AN INVESTIGATION INTO *SALIX FRAGILIS* AND THE SUBSEQUENT ENVIRONMENTAL IMPACTS OF ITS INTRODUCTION TO THE TASMANIAN LANDSCAPE

SCIENCE INVESTIGATION
Investigation Report

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PART ONE

AIM

To investigate and analyse how willow cuttings grow in both soil and water, in order to aid our investigation into how willows affect the Tasmanian environment.

THEORY

WHAT ARE WILLOWS?

Willows are a species of tree of the genus *Salix* (family Salicaceae). The plant originates from northern China but is now found widely throughout the northern hemisphere. Willows generally inhabit temperate areas that provide enough moisture and direct sunlight, making them often found next to lakes and ponds. The plants are fond of retentive soils, allowing the ground and surrounding areas to remain damp throughout the seasons. The species is characterised by its alternate long narrow leaves and catkins. Catkins are slim, cylindrical clusters of flowers that are arranged closely along a central drooping stem. These catkins allow the female flowers to be pollinated easily, as the pollen from the male flowers is blown by the wind and onto the flowers. Once the seeds have developed, they are dispersed by the wind.

Figure 1.1.1 A catkin on a crack willow tree.

The trees are either male or female, and the seed have long, silky hairs. There are around 400 species of willow worldwide. Willows are divided into 2 categories – tree willows and shrub willows. Tree willows grow large with on or more trunks and can typically reach 10-30 meters in height. They have brittle and fragile branches that break off easily. Shrub willows, otherwise known as sallows, vary from shrubs and bushes to small trees.

They normally have multiple stems and twigs which are harder to break than the tree willows.

Willows are thought to have arrived in Tasmania in the early 19th century. An interesting origin story surrounds the plant, as they are thought to have originated from the famous weeping willow over the grave of Napoleon at St Helena. Different varieties of the willow tree continued to be planted, mainly for aesthetic reasons and as a source of drought feed for livestock, up until the mid-late 20th century when the adverse effects of willows on the environment and their full impact on Tasmanian waterways came to light.

There are over a hundred species of willow in Australia, all of which are introduced and not native species. Many species of willow occur in Tasmania, including but not limited to grey willow or pussy willow (*Salix cinereal*), crack willow (*Salix fragilis*), basket willow (*Salix rubens*) and black willow (*Salix nigra*). Out of these species, the species of crack willows, or *Salix fragilis*, is the biggest problem in Tasmania. This is due to their distribution around the state, making them widespread. As a result of this, they are now considered 'naturalised' in most catchments. Crack willows are a declared weed in Tasmania under the Tasmanian Weed Management Act 1999. This means it is illegal to sell, spread or use them in any way. Because of these reasons, it has been chosen that this investigation shall focus around the crack willow, or *Salix fragilis*.

Willows are listed on Australia's "Weeds of national significance". This is a list of the 20 most invasive plant species in in Australia and focuses on "weeds that have degraded large portions of Australia's natural and productive landscape and require action at a national level to reduce their impacts. "

THE EFFECT OF WILLOWS ON TASMANIAN RIPARIAN HABITATS

Willows cause many problems in Tasmania. These include but are not limited to the below issues:

- Willows tend to have thinner branches than native plants. This means they drop their limbs more frequently. This is a safety concern for people and property. If willows are situated near homes or structures, lives may be in danger. People may come into the way of harm, especially young children. Additionally, dropped limbs in creeks and streams increase the likelihood of additional debris being caught. This can lead to localised flooding, riverbed and bank erosion, and potential damage to bridge and pipe infrastructure.
- Willows have large and extended root masses, which means once they are established, the roots tend to spread into the nearby watercourse. This slows down the flow of water, which leads to an accumulation of silt and sediment in the riverbed. The accumulation of sediment in the root systems can cause rivers to become shallower and wider. The willows root mass can also decrease the hydraulic capacity, which can also increase the risk of flooding.
- Willows large and extended root masses can also divert the natural flow of water towards other riverbanks, which can also lead to further erosion problems.

Figure 1.2.1 A tangle of limbs at the base of the crack willow in West Hobart from where we collected our samples. Due to their thin branches willows frequently drop branches which can be a danger to pedestrians and can also propagate causing new willows to grow.

- Unlike Native Tasmanian trees and vegetation (except for the *Nothofagus gunnii*), willows drop their leaves in winter. Willows have many leaves which creates a flush of organic matter that breaks down quickly. The breakdown of high levels of organic matter is a high oxygen-requiring process which reduces water quality and available oxygen. This can be harmful for other aquatic plants and fish that need the dissolved oxygen to survive. It can also encourage blackwater events.

Figure 1.2.2 A blackwater event in the Wakool River, Murray-Darling Basin, NSW. Blackwater events occur when flooding washes organic material into waterways, where it is consumed by bacteria, leading to a rise in dissolved carbon in the water. The water appears black due to the dissolved carbon compounds, such as tannins. Rising levels of dissolved carbon causes a sudden depletion of dissolved oxygen in water, which is essential for aquatic organisms that need to breathe underwater, such as the dead fish shown in the picture.

- The dropping of leaves on the ground becomes so thick it prevents other native plant species from growing, decreasing the biodiversity of riparian habitat. Native animals and insects are not adapted to this ground compost and attempting to eat it can cause them harm.
- Willows have a large surface area which creates significant shading. This reduces the amount of sunlight reaching the understory and prevents the growth of other native plant species. This also creates a lack of native vegetation species diversity. When there is only a species of plant (i.e. willows) that dominate a system, this is referred to as a monoculture. Monocultures are not favourable for native Australian/Tasmanian wildlife because it reduces the amount of food available for a variety of different species. I.e. If wildlife cannot feed or shelter in willows they cannot be supported in a willow-infested system.
- The thick, overhanging canopy of willows casts heavy shade which decreases the amount of solar radiation reaching the stream channel, which can affect stream temperatures. The sparse, open canopy of native gums and eucalyptus does not do this as they allow light to filter through.

- Unlike native species, willow flowers only provide nectar for introduced honeybees for a very short period of time as they have a brief flowering season. There are no records of native nectar consuming birds who feed from the trees.
- Willows need to be removed properly, (see below), otherwise erosion can actually increase. This means willows can be extremely expensive to remove, especially if they are well established.
- As willows are extremely invasive, once removed they need to be burnt or incinerated as they can still grow roots when stacked up in a pile.
- Unlike native trees, willows do not form hollows that native animals can live in, so they reduce native animal habitat by out-competing trees that do form hollows.
- The presence of willows has been proved to encourage invasive invertebrates- studies have shown that invasive invertebrates are more abundant in streams with willows.

Figure 1.2.3 Willows blocking a creek in the Southern Midlands, Tasmania. Willows such as these ones can grow into the stream bed where they are often surrounded by water and can eventually block the stream completely as shown here.

- Willows have a high-water uptake. They use a great volume of water which can impact greatly on small creeks that naturally have low volumes of water and creeks/rivers in drought affected areas. In drought ridden areas in Victoria willows have dried out entire streams.

- Crack willows grow extremely quickly in comparison to their native counterparts, at around 40 cm per year, which means they can easily get out of control.

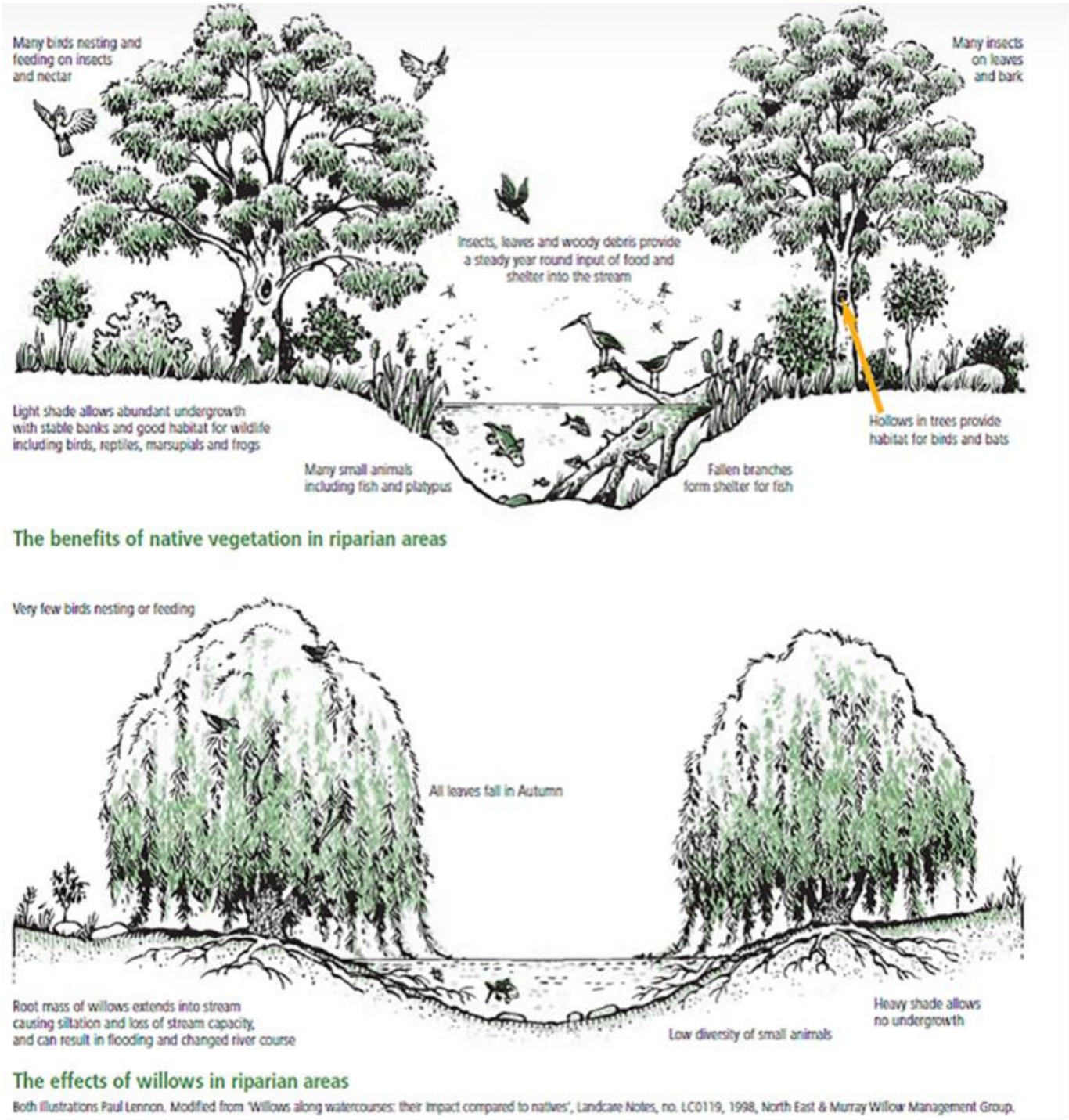


Figure 1.2.4 An information sheet comparing willows and native vegetation in riparian areas

EFFECT ON ASTACOPSIS GOULDI AND OTHER ANIMALS

These factors can be extremely damaging to native plants and wildlife, an example of this being the Tasmanian giant freshwater crayfish (*Astacopsis gouldi*), the largest freshwater invertebrate and largest freshwater crayfish species in the world. The species is not widely distributed, being only found in rivers below 400 m in the north-west, and to a lesser extent, the north-east of Tasmania.

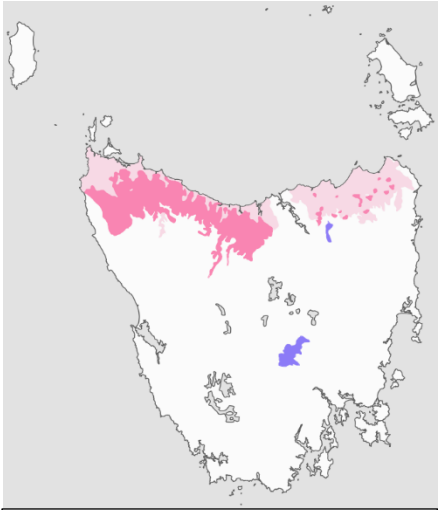


Figure 1.3.1 Above: A map of Tasmania showing the distribution of Astacopsis gouldi.

Dark Pink: Species likely to occur

Light Pink: Species may occur

Purple: translocated populations

Giant freshwater crayfishes preferred habitat is an intact catchment of several stream sizes including rivulets and the small creek headwaters flowing through a relatively undisturbed, well-vegetated catchment containing snags, pools and undercut (but not eroding) riverbanks. Tasmanian giant freshwater crayfishes diet varies with age, but mainly consists of decaying wood, leaves, aquatic plants, microbes, small fish, insects, rotting animal flesh and detritus. The water temperature should remain relatively constant and rarely exceed 18 C, have a high oxygen content and be clear of sediment. Adult crayfish take refuge in still, deep pools which are sheltered and well-shaded beneath submerged and decaying timber. Juveniles migrate into small stream/creek zones which are shallower but faster flowing with small pebbles and rocks to hide underneath. This includes semi-permanent creeks lined with overhanging vegetation.

Willows have had many adverse effects on the Tasmanian giant freshwater crayfish, including:

- Willows spread their roots into the riverbed which traps and accumulates sediment. Juvenile giant freshwater crayfish are slow-growing and vulnerable to predation from other animals, particularly from 0-6 years old. They rely on a

variety of different sized pebbles, cobble and boulders in a stream system as a form of cover to protect and defend themselves. When a significant sediment event occurs within a stream system it settles out over the pebble, cobble and boulders and fills in the gaps between the rocks. This essentially concretes the rocks into place making it impossible for juvenile crayfish to lift them. They are then forced to seek shelter in other streams which increases their risk of predation.

- Increased sediment also removes an essential food source. When a significant sediment event occurs within a stream system it also settles out over rotting leaves and wood which is the species' primary food source. They are then forced to seek other streams with enough woody debris and leaf litter to support them which also increases their risk of predation and potentially territorial problems with other crayfish. Crayfish have also been noted to prefer water with little suspended sediment and are sensitive to sedimentation.
- When willows drop their leaves in the winter, they create a large flush of organic matter which breaks down quickly. This reduces water quality and available oxygen for other aquatic species such as the giant freshwater crayfish. Dissolved oxygen levels can be an indication of how polluted the water is and how well the water can support aquatic life. Generally, higher dissolved oxygen level equals better water quality.
- Willows create significant shading which reduces the amount of sunlight reaching the understory and the river. Many aquatic plants that live in the water require sunlight to photosynthesise and without it, cannot grow. Many of these plants are essential parts of the giant freshwater crayfishes diet, which results in the crayfish having less to eat.

Due to overfishing, habitat degradation, arguably the introduction of willows and other environmental threats, the Tasmanian Giant freshwater crayfishes' conservation status is now listed as endangered. This means, according to World Wildlife Fund "*A species considered to be facing a very high risk of extinction in the wild".* Because of this, it has been illegal to catch the crayfish since 1988.

Figure 1.3.2 The Tasmanian Giant Freshwater Crayfishes conservation status is currently listed as endangered, which means they are under significant threat.

The Tasmanian Giant freshwater crayfish is just one of many Tasmanian species that have been negatively impacted by the introduction of willows, other examples include platypi, many types of fish, frogs, including the eastern banjo frog, brown tree frog, and insects.

Figure 1.3.3 A Giant Tasmanian Freshwater Crayfish in its natural habitat. The crayfish measure over 80 centimetres and can live to the age of 60.

EFFECT ON PLATYPUS POPULATION

Another animal negatively affected by willows is the platypus, (*Ornithorhynchus anatinus),* very similar to the reasons why the giant freshwater crayfish is affected. Like crayfish, platypuses live in intact catchments but are dispersed more widely around the state. Platypuses are also under threat, although their status is listed as "near threatened" rather than endangered like the crayfish. The platypus is also negatively affected by willows because of the following:

- Platypus eat small water animals such as insect larvae, which can easily be killed by increased river sediment levels from the willows
  -Increased sediment levels from the willows roots can make the water less clear and it harder for the platypus to move around in the stream.
  -Willows lower the dissolved oxygen levels in water which is dangerous for aquatic life such as the platypi.

The reason this investigation was conducted was because of inspiration from a previous research project on the platypus earlier in the year and the optimal water conditions for it. Different locations around Tasmania were tested to see if they met these conditions.

(Later we are going to post the video to YouTube so we can put a link here to it for people to watch.)

Figure 1.4.1 A platypus (Ornithorhynchus anatinus) in its natural habitat, a clear freshwater stream.

HOW WILLOWS SPREAD

There are only male crack willows in Tasmania, which means that they cannot sexually reproduce from seed germination on wet sediment, but instead reproduce asexually by propagating roots from broken branches and limbs. This is extremely problematic, as it only takes a single broken limb to fall, move down the stream and get stuck in a bank for a crack willow to start growing. It also means that all willows must be eradicated from an area at the same time to avoid the remaining willows reproducing and continuing the problem.

REMOVAL METHODS

There are two main methods used to remove willows in Tasmania: 'cut-stump and paint' or 'drill and fill'. These are used for more mature trees (older than 2 or 3 years) otherwise they can usually be pulled out without causing significant damage to the riverbank on which they are situated.

- Cut-stump and paint involves cutting mature willow trees at their bases so that the root system remains in the ground. The stump is then painted with glyphosate, a herbicide. Due to its sodium salt form, glyphosate can kill or regulate plant growth and is one of the most widely used herbicides in agriculture, forestry, domestic gardening and weeding. Glyphosate ensures that the willow tree itself will die and not resprout and reproduce but also provides an open area surrounding the tree for new native vegetation to grow and/or be planted. Leaving the stump in the ground means the willows root system will remain intact so the riverbank will hold in place so erosion will not become an issue. The trunks and limbs removed have to be taken away immediately and burnt to prevent them washing away, and getting caught up in the riverbed. Following this the limbs need to be burnt as they can still sprout roots stacked up in a pile.

- Drill and fill involves drilling holes around the limbs and trunk of the willow and squirting glyphosate into the holes to kill the tree in place. This again keeps the whole tree in place and the root system remains intact, so erosion is not a concern. Because the tree is dead it does not produce leaves to drop into nearby streams and when the tress falls over and/ or limbs are dropped they should not be able to grow roots.

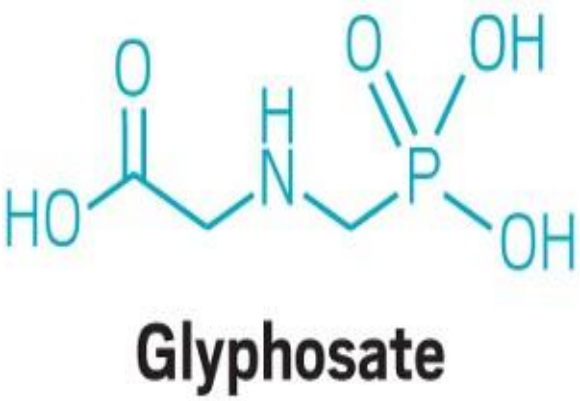


Figure 1.5.1 The chemical structure of glyphosate. Glyphosate comes in many forms, including an acid and several salts. It is a non-selective herbicide, meaning it will kill most plants. It does this by preventing the plants from making certain proteins that are needed for plant growth. Glyphosate also stops a specific enzyme pathway called the shikimic acid pathway which is necessary for plants such

It is important to note to only use glyphosate products registered for aquatic use when removing willows near waterways. Take care to remove any branches that have fallen onto the bank or stream once the removal process has been completed. You should also always make a plan when removing a high volume of willows. For example, the Derwent Catchment Project suggests the following:

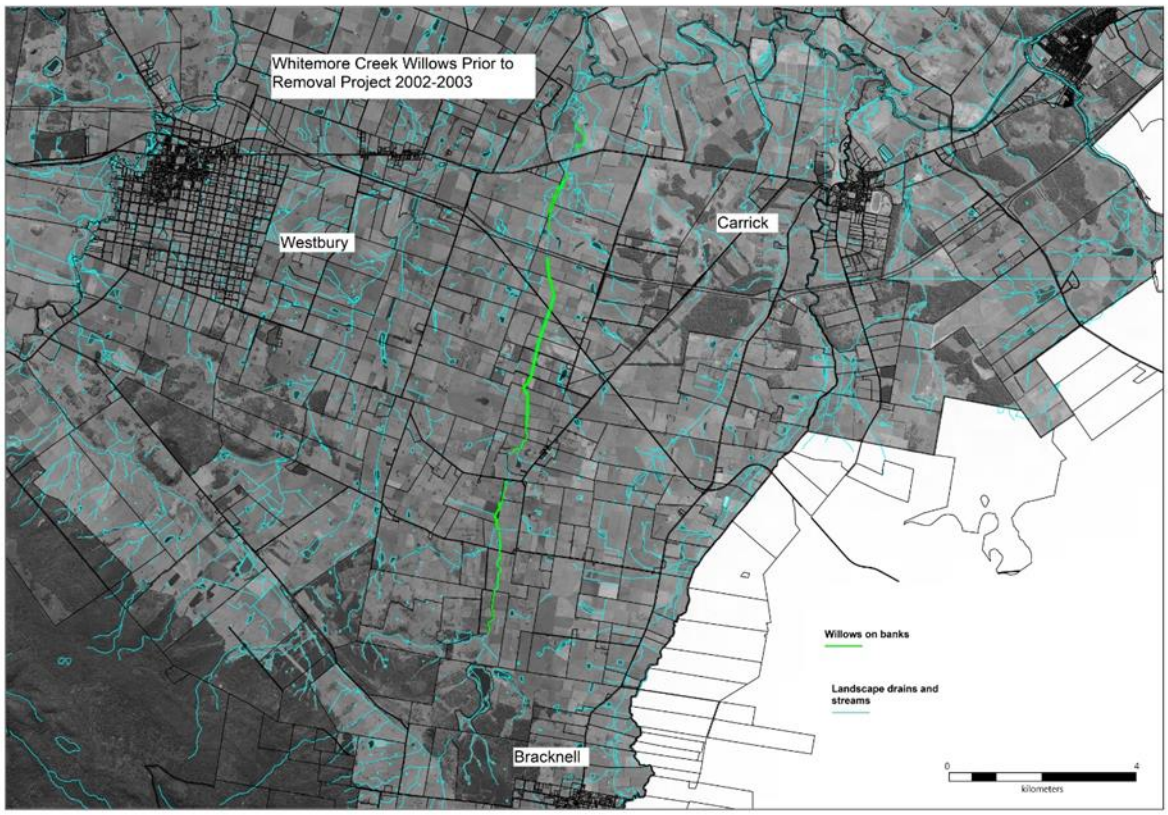
- Seek advice and make a long-term plan
- Work with your neighbours
- Start in the upper catchment and work downstream
- Remove dense willows in stages and revegetate riverbanks before moving on

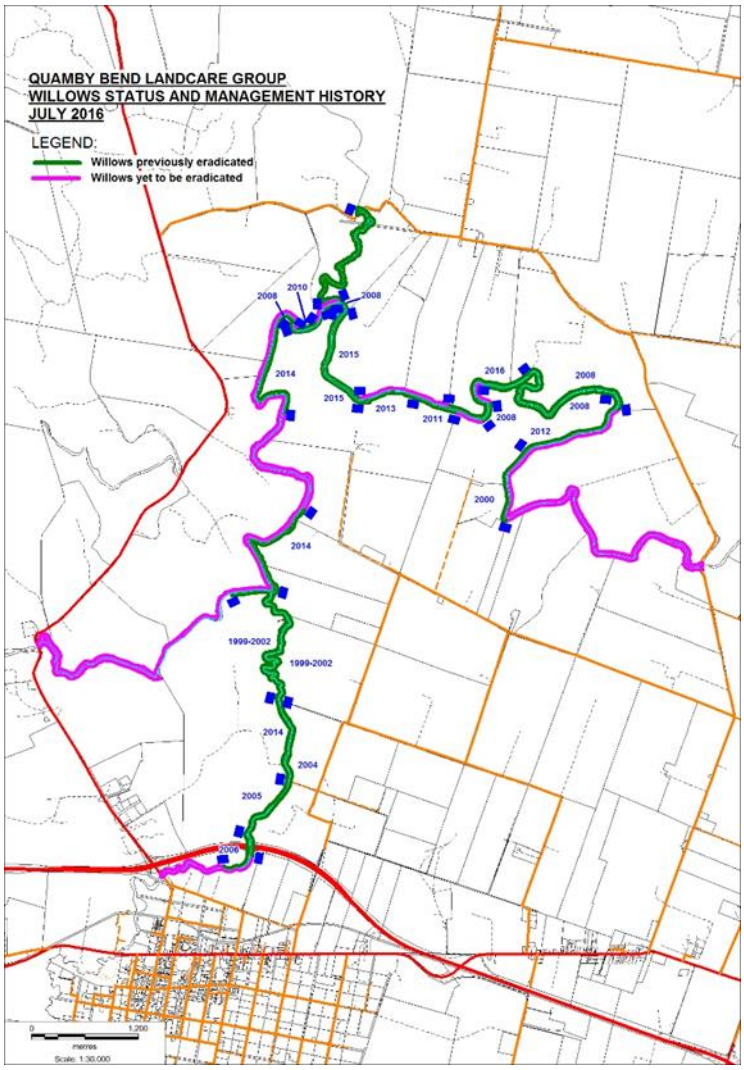
  The flow of the river will change once the willows are removed, and this can place greater pressure on restricted points downstream. In these cases, it may be better to start working on the lower end and progress upstream.
- Work on the inside bends first, establish native vegetation

PREVIOUS REMOVAL IN TASMANIA

Due to the complexity and nature of removing willows, it is usually a very costly and time-consuming process. To be completely effective, all willows have to be removed from an area at once. Willow management activities have occurred in a number of places in Tasmania in the last decade, some examples including:

- Ringarooma and Brid-Forester River Catchments
- Ringarooma Ramsar site
- McKerrows Marsh
- Whitmore creek
- Quamby bend
- Wynyard river
- Jordan River





Above: Figure 1.6.1 showing the removal of Willows in Whitemore Creek in 2002-2003.

Left: Figure 1.6.2 showing the removed and remaining Willows at Quamby Bend in 2016.

THE EXPERIMENT

In this experiment it is being observed how fast willows grow by simulating the Tasmanian Environment and growing willow cuttings in soil and water. Willow cuttings normally grow quickly and easily due to containing a natural hormone auxin in the bark that aids plant growth. It does this by moving to the darker side of the plant, causing the cells that grow there to grow larger than the cells on the other side of the plant. This produces a curving of the plant stem tip to grow towards the light, a movement also known as phototropism. Auxin promotes stem elongation and inhibit growth of lateral buds. It also promotes rooting, which is why willows root so quickly. This is why some common rooting solutions for other plants are actually made from boiled willow stems - because they have such high quantities of auxin.

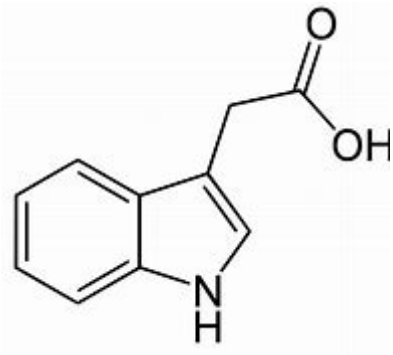


Figure 1.7.1 Auxin, a natural growth hormone present in

It is debatable whether willows will best propagate in water or soil. According to Gardening Guide magazine, “If you root your cutting in water, it develops roots that are best adapted to get what they need from water rather than from soil. If you move the plant immediately from water to soil, the plant may be stressed.” So whilst the water cuttings may grow faster, they might not be suited to soil if they were to be planted in it.

HYPOTHESIS

The plants will all grow somewhat, however the cuttings in the soil will grow faster and taller than those in the water, due to higher levels of nutrients.

VARIABLES

Dependant variable- growth/height

Independent variable- whether grown in soil or water

Control- size of pot, amount of water given, light exposed, time

MATERIALS

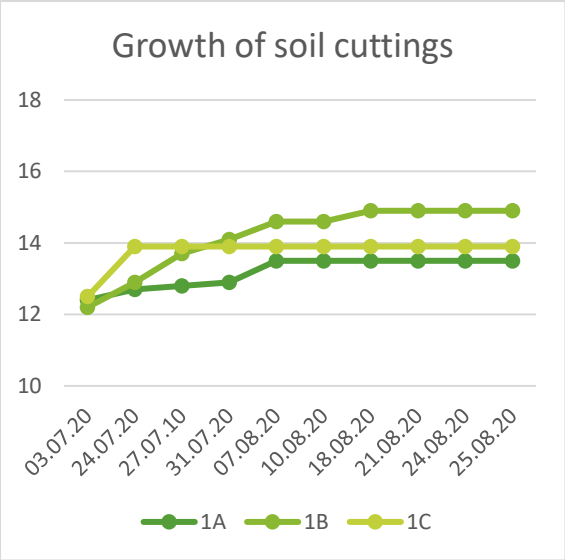
- Glass beaker x 3
- Pots x 3
- Disposable plastic gloves
- Lab coat
- Dust mask
- Safety goggles
- Pyrex test tube
- Potting mix
- Water
- Willow cuttings x 6

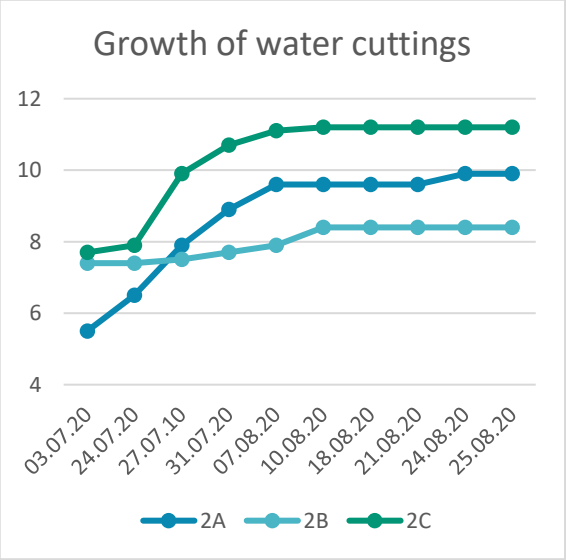
METHOD

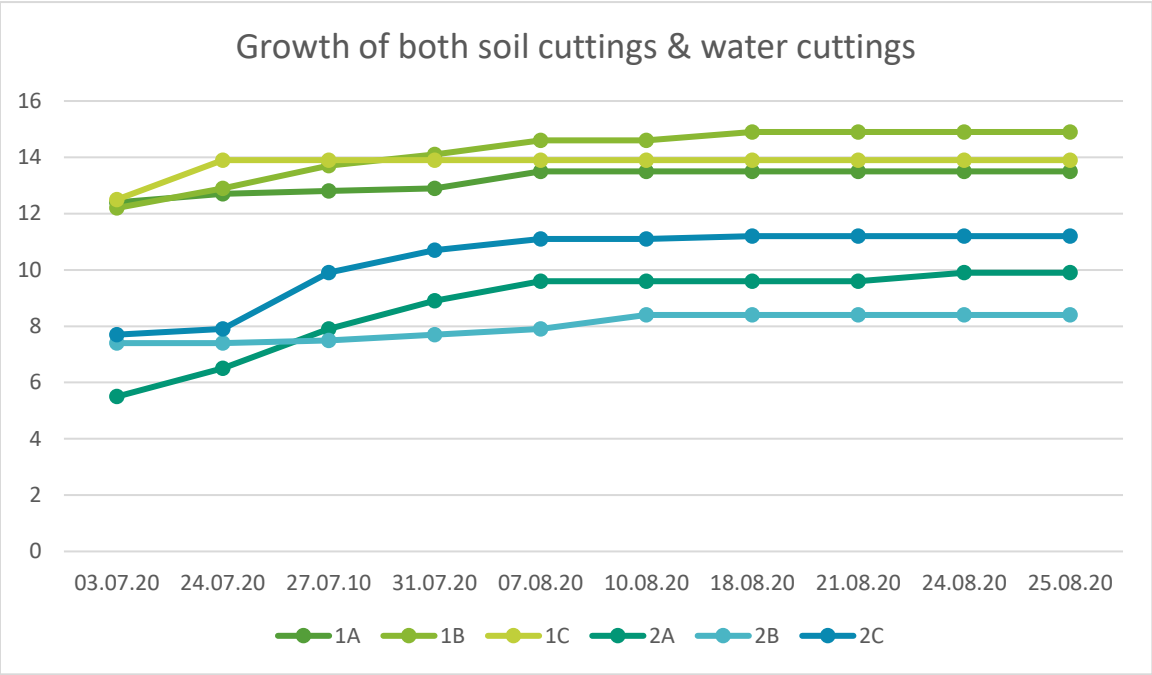
1. Collect equipment and materials.
2. Dress in lab coat, and put on safety googles, dust mask, and disposable plastic gloves (be mindful that most plastic gloves contain latex, please seek an alternative if reaction occurs).
3. Set out pots and beakers in an orderly fashion.
4. Place 1 willow cutting in each container, 3 in 3 pots & 3 in 3 beakers.
5. Fill each beaker with 80mL of water.
6. Fill each pot to the brim with potting soil.
7. Every few days, use a ruler to measure the water cuttings from the rim of the beaker to the tip of the cutting.
8. Every few days, use a ruler to measure the soil cuttings from the soil to the tip of the cutting.
9. Record these measurements.
10. Repeat steps 7-9 for a length of time.

RESULTS

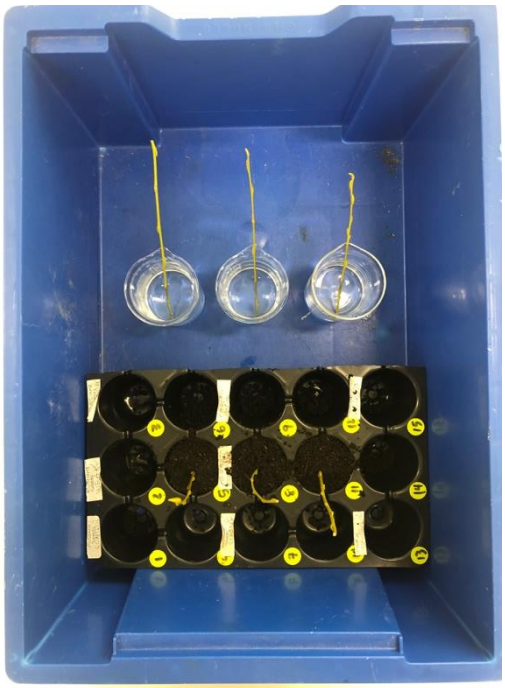
	03.07	24.07	27.07	31.07	07.08	10.08	18.08	21.08	24.08	25.08
1A	12.4	12.7	12.8	12.9	13.5	13.5	13.5	13.5	13.5	13.5
1B	12.2	12.9	13.7	14.1	14.6	14.6	14.9	14.9	14.9	14.9
1C	12.5	13.9	13.9	13.9	13.9	13.9	13.9	13.9	13.9	13.9
2A	5.5	6.5	7.9	8.9	9.6	9.6	9.6	9.6	9.9	9.9
2B	7.4	7.4	7.5	7.7	7.9	8.4	8.4	8.4	8.4	8.4
2C	7.7	7.9	9.9	10.7	11.1	11.2	11.2	11.2	11.2	11.2





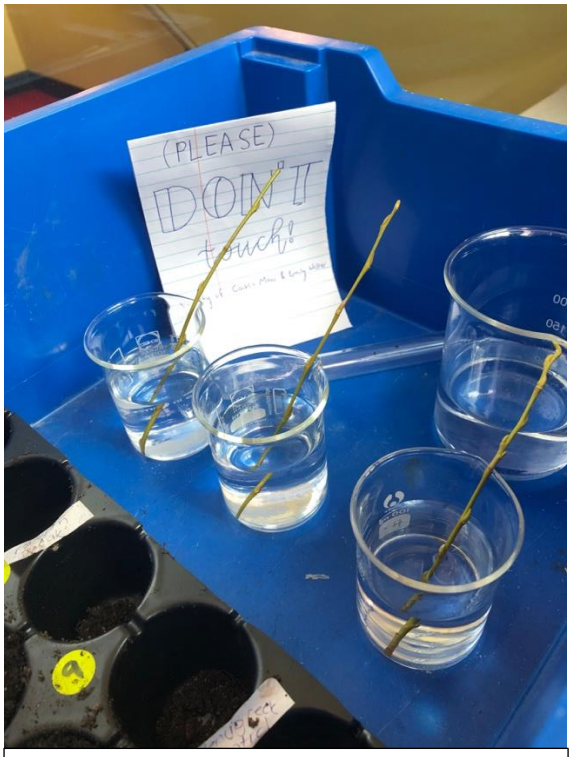


All six cuttings before the experiment



All six cuttings after being placed in their containers

3 July 2020 – Soil Cuttings



3 July 2020 – Water Cuttings

24 July 2020 – Soil Cuttings

24 July 2020 – Water Cuttings

27 July 2020 – Soil Cuttings

27 July 2020 – Water Cuttings

31 July 2020 – Soil Cuttings

31 July 2020 – Water Cuttings

4 August 2020 – Soil Cuttings

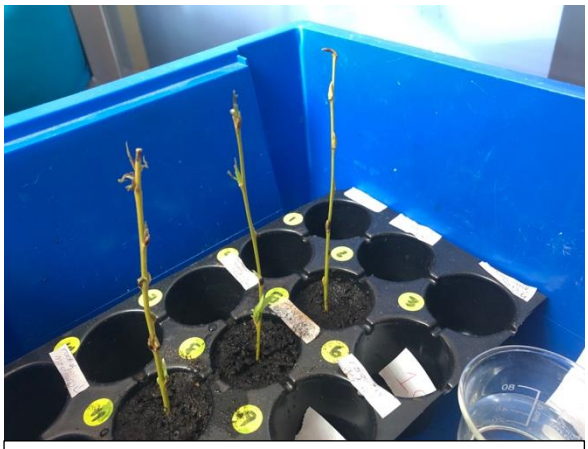
4 August 2020 – Water Cuttings

7 August 2020 – Soil Cuttings

7 August 2020 – Water Cuttings

10 August 2020 – Soil Cuttings

10 August 2020 – Water Cuttings



18 August 2020 – Soil Cuttings

18 August 2020 – Water Cuttings

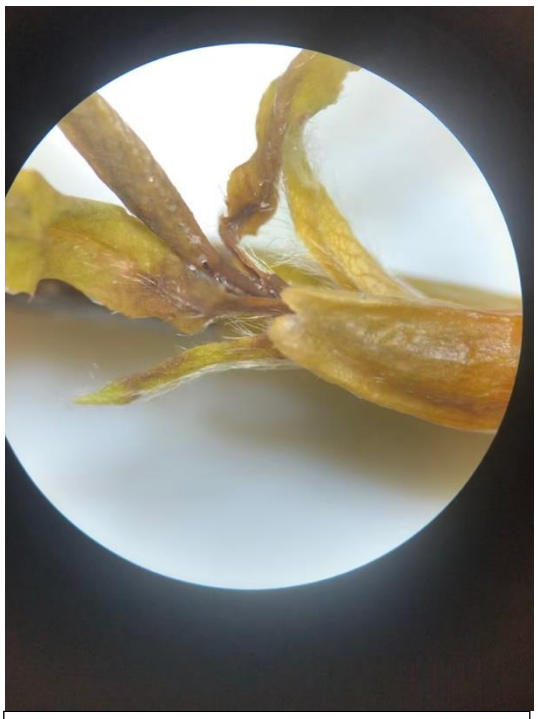
21 August 2020 – Soil Cuttings

21 August 2020 – Water Cuttings

24 August 2020 – Soil Cuttings

24 August 2020 – Water Cuttings

Close up image of leaf on Cutting 1A



Close up image of leaf on Cutting 1A

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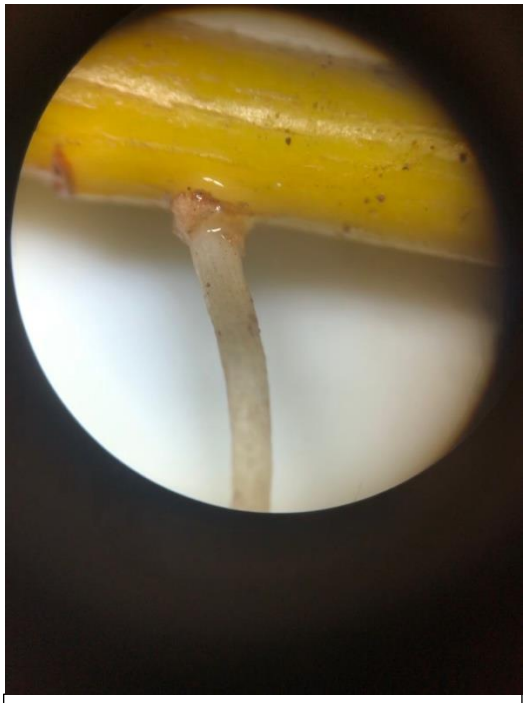
Close up image of leaf on Cutting 1A



Close up image of root on Cutting 1A

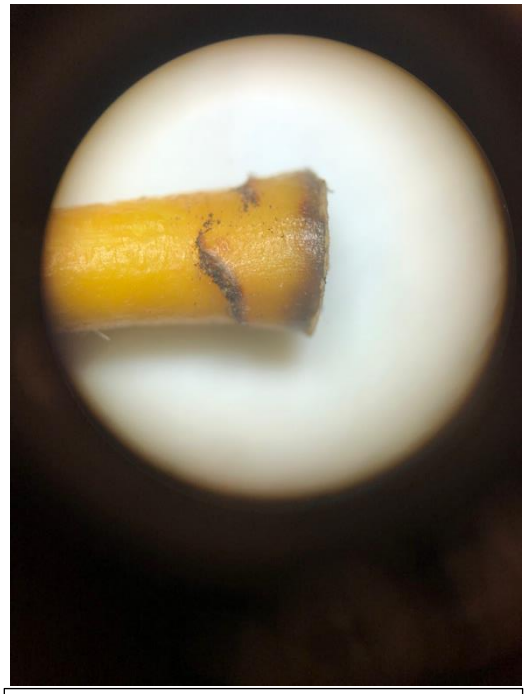
Close up image of root on Cutting 1A

Close up image of root on Cutting 1A

Close up image of stem tip on Cutting 1A

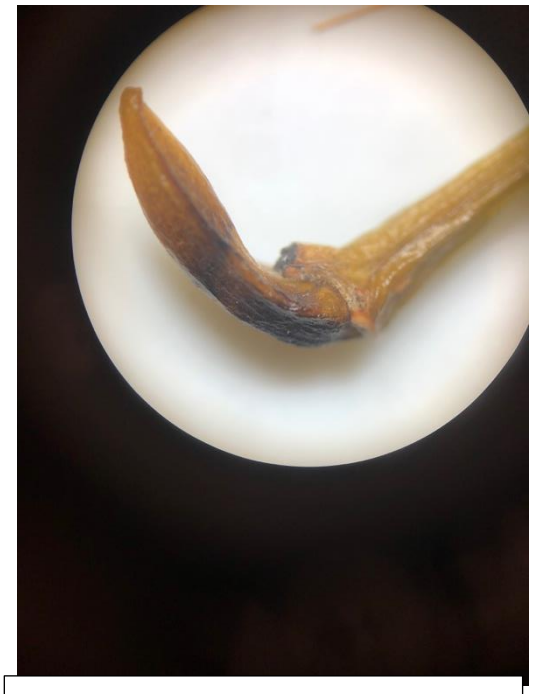
Close up image of newly budded leaf and its growth cap on Cutting 1B

Close up image of stem tip and leaves on Cutting 1B



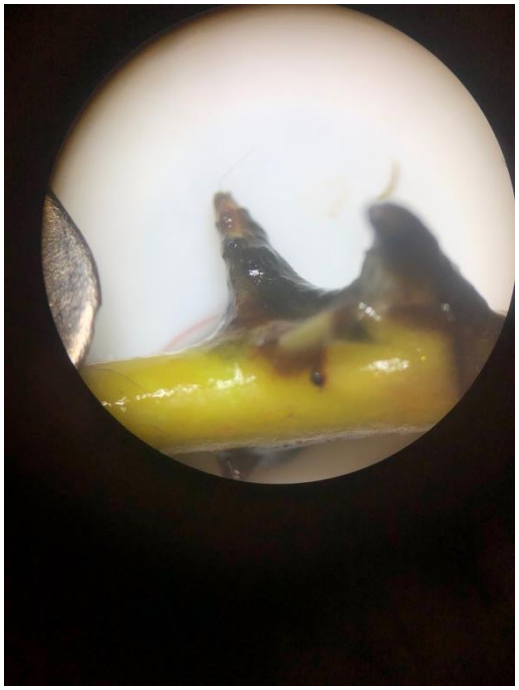
Close up image of stem base of Cutting 1C

Close up image of alive leaf sprout of Cutting 1C

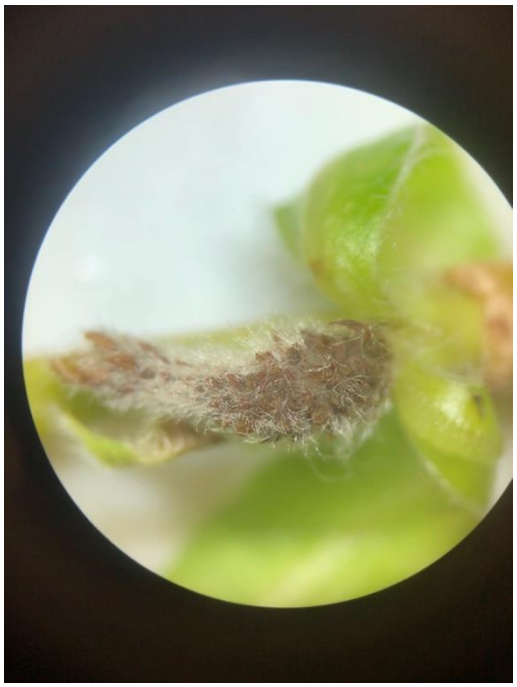


Close up image of stem tip of Cutting 1C

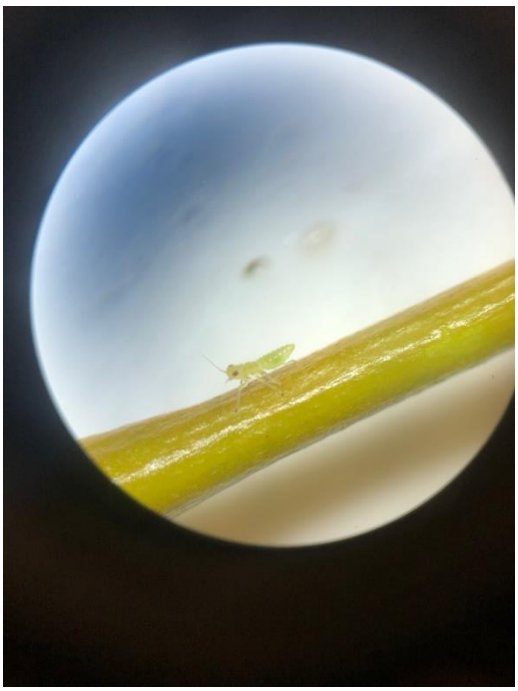
Close up image of dead leaf sprout of Cutting 1C



Close up image of newly budded root on Cutting 2A



Close up image of catkin on Cutting 2A



Close up image of insect found on the stem of Cutting 2A

Close up image of newly budded leaf and its growth cap on Cutting 2A

Close up image of roots on Cutting 2A

Close up image of newly budded leaves on Cutting 2B

Close up image of newly budded root on Cutting 2B

Close up image of root on Cutting 2C

Close up image of leaves and leaf bud on Cutting 2C

Close up image of roots on Cutting 2C

DISCUSSION OF EXPERIMENT

It is to be noted that the soil cuttings were measured from the surface of the dirt to the tip of the cuttings whereas the water cuttings were measured from the rim of the beaker to the tip of cuttings. This was done to ensure accurate measurements, as the level of the water may change throughout the course of the experiment. It is also recognised that potting soil is used only to replicate the conditions of the riverbanks in which willows would grow, and it is not the most accurate representation of this environment. This is because they two contain different things; potting mix usually consists of peat, composted bark, sand, perlite and limestone to tune their pH levels, whilst typical Tasmanian soil has large amounts of clay and a higher sodium level compared to most other soils.

It can be seen that the soil cuttings grew at a much slower rate than the water cuttings. Cutting 1A, the first soil cutting, grew at a steady pace to begin with. After approximately a month, its growth increased pace for about a week, then flattened out for the rest of the experiment, which was a period of about three weeks. In comparison, the Cutting 1B grew much faster at the start. It was shorter than Cutting 1A to begin with, but quickly surpassed it during the first month. After this first month, the growth levelled out for about half a week. After that point, it grew for a little over a week, remaining the tallest of all of the cuttings. From then on, its growth levelled out through to the end of the experiment. Cutting 1C had the worst growth out of all three soil cuttings. Whilst it grew much faster than the others in the first month, its growth levelled out almost immediately afterwards. It stayed at a constant height from that point on, withering slowly whilst the other cuttings continued to grow. Any leaves that had grown turned a dark brown and curled in up on themselves.

The water cuttings responded much better to their given conditions than the soil cuttings did. All three water cuttings grew to heights higher than the soil cuttings. Cutting 2A began as the shortest cutting by almost two centimetres. However, it quickly surpassed Cutting 2B in approximately a month.

Its growth levelled out for two weeks, then increased for three days, before finally levelling out for the remainder of the experiment's time frame. In contrast to the fast growth rate of Cutting 2A, Cutting 2B grew at a very slow pace. It only grew half a centimetre in the first month and was overtaken by Cutting 2A within that space of time. Then, the speed of growth picked up, with the cutting growing another half centimetre in only a week. After five weeks, the cutting was a centimetre taller than its original height. It was at this point that the plant stop growing. Its growth remained the same for the continuing weeks all the way through to the completion of the experiment. Cutting 2C, on the other hand, grew at a very similar pace to Cutting 2A. Whilst it only grew less than half a centimetre in the first three weeks, it quickly picked up pace, keeping within one and a half centimetres to two centimetres taller than the height of Cutting 2A. After about five and a half weeks, Cutting 2C had grown three and a half centimetres taller than its original height.

The hypothesis was somewhat supported as all plants did grow, albeit with some only growing less than a centimetre and a half. However, the majority of the hypothesis was disproven as the water cuttings grew faster and taller than the soil cuttings. This evidence suggests that willows are a hydrophilic plant, meaning they love water and respond better in aqueous environments. This is dangerous for the Tasmanian environment, because if willows congregate around water sources, the broken branches and limbs that break off as a reproductive means clog the waterways and negatively impact the aquatic ecosystems, such as reducing biodiversity and creating a monoculture in which aquatic life suffers.

CONCLUSION OF EXPERIMENT

At the conclusion of the experiment, the leaves and roots on the cuttings were counted and measured. The soil cuttings had very few leaves, with one having no leaves whatsoever, whilst the water cuttings all had well beyond a dozen leaves on each. The water cuttings also had many more roots than the soil cuttings, and the majority of the water cuttings' roots were longer than the soil cuttings' roots.

Cuttings in order from left to right and from bottom to top.

Cutting 1A

Cutting 1A had three dead leaves, as well as two roots. These roots were 0.7 centimetres long and 2.0 centimetres long. Cutting 1B had four alive leaves along with four dead leaves. It also had three roots, including two of the longest in the entire investigation. These three roots measured at 0.8 centimetres, 2.6 centimetres, and 3.6 centimetres. The last soil cutting, Cutting 1C, had no leaves and no roots. This was the cutting that died early on during the experiment due to unknown reasons.

Cutting 1B

Cutting 1C

Cutting 2A

Cutting 2B

Cutting 2C

The water cuttings grew much better than the soil cuttings. Cutting 2A had sixteen alive leaves. It also had grown one catkin, and it was the only cutting to do so. This cutting also had three roots, which measured 0.6 centimetres, 1.2 centimetres, and 1.9 centimetres. Cutting 2B had twenty alive leaves, the highest number of leaves that any single cutting grew. In fact, this cutting had more alive leaves than the combined number of dead and alive leaves grown by the soil cuttings. Cutting 2B also had two roots that grew to lengths of 0.4 centimetres and 0.8 centimetres. The last water cutting, Cutting 2C, had a total of fourteen alive leaves as well as six roots; the highest number of roots that any single cutting grew. These roots measured at 0.3 centimetres, 0.7 centimetres, 1.2 centimetres, 2.7 centimetres, 3.0 centimetres, and finally, 4.1 centimetres; which was the longest root grown in the entire investigation.

CONCLUSION

In conclusion, the cuttings grew best in environments that were high in aqueous content. The cuttings suffered when grown in soil-based containers but thrived when grown in just water. This corroborates known research regarding willows and their behaviour in different environments. It is known that willows prosper close to waterways and on riverbanks. This is supported by the research demonstrated by the given research.

PART TWO

AIM

To investigate and record how willow leaves (non-native leaves) and gum leaves (native leaves) affect the qualities of water, simulating the natural environment of rivers and aquatic habitats.

THEORY

THE EXPERIMENT

In this second experiment the Tasmanian environment is being simulated by putting alive willow leaves in one beaker, dead willow leaves in another beaker, native eucalyptus leaves in another beaker , and leaving one beaker empty. The first two beakers are simulating river water with willows and the next 2 beakers are simulating a river without willows.

There are some limitations to the experiment. The water that is used in the experiment is tap water, which usually contains added chemicals that river water does not. For example in Australia chlorine is added to tap water to kill microorganisms that may cause disease and fluoridate is also added to prevent tooth decay. River water contains many carbon compounds that tap water does not.

Also, unlike the simulation in the experiment, it would be rare to have a river with purely willows growing along it. In Tasmania, most rivers with willows have a mix of native trees as well, such as conifers like celery top pine and Huon pine, various varieties of eucalyptus and other shrubs and fauna. Over a long period of time, if willows are left to grow and are not controlled, they tend to take over and environment, reducing biodiversity to become a monoculture. This is rarely seen in Tasmania due to the introduction of willows only happening less than 200 years ago, which is not enough time for them to completely take over a landscape. However, if in this experiment native leaves and willow leaves were put

together it would be unknown what species was affecting the water more. For this reason, the two types have been separated so each separate variety impact can be accurately gauged.

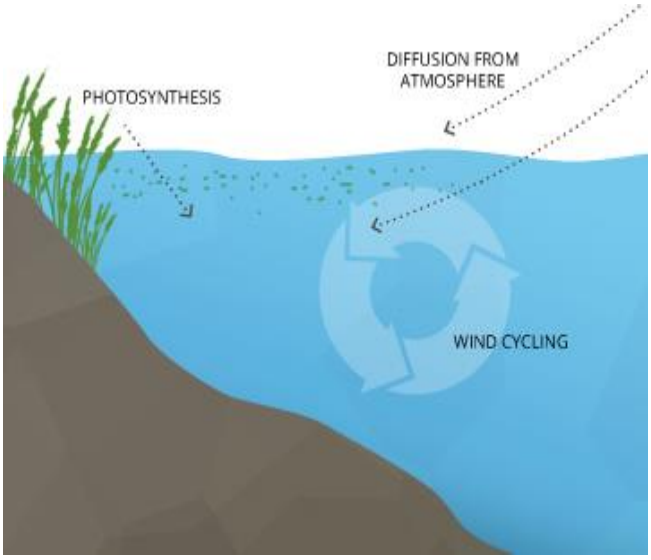
The water quality of each beaker will be tested each week to see how much the water was affected from week to week. Testing will be comprised of 4 different sectors: dissolved oxygen, turbidity, pH levels, and salinity.

DISSOLVED OXYGEN

Dissolved oxygen is a measure of how much oxygen is dissolved in the water and is available to living aquatic organisms. It refers to the level of free, non-compounded oxygen present in water or other liquids. Although water molecules contain an oxygen atom, this is not what is needed by aquatic organisms. Non-compound oxygen, or free oxygen (O2), is oxygen that is not bonded to any other element. Dissolved oxygen is the presence of these free O2 molecules within water. The bonded oxygen molecule in water (H2O) is in a compound and does not count toward dissolved oxygen levels. Only a small amount of oxygen, on average around ten molecules per million of water is actually dissolved oxygen. Rapidly moving water, such as flowing rivers and streams tend to contain a lot of dissolved oxygen, whilst stagnant and still bodies tend to contain less.

Dissolved oxygen is vital to many forms of aquatic life such as fish, crustaceans, bacteria, invertebrates and aquatic plants. These organisms use dissolves oxygen in respiration, similar to how humans need air to breathe. Fish and crustaceans obtain dissolved oxygen for respiration through their gills, while plant life and phytoplankton obtain dissolved oxygen for respiration when there is no light for photosynthesis . The amount of dissolved oxygen needed varies from creature to creature. Bottom feeders, crabs, oysters and worms need minimal amounts of dissolved oxygen (1-6 mg/L), while shallow water fish need higher levels (4-15 mg/L).

Microbes such as bacteria and fungi also require dissolved oxygen. They use dissolved oxygen to decompose organic material at the bottom of a body of water. Microbial decomposition is an important contributor to nutrient recycling.



Dissolved oxygen enters water through the air or as a plant by-product. From the air, oxygen can slowly diffuse across the water's surface from the surrounding atmosphere, or be mixed in quickly through aeration, whether natural or man-made 7. The aeration of water can be caused by wind (creating waves), rapids, waterfalls, ground water discharge or other forms of running water. Man-made causes of aeration vary from an aquarium air pump to a hand-turned waterwheel to a large dam. Dissolved oxygen is also produced as a waste product of photosynthesis from phytoplankton, algae, seaweed and other aquatic plants.

Figure 2.1.1 Oxygen enters a stream or similar body of water from the atmosphere or as a plant by-product.

Figure 2.1.2 A lake in which eutrophication has occurred which has lead to an algae bloom.

When willows lose their leaves in Autumn, most fall into the stream and create a huge flush of organic matter. Willows have hundreds of thousands of leaves per tree, so a river with many willows along its banks could have potentially millions of leaves falling into it. This can lead to potentially millions of willow leaves going into a single body of water.

When a lake or river has such an excess of organic matter a process known as eutrophication often occurs. This means that there is excessive richness of nutrients in the lake or body of water. Eutrophication normally leads to other environmental disasters, such as algal blooms, killing of fish and a chain reaction in the ecosystem. The latter happens because there is an overabundance of algae and plant matter. The excess algae and plant matter eventually decompose, producing large amounts of carbon dioxide. This lowers the pH of seawater, a process known as ocean acidification. Acidification slows the growth of fish and shellfish and can prevent shell formation in bivalve molluscs. If rivers with willows in them eventually lead to the ocean this can be extremely harmful.

TURBIDITY

Turbidity is a measure of water clarity through the number of particles present. It is a measurement of the amount of light that is scattered by material in the water when a light is shined through the water sample. The higher the intensity of scattered light, the higher the turbidity. Materials that can cause water to be turbid include clay, silt, organic matter, algae and other microscopic organisms. If water is clear, such as drinking water it will have low turbidity, whilst rivers will usually contain sediments and organic matter which leads to them having a higher turbidity. Common things that increase turbidity levels include sediment, silt, sand, decaying plant matter and algae.

Turbidity levels vary greatly from river to river, however most flowing rivers in Tasmania are relatively clear and have fairly low turbidity levels of around 20 NTU (Nephelometric turbidity units). It is not considered healthy for bodies of water to have large amount of turbidity as it can clog fishes gills, make it difficult for them to see and catch prey, and also may bury and kill eggs laid on the bottom of rivers.

As far as it is known, no research has been done on willow leaves effect on the turbidity of water. However, it is expected that because willows have thin, flimsy leaves they will break apart easily and increase the turbidity of the water.

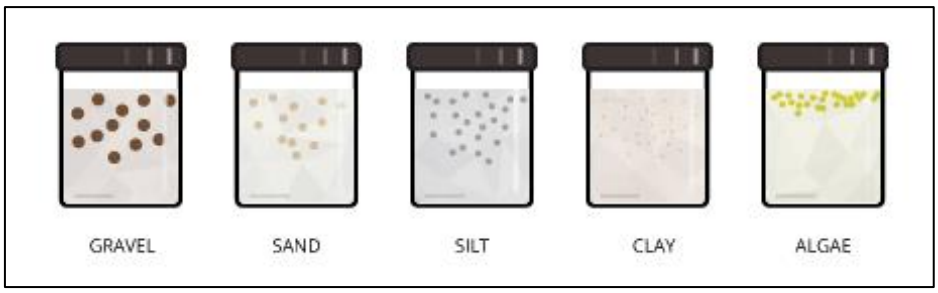


Figure 2.2.1 Suspended solids in reference to water clarity.

PH

pH is a measure of the relative amount of free hydrogen and hydroxyl ions in the water. It measures the acidity or alkalinity of a substance on a scale from 0-14, with 7 being neutral, a reading of less than 7 indicating acidity and a reading of greater than 7 being basic or alkaline. The Australian pH guidelines for a healthy body of fresh water is 6.5 - 9.0. The pH or acidity of Tasmanian streams is typically in the range 5.5 - 7.5. In some Tasmanian rivers, the humic acid, a chemical produced by decaying plants lowers the pH. For humic rich Tasmanian lakes and rivers, the pH range is 4.0 to 6.5. pH values in streams are influenced by a wide range of factors such as the surrounding geology, soil chemistry, vegetation and land use practices. As far as it could be found, no research has been conducted on how willow leaves affect the pH of a body of water.

SALINITY

The term salinity refers to the concentration of salts in water or soil. Salts are highly soluble in surface and groundwater and can be transported with water movement. There are two types of salinity:

Primary salinity refers to large salt deposits that are a natural feature of vast areas of the Australian landscape, stored deep in soils or as surface salt deposits and salt lakes.

Secondary salinity refers to additional salt transported to the soil surface or waterways, increased due to altered land use (vegetation clearance, poor land management, irrigation and industrial practices).

Salinity levels in streams should be less than1000 μS/cm to be considered healthy. Otherwise they can damage aquatic ecosystems. Since willow leaves don't contain any salt, it is expected that they won't have any effect on the salinity levels.

HYPOTHESIS

The alive willow leaves will affect the water to the highest degree, decreasing the amount of dissolved oxygen, increasing the salinity, increasing the turbidity, and lowering the pH (making the water more acidic). These leaves will then be followed by the gum leaves, then by the dead willow leaves, with the control affecting the water the least.

VARIABLES

Controlled: size of beakers, amount of water, amount of leaves, time of sampling

Dependent: concentration of particles in the water as shown by turbidity

Independent: different types of leaves; eucalyptus, green willow, dead willow

MATERIALS

-4 large 300ml beakers

-distilled water

-native eucalyptus gum leaves

-dead willow leaves

-live willow leaves

-tap water

-dissolved oxygen probe

-salinity probe

-turbidity measurer

-pH meter

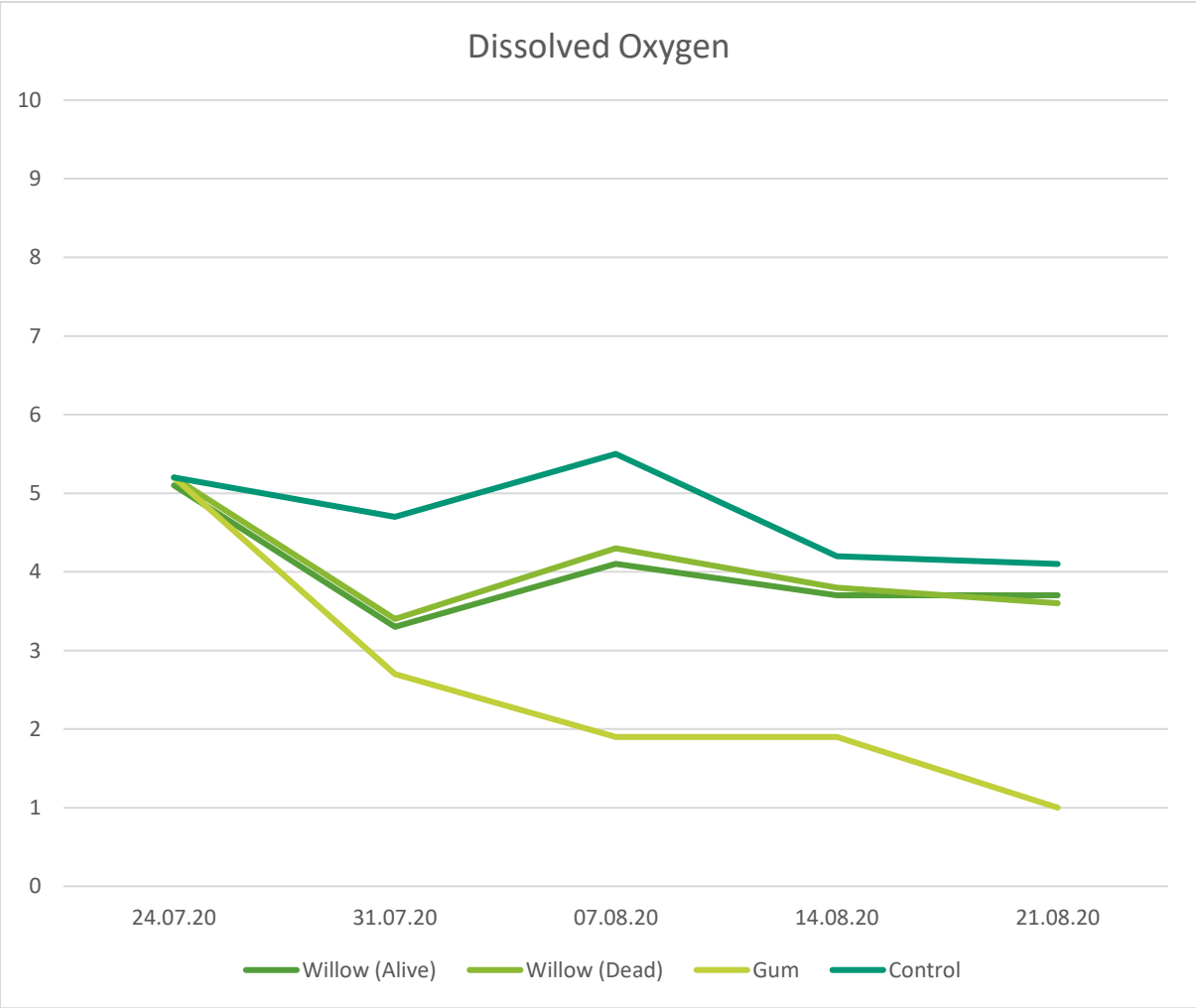
METHOD

1) Gather the four beakers and wash them with distilled water.
2) Put 5-6 native leaves (including gumnuts and branches) into the first beaker, 5-6 alive willow leaves (including catkins and stems) into the second beaker, 5-6 dead willow leaves (including catkins and stems) into the third beaker, and leave the final beaker empty.
3) Fill the beakers with 200 ml water each.
4) Store in a dry space with a constant temperature
5) Once a week for 5 weeks measure the turbidity, dissolved oxygen, ph. And salinity of each beaker. Record the results.

RESULTS

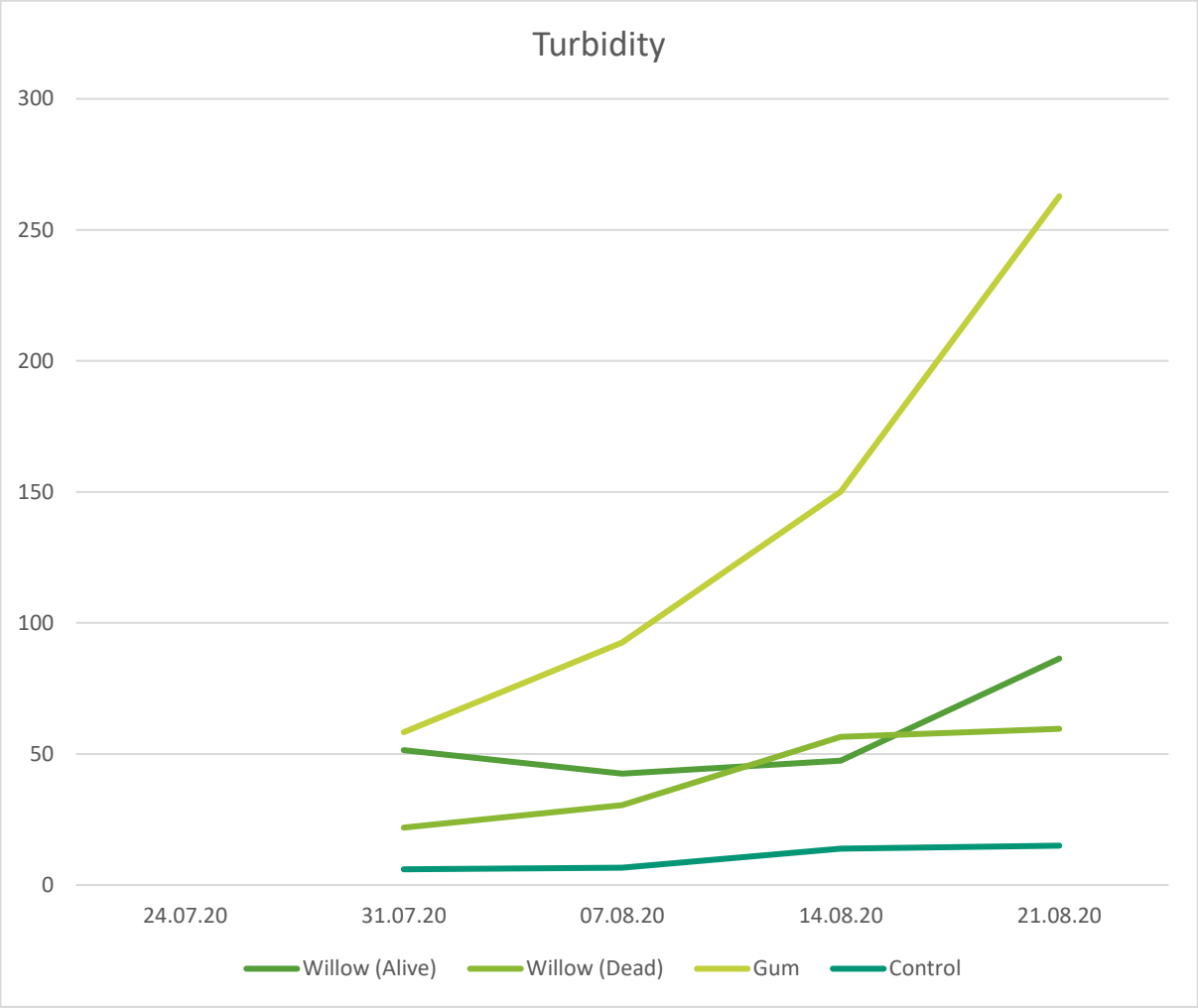
DISSOLVED OXYGEN:

	24.07.20	31.07.20	07.08.20	14.08.20	21.08.20
Willow (Alive)	5.1mg/L	3.3mg/L	4.1mg/L	3.7mg/L	3.7mg/L
Willow (Dead)	5.2mg/L	3.4mg/L	4.3mg/L	3.8mg/L	3.6mg/L
Gum	5.2mg/L	2.7mg/L	1.9mg/L	1.9mg/L	1.0mg/L
Control	5.2mg/L	4.7mg/L	5.5mg/L	4.2mg/L	4.1mg/L



TURBIDITY:

	24.07.20	31.07.20	07.08.20	14.08.20	21.08.20
Willow (Alive)	x	51.4NTU	42.5NTU	47.5NTU	86.4NTU
Willow (Dead)	x	21.9NTU	30.5NTU	56.5NTU	59.6NTU
Gum	x	58.3NTU	92.5NTU	150.1NTU	262.8NTU
Control	x	6.0NTU	6.6NTU	13.9NTU	15.0NTU

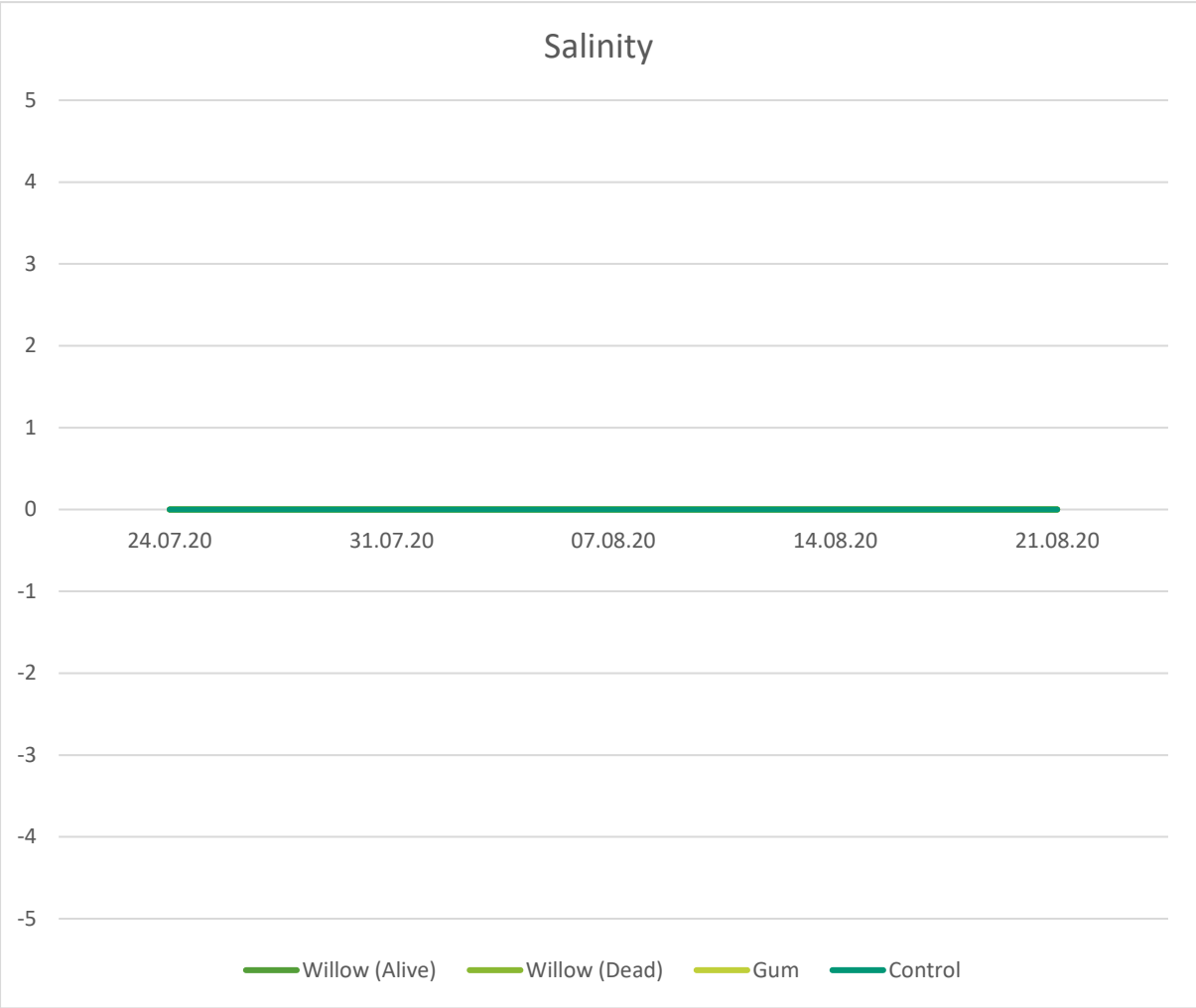


PH:

	24.07.20	31.07.20	07.08.20	14.08.20	21.08.20
Willow (Alive)	7.7	7.8	6.4	6.4	6.7
Willow (Dead)	8.0	8.6	6.4	6.4	6.4
Gum	8.0	7.1	6.9	6.5	6.4
Control	7.8	8.4	6.4	6.3	6.7

SALINITY:

	24.07.20	31.07.20	07.08.20	14.08.20	21.08.20
Willow (Alive)	0mg/L	0mg/L	0mg/L	0mg/L	0mg/L
Willow (Dead)	0mg/L	0mg/L	0mg/L	0mg/L	0mg/L
Gum	0mg/L	0mg/L	0mg/L	0mg/L	0mg/L
Control	0mg/L	0mg/L	0mg/L	0mg/L	0mg/L



DISCUSSION

PREPARATION OF EXPERIMENT

In the preparation of this experiment, all equipment was collected and laid out. The four beakers were filled with 80ml of water, and the leaves were added to three of them: gum leaves to one, alive willow leaves to another one, and dead willow leaves to the last one. One beaker was left empty as a control.

At this point, all water in the beakers was clear, and the leaves had not had any effect.

DISCUSSION OF EXPERIMENTAL PROCESS

Recordings were taken approximately every week for a period of about a month. All four sectors of water quality were tested: dissolved oxygen, turbidity, pH levels, and salinity. There were five recordings in total.

Some of the results don't seem to align with common sense or logic, such as how the pH levels of three of the four samples increased before decreasing. Any inconsistencies within the experiments in regard to readings were on account of the tools use to take such readings. As the tools were used by others between the times they were used for this experiment, underlying settings changes or alternate calibrations may have occurred without knowledge to those overseeing the experiment. All readings are analysed with this knowledge in mind, and the following interpretation of the results of this experiment must be scrutinised under this lens.

All samples decreased the amount of dissolved oxygen in the water. The dissolved oxygen levels in the control sample only decreased a small amount, approximately 1mg/L. This was due to the fact that there were no leaves or other organic material in the sample. The dissolved oxygen levels deceased slightly because the water was stagnant and therefore was not getting a constant flow of oxygen like a river environment would have happen. The dissolved oxygen levels of the alive willow leaves and the dead willow leaves decreased in a very similar fashion and at a very similar rate to each other. The measurements of these two samples stayed within 0.2mg/L the entire experiment, with the dissolved oxygen level of the dead willow leaves remaining higher throughout the experiment until the very end. The final results of the alive willow leaves and the dead willow leaves were within 0.1mg/L, at measurements of 3.7mg/L and 3.6 mg/L respectively. The reason these samples had very similar effects to each other is because both samples had willow leaves. The chemicals and organic content of the willows has little variation between alive leaves and dead leaves. This means that the proceedings of these two samples in the experiment remained very similar throughout.

The gum leaves affected the water quality the most, with the measurements dropping from 5.2mg/L to 1.0mg/L. This was the biggest change in any of the water samples in regards to dissolved oxygen. This is likely due to organic content in the gum leaves.

The second area of water quality tested was turbidity. The turbidity of the water samples was only tested four times instead of five. This was because the tool for measuring turbidity was not available for use in the first week of the experiment. As such, results for this sector of water quality is only available for the last three weeks of the experiment rather than the full month. The turbidity of the control sample was affected the least, with the measurements increasing from 6NTU to 15NTU. This was simply to do with the amount of dust that had settled on the surface of the control water sample throughout the experiment. When the sample was stirred in preparation for turbidity testing, those dust particles became submerged in the water and affected the turbidity reading. There was a greater difference in the turbidity measurements between the alive willow leaves sample and the dead willow leaves sample than the dissolved oxygen measurements. The alive willow leaves sample had a much higher turbidity reading than the dead willow leaves sample. The turbidity of the alive willow leaves sample dropped after a week then rose again, whilst the turbidity of the dead willow leaves sample increased throughout. This drop in the alive willow leaves sample measurements may have been due to the sample not being stirred properly before being tested, as the particles in the water would have sunk to the bottom. However, apart from this, the measurements seem relatively accurate. The final sample, the gum leaves water beaker, had turbidity readings that skyrocketed above the rest. Over the three weeks, it increased exponentially to the point where, at the conclusion of the experiment, the final reading was over double the second-highest reading. This high reading was not due to physical particles that had broken off from the leaves, rather it was due to the large concentration of tannic acid that leeched into the water sample.

pH level was the third water quality sector that was tested. There were some issues with the pH testing equipment, therefore these readings were a combination of that of pH tester paper as well as that of a pH meter. These issues can be clearly seen in the second and fifth readings of this experiment. At these points, the measurements were at an increase from the measurement previous. This makes no logical sense for the water sample to become more alkaline then drop to being more acidic, before becoming more alkaline again. It is thought that these inconsistencies are due to equipment and technical issues. All the readings were similar at the beginning of the experiment, reading as slightly alkaline, which is not unexpected with Hobart tap water. Springs are typically natural sources of alkaline water, and as the majority of Hobartian tap water is fed from Kunanyi, it comes as no surprise that the pH of the water is slightly higher than neutral.

The salinity levels were unchanging throughout the entire experiment. Over the span of the five weeks, no saline content was measured, therefore all measurements of all four samples sat at 0mg/L.

CONCLUSION OF EXPERIMENT

At the conclusion of the experiment, the water in three of the beakers appeared significantly different.

The water in which the gum leaves resided was a deep red colour. It was semi-transparent and had a strong aromatic odour that smelled tangy. The water from the beaker which contained the alive willow leaves was slightly murky but still clear. It had some particles of broken leaves swirling around and was a light brown colour. The water in dead willow leaves beaker stayed relatively the same, decently clear with a slight brown tinge. The fourth and final beaker, the control beaker with only water stayed relatively the same in appearance.

After the experiment's conclusion, the remaining organic matter from the beakers was laid out and analysed. The gum leaves were completely intact and had retained their original green colouring with a few yellow patches. The leaves had not broken down in any way. The willow leaves that had been alive and a light green colour at the start of the experiment had turned a dark brown. The leaves had broken down somewhat, with jagged edges and pieces missing. The leaves were brittle and broke apart easily under inspection. The willow leaves that had been dead the entire experiment remained the same in colour, a dark deep brown. However, they were almost completely broken down, the edges uneven and pieces breaking off onto the table.

TAKING IT FURTHER

Whilst the hypothesis was not supported, this does not inherently mean that gum leaves, and other native leaves, are worse for the environment than willow leaves. When considering the shedding of leaves in regard to each type of tree, willow trees shed much more than gum trees. This means that, whilst gum leaves have a greater effect on water quality individually, the concentration of willow leaves in waterways would mean that willow trees have a more detrimental impact to the Tasmanian aquatic environment than gum trees.

In the future, this original experiment could be taken further by testing different concentrations of willow leaves in water. For example, one beaker could have with 1 leaf, the next could have 5, the next could have 10, the next could have 20 and the final one could have 40. Then, the four sectors that were tested in the original experiment could be tested again; dissolved oxygen, pH, turbidity and salinity. It could be observed how the different concentrations of willow leaves affect the water differently. This will allow the opportunity for greater understanding in regard to the full effects that willows have on the quality of water, which could then be applied to real-world scenarios such as rivers and waterways with willows near them.

CONCLUSION

As observed by the results, it is obvious that the gum leaves have the greatest effect on water quality, especially in three of the four sectors of water quality: dissolved oxygen levels, pH readings, and turbidity readings. This disproves and does not support the original hypothesis of this experiment, which stated that the alive willow leaves would have the greatest impact on all four sectors of water quality, be decreasing the amount of dissolved oxygen, increasing the level of salinity, increasing the turbidity, and by acidifying the water (decreasing the pH level).

However, the second half of the hypothesis regarding the effects of the dead willow leaves water sample and the control water sample on water quality was supported by this experiment.

OVERALL CONCLUSION

This investigation allowed for a deeper understanding into how willows affect the Tasmanian landscape and aquatic environment.

ACKNOWLEDGEMENTS

We would like to acknowledge those who have assisted us throughout this investigation.

Firstly, we would like to acknowledge our Science teacher, Mrs Heather Omant. She has been our teacher for many years, and we owe her a great deal. We thank her for the phenomenal amount of time and effort she has put into supporting us as scientists and as students.

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- Greg Lundstrom
- Polly Buchhorn
- Michelle Storer
- Alister Hazeldine

We acknowledge the time, effort, and resources the above people have spent assisting us. We give thanks to them and greatly appreciate their contributions to this scientific endeavour. Their knowledge and expertise have been invaluable over the course of this investigation.

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- Louise Lieschke
- Bill Crawford
- Greg Robertson
- Denise Colvin
- Darcy Vickers
- Liz Quinn
- Chris Johns
- Glenys Nicholls

LOGBOOK

DATE	TIME	DETAILS	PEOPLE	TOTAL
29.05.20	1h 40m	Idea thinking/brainstorming	E	
29.05.20	1h 40m	Idea thinking/brainstorming	C	
01.06.20	1h 45m	Idea thinking/brainstorming	E	
01.06.20	1h 45m	Idea thinking/brainstorming	C	
05.06.20	1h 50m	Writing letters to council	E	
05.06.20	1h 50m	Writing letters to council	C	
05.06.20	0h 30m	Sending emails	E	
06.06.20	0h 30m	Sending emails	E	
11.06.20	0h 30m	Discussing availability of data with parents	C	
12.06.20	1h 40m	Replying to emails	E	
12.06.20	1h 40m	Replying to emails	C	
15.06.20	1h 45m	Replying to emails + collecting data	E	
15.06.20	1h 45m	Replying to emails + collecting data	C	
16.06.20	0h 10m	Discussing with parents	C	
19.06.20	1h 50m	Replying to emails + planning for Part 1	E	
19.06.20	1h 50m	Replying to emails + planning for Part 1	C	
26.06.20	1h 40m	Starting experiment + drafting report + doing risk assessment	E	
26.06.20	1h 40m	Starting experiment + drafting report + doing risk assessment	C	
29.06.20	1h 45m	Completing risk assessment + watering plants + theory	E	
29.06.20	1h 45m	Completing risk assessment + watering plants + theory	C	
03.07.20	1h 40m	Measuring + recording for Part 1	E	
03.07.20	1h 40m	Measuring + recording for Part 1	C	
24.07.20	1h 40m	Starting Part 2 of investigation	E	
24.07.20	1h 40m	Starting Part 2 of investigation	C	
27.07.20	1h 45m	Recording results so far, creating graphs	E	
27.07.20	1h 45m	Recording results so far, creating graphs	C	
31.07.20	1h 40m	Measuring cuttings, writing report, testing water	C	
03.08.20	40m	Researching	C	

05.08.20	45m	Writing theory	E	
07.08.20	1h 30m	Measuring cuttings, testing water, writing report	E	
07.08.20	1h 30m	Measuring cuttings, testing water, writing report	C	
07.08.20	1h 25m	Writing report	E	
07.08.20	50m	Writing report	E	
09.08.20	1h	Writing report	E	
09.08.20	30m	Brainstorming	C	
10.08.20	1h 35m	Writing report, measuring willows	E	
10.08.20	1h 35m	Writing report, measuring willows	C	
11.08.20	10m	Adding to graphs + data	C	
12.08.20	30 m	Writing report	E	
12.08.20	20m	Discussion and brainstorm regarding report	C	
13.08.20	15m	Amending and altering incorrect data in graphs	C	
14.08.20	1h 45m	Writing report	E	
14.08.20	1h 45m	Writing report	C	
15.08.20	1h	Writing report	E	
16.08.20	1h 10m	Writing report	E	
18.08.20	1h 45m	Writing report, measuring cuttings collecting results.	E	
18.08.20	1h 45m	Writing report, measuring cuttings collecting results.	C	
18.08.20	30m	Writing report	E	
19.08.20	25m	Writing report	E	
21.08.20	1h 45m	Writing report	E	
21.08.20	1h 45m	Writing report	C	
23.08.20	20m	Adding to discussion + finishing references	C	
24.08.20	1h 50m	Finishing 1st experiment	E	
24.08.20	1h 50m	Writing report	C	
25.08.20	30m	Writing report	C	
25.08.20	1h 50m	Finishing Part 1 Experiment	E	
28.08.20	1h 55m	Finishing 2nd experiment	E	
28.08.20	1h 55m	Adding photos	C	
28.08.20	1h 45m	Writing report	E	
28.08.20	1h 45m	Writing report	C	
30.08.20	4h 30m	Downloading photos	C	

01.09.20	2h 00m	Preparing proposal	E	
01.09.20	2h 00m	Writing discussion	C	
01.09.20	1h 00m	Writing discussion	C	
01.09.20	1h 00m	Writing theory	E	
02.09.20	30m	Calculating times	C	96h 00m
02.09.20	1h	Writing goals + writing discussion	E	
02.09.20	1h	Calculating	C	98h 00m
07.09.20	1h 45m	Writing report	C	
07.09.20	50m	Writing report	E	
08.09.20	1h 50m	Writing report	C	
08.09.20	1h 50m	Writing report	E	
08.09.20	20m	Writing discussion	C	
08.09.20	3h 40m	Adding photos to Part 1 + graphs to Part 2	C	
09.09.20	1h	Finishing addition of photos + captions to Part 1	C	
11.09.20	1h 30m	Writing discussion part 2	C	
11.09.20	1h 30m	Writing discussion part 2	E	112h 15m
11.09.20	1h 30m	Writing part 2 theory	C	
11.09.20	1h 30m	Writing part 2 theory	E	
12.09.20	3h 40m	Writing Part 2 discussion	C	
15.09.20	1h 40m	Finishing discussions	C	
18.09,20	1h 40m	Finishing theory	E	
18.09.20	1h 40m	Finishing discussions	C	
20.09.20	1h	Finalising report	E	124h 55m
13.01.20	5m	Finalising time log	C	125h

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RISK ASSESSMENT **St Mary's College, Hobart Students**

An investigation into salix fragilis and its effects on the Tasmanian environment

Written by: Caitlin Marr, Emily Walter **Commenced on:** 29 Jun 2020 **Expires:** 29 Sep 2021

Classes for which experiment is required

Teacher:	**Year Group:**	**Room**	**Period**	**Date**
Mrs Heather Ornant (training code 1)	10	S324	1&2	Fri 3/7/20

Procedure or reference, including variations

refer to report

Equipment to be used

glass beaker, 250 mL to 1 L

Potential hazards
Breakage of beaker. Cuts from chipped rims.

Standard handling procedures
Inspect and discard any chipped or cracked beakers, no matter how small the damage. Sweep up broken glass with brush and dustpan; do not use fingers.

disposable plastic gloves

Potential hazards
ALLERGY ALERT. May easily be punctured, allowing entry of liquid. Latex gloves may cause an allergic reaction to some people; check for latex allergies before use. Check for talc allergies, if gloves are powdered with talc. Organic solvents may damage gloves.

Standard handling procedures
Take care not to puncture. Check for punctures before use. Use a type of glove that is suitable for the chemicals to be used.

lab coat

Potential hazards
Flammable. Sleeves may catch on objects and knock them over.

Standard handling procedures
Clean regularly. Kep clear of naked flames.

dust mask

Potential hazards
Provides limited protection against large airborne particles. Typically ~60% effective, due to poor design, leading to poor fit to face, and large pore size in mask material.

Standard handling procedures
Use a N95 or P2 rated mask in preference to a cheap dust mask, since it is much more effective in preventing inhalation of particles.

safety goggles

Potential hazards
May transfer pathogens from one user to the next, e.g. eye infections, flu or coronavirus, which may enter the body through the conjunctiva. Scratched or dirty goggles may hinder vision, causing headaches during prolonged use.

Standard handling procedures
Each student should preferably have own safety goggles. If safety goggles are shared, they should be disinfected between use. Safety goggles may be stored in a tank of detergent solution and removed as needed, rinsed and dried before use. Avoid scratching lenses during storage. Check and, if necessary, clean goggles before each use. Ensure that the safety goggles fit the shape of the face and provide protection around the edges, especially at the bottom (against upward splashes of liquid).

medium borosilicate glass test tube, ~150 mm x 15 mm (Pyrex test tube, ~150 mm x 15 mm)

Potential hazards
Breakage of test tubes. Cuts from chipped test-tube rims. Small test tubes more likely to eject material during exothermic reactions.

Standard handling procedures
Inspect and discard any damaged test tubes. Sweep up broken glass with brush and dustpan; do not use fingers. Do not insert finger in test tube, since it may become stuck and swell. Borosilicate test tubes are generally recommended if the contents are to be heated. Rimless borosilicate test tubes are known as "ignition tubes", but offer no advantage over tubes with rims for heating solids over a Bunsen flame.

Biologicals and food to be used

potting mix

Potential hazards	*Standard handling procedures*
Possibility of microbial contamination, including Legionella. Check for anyone with particular allergies or who is immunocompromised.	Wear gloves and a well-fitting face mask (certified N95 or P2; not a cheap "dust mask"). The day before, open bag outside in a well-ventilated area, pour carefully into a wheelbarrow and dampen slightly to reduce dust production. Wash hands thoroughly after each use. Ensure students wash hands thoroughly.

water, 43.5°C or less (cold-warm)

Potential hazards	*Standard handling procedures*
Water below 43.5°C is generally considered safe for adults and children. Cold water causes numbness and hypothermia if exposure is prolonged.	Water spilled on the floor may be a slip hazard. Do not drink in the classroom, due to the possibility of contamination.

Others

willow

Knowledge

I/we have read and understood the potential hazards and standard handling procedures of all the equipment, chemicals and biological items, including living organisms.
I/we have read and understood the Safety Data Sheets for all hazardous chemicals used in the experiment.
I/we have copies of the Safety Data Sheets of all the hazardous chemicals available in or near the laboratory.

Agreement by student(s)

I/we, Caitlin Marr, Emily Walter, agree to conduct this experiment safely in accordance with school rules and teacher instructions.

Risk assessment

I/we have considered the risks of:

fire or explosion	breakage of equipment	exposure to pathogens	waste disposal
chemicals in eyes	injuries from equipment	injuries from animals	improper labelling/storage
inhalation of gas/dust	rotating equipment	intense light/lasers	inappropriate behaviour
chemicals on skin	electrical shock	UV, IR, nuclear radiation	communication issues
ingestion of chemicals	vibration or noise	pressure inside equipment	allergies
runaway reaction	sharp objects	heavy lifting	special needs
heat or cold	falling or flying objects	slipping, tripping, falling	other risks

Assessment by Student(s)

I/we have assessed the risks associated with performing this experiment in the classroom on the basis of likelihood and consequences using the School's risk matrix, according to International Organization for Standardization Standard ISO 31000:2018.

I/we consider the inherent level of risk (risk level without control measures) to be:

Low risk Medium risk High risk Extreme risk

Risks will therefore be managed by routine procedures in the classroom.

Certification by Teacher

I have assessed the risks associated with performing this experiment in the classroom on the basis of likelihood and consequences using the School's risk matrix, according to International Organization for Standardization Standard ISO 31000:2018. I confirm that the risk level and control measures entered by student(s) above are correct and appropriate.

Electronic Signature: Heather Omant **Date:** 29 Jun 2020

You have provided an electronic signature which is the equivalent of signing your name with a pen and as such will constitute a legally binding agreement between the relevant parties. We can give no warranty in respect to fraud or security breach resulting from the use of an electronic signature.

Certification by Laboratory Technician

I have assessed the risks associated with preparing the equipment, chemicals and and biological items, including living organisms, for this experiment and subsequently cleaning up after the experiment and disposing of wastes, on the basis of likelihood and consequences using the School's risk matrix, according to International Organization for Standardization Standard ISO 31000:2018.

I consider the inherent level of risk (risk level without control measures) to be:

Low risk Medium risk High risk Extreme risk

Risks will therefore be managed by routine procedures in the laboratory.

Electronic Signature: Nina Jones **Date:** 29 Jun 2020

You have provided an electronic signature which is the equivalent of signing your name with a pen and as such will constitute a legally binding agreement between the relevant parties. We can give no warranty in respect to fraud or security breach resulting from the use of an electronic signature.

Monitoring and review

This risk assessment will be monitored using comments below and will be reviewed within 15 months from the date of certification.

Attach further pages as required