres until the end of transect is reached.

Done by: Joline Lalime, Mackay North SHS

◊ Estimate populations using belt transect technique in mid-intertidal zone

Aim: To use a belt transect as a method for obtaining a representational survey of a community.

Materials:

- 50m tape measure
- Surveyors tape
- 4 pegs
- Pen/pencil
- Plain paper

Method:

1. Choose a suitable area of the mid-intertidal zone for the study site.
2. Measure out a 1 metre and 5 metre box and mark the corners with pegs
3. Observe and record all organisms within the study area
4. Remove pegs and return the equipment.

Done by: Joline Lalime, Mackay North SHS

◊ Estimate populations using quadrats

Aim: To use a belt transect as a method for obtaining a representational survey of a community.

Materials:

- Quadrat (1x1m) (Quadrats can be made out of PVC pipes, wire hangers bent into squares, hula-hoops, wooden dowels or even cardboard. Use monofilament or string to section off the quadrat into a set number of squares or intercepts.)
- Pen/pencil
- Plain paper

Method:
Quadrat sampling can be done at random, on a grid pattern or at set points along a transect line.

At random:

1. In a sample area chosen by your supervisor, place your quadrat at random. You can do this by throwing the quadrat over your shoulder or simply placing it in any location.
2. Count the number of plants present for the nominated plant species and record these numbers on the recording sheet.
3. Repeat the above process until you have covered ten areas using your quadrat.
   Note: Using your quadrat in 10 different areas will equal 10m².
4. On your Recording Sheet add up the total number of plants you recorded for each species. This will give you the population density for 10m².
5. You can then easily calculate the estimated population density for 100m² by multiplying your answer by 10.
6. Calculate the size of population using following formula: $N = \frac{A}{a}n$

   N… estimated size of entire population
   A… area of entire population
   a… area of sample size
   n… number of individuals in the sampling area

Along the transect line:

1. Lay transect line.
2. Place a quadrat along pre-determined points along a transect line.
3. You can record data by using two different techniques:
   a. **Visually estimate** the percent each item (organism/species) takes up within the frame (e.g. 75% species A, 25% species B). The quadrat divisions can also be used to estimate what is in each of the smaller squares (e.g. 12 squares species A, 5 squares species B, 0.5 squares species C, etc.). Organisms that take up very little area can be recorded as <1% or <0.5 squares.
   b. **Point intercept with the quadrat** method. Record what is underneath each intersection within the quadrat frame.

Source:
Janet Galle and Patricia Warren; Exploring Ecology: 49 Ready-to-Use Activities for Grades 4–8
Video link: https://www.youtube.com/watch?v=mDMOIpXVbVM
Extra resource: https://www.hawaii.edu/gk-12/opihi/classroom/measuring.pdf

Done by: Joline Lalime, Mackay North SHS

◊ Estimate population density using quadrat technique in mid-intertidal zone

Aim: To use quadrats to estimate population density.

Materials:

- Quadrat (50cm x 50cm)
- Tape measure
- Field identification guide
- Thermometer
- Anemometer
- Light intensity meter
- Salinity probe
- Pen/pencil & paper OR recording device (Example: Epicollect5 can be set up by the teacher on the computer, students can use Epicollect5 app to record information in the field)

Method:

1. Proceed to the lower intertidal zone (closest to the low water mark).
2. Choose a site with some barnacles and place the quadrat over the habitat.
3. Count each square that has at least one barnacle present in it.
4. Record the total number of squares with barnacle cover.
5. Move to the mid-intertidal zone, repeat steps 2 –4.
6. Move to the upper intertidal zone, repeat steps 2 – 4.
7. Record any qualitative observations relevant to the study.

Done by: Joline Lalime, Mackay North SHS

◊ Estimate population diversity using quadrat technique in intertidal zone

Aim: To use quadrats to obtain data for estimation of population diversity.

Materials:

- Quadrat (50cm x 50cm)
- Tape measure
- Field identification guide
- Thermometer
- Anemometer
- Light intensity meter
- Salinity probe
- Pen/pencil & paper OR recording device (Example: Epicollect5 can be set up by the teacher on the computer, students can use Epicollect5 app to record information in the field)

Method:

1. Proceed to the lower intertidal zone (closest to the low water mark).
2. Choose a random location for the quadrat.
3. Identify different types of organisms visible within the quadrat. Use the identification card to identify unknown species if possible. Record. Photograph any unidentifiable species for later research.
4. Make a tally count of each type of organism. Record.
5. Move the quadrat to another location within the lower intertidal zone.
6. Record low tide zone abiotic data (salinity, temperature, light etc). Record.
7. Repeat steps 3 -5 (total of three quadrat counts in this zone).
8. Move to the mid-intertidal zone, repeat steps 2 –5.
9. Record mid-tide abiotic data (salinity, temperature, light etc). Record.
10. Move to the upper intertidal zone, repeat steps 2 – 5.
11. Record upper intertidal abiotic data (salinity, temperature, light etc). Record.

Done by: Joline Lalime, Mackay North SHS

Suggested practical:

- ♦ Identify to a genus level (using field guides)

◊ Mangrove identification

Aim: To identify mangrove species to genus level.

Materials:

- Video camera provided by MangroveWatch
- Tripod for stability
- Equipment/ Access for land or water (boat) based survey
- Mangrove habitat e.g. river edge

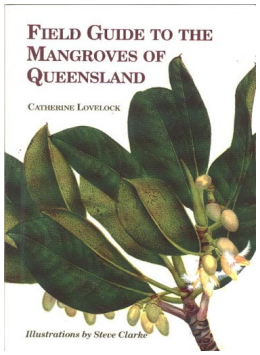
Method:

1. Excursion to an area with at least two mangrove species
2. Use guide to identify genus
   Contact MangroveWatch and be trained in methodology
3. Use the standardised method to assess shoreline mangrove condition and change over time – the Shoreline Video Assessment Method (S-VAM).
4. Conduct geo-tagged video survey and send to MangroveWatch scientific team

Sources: http://mangrovewatch.org.au/

- There are also MangroveWatch Lesson Plans and Activities at COOL AUSTRALIA - https://www.coolaustralia.org/ It requires a FREE account.
- Australian Institute of Marine Science (AIMS): Field guide to the mangroves of Queensland Field guide to the mangroves of Queensland (AIMS)

The authoritative guide to Australia's mangrove plants.
Written by Norm Duke.
Designed by Diana Kleine.
The University of Queensland,
Centre for Marine Studies, Brisbane.
Published by The University of Queensland and Norman C Duke, June 2006
ISBN 0 646 46196 6

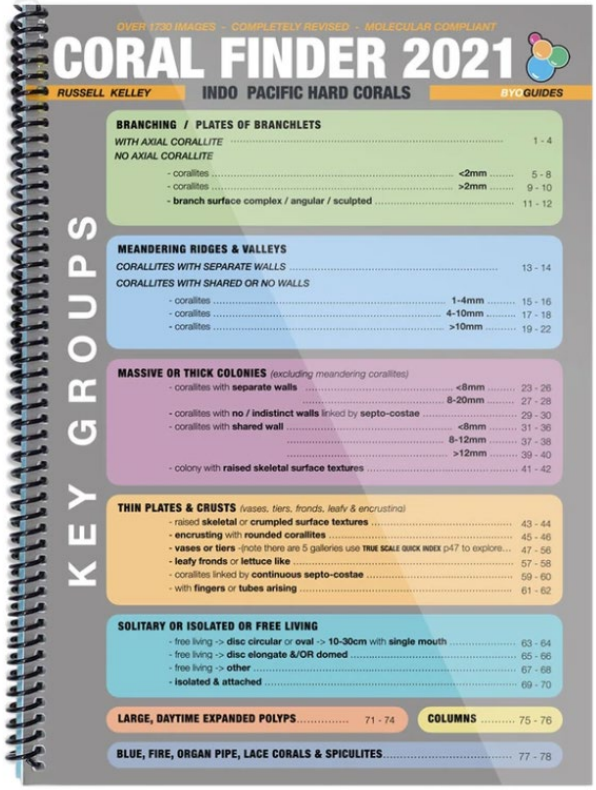


◊ Coral identification

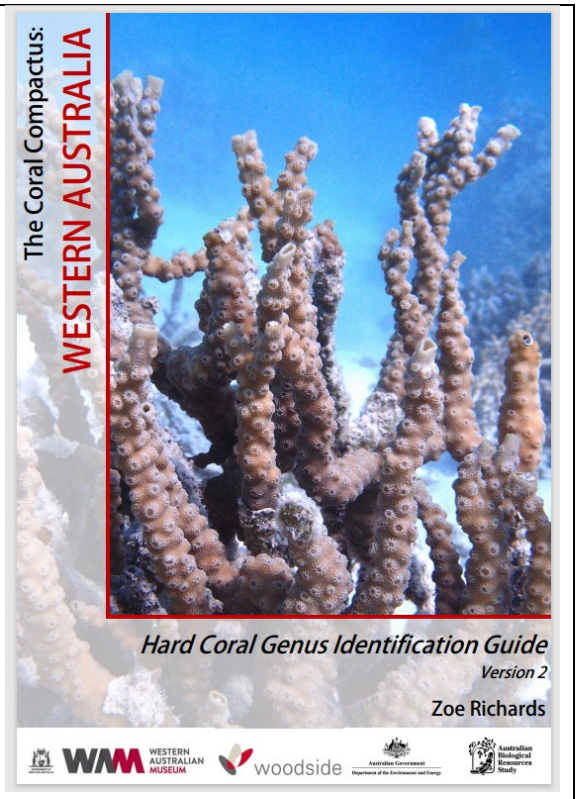
Aim: To identify corals to genus level.

Materials:

- Field guide to corals



https://byoguides.com/



http://museum.wa.gov.au/research/research-areas/aquatic-zoology/wa-coral-field-guides

Method:

1. Excursion to a location with various corals
2. Use guide to identify genus (it is waterproof)

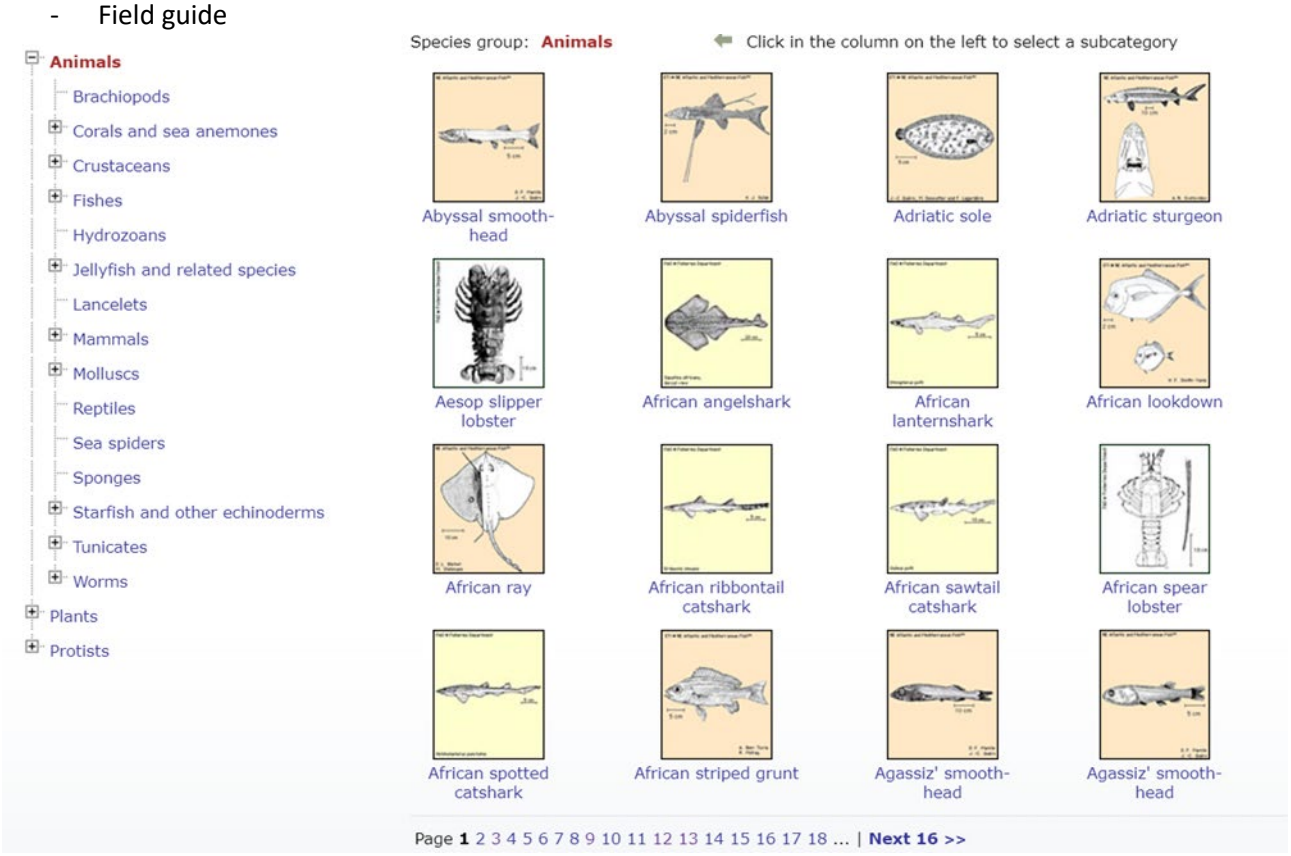
Source: BYO Guides

◊ General marine species identification

Aim: To identify general marine species.

Materials:

- Field guide



Source: http://species-identification.org/

Suggested practical:

- ♦ Measure biotic factors related to marine environments

Aim: Gain an understanding of biotic factors related to marine environments using a range of field equipment. Biotic factors include: include animals, plants, fungi, bacteria, and protists.

Materials:
Similar to population dynamics practical

Method:
Similar to population dynamics practical using quadrats, transects and water sampling with observation of marine invertebrates under microscopes.

Sources: https://stemlyndalesc.weebly.com/how-can-biotic-factors-be-measured.html

https://www.arcgis.com/apps/MapJournal/index.html?appid=2a1f4f01d545432798a498899780e399

Suggested practical:

- In-field mapping of food webs via gut analysis to determine food source

◊

Aim:

Materials:

Method:

Mandatory practical:

♦ Factors of population dynamics

◊

Aim:

Materials:

Method:

Suggested practical:

- Physical structures of a specific marine organism

◊

Aim:

Materials:

Method:

Unit 3

Marine Systems - Connections & Change

Source: Mat Miller, titled *Equilibrium*

Suggested practical:

♦ Identify coral genus

◊ Identifying coral type using Coral Health Chart

Aim: To identify coral types using the Coral Health Chart.

Materials:

- Booties
- Hat, sunscreen and water bottle
- Coral Health Chart
- Waterproof slate or paper with pencil
- Underwater camera (if available)
- Viewing tube (if available)
- Waterproof ID guide (if available)
- ID reference books (if available)

Method:

1. This activity should be conducted at low tide. Check tides timetable and plan this activity accordingly.
2. Because identifying coral species is very difficult, CoralWatch classifies coral types in easy identifiable groups. For this purpose, coral types are described by the overall shape ('growth form') of coral colonies. There are hundreds of different coral species, each with unique shapes and colours. Sometimes the colour or shape may not clearly match the options on the Coral Health Chart. If it is difficult to select a colour or coral type, simply record the closest match. If a particular coral is too difficult to classify, just skip this coral and move on to the next one.
3. CoralWatch classifies most free living, tabular, encrusting and foliaceous corals as plates.
4. CoralWatch classifies most digitate, bushy and staghorn corals as branching.

1. Choose a random coral.
2. Record coral type. Coral types are described by the overall shape (growth form) of coral colonies. Using the Coral Health Chart select the following four coral types or record what you think is the closest.

Boulder (BO)

Any massive or rounded coral, such as some Porites species.

Branching (BR)

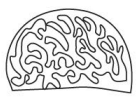
Any branching coral, such as some Acropora species.

Plate (PL)

Any plate-like shape coral, such as tabular Acropora species.

Soft (SO)

Any corals lacking a hard skeleton, such as Sarcophyton species.



The Coral Health Chart uses four coral types to classify corals. Branching refers to any branching coral, such as Acropora and Pocillopora species. Boulder refers to any massive or rounded corals, such as brain corals and some Platygyra and Porites species. Plate refers to any coral that forms a

plate-like shape, such as tabular Acropora species, and the soft category refers to corals lacking a hard skeleton, e.g. Sarcophytum species.

Discuss:

1. Are there mainly soft or hard corals in your local area?
2. In which zone do most corals appear to be?
3. What is the most common coral growth form?
4. What is the most common species of coral? Use your coral ID card.

Sourced from: CoralWatch, The University of Queensland,
https://coralwatch.org/index.php/monitoring/using-the-chart/
https://coralwatch.org/wp-content/uploads/2018/11/Field-worksheet-2_Identifying-coral-type-coral-health-chart_CoralWatch-Moreton-Bay_YR7-science.pdf

Done by: Nika Langham, Pioneer SHS

◊ Coral Classification using photos and identification key

Aim: To classify a specific coral to genus level only, using a relevant identification key

Materials:

- Photos of corals (printed in colour and laminated- for the ease of sharing between students)- Separate pdf file with photos and answers (CoralWatch resource)
- Coral finder guide (Russel Kelly)
- Coral Health Chart

Method:

1. Read through the coral lingo in the Coral finder guide. Familiarise yourself with terms like septa, costa, septacostae, corallite, polys, walls.
2. Choose a coral you want to identify.
3. Choose the key group (branching, meandering, massive, thin, solitary, large polyps, columns, lace corals) on the front page of the coral finder that best describes your call.
4. Answer the text prompts (shared/separate walls, size of corallites e.g.) and select a look-alike page.
5. On the look-a-like page compare the different options. If you can't find your coral, try an alternative key group.

Adapted from: CoralWatch, The University of Queensland, https://coralwatch.org/wp-content/uploads/2019/03/Coral-classification_CoralWatch_Marine-Science_Unit-3_2019.pdf

Other sources:
Coral Finder, Russel Kelley, www.byoguides.com/coralfinder/
Corals of the World, Veron 2000, www.coralsoftheworld.org/page/home/
Hard Coral Genus Identification Guide, Zoe Richards, http://museum.wa.gov.au/kimberley/sites/default/files/WA%20Coral%20CompactusV2_May2018.pdf

Done by: Nika Langham, Pioneer SHS

Suggested practical:

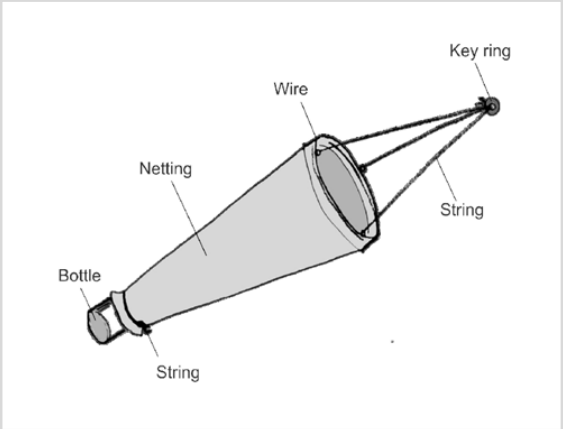
♦ Classify plankton using field work techniques

◊ Catching plankton

Aim: To collect living plankton from the local river or estuary using plankton net.

Materials:

- Plankton net
- Collecting bottle



Method:

3. You'll need to find a body of water and then devise a way to move the net through the water. Usually scientists tow a plankton net behind a boat. Since you probably don't have access to a boat, what are some other ways you might have to move water through the plankton net?
4. If you decide to tow your plankton net, tie a length of strong string to the bridal ring so that you can pull your net through the water. The plankton will become concentrated in the bottle.
5. When you have finished towing the plankton net, rinse the inside of the net with water so that plankton on the inside of the net get washed down into the collection bottle. When you are done, untie the string around the mouth of the bottle and carefully remove the bottle.
6. View the water contents in your collecting bottle. Do you observe particle motion even after the water has stopped moving? If you do, then those are your zooplankton!

Sourced from: https://savetheirl.org/teacher-resources/05_Plankton_TG.pdf

◊ Identifying plankton

Aim: To observe and identify phytoplankton and zooplankton.

Reason for activity:
Plankton provides ideal indicators of ecosystem health and ecological change because it is abundant, short-lived, not harvested, and sensitive to changes in temperature, acidity and nutrients. Plankton has thus been used as indicators for climate change, eutrophication, fisheries, invasive species, ecosystem health and biodiversity.

Materials:

- Compound microscope
- Dissecting microscope
- Plankton sample (see preparation)
- Petri dish
- Microscope slides
- Eye dropper
- Plankton Identification Chart

Method:

1. Use an eye dropper to collect a few drops of the sample and place in a petri dish. Observe the sample with a dissecting microscope. Since the plankton can move up and down in the drop, you will need to refocus your microscope to see plankton at different levels.
2. Many of the organisms are too small to be seen with the dissecting microscope. You can prepare a slide of the sample and observe it under a compound scope with low and high-power objectives. Make sure that you save all of the sample.
3. To prepare the students to distinguish different plankton types, you may wish to discuss how to differentiate ocean plants from animals. Will they have different colours? Structure? Behaviour? You may want to alert them that distinguishing characteristics for land-based plants and animals are usually much different than those for plankton.
4. Observe your sample for the following:
    a. most abundant organisms
    b. variations in shape, colour, and swimming ability
    c. types of appendages seen on various plankton
    d. eggs
    e. larval and juvenile forms of crustaceans and fish
5. Select the most common organisms from your sample. Record the following information on your data sheet:
    a. a detailed drawing of the specimen
    b. identification of your specimen using the Plankton Identification Chart

6. Repeat this procedure with as many different organisms as time permits. Make sure that you have at least:
    a. two different kinds of phytoplankton (plants)
    b. four different kinds of zooplankton (animals)
    c. one diatom, one dinoflagellate, and one form of permanent zooplankton

Adapted from: Kolb, James A., Project Director. Marine Biology and Oceanography Grades 9 - 12. Marine Science Project: FOR SEA. Marine Science Center. Poulsbo, WA. 1986. https://savetheirl.org/teacher-resources/05_Plankton_TG.pdf

Suggested practical:

- ♦ Investigate zooxanthellae using microscope

◊ Investigating zooxanthellae in corals

Aim: To identify and describe the symbiotic relationships in a coral colony (polyp interconnections and zooxanthellae).

Materials:

- Small coral fragment/ anemone tissue (Don't collect from the wild unless you have a permit.)
- 1 pipette (1 ml) and disposable tips
- Cutters
- Airpik or Aaterpik
- Airpik tips
- Small zip lock
- 5ml of PBS or filtered seawater
- Calipers (to measure coral)
- Haemocytometer
- Haemocytometer cover slips
- Microscope
- Pipettes
- Tissues for cleaning

Method:

1. Remove small coral fragment or Anemone tentacle. Don't collect from the wild unless you have a permit.
2. Add 5ml of PBS or seawater into zip lock bag.
3. Place coral fragment into the zip lock bag.
4. Use the airpik/waterpik to clear a surface of the coral (aim for a circle or square so you can estimate the surface area).
5. Make sure you get all of the tissue off the area of coral.
6. If possible complete the following step) Centrifuge the solution to concentrate the algal pellet and remove the excess solution/supernatant from above the pellet. Then resuspend pellet in known volume of seawater /PBS,
7. Measure (if step 6 is not completed) and Mix the pellet solution with a pipette to ensure its thoroughly mixed.
8. Place a cover glass on the haemocytometer (counting chamber) and pipette a known volume at the edge of the cover slip/haemocytometer and allow surface tension and pressure to drawn solution across the measuring area.

9. View the sample under the microscope and count the cells between the double lines in square of 20 divisions (5 squares). Keep to regular square count between samples. For example, use 3 or 5 squares in your counting regime. Clean and repeat 3-5 times with fresh subsamples and take the average of the counts as a measure per volume. If there are cells that are on the double line borderline, and more than their half inside then include these in the count. NB: If there are too many cells in your solution or they are clumped together, then remix and dilute further. Likewise, if there are very few cells then concentrate the main sample.

Sourced from: CoralWatch, The University of Queensland, https://coralwatch.org/wp-content/uploads/2019/03/Symbiosis_CoralWatch_Marine-Science_Unit-3_2019.pdf

◊ Investigating zooxanthellae in anemone

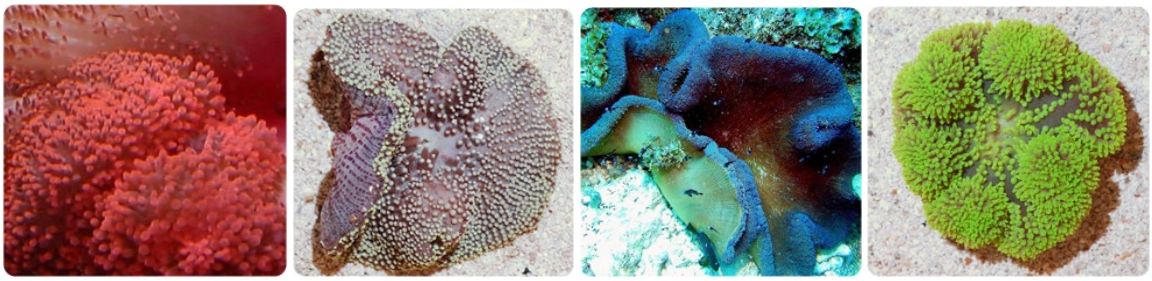
Aim: To investigate the appearance and quantity of zooxanthellae in an anemone.

Materials:

- One anemone tentacle or several small animals (depending on the number of students involved in the experiment)
  You can use anemone that clownfish use (a variety of species, most common Heteractis spp or Stichodactyla spp) or another anemone species (*Aiptasia pulchella*).

Heteractis spp

Heteractis crispa *Heteractis magnifica*



Stichodactyla haddoni *Stichodactyla mertensii* *Stichodactyla gigantea* *Stichodactyla gigantea*

Aiptasia pulchella in aquarium

Aiptasia pulchella in vial

If you have an aquarium you can use the anemone in there or buy one from the shop. Don't collect from the wild unless you have a permit.

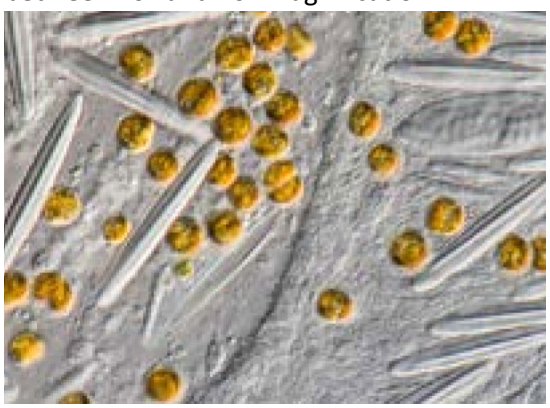
- Magnesium chloride (0.36M)
- 5mL of PBS or filtered seawater
- Scalpel/cutters
- Pipette (1 mL)
- 1 vial
- Microscope
- Slides and cover slips
- Tissues for cleaning

Method:
Depending on species available and amount of people involved in your experiment, decide if you just need one tentacle or several small animals.

The photos of this experiment show *Aiptasia pulchella* species and individual animals were fairly small.

1. To relax the tentacles, use magnesium chloride (FC: 0.36 M).
2. Cut one tentacle and/or placed this together with one small animal in between cover slips.

3. Looking under the microscope zooxanthella and nematocyst should be able to be identified between 10x and 40x magnification.



The golden-brown circles are Symbiodinium (endosymbiotic dinoflagellates).

The elongated light grey cells are stinging cells, nematocyst.

NB: Makes sure no saltwater /PBS comes into contact with the microscope as this will cause corrosion and damage

Sourced from: CoralWatch, The University of Queensland, https://coralwatch.org/wp-content/uploads/2019/03/Symbiosis_CoralWatch_Marine-Science_Unit-3_2019.pdf

Unit 3: Marine systems- connections and change

Suggested practical:

- Coral diversity (using transect technique)

Aim: To investigate the diversity of corals using transect surveys

Materials:

- Snorkel, mask, fins (if snorkelling)
- Booties, hat, sunscreen (if reef-walking)
- Coral Health Chart-
- Waterproof data slate with pencil
- Viewing tube (if available)

Method:

3. If using only one transect, select an area that represents the overall coral cover/diversity. Try not to be bias and select an area with higher or lower diversity.
4. If possible, lay out a tape measure or string with measurements marked on it.
5. To decide what intervals to use on the transect tape you should consider the coral cover in your area. With high coral density you could collect data every half a meter, meter or with low density it might be best to take 2-meter intervals.
6. Swim or walk along the tape, and at every interval (decided in the previous step) choose the closest coral. Record coral type. Aim to collect coral type data for 20 different coral colonies.

Adapted from: CoralWatch, The University of Queensland, https://coralwatch.org/wp-content/uploads/2018/11/Teacher-guide_Measuring-coral-health-using-transect-surveys_CoralWatch-Moreton-Bay_YR7-science.pdf

Done by: Nika Langham, Pioneer SHS

Unit 3: Marine systems- connections and change

Mandatory practical:

- ♦ Connectivity in a habitat: Impact of water quality on reef health

Aim: To investigate the impact of water quality on reef health.

Materials:
This practical is based on collecting the transect data online & measuring water parameters (see Mandatory practical: Water quality tests)

Method:

Determine coral cover for three different reefs:

3. Go to http://globalreefrecord.org/data and choose a reef and transect you will work off.
4. Find quadrats (birds-eye photos) of the selected reef (got to "Explore transect" and click on 'Quadrat").
5. Determine coral cover for each quadrat.
6. Calculate mean coral cover for the reef.
7. Repeat steps 1 to 4 for two more reefs.

Determine the water quality index for three different reefs:

1. Measure as many parameters as possible from the following DO, Fecal Coli, pH, BOD, Temperature, Total phosphate, Nitrate, Turbidity, Total solids
2. Put the data into the online calculator http://home.eng.iastate.edu/~dslutz/dmrwqn/water_quality_index_calc.htm which will calculate your water quality index.
3.

Once you determined the coral cover and water quality index for all three reefs, find if there is a correlation between coral cover and water quality.

1. Use Excel to make a scatter plot.
2. Add the trendline.
3. Use correlation index and coefficient of determination to discuss the relationship between the two variables.

Adapted from: Year 12 Marine science student workbook that can be purchased which can be purchased from http://www.marineeducation.com.au/

Mandatory practical:

♦ Effects an altered ocean pH has on marine carbonate structures

◊ Where did my shell go?

Aim: To investigate the effect of increased CO_2 in the water on marine carbonate structures.

Materials:

- 2 cups of 250 ml of tap water.
- 2 cups of 250 ml plain seltzer (carbonated water) or use Soda Stream
- 2 cups of HCl or white vinegar
- 3 cups with .2 grams of calcium carbonate powder
- 1 cup with 3 small shells from marine organisms
- Petri dishes (to cover the cups with carbonated water) or glad wrap
- Optional: pH meter or pH indicator solution, chalk (not non-dusting)

Method:

1. Fill two cups with 250ml of tap water. Measure and record pH. Cover with a lid.
2. Fill another two cups with carbonated water. (make carbonated water with Soda stream) Measure and record pH. Cover with a lid.
3. Fill another two cups with vinegar (or HCl). Measure and record pH. Cover with a lid.

---

4. Hypothesize; what will happen when calcium carbonate powder is mixed into tap water, into carbonated water, and into vinegar. Write down your hypothesis.
5. Add the calcium carbonate ($CaCO_3$) to the containers with the tap water, carbonated water, and vinegar and then stir. Cover with lids. Make observations for 1-3 minutes and record what you observe.
6. Test pH of the three solutions where calcium carbonate powder was added.

---

7. Weigh and record the mass of all shells. (Make sure the shells are free from dust and dry.)
8. Add one shell to the container with tap water, another to the container with carbonated water, and third to the vinegar. Cover with a Petri dish lids.
9. Be sure to observe and record the differences in all solutions.
10. Review your hypothesis, determine if it was correct, and discuss results.
11. After a week, remove all shells from the solutions, dry and weigh them. Record the weight. Test pH of the solutions.

---

12. Research. Explain how CO_2 makes the ocean more acidic and less alkaline, and what happens to calcium carbonate when in an acidic solution. Explain how acidification affects marine animals that create calcium carbonate to make shells and other skeletal structures. Explain the chemical reactions that take place to make oceans acidic and how acidification dissolves calcium carbonate and even prevents calcium carbonate from forming.

Original experiment from: https://www.rgs.org/CMSPages/GetFile.aspx?nodeguid=b13ca35a-bd22-4b62-8a13-e6fb618ab001&lang=en-GB

Done by: Nika Langham, Pioneer SHS, Mackay

◊ Where did my shell go? (variation 2)

Materials (for each group):

- 1 cup of 250 mL of tap water.
- 1 cup of 250 mL plain seltzer (carbonated water).
- 2 cups with 0.2 grams of calcium carbonate powder.

Optional: heat plate, cups of ice, pH meter or pH indicator solution, chalk (not non-dusting), and shells found on the beach from various marine organisms.

Method:

1. Briefly describe ocean acidification and that it is causing the shells of marine animals--such as corals, snails, clams, and many other creatures-- to dissolve or not even form at all.
2. Provide each group of students with four containers-- one with 250 mL of tap water, one with 250 mL of seltzer water, and two with 0.2 grams of calcium carbonate powder each.
3. Ask students to explain what the seltzer water represents in this experiment about ocean acidification (represents ocean water with an excess of CO_2 from the burning of fossil fuels in Earth's atmosphere) and what the calcium carbonate powder represents (the chemical that makes up the shells and other skeletal structures of marine animals). The tap water represents a control in this experiment.
4. Ask students to hypothesize what they think will happen when calcium carbonate powder is mixed into tap water and into seltzer water. Ask students to write their hypothesis.
5. Have students next add the calcium carbonate ($CaCO_3$) to the containers with the tap water and seltzer water and then stir. Students should make observations for 1-3 minutes and record what they observe.
6. Be sure that students observe and record the differences in the two solutions. (In the tap water, the calcium carbonate will super saturate the water, making the water cloudy; some excess calcium will sink to the bottom. In the seltzer water, the calcium carbonate will dissolve within a few minutes. The solution should go from cloudy to clear; left over calcium will sink to the bottom.)
7. Ask students to review their hypothesis, determine if it was correct, and discuss results.
8. Ask students to research (Internet, chemistry books, or the background given in this lesson plan) how CO_2 makes the ocean more acidic and less alkaline, and what happens to calcium carbonate when in an acidic solution. Students should explain how acidification affects marine animals that create calcium carbonate to make shells and other skeletal structures. Depending on the grade level, ask students to explain the chemical reactions that take place to make oceans acidic and how acidification dissolves calcium carbonate and even prevents calcium carbonate from forming.
9. Optional- Have students test the pH of the two different waters to observe the differences in pH levels before adding the calcium carbonate. Be sure to tell students that seltzer is a super-saturated solution of CO_2 (so much CO_2 is added to water to help create the super fizz people prefer in seltzer). This is the same CO_2 being absorbed by our oceans, making them acidic (an excess of H^+ ions) but so much that the solution is mostly carbonic acid. Students can observe the excess CO_2 gas (bubbles)that escape from the solution.

From: http://njseagrant.org/wp-content/uploads/2014/02/Ocean-Acidification.pdf

Suggested practical:

- CO_2 lowers pH of solution

◊ Gas exchange between ocean and atmosphere

Aim: To investigate the exchange of gases between atmosphere and water.

Materials:

- 3 bottles or flasks of the same size (any material or size will do)
- 3 beakers or drinking glasses large enough for the bottles to fit in
- 1 drinking straw
- 1 small pipette
- universal indicator (McCrumb)
- water: distilled water, tap water or seawater are suitable
- hot water
- ice cubes

Method:

1. Fill the bottles half-full with water. Use either distilled water, tap water or seawater in all three bottles. Put several drops of indicator solution into the bottles with the pipette until the water in the bottles has reached the same shade of green. Be sure to add the same amount of indicator in all bottles. The amount of indicator depends on what water is used and its volume.
2. The air you exhale contains carbon dioxide. Use the drinking straw to blow air into the bottles until the colour changes from green to yellow. This means that the pH of the water has changed from basic to acidic. Keep blowing air through the straw until there is no further colour change.
3. Fill one beaker half-full with water at room temperature, the second beaker half-full with hot water and the last beaker with ice water.
4. Put one uncapped bottle in each beaker and observe the change in colour of the water inside the bottles. Depending on the temperature of your water bath, this may take about an hour until the change becomes visible.

Discuss:

- Did the colour of the water in the bottles change?
- What does the colour change in the bottles mean?

Sourced from: https://www.bioacid.de/wp-content/uploads/2017/04/BIOACID_Experiments_en.pdf
Done by: Brad Lawrence, Science Teacher, Redlands College

◊ Acidification in the boundary layer between ocean and atmosphere

Aim: To investigate the gas exchange at the interface between water and air &the effect of temperature on the exchange.

Materials:

- 6 salad bowls of the same diameter
- Distilled water at room temperature
- Frozen distilled water (ice cubes)
- 12 floating candles or tealight candles, preferably white
- Matches
- White background (paper, napkin, kitchen towel)
- Universal indicator (McCrumb)
- Pipette

To save on materials, the experiment can be carried out in parts one after the other. In this case, only two salad bowls of the same size and four candles are necessary. It is essential to use water which has equilibrated with the surrounding air. Place the water in an open container and leave it to stand in the room overnight, better still outdoors. To speed the process up, the water can be aerated using an aquarium pump.

Method:

1. Place three bowls on top of the white paper. Fill these with the same volume of distilled water. These bowls represent three oceans. Cool one bowl by adding the frozen distilled water (ice cubes). It is important that all three bowls are filled to the same level, about two-thirds. Add the same amount of indicator to each bowl. Make sure that they have the same intensity of green colour.
2. Light eight candles and place four in the bowl with frozen distilled water and four in another bowl. Put the four unlit candles in the third bowl. This will serve as your control. Cover the bowls with the remaining empty bowls. Note the colour of the water at the start of the experiment.
3. Watch how the candles burn out and how the colour of the water in the bowls changes. This will take a few minutes. Look closely at the boundary between air and water. Can you explain why the candles burn out? What gas do the candles release when they burn? If necessary, explain that the candles burn out because they had used up the oxygen trapped in the bowls and that they release carbon dioxide when they burn.

Discuss:

- What colour change did you observe? What does it indicate?
- Where does the colour change take place? Did all the water in the bowl change its colour? What does this imply in relation to the oceans?
- In which bowl was the colour change more visible? What is the reason for this?

Sourced from: https://www.bioacid.de/wp-content/uploads/2017/04/BIOACID_Experiments_en.pdf
Done by: Brad Lawrence, Science Teacher, Redlands College

◊ Acidification of the ocean

Aim: To investigate the diffusion of CO_2 from air into the water.

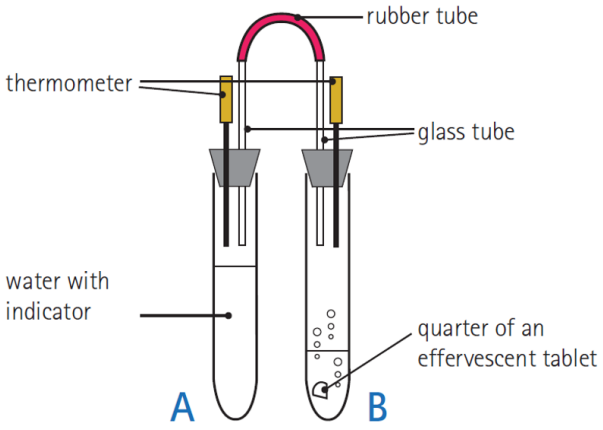
Materials:

- 2 large test tubes
- 2 rubber stoppers with 2 holes for the test tubes
- 2 glass tubes which fit into the holes in the rubber stoppers
- 2 thermometers
- A small amount of modelling clay
- 1 rubber tube
- Test tube rack
- Universal indicator (McCrumb)
- Tap water or seawater, and distilled water at room temperature
- Effervescent vitamin tablets, split into quarters

Method:

1. Insert one glass tube and a thermometer into the holes of each rubber stopper. Connect the two glass tubes in each plug with the rubber tubing.
2. Close the test tubes with the rubber stoppers. Make your setup airtight using modelling clay to plug the air space on the holes of the stopper where you inserted the thermometers and the glass tubes. Note down the temperature of the air inside the test tubes as soon as it has stabilised.
3. Fill one test tube half-full with distilled water and add some drops of indicator until the water is a dark shade of green. Replace the stopper making sure that the thermometer does not touch the water. Place the test tube in the rack. This is test tube A.
4. Put one quarter of an effervescent tablet in the other test tube and add a generous amount of tap water to cover the tablet. Close the test tube immediately with the rubber stopper. Place the test tube in the rack next to the first one. This is test tube B.
5. Note down the temperature of the air inside the two test tubes.
6. Look at the colour of the water with the indicator in test tube A. Do you already notice a change?
7. Put another quarter of the effervescent tablet into test tube B and close it immediately. Wait about 3 to 5 minutes and observe the colour of the water in test tube A again. Do you observe a change now? Keep adding a quarter of an effervescent tablet until the colour changes waiting for at least 5 minutes after each addition. Write down the temperature every time you add another quarter.
8. Repeat the experiment with seawater or tap water.

Set up:



Results:

observation												
addition of effervescent tablets	0		1/4		1/2		3/4		1		1 1/4	
	temp (°C)	colour /pH	temp (°C)	colour /pH	temp (°C)	colour /pH	temp (°C)	colour /pH	temp (°C)	colour /pH	temp (°C)	colour /pH
distilled water												
tap water												
seawater												

Discuss:

- Describe any changes in colour of the water in test tube A after the first quarter of an effervescent tablet was dissolved in test tube B. What happened after adding another quarter? And another? What does the visible change mean?
- What happened to the temperature in the test tubes after the effervescent tablets were added? What does this temperature rise indicate?
- At what temperature does the colour change at the surface of the distilled water become visible? How does this compare to when tap water or seawater is used? Can you explain any differences?

Sourced from: https://www.bioacid.de/wp-content/uploads/2017/04/BIOACID_Experiments_en.pdf
Done by: Brad Lawrence, Science Teacher, Redlands College

◊ Ocean acidification in a cup

Aim: To investigate how the diffusion of a gas into a liquid can cause ocean acidification.

This activity models part of the short-term carbon cycle—specifically the interaction between our atmosphere and the ocean's surface.

Materials:

- Safety goggles
- An acid-base indicator such as bromothymol blue, diluted with water: 8 mL bromothymol blue (0.04% aqueous) to 1 L of water (see the Teaching Tips section below for alternative acid-base indicators, including one made from cabbage juice)
- Two clear 10-oz plastic cups (the tall ones)
- Paper cups, 3-oz size (you'll only use one in the experiment, but keep a few extras at hand just in case)
- Masking tape
- Plain white paper
- Permanent marker
- Baking soda
- White vinegar
- Two Petri dishes to use as lids for the plastic cups
- Graduated cylinder or measuring spoons
- Gram scale or measuring spoons

Method:

1. Put on your safety goggles.
2. Pour 40–50 mL of acid-base indicator solution into each of the two clear plastic cups.
3. Add 1/2 teaspoon (2g) of baking soda to the paper cup.
4. Tape the paper cup inside one of the clear plastic cups containing the indicator solution so that the top of the paper cup is about 1/2 inch (roughly 1 cm) below the top of the plastic cup. Make sure the bottom of the paper cup is not touching the surface of the liquid in the plastic cup—you don't want the paper cut to get wet. The second plastic cup containing indicator solution will be your control.
5. Place both clear plastic cups onto a sheet of white paper and arrange another piece of white paper behind the cups as a backdrop (this makes it easier to see the change).
6. Carefully add 1 teaspoon (about 5-6 mL) of white vinegar to the paper cup containing the baking soda (image below). Be very careful not to spill any vinegar into the indicator solution. Immediately place a Petri dish over the top of each plastic cup.
7. Position yourself so you are at eye level with the surface of the indicator solution and look closely. What do you see? Where is the colour change taking place?

Original experiment from: https://www.exploratorium.edu/snacks/ocean-acidification-in-cup?media=7385

Done by: Nika Langham, Pioneer SHS, Mackay

◊ Increased CO_2 concentration in the air and pH of water

Aim: To investigate (compare) the effects of rising CO_2 levels in the air on pH of seawater and distilled water.

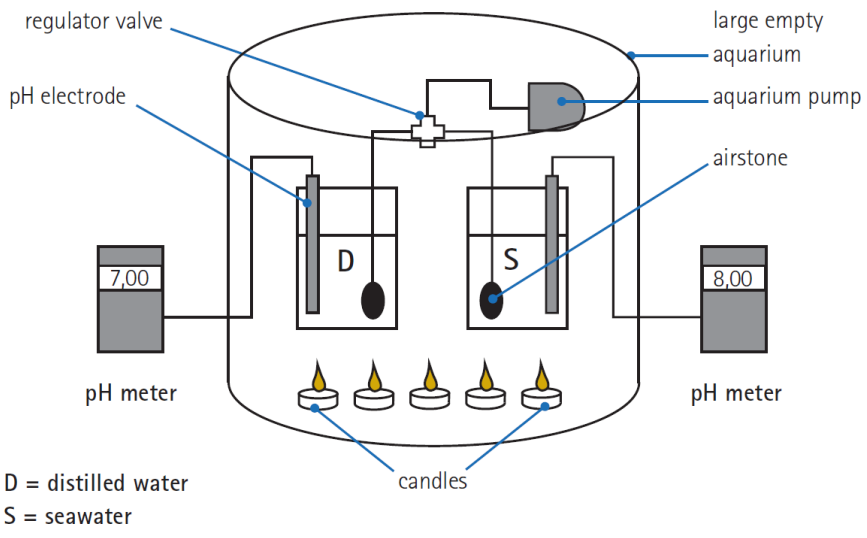
Materials:

- Large transparent basin or a large aquarium
- 2 beakers (500 mL)
- 2 pH meters
- Aquarium pump
- Plastic tubing and 2 air stones with regulator valves
- seawater*, distilled water*
  *Leave the water to stand overnight in an open container to allow it to equilibrate with the surrounding air. Aerating the water with the aquarium pump is also advisable.
- 5 tealight candles
- Matches
- stopwatch

Method:

1. Fill one beaker half-full with seawater and the other half full with distilled water. Aerate the water in the beakers with the air stones connected to the aquarium pump. Regulate the aeration with the valves so that both beakers are bubbled with air to the same degree.
2. Place one pH electrode in each beaker and connect these to the pH meters. Make an initial reading and record this in the table.
3. Light the candles and place them next to the beakers. Place the entire setup under the inverted aquarium. The aquarium pump should also be inside the aquarium. If the aquarium is not big enough, the pH meters can be placed outside.
4. Record the pH of the water in the beakers for 10 minutes at 1-minute intervals. Note down the time when all the candles burn out.
5. Make a graph of the results you recorded in the table.

Set up:

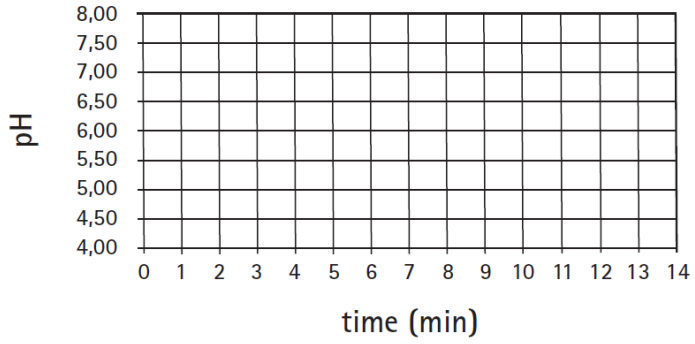


Results:

Change of pH of distilled water and seawater

pH of the water in the beakers		
time (min)	pH	
	seawater	distilled water
0		
1		
2		
3		
4		
5		
6		
7		
8		
9		
10		
11		
12		
13		
14		

Change of pH over time



Discuss:

- In which beaker was there a greater change in pH? Did the water become more basic or acidic?
- Many aquatic organisms cannot tolerate big pH changes in their environment. What do you think will be the effect of a high CO_2 environment to organisms in a lake and in the ocean?
- Why should the water in the beakers be aerated? Why should the pump also be placed inside the inverted aquarium?
- What do the burning candles do to the air inside the aquarium? Where does the oxygen go? What gas do the burning candles produce?

Sourced from: https://www.bioacid.de/wp-content/uploads/2017/04/BIOACID_Experiments_en.pdf
Done by: Brad Lawrence, Science Teacher, Redlands College

Suggested practical:

♦ Changes in temperature and salinity affect the solubility of CO2 in aqueous saline solutions

◊ Temperature and solubility of CO_2

Aim: To investigate how does temperature affect the solubility of CO_2 in water.

Materials:

- 500 mL graduated cylinder
- Funnel
- Petri dish cover
- Transparent basin or an aquarium
- Stand and Clamp
- Ice cubes/ cold water
- Water heater/Warm water
- Effervescent (Fizz) tablet

Method:

1. Fill the basin half-full with cold water. Place the stand beside the basin.
2. Fill the graduated cylinder to the brim with cold water and place it carefully upside down in the basin. Be sure that no water spills out of the cylinder so that no air bubble is formed. To do this, cover the mouth of the full cylinder with a Petri dish. Invert the cylinder and immerse this in the basin. Remove the Petri dish after the mouth of the cylinder is already underwater.
3. Secure the graduated cylinder with the clamp to the stand and place the funnel in the mouth of the cylinder taking care that there is minimal space between the funnel and the opening of the cylinder (Fig. 1). Just holding the graduated cylinder in place can do this. (Fig. 2)

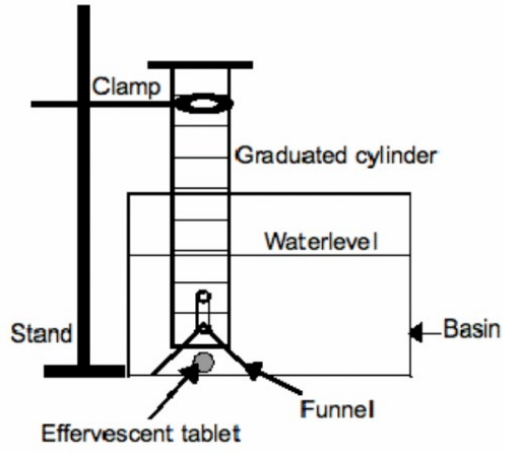


Figure 1. Experimental Set-up

Fig. 2 Hold the graduated cylinder in place

4. Place an effervescent tablet carefully under the funnel. Be sure your hands are dry. The effervescent tablets dissolve fat in water. Observe how an air space develops on top of the upside-down cylinder. Record the volume of the air space formed in the table below. Make several trials.
5. Repeat the same procedure with warm water and record your results in the table. What happens with the air space when warm water is used? (Fig.3)

Fig. 3 The air space formed at the top of the cylinder is smaller when cold water is used (left photo, note the ice cubes). When the water is warm a bigger air space is formed (above)

Sourced from: http://www.carboeurope.org/education/CS_Materials/CO2solubility.pdf
Done by: Joline Lalime, Mackay North SHS

◊ Temperature and solubility of CO_2

Aim: To investigate how does the change of water temperature affect solubility of CO_2 in water.

Materials:

- Large transparent bowl or an aquarium
- 250 mL transparent graduated cylinder
- Funnel
- Petri dish covers
- Stand and clamp (optional)
- Ice cubes / cold water
- Water heater / warm water
- Effervescent tablets

Method:

It is easier to compare the experiment with warm and cold water if you do both experiments simultaneously. Colour the water with food colouring (e.g. blue and red) to distinguish the warm and cold experiments and see the results more effectively.

1. Fill the bowl half-full with cold or warm water.
2. Fill the graduated cylinder to the brim with water from the bowl and place it carefully upside down in the bowl. Be sure that no water spills out of the cylinder so that no air bubbles are formed. To do this, cover the opening of the full cylinder with a petri dish, invert the cylinder and immerse this in the bowl. Remove the petri dish after the mouth of the cylinder is already under water.
3. Tilt the graduated cylinder and place the funnel in the opening of the cylinder taking care that there is minimal space between the funnel and the opening of the cylinder and hold it in place. Eventually, the cylinder should sit safely on the funnel.
4. Place an effervescent tablet carefully but quickly under the funnel. This dissolves immediately in the water. Observe how an air pocket develops at the top of the upside-down cylinder. Record the volume of the air space formed in the table below. Repeat the trial several times. For each trial discard the used water and replace this with the same volume of fresh water.
5. Compare the results you get when using warm and cold water and record your results in the table. What happens with the air space when warm water is used?

Results:

formation of an air space inside the graduated cylinder		
number of effervescent tablets	volume of air space formed (ml)	
trial number	cold water	warm water
1		
2		

Discuss:

- Which produces a larger volume of air space inside the graduated cylinder: cold water or warm water?
- What will be the consequence of a warming ocean? How will this affect the role of the oceans as a CO_2 sink?
- In which areas of the world will you expect more CO_2 uptake into the ocean? Where will it be less?

Sourced from: https://www.bioacid.de/wp-content/uploads/2017/04/BIOACID_Experiments_en.pdf
Done by: Brad Lawrence, Science Teacher, Redlands College

◊ pH regulation in seawater

Aim: To investigate the role of carbonate (CO_3^{2-}) and bicarbonate (HCO_3^-) in regulating pH of seawater.

Materials:

- 3 to 6 bottles of the same size with lids
- Distilled water
- Seawater
- 6 drinking straws
- Sodium carbonate (Na_2CO_3)
- Sodium bicarbonate (baking soda, $NaHCO_3$)
- Universal indicator (McCrumb) with colour chart
- Pipette
- Eggshells (optional)

Method:

1. Fill three bottles with the equal amounts of seawater and the other three with the same amount of distilled water. Add a small pinch of sodium carbonate to one of the bottles containing seawater and to one containing distilled water. Do the same for the sodium bicarbonate.
2. Compare the colours of the water inside the bottles. Do you see any differences?
3. Add 20 drops of the McCrumb indicator solution to each bottle. Determine the pH of the water in each bottle using the colour chart. Can you see any differences now?
4. Insert drinking straws through the openings of the bottles making sure that they are immersed in the water.
5. Blow air through the drinking straw to introduce CO_2 into the bottles. Try to blow uniformly for 20 seconds into each bottle. Replace the lids of the bottles and determine the pH. Record the pH in the table below.
6. Repeat step 5 but instead blow for 40 seconds and 60 seconds into the bottles. Do you notice any differences?

Results:

	estimated pH (see colour chart)				
water samples	initial colour	after addition of indicator	after blowing for 20 seconds	after blowing for 40 seconds	after blowing for 60 seconds
seawater					
seawater with $NaHCO_3$					
seawater with Na_2CO_3					
distilled water					
distilled water with $NaHCO_3$					
distilled water with Na_2CO_3					

Discuss:

- Do you notice any differences in the appearance of the water in the different bottles before the addition of the indicator solution?
- Compare the pH of the water in the different bottles. How did the addition of sodium carbonate and sodium bicarbonate to seawater and distilled water prior to bubbling with CO_2 affect the pH?
- What happens to the pH of the water in the bottles after blowing in CO_2 into them? Does addition of sodium carbonate and sodium bicarbonate to seawater and to distilled water make any difference with regard to pH changes?
- Eggshells are made of $CaCO_3$. Add powdered eggshells to the acidic seawater and to the acidic distilled water. Do you notice any changes in the pH? Why? What is the difference between the carbonate - in the powdered eggshell and in Na_2CO_3?
- Can you now make a conclusion about the roles of bicarbonate and carbonate - in the buffering capacity of seawater?

Sourced from: https://www.bioacid.de/wp-content/uploads/2017/04/BIOACID_Experiments_en.pdf
Done by: Brad Lawrence, Science Teacher, Redlands College

◊ Buffering process of seawater

Part 1: Buffering atmospheric CO_2

Aim: To investigate how seawater and fresh water buffer carbon dioxide gas.

Materials:

- 150 mL seawater
- 150 mL tap water
- pH meter
- Drinking straw
- Stopwatch
- 2 x 250 mL beakers
- 250 mL measuring cylinder

Method:

1. Place 150 mL of tap water in one beaker and 150 mL of seawater into the other.
2. Measure the pH of the tap water using the pH meter and record your reading in the table below.
3. Use the straw to blow continuously into the water for five seconds. It is important to blow at a constant rate.
4. Measure the pH again and record.
5. Blow for another five seconds and record the pH again. The same person should blow each time. Repeat this procedure until you reach a total of 100 seconds of blowing.
6. Now repeat steps 2 to 5 for the seawater, using the same person as you used in the original trial.

Results:

TOTAL TIME BLOWING (s)	pH OF TAP WATER	pH OF SEAWATER	TOTAL TIME BLOWING (s)	pH OF TAP WATER	pH OF SEAWATER
0			55		
5			60		
10			65		
15			70		
20			75		
25			80		
30			85		
35			90		
40			95		
45			100		
50					

Discuss:

- Which of the solutions that you tested was the most effective buffer? Explain your answer.
- One variable difficult to control in this experiment is blowing the carbon dioxide into the water at a constant rate. It is also not a life-like way of exposing the seawater to atmospheric carbon dioxide. Suggest a better method.

Sourced from: https://www.uwa.edu.au/science/-/media/Faculties/Science/Docs/Activity-How-does-the-ocean-buffer.pdf
https://www.uwa.edu.au/study/-/media/Faculties/Science/Docs/Researching-ocean-buffering.pdf
Done by: Jenny Strodl, Marine Science, Science and Maths Teacher, St Peters Lutheran College

Part 2: The role of carbonate and bicarbonate in buffering seawater

Aim: To investigate how seawater and fresh water buffer carbon dioxide gas.

Materials:

- 450 mL seawater
- pH meter
- Drinking straw
- Stopwatch
- 250 mL measuring cylinder
- Glass stirring rod
- 3 x 250 mL beakers
- $CaCO_3$ (lumps)
- 0.5 g Na_2CO_3
- 0.5 g $NaHCO_3$

Method:

1. Place 150 mL of seawater into each of the three beakers.
2. Add a small lump of $CaCO_3$ (approximately 0.5 g) to the first beaker, 0.5g Na_2CO_3 to the second beaker and 0.5 g $NaHCO_3$ to the third beaker. Stir each beaker.
3. Measure the pH of the first beaker using the pH meter and record the measurement in the following table.
4. Blow through a straw into the seawater in the first beaker for 5 seconds. (Once again it is better to use the same person for blowing through the straw in all the trials.) Measure the pH and record the result.
5. Repeat step 4 until you have been blowing for a total of 100 seconds.
6. Repeat step 3 to 5 for each of the other beakers.

Results:

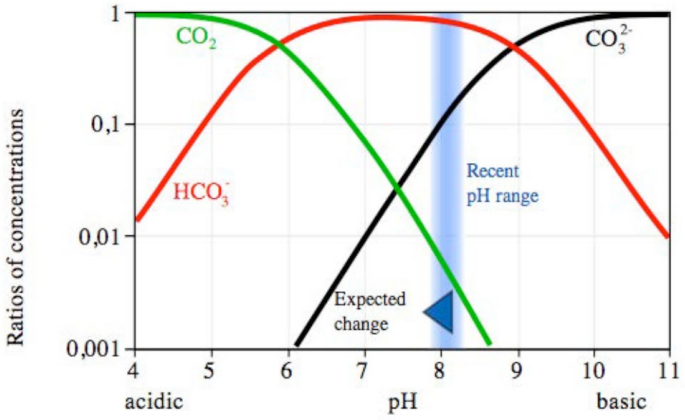
TOTAL TIME BLOWING (s)	pH OF SEAWATER CONTAINING $CaCO_3$	pH OF SEAWATER CONTAINING Na_2CO_3	pH OF SEAWATER CONTAINING $NaHCO_3$
0			
5			
10			
15			
20			
25			
30			
35			
40			
45			
50			
55			
60			
65			
70			
75			
80			
85			
90			
95			
100			

Discuss:

- How did the addition of $CaCO_3$, Na_2CO_3 and $NaHCO_3$ affect the pH of the seawater before carbon dioxide was bubbled through? Why is this?
- How did Na_2CO_3 and $NaHCO_3$ affect buffering capacity? Compare your results with the seawater from part one.
- Why does solid calcium carbonate not have much effect on buffering capacity compared with Na_2CO_3?
- If scientists added calcium carbonate to the ocean, do you think it would help improve its buffering capacity? Would it be a practical solution? Explain why or why not.
- Sea water is buffered according to the equilibrium reactions below:

  $$CO_2 + H_2O \rightleftharpoons H_2CO_3 \rightleftharpoons H^+ + HCO_3^- \rightleftharpoons 2H^+ + CO_3^{2-}$$

  Use this equation and Le Chatelier's principle to explain the following:
  a. Why does bubbling CO2 through seawater decrease its pH?
  b. Why does adding Na_2CO_3 to seawater increase its pH?
  c. Why does adding $NaHCO_3$ to seawater increase its pH?
- Identify the conjugate acid base pairs in the equation above.
- The diagram below shows the distribution of carbon dioxide, bicarbonate ions and carbonate ions in seawater.



  Why isn't the proportion of carbonic acid shown on this graph?
- Organisms with calcium carbonate shells produce their shells according to the equation:

  $$Ca^{2+}_{(aq)} + CO^{2-}_{3(aq)} \rightarrow CaCO_{3(s)} \text{ and } HCO^-_{3(aq)} \rightleftarrows H^+_{(aq)} + CO^{2-}_{3(aq)}$$

  Use the graph to explain why these marine organisms won't be able to build shells if pH decreases.

Sourced from: https://www.uwa.edu.au/science/-/media/Faculties/Science/Docs/Activity-How-does-the-ocean-buffer.pdf
Done by: Jenny Strodl, Marine Science, Science and Maths Teacher, St Peters Lutheran College

Suggested practical:

♦ Effect of CO_2 on planktonic organisms

◊ CO2 fertilization of marine microalgae cultures *(Dunaliella sp.)*

Aim: To investigate how the concentration of CO_2 in water affects marine macroalgae.

Materials:

- Erlenmeyer culture flasks (200 mL)
- Rubber stoppers with one-hole fitting into the mouths of the flasks (or cotton balls wrapped into transparent films)
- Pipettes
- Filter paper
- Glass fibre filter (GF/F)
- Drinking straws
- Culture water: pond or lake water, seawater (salinity: 35), brackish water (salinity: 15)
- Nutrients for algae culture: nitrate (NO_3^-), phosphate (PO_4^{3-}), silicate ($Si(OH)_4$), vitamin mix, metal mix (If nutrients are not available, a plant fertilizer with high nitrate and phosphate content will also do. Use 5 – 10 millilitres per litre of culture water.)
- Stock culture of freshwater or marine microalgae, e.g. *Dunaliella sp.* (If no stock culture is available, natural populations will do. Use a plankton net to gather phytoplankton from a lake, pond or from the sea. It may take a little more time until results can be seen with natural populations. Natural *Dunaliella sp*. does not require silicate.)

Method:

1. Filter the culture water first with the paper filters and then with the GF/F filters. Filter sterilization is optional, although this minimises bacterial and foreign algal contamination.
2. Prepare common culture water for all the treatments by adding the nutrients to the filtered water. Here, one mL of each solution from nitrate, phosphate, silicate and metal mix plus 500 µL of the vitamin mix is used for 1 L filtered culture water.
3. Add about 5 mL of the stock algal culture, for example Dunaliella sp. to the prepared culture medium. The exact amount depends on the density of the algal culture. The more of a stock culture you add, the faster the new culture will grow. If you run this experiment in winter, it is better to use a bit more than in summer when higher temperatures and light promote growth.
4. Rinse and then fill the culture flasks with the prepared algal culture up to the 150 mL mark. Then you can decide what treatments you would like to make.
   For example:
   Treatment 1 (T1): CO_2 limited - flask covered tightly
   Treatment 2 (T2): CO_2 fertilised - flask covered with a cotton wad made airtight by wrapping it in transparent film with a straw inserted in the middle for CO_2 introduction by blowing
   Treatment 3 (T3): control (no CO_2 fertilisation) - flask covered with a cotton wad
5. Make three replicates of each treatment. Place the flasks in a bright place but not in direct sunlight. Treatments 1 and 3 should be gently shaken at least twice daily. For treatment 2, air should be blown into the culture for 30 – 60 seconds twice a day.

Discuss:

- Observe the algae cultures over two weeks. How do the colours in the flasks change?
- What do the changes tell you about the growth of the algae under the different treatments?

Sourced from: https://www.bioacid.de/wp-content/uploads/2017/04/BIOACID_Experiments_en.pdf

Unit 4

Ocean Issues & Resource Management

Source: SpringerLink

Mandatory practical:

- ♦ Lincoln index in a modelled capture-recapture scenario

◊ Catch per unit effort

Aim: To investigate how the catch rates change as the population declines.

Materials:

- Bath tub
- Crays or fish or substitute – 200
- Nets (range)
- Excel table

Method:

Sourced from: https://www.youtube.com/watch?v=yLXEYWZnUgA&t=186s
Done by: Chris Pacey, St Patrick's College Townsville

◊ Capture-recapture

Aim: To simulate capture-recapture method and estimate population size.

Materials:

- 50 beans
- Marking pen
- Plastic container with lid

Method:

1. Mark 20 of the beans with the marker.
2. Place all 50 beans in the container and cover with the lid. Gently shake the container to mix the beans thoroughly (do not shake too hard or your beans will break apart). Take the lid off and without looking, withdraw 10 beans. Count the number of marked beans and record in the data table.
3. Put the 10 beans back in the container and replace the lid. Shake the container again and withdraw a second set of beans. Again, record the number of marked beans in the sample in the data table. Continue in this manner until you have completed 10 trials.
4. Estimate the population size for each trial using the Lincoln Index by multiplying the number of beans marked (20) by the number of beans in each sample (10) and dividing the product by the number of marked beans (recaptured) for that trial. The result is an estimation of population size. When you have done this for all 10 trials, find the average by adding the estimates and dividing by 10.

$$P = (N_1 \times N_2)/R$$

P = total size of population
N_1 = size of first sample (all marked)
N_2 = size of second sample (recapture: some will be marked, some won't)
R = number of marked individuals recaptured in second sample

DATA:

Trial #	# marked beans in sample	population estimate
1		
2		
3		
4		
5		
6		
7		
8		
9		
10		

Discussion:

- How does the average value compare to the actual population size of 50?
- If there is a difference, explain what might cause the difference.
- What problems might scientists encounter in using this method in the field that you would not have encountered in the simulation?

Source:
https://www.birdvilleschools.net/cms/lib/TX01000797/Centricity/Domain/3134/Estimating%20Population%20Size%20Activity%20beans%20%20pillbugs.pdf

Secondary sources:
https://apeslabreportsbva.weebly.com/catch--release-lab.html
https://www.biologycorner.com/worksheets/estimating_population_size.html

Done by: Chris Pacey, St Patrick's College Townsville

◊ Estimating population size using capture-recapture method and Lincoln index

Aim: To estimate population size of mobile animals & to investigate how sampling size affects accuracy of the estimate.

Materials:

- Popping corn kernels ('our population')
- Marking pen
- Plastic container with lid
- Measuring spoon (1tbsp, ½ tsp)

Method:

1. Obtain a container with 'individuals' (popping corn). Count the number of all kernels.
2. Obtain a sampler (1 tablespoon).
3. Collect a sample of individuals by filling the measuring spoon.
4. Count and record the number collected.
5. Mark each collected individual with a marker. After marking, return the marked individuals to the population and thoroughly mix the individuals.
6. Draw out a second sample.
   Record the number of marked and unmarked beans, but do not return them back to the population.
7. Estimate population size using Lincoln index:

$$N = \frac{Mn}{m}$$

N… population estimate
M… marked individuals released
n… size of second sample
m… marked animals captured

8. Change the size of the measuring spoon (½ tsp).
9. Repeat steps 3-7

10. Collate the data from all groups.
11. Calculate a mean for the estimated population size using 1 tablespoon and a mean for the estimated population size using ½ teaspoon.

Data table:

Sample (using 1 tablespoon)	**Number caught**	**Caught marked**	**Caught unmarked**
1			
2			
Sample (using ½ teaspoon)	**Number caught**	**Caught marked**	**Caught unmarked**
1			
2			

Discuss:

The Lincoln method is fairly simple, and its calculations are straightforward, but it does depend on several assumptions. Violating the conditions of the Lincoln model can seriously affect the accuracy of your estimate, so it is very important to bear these assumptions in mind as you interpret your results:

1. Individuals with marks have the same probability of survival as other members of the population.
2. Births and deaths do not occur in significant numbers between the time of release and the time of recapture.
3. Immigration and emigration do not occur in significant numbers between the time of release and the time of recapture.
4. Marked individuals mix randomly with the population at large.
5. Marked animals are neither easier, nor harder, to capture a second time.
6. Marks do not come off of your marked organisms.
7. Recapture rates are high enough to support an accurate estimate.

- How do your estimates compare to the true population size?
- If your estimated value differs to the true value, why do you think this might have occurred?
- How do calculated means for using 1 teaspoon and ½ teaspoon compare to the true population size?
- Which of the calculated means is a better estimate of the population size?
- If immigrations occurred in a natural population being studied, how would this influence the reliability of your estimate of population size determined using mark-recapture technique? Would your population estimate be too high or too low, or would you be able to predict how your estimate would be biased? Remember: migration consists of both immigration and emigration.

Source:

Estimating Population Size: Mark-Recapture
https://www.deanza.edu/faculty/heyerbruce/b6c_pdf/3b_Estimating%20Population%20Size.pdf
A Method of Population Estimation: Mark & Recapture https://www.radford.edu/~jkell/mark_rec103.pdf

Done by: Nika Langham, Pioneer SHS, Mackay

◊ Estimating population size using capture-recapture method and Schnabel index

Aim: To estimate population size of mobile animals using Schnabel index.

Background: Assumption 7 (from calculating population size using Lincoln index, see previous practical) is often violated, because it is difficult to generate sufficient recaptures in large populations. The Lincoln calculation tends to overestimate the population size, especially if the number of recaptures is small. To provide a better estimate, ecologists sometimes use multiple marks and recaptures and the Schnabel index to estimate population size.

Materials:

- Popping corn kernels ('our population')
- Marking pen
- Plastic container with lid
- Measuring spoon (½ tsp)

Method:

1. Obtain a container with 'individuals' (popping corn). Count the number of all kernels.
2. Obtain a sampler (½ tsp).
3. Collect a sample of individuals by filling the measuring spoon.
4. Count and record the number collected.
5. Mark each collected individual with a marker. After marking, return the marked individuals to the population and thoroughly mix the individuals.
6. Draw out a second sample.
   Record the number of marked and unmarked beans, but do not return them back to the population.
7. Mark each unmarked individual, return this sample back to the population, and thoroughly mix.
8. Draw out a third sample.
9. Again, count and record the number of marked and unmarked beans.
10. Mark the unmarked beans and return the sample to the population.
11. Draw out a fourth sample.
12. Count and record the marked and unmarked individuals.
13. Calculate population size using Schnabel index:

$$N = \frac{\sum_{i=1}^{m} M_i C_i}{\sum_{i=1}^{m} R_i}$$

M_i... total number of previously marked animals at time
C_i... number caught at time
R_i... number of marked animals caught at time i

Data table:

Sample (using ½ teaspoon) S.I. (i)	**Caught** S.I. (C_i)	**Caught marked** S.I. (R_i)	**Caught unmarked**	**Total number of previously marked** S.I (M_i)
1				
2				
3				
4				

Discuss:

- Did the Schnabel index give you a better estimate of the actual population size than did the Lincoln index? Why?

Source:
Estimating Population Size: Mark-Recapture
https://www.deanza.edu/faculty/heyerbruce/b6c_pdf/3b_Estimating%20Population%20Size.pdf
A Method of Population Estimation: Mark & Recapture https://www.radford.edu/~jkell/mark_rec103.pdf

Done by: Nika Langham, Pioneer SHS, Mackay

Suggested practical:

♦ Life history of a fish (otoliths)

Aim: To investigate the life history of a fish by reviewing otoliths using a microscope.

Materials:
Note: materials required will depend on whether otoliths are sourced already extracted or whether they need to be extracted from the head of the fish.
For extraction:

- Fish head(s)
- Forceps
- Sharp knife/ 20-25mm wide blade
- Chisel
- Scalpel
- Bright light/ headlamp
- Distilled water (to clean otolith)

For reading of the otolith:

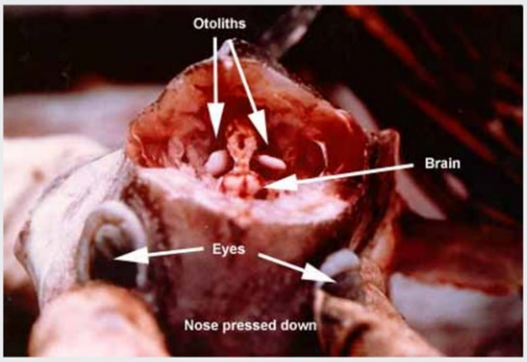
- Microscope
- Petri dish
- forceps

Method:

Method of Extraction:
There are several methods of extraction – the following is directly from Fisheries & Oceans Canada (2018) – full reference below:
Otoliths of adult fish can generally be removed with nothing more than a sharp fish knife and a pair of forceps or tweezers. With a little practice, the large pair of otoliths (the sagittate) can be removed in 15 seconds.
Marine fish such as cod and haddock have otoliths which are relatively large and therefore easy to find (about 1 cm long in a 30 cm long fish). Smaller fish, such as minnows, may require the use of a microscope.

To remove a pair of otoliths:

1. use a knife with at least a 15 to 20 cm blade
    - the blade should be as sharp as possible
    - you'll also need a pair of forceps or tweezers about 10 cm long
2. grip the head of the fish by putting your thumb and forefinger in its eye sockets
    - lay the body of the fish on a counter with the tail pointing away from you
3. put the knife blade on the top of the fish's head about 1 eye diameter behind the eyes
    - slant the blade **away** from you, at about a 30° angle
4. slice back and down about 1 head length
    - you should feel the knife cut through the top of the skull
    - for flatfish and some other species, a vertical cut through the top of the skull directly over the preopercle (the curved line 3/4 of the way back on the gill flap) also works well



Head and skull of a fish cut open to expose the brain (central white portion of image) and sagittal otoliths on either side.

Fisheries & Oceans Canada (2018)

5. check to see if you've cut the top off the skull
    - if you haven't, make another slightly deeper cut
    - an ideal cut removes the top of the skull, revealing the full length of the soft white brain underneath
    - note that the brain joins the much narrower (but still white) spinal cord at the rear
    - once the brain is visible, expose the brain even more by pressing the nose and body down and towards each other
    - this should snap a portion of the skull, and push the brain and otoliths up
    - very often, this exposes the otoliths and allows them to be removed immediately
6. push the rear of the brain to one side, or cut it out all together
    - the large pair of otoliths should be visible underneath the rear of the brain, still inside the skull
    - they may or may not be resting inside hollows in the base of the skull
7. use forceps to pull out both otoliths
    - they won't be attached to anything other than soft tissue
    - clean off the otoliths with water or your fingers
    - store dry in a paper envelope until you're ready to age them

Method for Review of Otolith:

a) Ensure that the otolith is clean and in good condition. Handle with care.
b) The otolith may be observed whole or it may need to be sectioned
c) To section, cut a thin slice through the core (may require specialised equipment)
d) Count the number of opaque bands between the core and the otolith edge

How to Estimate Fish Age:

Otoliths (ear bones) help fish orientate themselves and maintain balance, acting like our middle ear. Otoliths are composed of a form of calcium carbonate and protein which is laid down at different rates throughout a fish's life. This process leaves bands (alternating opaque and translucent bands) on the otolith like the growth rings in a tree.

Sourced from:

- Removal of Fish Otolith:
  1. Fisheries & Oceans Canada (2018) Otoliths, removal and aging Fisheries & Oceans Canada Website: https://www.dfo-mpo.gc.ca/science/aah-saa/otoliths/students/removal-prelevement-eng.html .
  2. Wakefield, C.B., D.K. Boddington & S.J. Newman (2016) Rapid Lateral Extraction of Otoliths that Maintains the Integrity of Fish Product to Improve Access to Catches and Reduce Potential Sampling Biases. The Open Fish Science Journal. 9:26-28. https://www.researchgate.net/publication/303240456

- How to Estimate Age:
  3. Department of Agriculture & Fisheries (2019) Estimated Fish Age. Queensland Government. https://www.daf.qld.gov.au/business-priorities/fisheries/monitoring-research/monitoring-reporting/commercial-fisheries/species-specific/estimated-fish-age
  4. Marino, J.C., A.J. Fowler, Z.A. Doubleday, G.L. Grammar & B.M. Gillanders. (2019) Using otolith chronologies to understand long-term trends and extrinsic drivers of growth in fisheries. Ecosphere 10(1)e02553. https://esajournals.onlinelibrary.wiley.com/doi/epdf/10.1002/ecs2.2553
  5. Morais, P., E. Dias, I.Cerveira, S.M. Carlson, R.C. Johnson & A.M. Sturrock (2018) How Scientists Reveal the Secret Migrations of Fish. Frontier for Young Minds. https://kids.frontiersin.org/article/10.3389/frym.2018.00067 .
  6. O'Sullivan, S. (2007) Fisheries Long Term Monitoring Program: Fish Age Estimation Review. Queensland Government. P28 http://era.daf.qld.gov.au/id/eprint/6430/1/FishAge-Review.pdf
  7. Ramel, G. (2021) Fish Skeleton 101: The Evolution of these Bony Structures Earth Life. https://www.earthlife.net/fish/skeleton.html
  8. The National Institute of Water and Atmospheric Research (NIWA) (2012) Determining the age of fish. NIWA Science Website. https://niwa.co.nz/fisheries/research-projects/determining-the-age-of-fish .
  9. Golf States Marine Fisheries Commission (2003) Otolith Removal, cleaning, and prep.pdf https://www.gsmfc.org/pubs/ijf/otolith/S_Section%20003%20.pdf
  10. Video: Age determination of otoliths with the crack and burn method. https://www.youtube.com/watch?v=aDKaBJLar_w

Done by: Unknown whether any Marine Science teacher has successfully done this practical

Suggested practical:

♦ Presence of microplastics

◊ Presence of microplastics in water

Aim: To analyse a water sample to identify the presence of microplastics.

Materials:

- 200 ml container (plastic or glass) to collect water samples
- Magnetite (may be sourced from old copier cartridges)
- Cooking Oil (experiment may work without this)
- Magnet
- Microscopes
- Petri-dishes

Method:

1. Collect several water samples: from creek, river, estuary, beach zone
2. Add 10ml oil to 200 ml water sample and stir gently
3. Add 5g of magnetite and stir gently
4. Dip the magnet into the solution and remove the magnetite-oil-microplastic mix
5. Scrape the mix into a petri dish
6. Observe under a microscope

Note: in the original experiments the author built a visible light spectrometer so he could accurately measure the density of microplastic in his test solutions. There are links below on this part of the experiment.

Original water experiment based on: An investigation into the removal of microplastics from water using Ferro fluids, by Fionn Ferreira - https://www.youtube.com/watch?v=jmfIkj6V7Ns

More information can also be found here: https://abstracts.societyforscience.org/Home/PrintPdf/14829# and here: https://www.edinst.com/blog/the-beer-lambert-law/. More information on the Beer Lambert Law (of light attenuation) if you want to use this, can be found here: https://www.edinst.com/blog/the-beer-lambert-law/

Done by: Joline Lalime, Mackay North SHS, Mackay

◊ Presence of microplastics in sand

Aim: To analyse a sand sample to identify the presence of microplastics.

Materials:

- 50cm x 50cm square of fly screen (can frame it to make it more stable)
- 200ml plastic containers to collect samples
- Microscopes
- Petri-dishes

Method:

1. Collect several sand samples: from a beach, estuary or river
2. Sift each sample through the fly screen
3. Put the resulting "waste" into a container & label it (where it was from)
4. Go back to the lab
5. Take a sample from a container and put it in a petri dish
6. Observe under a microscope

Original sand experiment based on: An investigation into the removal of microplastics from sand using electrostatic properties of plastic https://www.youtube.com/watch?v=KaC1vDYGpFc
More information can also be found here:
https://www.researchgate.net/publication/321194100_A_new_approach_in_separating_microplastics_from_environmental_samples_based_on_their_electrostatic_behavior
Done by: Joline Lalime, Mackay North SHS, Mackay

Unit 4: Ocean issues and resource management

Suggested practical:

- **Factors that affect the growth rate of an aquaculture species**

Aim: To investigate factors that affect the growth rate of an aquaculture species. This may include: biotic (crowding, territorial or dominant behaviour, stress, waste production, metabolic rate) OR abiotic (dissolved oxygen, nitrites, ammonia, temperature, pH) Or systemic (volume, exchange rates, dead spots).

Materials:

- Access to aquaculture facility / build own aquaculture small-scale
-

Method:

1 xxxx

Sourced from:

1. Department of Agriculture, Water & the Environment (2019) Aquaculture. DAWE Australian Government. https://www.agriculture.gov.au/fisheries/aquaculture
2.

Done by: ??

◊ Effect of salinity on hatching success of artemia

Aim:

Materials:
Each group:

- Set of 3 bottles with different salinity water
- Artemia for each bottle
- Aerator/hosing
- Stand (retort stand or similar)

Method:

1.

Resources:
Brine shrimp project: https://learning-center.homesciencetools.com/article/brine-shrimp-science-project/
Video: DIY brine shrimp hatchery https://www.youtube.com/watch?v=G2BKVhFgpVU

Done by: Chris Pacey, St Patrick's College, Townsville