STANDARD OPERATING PROCEDURES (SOPs)

A compilation of SOPs developed by Science ASSIST for Australian schools

Introduction

This compilation of Standard Operating Procedures (SOPs) has been created from the SOPs posted on the Science ASSIST website prior to its closure in December 2021. They are grouped by their science area focus and hyperlinked from the contents page to enable easy navigation.

They have been compiled so that they can continue to be available to support schools after the closure of the Science ASSIST website in December 2021. Note: The SOPs have not been revised since the date of publication in the footer, so many of the links to further information may no longer be current.

The SOPs include the following information: introduction, context, safety notes, regulations, licences and permits, equipment, troubleshooting, waste disposal, related material and references to surround the actual procedure, which is contained in part 6.

These Standard Operating Procedures (SOPs) were produced by the Science ASSIST project which was managed by the Australian Science Teachers Association (ASTA) in consultation with the Science Education Technicians Association (SETA).

Disclaimer: Science ASSIST accepts no liability for any damages to individuals or property arising directly or indirectly from using these resources. All content is provided in good faith and on the basis that readers will be responsible for making their own assessment of the matters discussed herein, and that they should verify all relevant representations, statements and information. Risk assessments and appropriate safety precautions must always be undertaken and are the sole responsibility of the user.

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STANDARD OPERATING PROCEDURE:

Diluting concentrated acetic acid

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

Acetic acid is an organic acid with chemical formula CH_3COOH. Acetic acid containing less than 1% water is commonly referred to as 'glacial acetic acid', so-called as it resembles ice crystals when it freezes, which it does at just under 17°C. The concentrated acid is a colourless, corrosive liquid with a pungent odour, and should be handled with care.

The name 'acetic' is derived from the Latin *acetum* meaning 'sour wine'. The production of acetic acid dates back to at least 10000 BC with the emergence of the practice of winemaking. While industrial-grade acetic acid is synthesised from fossil fuels, food-grade vinegar, which typically contains 4-8% acetic acid, is still made through the fermentation of sugars by yeast to give ethanol, which is then oxidised to acetic acid by bacteria of the genus Acetobacter.

In senior school chemistry, concentrated acetic acid is used in the preparation of acetate esters. In dilute form in junior and senior school science, acetic acid is used as an example of a weak organic acid.

The concentrated acid is flammable and combustible and should be handled away from flames or sparks; it is also hygroscopic, which means that it absorbs water from the air.

Synonyms: ethanoic acid, glacial acetic acid, vinegar acid, methanecarboxylic acid, ethylic acid

2. Context

- These instructions are for the use of experienced teachers and technicians only.
- Do not make up a dilution for the first time without seeking practical advice from an experienced colleague.
- Students must not be asked to make up dilutions from concentrated acetic acid.

3. Safety notes

- This activity may only be carried out with appropriate facilities available i.e. running water, fume cupboard, chemical safety/eyewash station and relevant Personal Protective Equipment (PPE).
- Avoid contact with skin and eyes, and avoid breathing fumes. Concentrated and high molarity acetic acid liquid can cause severe burns and eye damage. Fumes of concentrated acetic acid cause irritation to the eyes and respiratory system.
- Always carry large bottles of concentrated acid either in an approved carrier or by firmly grasping the body of the bottle with one hand and placing the other hand underneath the bottle. Do not carry by the neck or lid. Do not rush.
- Always make up dilutions in a fume cupboard.

- Ensure that glassware is free from chips and cracks before use.
- For first aid, accident and spill procedures refer to SDS before performing the dilution.
- Note that the concentrated acid should be stored in bunding (secondary containment), segregated from strong bases and oxidising agents, including nitric acid, and segregated from other acids. See the SDS for further details of incompatibilities.
- Pure (glacial) acetic acid has a melting point of 16.7°C and may freeze in cool weather. The frozen acid can be melted by placing the bottle in a plastic bag in a bath of warm water.
- **Always add concentrated acid to water (never water to acid).**

4. Regulations, licences and permits

Not applicable.

5. Equipment

- Fume cupboard
- PPE (lab coat, safety glasses or face shield, acetic acid resistant gloves (butyl-rubber gloves are well-suited for the handling of concentrated acetic acid; nitrile, neoprene/latex or latex gloves also provide good splash protection against the concentrated acid), closed-in shoes.
- Concentrated acetic acid
- Distilled/de-ionised water and wash bottle
- Large (2L) borosilicate glass beaker for diluting the acid
- Small glass measuring cylinder
- Large (1L) glass measuring cylinder or volumetric flask
- Glass stirring rod or magnetic stirrer and magnetic stirring bar
- Pre-labelled storage bottle

Note: For laboratory `Stock' solutions use measuring cylinders. For greater accuracy use volumetric flasks and pipettes.

6. Operating procedure

To make 1 litre of stock solution:

1. Wear PPE and work at a fume cupboard.
2. Into the large beaker place about 650mL of distilled water (or an amount such that the volume of water combined with the volume of concentrated acid to be added does not exceed about 850mL).
3. Carefully measure the required volume of concentrated acid in the small measuring cylinder; see table below. (If your concentrated acid is stored in a large 2.5L Winchester bottle, firstly pour some into a smaller bottle or beaker to be able to safely pour into the measuring cylinder as handling liquids in smaller containers is safer and easier.) *Hint:* keep the label of the acid bottle uppermost when pouring and clean up any spilt liquid from the outside of the bottle.

(Operating procedure cont….)

4. Add the concentrated acid slowly to the water with stirring. Dissolution of the concentrated acid will generate heat.
5. With distilled water from the wash bottle, rinse the remaining acid from the small measuring cylinder into the solution in the large beaker.
6. When the solution has cooled to room temperature, transfer it to a 1L measuring cylinder or volumetric flask. Rinse the large beaker and stirring rod or magnetic stirring bar with distilled water and add the rinsings to the solution in the measuring cylinder or volumetric flask. Make up the volume to 1 litre.
7. Pour this solution into the pre-labelled bottle.
8. On completion of the activity, clean up spills or splashes with plenty of water and thoroughly clean all used equipment and fume cupboard. All glassware that may be contaminated with concentrated acid should be rinsed with water BEFORE removing it from the fume cupboard.

Table: Volume of concentrated acetic acid required to prepare 1L of dilute solution

Molarity required	Volume of concentrated acetic acid (mL)[a]	Volume of concentrated acetic acid (mL)[b]
0.01M	0.6[c]	0.6[c]
0.1M	5.8[c]	6.4[c]
0.5M	29	32
1M	58	64
2M	116	127

[a] Based upon a 99% solution, approximately 17.3M.
[b] Based upon a 90% solution, approximately 15.7M.
[c] 0.5M solution can be prepared by a 1 part in 2 dilution of the 1M solution, a 0.1M solution by a 1 part in 10 dilution of the 1M solution, and a 0.01M solution by a 1 part in 10 dilution of the 0.1M solution.

Trouble shooting/emergencies

- First aid: See latest SDS for more detailed information
  - **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give plenty of water to drink. Seek urgent medical attention.
  - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek medical attention.
  - **Skin/clothes:** Remove contaminated clothing and drench the area with excess water under a safety shower. Seek medical attention.
  - **If inhaled:** Remove to fresh air and seek medical attention if symptoms persist.
  - For further advice contact the Poisons Information Centre on 131 126.
- Check plastic bottle caps for corrosion regularly and ensure bottles are tightly screwed closed.

8. Waste disposal

- Clean up any small spillages immediately with plenty of water (larger spills should be neutralised). When fuming stops, sweep/mop up. Spills that are too large to neutralise safely should be absorbed with non-combustible material such as dry sand or vermiculite and placed in a labelled container for collection and disposal by a registered hazardous waste disposal company.
- To neutralise concentrated acid, first dilute the acid by adding it carefully to a larger volume of water. Neutralise the solution with sodium bicarbonate, sodium carbonate or a 50:50 mixture of sodium carbonate and calcium hydroxide so that the pH is in the range pH 6-8. Exercise care as heat and corrosive fumes are produced. Use an indicator such as Universal Indicator to determine the pH. Wash the neutralised diluted solution to waste down the sink with excess cold water.
- Large quantities of waste acetic acid need to be collected and disposed of by a registered hazardous waste disposal company.

9. Related material

- Manufacturer's Safety Data Sheet
- Risk Assessment

References:

'Chemical Glove Guide', Ansell website http://ppe.ansell.com.au/chemical-glove-guide (Accessed June 2015)

Chemwatch GoldFFX, 2013, Material Safety Data Sheet 2789-3 Version 8.1.1.1, Acetic acid glacial https://jr.chemwatch.net/ (Subscription required. June 2015)

Cheryan, M. Acetic Acid Production, in Encyclopedia of Microbiology, (M. Schaechter, Editor-in-Chief) 3rd Edition, Elsevier Inc. (2009), Academic Press, pp. 145-149.

CLEAPSS, 2014, Hazcard 38A Ethanoic acid, methanoic acid and their salts, http://www.cleapss.org.uk/attachments/article/0/HC-038A.pdf?Secondary/Science/Hazcards/ (Subscription required. Accessed June 2015)

CLEAPSS, 2014, The CLEAPSS Recipe Book,39 Ethanoic acid and propanoic acid solutions, http://www.cleapss.org.uk/attachments/article/0/RB_039_Ethanoic%20&%20propanoic%20acidsOOct2014.pdf?Secondary/Science/Recipe%20Book/ (Subscription required. Accessed June 2015)

'Material Safety Data Sheet – Acetic acid 89-100%', January 2011, Chem-supply website https://www.chemsupply.com.au/documents/AA0091CH0J.pdf (Accessed June 2015)

History of reviews:

Date	Version Number	Notes
Jun 2015	Version 1.0	
Nov 2016	Version 2.0	Added magnetic stirrer and magnetic stirring bar to equipment Additional instructions for rinsing the stirring rod/magnetic stirring bar

STANDARD OPERATING PROCEDURE:

Diluting concentrated hydrochloric acid

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

Concentrated hydrochloric acid (HCl) is a hazardous chemical and must be handled with care. It is a strong inorganic acid which is highly corrosive, poisonous, and toxic. It is a colourless or slightly yellow liquid with a strong pungent odour and produces fumes at high concentrations. Concentrated hydrochloric acid is normally purchased from a supplier as either 32% or 36%.

The dilution of hydrochloric acid is an exothermic (heat producing) reaction.

Synonyms: Muriatic acid; hydrogen chloride solution.

2. Context

- These instructions are for the use of experienced science teachers and technicians only.
- Do not make up a dilution for the first time without seeking practical advice from an experienced colleague.

3. Safety Notes

- This activity may only be carried out with appropriate facilities available i.e. running water, fume cupboard, chemical safety/eyewash station and relevant Personal Protective Equipment (PPE).
- Avoid contact with skin and eyes, and avoid breathing fumes. Concentrated and high molarity hydrochloric acid causes severe burns and eye damage.
- Always carry large bottles of concentrated acid either in an approved carrier or by firmly grasping the body of the bottle with one hand and placing the other hand underneath the bottle. Do not carry by the neck or lid. Do not rush.
- Always make up dilutions in a fume cupboard.
- Ensure that glassware is free from chips and cracks before use.
- For first aid, accident and spill procedures refer to SDS before performing the dilution.
- **Always add concentrated acid to water (never water to acid).**

4. Regulations, Licences and Permits

Not applicable

5. Equipment

- Fume cupboard
- PPE (lab coat, safety glasses or face shield, acid resistant e.g. PVC or nitrile gloves, closed in shoes)
- Concentrated HCl
- Distilled/de-ionised water and wash bottle
- Large (2L) borosilicate glass beaker for diluting the acid
- Small glass measuring cylinder
- Large (1L) glass measuring cylinder or volumetric flask
- Glass stirring rod or magnetic stirrer and magnetic stirring bar
- Pre-labelled storage bottle

Note: For laboratory `Stock' solutions use measuring cylinders. For greater accuracy use volumetric flasks and pipettes.

6. Operating Procedure

To make 1 litre of stock solution:

1. Wear PPE and work at a fume cupboard.
2. Into the large beaker place about 650mL of distilled water (or an amount such that the volume of water combined with the volume of concentrated acid to be added does not exceed about 850mL).
3. Measure the required volume of concentrated acid in a small measuring cylinder, see table below. (If your concentrated acid is stored in a large 2.5L Winchester bottle, firstly pour some into a smaller bottle or beaker to be able to safely pour into the measuring cylinder). Hint: keep the label of the acid bottle uppermost when pouring and clean up any spilt liquid from the outside of the bottle.
4. Slowly add the concentrated acid to the water whilst stirring.
5. Rinse out the small measuring cylinder with distilled water using the wash bottle.
6. When the acid is well mixed and the solution has cooled, transfer it to a 1 litre measuring cylinder or volumetric flask. Rinse the large beaker and stirring rod or magnetic stirring bar with distilled water and add the rinsings to the solution in the measuring cylinder or volumetric flask. Make up the volume to 1 litre.
7. Pour this solution into the pre-labelled bottle.
8. On completion of the activity, clean up spills or splashes with plenty of water and thoroughly clean all used equipment and fume cupboard. All glassware that may be contaminated with concentrated acid should be rinsed with water BEFORE removing it from the fume cupboard.

Table: Volume of concentrated HCl (32% or 36%) required to prepare 1L of dilute solution

Concentration of HCl	Molarity required	Volume of concentrated HCl (mL)
32%	0.01M	0.98[a]
	0.1M	9.8[a]
	0.5M	49[a]
	1M	98
	2M	196
36%	0.5M	43
	1M	86
	2M	172

[a] 0.5M solution can be prepared by a 1 part in 2 dilution of the 1M solution, a 0.1M solution by a 1 part in 10 dilution of the 1M solution, and a 0.01M solution by a 1 part in 10 dilution of the 0.1M solution.

7. Trouble shooting/Emergencies

- First aid: See latest SDS for more detailed information
  - **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention.
  - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek urgent medical attention.
  - **If on skin/clothes:** Remove contaminated clothes and drench the area with excess water under a safety shower. Seek medical attention.
  - **If inhaled:** Remove to fresh air and seek urgent medical attention if breathing difficulties are obvious.
  - For further advice contact the Poisons Information Centre on 131126.
- Check plastic bottle caps for corrosion regularly and ensure bottles are tightly screwed closed.

8. Waste Disposal

- Clean up any small spillages immediately with plenty of water. (Larger spills should be neutralised).
- To neutralise concentrated acid, first dilute the acid by adding it carefully to a larger volume of water. Neutralise waste by addition of sodium bicarbonate until no further fizzing occurs so that the pH is in the range pH 6-8. Use an indicator such as Universal Indicator to determine pH. Wash the neutralised diluted solution to waste down the sink with excess cold water.
- Larger quantities will need to be collected and disposed of by a registered hazardous waste disposal company.

9. Related Material

- Manufacturer's Safety Data Sheet
- Risk Assessment.

References:

Chem-supply. 2012. *Safety Data Sheet Hydrochloric acid 25-36%*. http://chemsupply.customer-self-service.com/images/HL0201CH34.pdf (accessed March 2014)

Chemwatch Gold. 2012. *Long Safety Data Sheet: Hydrochloric Acid*. http://www.chemwatch.net (Subscription required accessed March 2014).

Dungey, B. 2002. *The Laboratory: A Science Reference and Preparation Manual for Schools.* Bayswater, Vic. Contemporary Press Pty. Ltd.

History of reviews:

Date	Version Number	Notes
April 2014	Version 1.0	
Nov 2016	Version 2.0	Additional instructions for rinsing the stirring rod/magnetic stirring bar

STANDARD OPERATING PROCEDURE:

Diluting concentrated nitric acid

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

Concentrated nitric acid is a clear, colourless to pale yellow liquid with a strong, sharp odour. It is both a very strong acid and a powerful oxidising agent, which must be handled with great care. The concentrated acid oxidises metals and organic material with the formation of nitrogen oxides, including nitrogen dioxide (NO_2), a reddish brown, acrid-smelling, highly toxic gas. In diluted solutions (<2M), nitric acid behaves as a strong acid in its reaction with metals; however, only in the reaction of the dilute acid with magnesium is hydrogen gas itself liberated.

'Concentrated nitric acid' refers to a 70% solution of nitric acid. Concentrations above 90% are called 'fuming nitric acid' and should not be used in schools.

Nitric acid is used industrially in the manufacture of ammonium nitrate, in nitration reactions with organic compounds, as a pickling agent for stainless steel, in the etching of metals, and in the production of explosives.

Due to its highly corrosive and oxidising properties, all handling of concentrated nitric acid must be carried out in a fume cupboard. The dilution of nitric acid is an exothermic (heat producing) reaction.

Synonyms: hydrogen nitrate, aqua fortis, spirit of niter, engraver's acid, azotic acid.

2. Context

- These instructions are for the use of experienced teachers and technicians only.
- Do not make up a dilution for the first time without seeking practical advice from an experienced colleague.
- Students must not be asked to make up dilutions from concentrated nitric acid.

3. Safety notes

- This activity may only be carried out with appropriate facilities available i.e. running water, fume cupboard, chemical safety/eyewash station and relevant Personal Protective Equipment (PPE).
- Avoid contact with skin and eyes, and avoid breathing fumes. Concentrated and high molarity nitric acid fumes and liquid cause severe burns and eye damage. Fumes of concentrated nitric acid are irritating to the respiratory system.
- Always carry large bottles of concentrated acid either in an approved carrier or by firmly grasping the body of the bottle with one hand and placing the other hand underneath the bottle. Do not carry by the neck or lid. Do not rush.
- Always make up dilutions in a fume cupboard.

- Ensure that glassware is free from chips and cracks before use.
- For first aid, accident and spill procedures refer to SDS before performing the dilution.
- Exposure to light can cause decomposition of nitric acid with formation of nitrogen oxides, which colour the acid yellow to brown. The concentrated acid should be stored in amber bottles away from light, in bunding (secondary containment), segregated from combustible material such as glacial acetic acid. See the SDS for further details of incompatibilities.
- **Always add concentrated acid to water (never water to acid).**

4. Regulations, licences and permits

Not applicable.

5. Equipment

- Fume cupboard
- PPE (lab coat, safety glasses or face shield, nitric acid resistant gloves e.g. neoprene, butyl rubber or neoprene/latex blend gloves, closed in shoes). Note that nitrile and latex gloves are not recommended for use with concentrated nitric acid, but have good resistance to dilute nitric acid. Nitrile gloves may be used for 'splash protection' but if contaminated with concentrated acid, the gloves should be removed and hands should be washed.
- Concentrated nitric acid
- Distilled/de-ionised water and wash bottle
- Large (2L) borosilicate glass beaker for diluting the acid
- Small glass measuring cylinder
- Large (1L) glass measuring cylinder or volumetric flask
- Glass stirring rod or magnetic stirrer and magnetic stirring bar
- Pre-labelled storage bottle

Note: For laboratory `Stock' solutions use measuring cylinders. For greater accuracy use volumetric flasks and pipettes.

6. Operating procedure

To make 1 litre of stock solution:

1. Wear PPE and work at a fume cupboard.
2. Into the large beaker place about 650mL of distilled water (or an amount such that the volume of water combined with the volume of concentrated acid to be added does not exceed about 850mL).
3. Carefully measure the required volume of concentrated acid in the small measuring cylinder; see table below. (If your concentrated acid is stored in a large 2.5L Winchester bottle, firstly pour some into a smaller bottle or beaker to be able to safely pour into the measuring cylinder as handling liquids in smaller containers is safer and easier.) *Hint:* keep the label of the acid bottle uppermost when pouring and clean up any spilt liquid from the outside of the bottle.

(Operating procedure cont….)

4. Add the concentrated acid slowly to the water with stirring. Dissolution of the concentrated acid will generate heat.
5. With distilled water from the wash bottle, rinse the remaining acid from the small measuring cylinder into the solution in the large beaker.
6. When the solution has cooled to room temperature, transfer it to a 1L measuring cylinder or volumetric flask. Rinse the large beaker and stirring rod or magnetic stirring bar with distilled water and add the rinsings to the solution in the measuring cylinder or volumetric flask. Make up the volume to 1litre.
7. Pour this solution into the pre-labelled bottle.
8. On completion of the activity, clean up spills or splashes with plenty of water and thoroughly clean all used equipment and fume cupboard. All glassware that may be contaminated with concentrated acid should be rinsed with water BEFORE removing it from the fume cupboard.

Table: Volume of concentrated HNO_3 required to prepare 1L of dilute solution

Molarity required	Volume of concentrated HNO_3 (mL)[a]
0.01M	0.6[b]
0.1M	6.4[b]
0.5M	32[b]
1M	64
2M	127

[a] Based upon a 70% solution, approximately 15.7M.
[b] 0.5M solution can be prepared by a 1 part in 2 dilution of the 1M solution, a 0.1M solution by a 1 part in 10 dilution of the 1M solution, and a 0.01M solution by a 1 part in 10 dilution of the 0.1M solution.

7. Trouble shooting/emergencies

- First aid: See latest SDS for more detailed information
  - **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give plenty of water to drink. Seek urgent medical attention.
  - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek urgent medical attention.
  - **Skin/clothes:** Immediately flush skin and clothes with excess water under a safety shower. Remove contaminated clothing. Seek medical attention.
  - **If inhaled:** Remove to fresh air. Seek medical attention.
  - For further advice contact the Poisons Information Centre on 131 126.
- Check plastic bottle caps for corrosion regularly and ensure bottles are tightly screwed closed.

8. Waste disposal

- Clean up any small spillages immediately with plenty of water (larger spills should be neutralised). When fuming stops, sweep/mop up. Spills that are too large to neutralise safely should be absorbed with dry sand or vermiculite and placed in a labelled container for collection and disposal by a registered hazardous waste disposal company. Do not absorb spill with combustible materials such as sawdust, paper or clothing as fire may result.
- To neutralise concentrated acid, first dilute the acid by adding it carefully to a larger volume of water. Neutralise the solution with sodium bicarbonate, sodium carbonate or a 50:50 mixture of sodium carbonate and calcium hydroxide so that the pH is in the range pH 6-8. Exercise care as heat & corrosive fumes are produced. Use an indicator such as Universal Indicator to determine the pH. Wash the neutralised diluted solution to waste down the sink with excess cold water.
- Large quantities of waste nitric acid need to be collected and disposed of by a registered hazardous waste disposal company.

9. Related material

- Manufacturer's Safety Data Sheet
- Risk Assessment

References:

Chem-supply, 2012, Safety Data Sheet Nitric acid 68-70%, http://chemsupply.customer-self-service.com/images/NT0011CHGO.pdf (Accessed June 2015)

Chemwatch GoldFFX, 2013, Material Safety Data Sheet 6632-54 Version 2.1.1.1, Nitric Acid 70%, https://jr.chemwatch.net/ (Subscription required. Accessed June 2015)

CLEAPSS, 2007, Hazcard 67 Nitric(V) acid, http://www.cleapss.org.uk/attachments/article/0/067.pdf?Secondary/Science/Hazcards/ (Subscription required. Accessed June 2015)

CLEAPSS, 2011, The CLEAPSS Recipe Book,61 Nitric(V) acid, http://www.cleapss.org.uk/attachments/article/0/RB61.pdf?Secondary/Science/Recipe%20Book/ (Subscription required. Accessed June 2015)

Greenwood,N.N. and Earnshaw,A. 1984, Chemistry of the Elements, Pergamon Press: Oxford, pp. 536-545.

Evans, U. R., Behaviour of metals in nitric acid, Trans. Faraday Soc., **40**, 1944, p. 120.

History of reviews:

Date	Version Number	Notes
Jun 2015	Version 1.0	
Nov 2016	Version 2.0	Added magnetic stirrer and magnetic stirring bar to equipment Additional instructions for rinsing the stirring rod/magnetic stirring bar

STANDARD OPERATING PROCEDURE:

Diluting concentrated sulphuric acid

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

Concentrated sulphuric acid (H_2SO_4) is a hazardous, highly corrosive chemical, which must be handled with great care. It is a clear, colourless, viscous (thick, oily) liquid when pure, but has a yellowish to brown tinge when contaminated with impurities such as organic material. It has a higher density than water and is hygroscopic, which means that it absorbs water from the air.

The dilution of sulphuric acid is a highly exothermic (heat generating) reaction.

Synonyms: hydrogen sulphate, oil of vitriol.

2. Context

- These instructions are for the use of experienced teachers and technicians only.
- Do not make up a dilution for the first time without seeking practical advice from an experienced colleague.
- Students must not be asked to make up dilutions from concentrated sulphuric acid.

3. Safety notes

- This activity may only be carried out with appropriate facilities available i.e. running water, fume cupboard, chemical safety/eyewash station and relevant Personal Protective Equipment (PPE).
- Avoid contact with skin and eyes, and avoid breathing fumes. Concentrated and high molarity sulphuric acid causes severe burns and eye damage.
- Always carry large bottles of concentrated acid either in an approved carrier or by firmly grasping the body of the bottle with one hand and placing the other hand underneath the bottle. Do not carry by the neck or lid. Do not rush.
- Always make up dilutions in a fume cupboard.
- Ensure that glassware is free from chips and cracks before use.
- For first aid, accident and spill procedures refer to SDS before performing the dilution.
- **Dilution of sulphuric acid liberates much heat and can cause a glass beaker to crack so must be conducted using a water bath to cool the solution.**
- **Always add concentrated acid to water (never water to acid).**

4. Regulations, licences and permits

Not applicable

5. Equipment

- Fume cupboard
- PPE (lab coat, safety glasses or face shield, sulphuric acid resistant gloves e.g. neoprene or PVC gloves, closed in shoes). Note that nitrile and latex gloves have poor resistance to concentrated sulfuric acid, but good resistance to dilute sulphuric acid. Nitrile gloves may be used for 'splash protection' but if contaminated with concentrated acid, the gloves should be removed and hands washed.
- Concentrated sulphuric acid
- Distilled/de-ionised water and wash bottle
- Large (2L) borosilicate glass beaker for diluting the acid
- Small glass measuring cylinder
- Large (1L) glass measuring cylinder or volumetric flask
- Glass stirring rod
- Large trough or bucket containing cold water to act as a water bath to cool the diluted acid
- Pre-labelled storage bottle

Note: For laboratory `Stock' solutions use measuring cylinders. For greater accuracy use volumetric flasks and pipettes.

6. Operating procedure

To make 1 litre of stock solution:

1. Wear PPE and work at a fume cupboard.
2. Into the large beaker place about 650mL of distilled water (or an amount such that the volume of water combined with the volume of concentrated acid to be added does not exceed about 850mL).
3. Place the beaker into a cold water bath and ensure that it will not tip. Ensure the cold water in the bath comes about equal to the level of the water inside the beaker.
4. Carefully measure the required volume of concentrated acid in the small measuring cylinder; see table below. (If your concentrated acid is stored in a large 2.5L Winchester bottle, firstly pour some into a smaller bottle or beaker to be able to safely pour into the measuring cylinder as handling liquids in smaller containers is safer and easier.) Hint: keep the label of the acid bottle uppermost when pouring and clean up any spilt liquid from the outside of the bottle.
5. Very slowly add the concentrated acid to the water with constant stirring. It is important to stir whilst adding the acid to the water to avoid a layer of concentrated acid forming at the bottom of the beaker creating a temperature gradient.
6. Rinse out the small measuring cylinder with distilled water using the wash bottle.
7. When the acid is well mixed leave the diluted acid in the water bath until it has cooled to room temperature. Note: Do not handle the diluted mixture until cooled. The cold water in the water bath may have to be changed.

(Operating procedure cont….)

8. When the solution has cooled to room temperature, transfer it to a 1 litre measuring cylinder or volumetric flask. Rinse the large beaker and stirring rod or magnetic stirring bar with distilled water and add the rinsings to the solution in the measuring cylinder or volumetric flask. Make up the volume to 1 litre.
9. Pour this solution into the pre-labelled bottle.
10. On completion of the activity, clean up spills or splashes with plenty of water and thoroughly clean all used equipment and fume cupboard. All glassware, which may be contaminated with concentrated acid, should be rinsed with water BEFORE removing it from the fume cupboard.

Table: Volume of concentrated H_2SO_4 required to prepare 1L of dilute solution

Molarity required	Volume of concentrated H_2SO_4 (mL)[a]
0.01M	0.55[b]
0.1M	5.5[b]
0.5M	27.5[b]
1M	55
2M	110

[a] Based upon a 98% solution, approximately 18.2M
[b] 0.5M solution can be prepared by a 1 part in 2 dilution of the 1M solution, a 0.1M solution by a 1 part in 10 dilution of the 1M solution, and a 0.01M solution by a 1 part in 10 dilution of the 0.1M solution.

7. Trouble shooting/emergencies

- First aid: See latest SDS for more detailed information
  - **<u>If swallowed</u>:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention.
  - **<u>If in eyes</u>:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek medical attention.
  - **<u>Skin/clothes</u>:** If spilt on skin or clothes quickly wipe off with a dry cloth to absorb as much liquid as possible. Remove contaminated clothes and drench the area with excess water under a safety shower. Seek medical attention.
  - **<u>If inhaled</u>:** Remove to fresh air and seek medical attention if symptoms persist.
  - For further advice contact the Poisons Information Centre on 131 126.
- Check plastic bottle caps for corrosion regularly and ensure bottles are tightly screwed closed.

8. Waste disposal

- Clean up any small spillages immediately with plenty of water (larger spills should be neutralised). When fuming stops sweep/mop up.
- To neutralise concentrated acid, first dilute the acid by adding it carefully to a larger volume of water. Neutralise waste with sodium bicarbonate or a 50:50 mixture of sodium carbonate and calcium hydroxide so that the pH is in the range pH 6-8. Exercise care as heat & corrosive fumes are produced. Use an indicator such as Universal Indicator to determine the pH. Wash the neutralised diluted solution to waste down the sink with excess cold water.
- Larger quantities will need to be collected and disposed of by a registered hazardous waste disposal company.

9. Related material

- Manufacturer's Safety Data Sheet
- Risk Assessment

References:

Chem-supply. 2013. *Safety Data Sheet Sulfuric acid 52-98%*. http://chemsupply.customer-self-service.com/images/SA0081CH72.pdf (accessed March 2014)

Chemwatch Gold. 2012. *Long Safety Data Sheet: Sulfuric Acid 18M, 98%.* http://www.chemwatch.net (Subscription required. Accessed March 2014)

CLEAPSS. 2007. *Hazcard 98A Sulfuric(VI) acid.* http://www.cleapss.org.uk/attachments/article/0/098A.pdf?Secondary/Science/Hazcards/ (Subscription required. Accessed March 2014)

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NSW Department of Education and Training (2010) *Working In Science Manual. Professional Learning and Leadership Development Directorate 2010.* Sydney: NSW

NSW Department of Education and Training (2013). *Chemical Safety in Schools (CSIS)* Sydney: NSW - DET Intranet, http://www.dec.nsw.gov.au/ (Login required. Accessed March 2014)

History of reviews:

Date	Version Number	Notes
April 2014	Version 1.0	
Nov 2016	Version 2.0	Additional instructions for rinsing the stirring rod/magnetic stirring bar

STANDARD OPERATING PROCEDURE:

Preparing sodium hydroxide solutions

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

Sodium hydroxide (NaOH) is a hazardous chemical and must be treated with care. It is highly corrosive to skin and eyes and extremely toxic if ingested. The solid can be purchased as pellets, flakes or mini pearls. It is very hygroscopic (absorbs water from the air) and will also absorb carbon dioxide from the air and therefore cannot be used as a primary standard for titrations. To minimise its absorption of water and carbon dioxide, it should be kept in a tightly closed container and left open for as little time as possible.

Dissolving sodium hydroxide in water is an exothermic (heat producing) reaction.

Synonyms: caustic soda, lye, soda lye.

2. Context

- These instructions are for the use of experienced teachers and technicians only.
- Do not make up a solution for the first time without seeking practical advice from an experienced colleague.

3. Safety Notes

- This activity may only be carried out with appropriate facilities available i.e. running water, chemical safety/eyewash station and relevant Personal Protective Equipment (PPE)
- Conduct procedure in a well-ventilated area or fume cupboard.
- Avoid contact with skin and eyes, and avoid breathing fumes.
- The preparation of sodium hydroxide solutions liberates heat and may produce caustic fumes/vapours.
- Ensure that glassware is free from chips and cracks before use.
- For first aid, accident and spill procedures refer to SDS before making a solution.
- **Always add solid sodium hydroxide to water (never water to sodium hydroxide).**

4. Regulations, Licences and Permits

Not applicable

5. Equipment

- Fume cupboard (preferable) or a well-ventilated area
- PPE (lab coat, safety glasses or face shield, chemical resistant gloves e.g. nitrile or latex, closed in shoes)
- Sodium hydroxide

- Distilled/de-ionised water and wash bottle
- Large (2L) borosilicate glass beaker (a capacity of 50% greater than the volume required)
- Small borosilicate glass beaker (for weighing sodium hydroxide)
- Large (1L) glass measuring cylinder or volumetric flask
- Stirring rod or magnetic stirrer and magnetic stirring bar
- Pre-labelled storage bottle with a plastic screw cap lid or stopper.
- Large trough or bucket containing cold water to act as a water bath to cool solutions of 1M or stronger

Note: For laboratory `Stock' solutions use measuring cylinders. For greater accuracy use volumetric flasks and pipette. Sodium hydroxide cannot be used as a primary standard.

6. Operating Procedure

To make 1 litre of stock solution:

1. Wear PPE and work at a fume cupboard or in a well-ventilated area.
2. Place about 650mL of distilled water (or about two-thirds of the final required volume) into the large beaker.
3. Place the beaker into a cold water bath and ensure that it will not tip. If using a magnetic stirrer, set the water bath on the magnetic stirrer before placing the beaker into the bath.
4. Weigh the required amount of sodium hydroxide in the small beaker; see table below.
5. Add small amounts at a time of sodium hydroxide to the solution in the large beaker, stirring with each addition.
6. Check the temperature of the solution to see that it is not too hot before each addition.
7. When all the sodium hydroxide has been added rinse the small beaker with water from the wash bottle.
8. When the solution has cooled to room temperature, transfer it to a 1 litre measuring cylinder or volumetric flask. Rinse the large beaker and stirring rod or magnetic stirring bar with distilled water and add the rinsings to the solution in the measuring cylinder or volumetric flask. Make up the volume to 1 litre.
9. Pour this solution into the pre-labelled bottle that has a plastic screw cap lid (or stopper).
10. On completion of the activity, clean up spills or splashes with plenty of water and thoroughly clean all used equipment and the bench or fume cupboard surface.

Table: Mass of NaOH required to prepare 1L of dilute solution

Molarity required	Mass of solid NaOH
0.1M[a]	4.0g
0.5M[a]	20.0g
1M	40.0g
2M	80.0g
6M[b]	240.0g

[a] 0.5M solution can be prepared by a 1 part in 2 dilution of the 1M solution and a 0.1M solution by a 1 part in 10 dilution of the 1M solution.
[b] CAUTION: creates a lot of heat

7. Trouble shooting/Emergencies

- First aid: See latest SDS for more detailed information
  - **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention.
  - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek urgent medical attention.
  - **If on skin/clothes:** Remove contaminated clothes and drench the area with excess water under a safety shower. Seek medical attention.
  - **If inhaled:** Remove to fresh air and seek urgent medical attention if breathing difficulties are obvious.
  - For further advice contact the Poisons Information Centre on 131126.
- To minimise absorption of water and carbon dioxide from the air, the container of solid sodium hydroxide should be kept tightly closed when not in use.
- Make sure the solution is well mixed. On dissolving, the more concentrated (and denser) solution may sit on the bottom.
- **Ensure that the solution is not stored in a glass bottle with a ground glass stopper.** Strong alkali solutions can 'freeze' ground glass stoppers in place.

8. Waste Disposal

- Clean up any small spillages immediately with plenty of water. (Larger spills should be neutralised with an acid).
- Concentrated solutions should first be diluted by addition to a larger volume of water. Neutralise waste with dilute acetic acid (or vinegar), citric acid or dilute hydrochloric acid, so that the pH is in the range pH 6-8. Use an indicator such as Universal Indicator to determine the pH. Wash the neutralised diluted solution to waste down the sink with excess cold water.
- Larger quantities will need to be collected and disposed of by a registered hazardous waste disposal company.

9. Related Material

- Manufacturer's safety data sheet.
- Risk Assessment.

References:

Chemwatch Gold. 2011. *Chemwatch Independent Material Safety Data Sheet: Sodium hydroxide.* http://www.chemwatch.net (Subscription required accessed February 2014)

Risk Assess. 2014. *Risk Assessment for making 1M NaOH* http://www.riskassess.com.au/ (Subscription required accessed February 2014)

History of reviews:

Date	Version Number	Notes
April 2014	Version 1.0	
Nov 2015	Version 2.0	Text box for 6. Operating Procedure enlarged to reveal footnote 'b' Additional synonyms included
Nov 2016	Version 3.0	Additional instructions for rinsing the stirring rod/magnetic stirring bar

STANDARD OPERATING PROCEDURE:

Handling dry ice (solid carbon dioxide)

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

Dry ice is solid carbon dioxide (CO_2). Under normal classroom conditions dry ice changes directly from the solid to carbon dioxide gas, without going through the liquid phase. The phase change of a solid transforming directly into a gas, without passing through the liquid phase, is called sublimation. The properties of dry ice and carbon dioxide form the basis for many interesting classroom demonstrations such as the process of sublimation, cryogenics, fog effects and extinguishing a flame.

2. Context

- These instructions are for the use of experienced science teachers and technicians only.
- The use of dry ice is for demonstration purposes only.

3. Safety Notes

- The sublimation temperature of dry ice is -78.5°C. Contact of dry ice with the skin may result in frostbite or cold burns.
- Dry ice must be used and stored in a well-ventilated area. A concentration of carbon dioxide in air of greater than 1.5% can cause headache, nausea and vomiting and may lead to unconsciousness.
- Dry ice should be stored in an insulated and secure container, which has a loose-fitting lid (e.g. a foam cooler box). Containers should be vented periodically to avoid the build-up of gas. Dry ice must not be stored in a sealed container as the build-up of pressure from sublimation could cause the container to rupture or explode.
- Dry ice may be stored in a running fume cupboard if ventilation is otherwise inadequate.
- Protect eyes, face and skin from contact with dry ice. Safety glasses, thermally insulated gloves, a lab coat and closed shoes should be worn and tongs should be used to pick up pieces of dry ice.
- Carbon dioxide is more dense than air and may accumulate in low, confined spaces with poor ventilation.
- Dry ice is not classified into any hazard class in the GHS. However, the Safe Work Australia *Labelling of Hazardous Chemicals* Code of Practice recommends that containers be labelled with the quantity of dry ice contained and information regarding the asphyxiation hazard and safe handling to avoid cold burns. See the Code of Practice for examples of labels for containers of dry ice.

- Transport in a private vehicle should be avoided; where possible, delivery of dry ice should be arranged with the supplier. If the dry ice is to be transported in a private vehicle, only small amounts (up to 5kg) at a time should be purchased. The dry ice should be collected in an insulated container with a loose-fitting lid and the container securely placed in a compartment of the vehicle, which is segregated from the driver's compartment. Good ventilation to the driver's compartment must be ensured in case of leakage of carbon dioxide gas into the driver's compartment.

4. Regulations, Licences and Permits

Not applicable

5. Equipment

- Insulated storage container, such as a foam cooler box
- Tongs
- PPE: safety glasses, thermally insulated gloves, lab coat or overalls, closed shoes
- Safety screen for use in class demonstrations

6. Operating Procedure

1. Wear PPE and work in a well-ventilated area.
2. Avoid cold burns by wearing gloves and using tongs to pick up pieces of dry ice.

7. Trouble shooting/Emergencies

- First Aid: See latest SDS for more detailed information
  - In case of frostbite or cold burns, flush skin with warm (30°C) water for 15 minutes. Apply a sterile dressing. Seek medical attention. Do not apply hot water or radiant heat.
  - In case of contact with the eye, irrigate eye with tepid water for 15 minutes. Seek medical attention immediately.
  - In case of inhalation, remove patient to well-ventilated area. Apply artificial respiration if not breathing. Seek medical attention.
  - For further advice contact the Poisons Information Centre on 131126.

8. Waste Disposal

- Unused dry ice may be allowed to sublime in a well-ventilated area.

9. Related Material

- SDS
- Risk Assessment.

References:

Safe Work Australia. 2011. 'Model Code of Practice Labelling of Hazardous Chemicals' http://www.safeworkaustralia.gov.au/sites/swa/about/publications/pages/labelling-hazardous-chemicals-cop (Accessed April 2014)

Air Liquide Australia Limited, Material Safety Data Sheet AL066: Carbon dioxide, solid (CO_2), Dry Ice, Revised edition number 7, MSDS date 8 November 2012. http://docs.airliquide.com.au/msdsau/AL066.pdf

Risk Management Technologies, Perth, WA, BOC Limited (Australia) Safety Data Sheet #033: Solid Carbon dioxide, Revision 2, SDS date 8 January 2014 http://msds.chemalert.com/?id=21&file=0008513_001_001.pdf

Information on the properties and uses of dry ice: http://science.howstuffworks.com/innovation/science-questions/question264.htm

Date	Version Number	Notes
April 2014	Version 1.0	
Jan 2016	Version 2.0	Activities added

Activities with dry ice (solid carbon dioxide)

These activities are for the use of experienced science teachers and technicians as demonstrations only, and involve the processes of sublimation, cryogenics, fog effects and extinguishing a flame. Activities involving a sealed system such as the balloon and the popping film canister should not be carried out. All run the risk of exploding and showering students with dry ice and other shrapnel. None of the activities should be brought close to the face or ears.

Some activities that involve the ***sublimation*** process are:

Awesome bubbles

You will need:

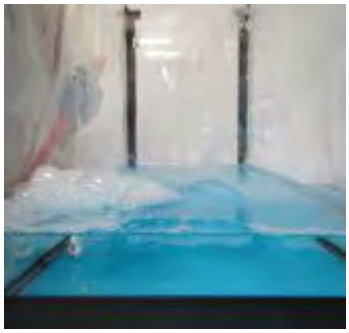
- gloves/tongs
- glass measuring cylinder
- liquid detergent
- food colouring optional •

dry ice

Method

1. Fill a 250 mL glass measuring cylinder with 150 mL warm water.
2. Add a squirt of dishwashing liquid to the water followed by a few drops of food colouring.
3. Place 2–3 pieces of dry ice into the soapy water and watch as the bubbles climb out of the cylinder with a burst of ‘smoke.
4. For an eerie glow, add a glow stick into the water along with the dry ice.

Floating bubbles



You will need:

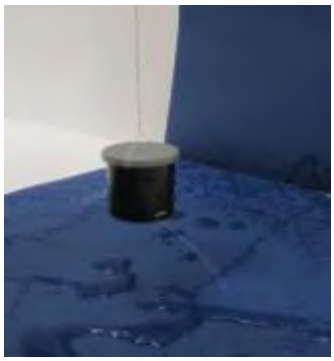
- gloves/tongs
- aquarium
- bubbles solution and wand
- dry ice

Method:

1. Fill the bottom of a fish aquarium (60 L) with 4 litres of warm water.
2. Add 2–3 pieces of dry ice.
3. Blow a few bubbles into the aquarium, using a bubble wand and bubbles solution.

Bubbles will appear to float in mid-air on a cushion of invisible carbon dioxide gas.

Hero's engine or film canister engine



You will need:

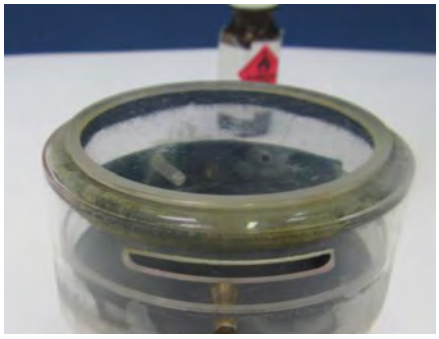
- gloves/tongs
- film canister
- tape
- cotton string
- paper towel, dry ice
- scissors

Method:

1. Pierce two holes on the opposite sides of a film canister near the bottom
2. Tie a loop in a piece of string and tape to the inside of the cap of the canister. You should be able to suspend the canister from the string.
3. Add 1 piece of dry ice and 5 mL of warm water to the canister and hold it in the air over some paper towels or absorbent pads.
4. Lift the canister by the thread and watch the sprinkler effect.

Cloud chamber

1. The diffusion cloud chamber can be used to observe Alpha or Beta particles in the presence of dry ice.
2. If a commercial cloud chamber is not available, a simple one can easily be made in the lab.
3. Firstly, cover the base of a 200–250 g glass jar and one third of the sides with black felt.
4. Cut out a piece of cardboard larger than the top of the dish.
5. Soak the felt in isopropyl alcohol until it is saturated and drain off any excess alcohol.
6. Place the cardboard on the glass container and rub the top with your hand.
7. Place the container on some dry ice.
8. Shine a flashlight through the side of the container and observe the vapour trails.



The alcohol absorbed by the felt is at room temperature and is slowly evaporating into the air but as the evaporated alcohol sinks toward the dry ice, it cools down and wants to turn back into a liquid.

At the end of this activity, place the glass container in the fume cupboard with the fan on. This will allow the isopropyl alcohol from the felt to evaporate over time.

Singing spoon

You will need:

- gloves
- spoon
- dry ice

Method

1. Hold a warm spoon on a piece of dry ice and listen to the spoon screaming loudly as the dry ice absorbs the heat from the spoon and sublimates.

The gas pressure pushes the spoon away from the dry ice and ceases sublimation. The spoon then comes back into contact with the dry ice and the cycle starts again.

This whole process is so quick that it results in the vibration of the spoon molecules and thus the singing/screaming sound.

Bubbling and smoking water

This activity involves **sublimation, fog effect** and can also be used to demonstrate the **flame extinguishing** process.

You will need:

- gloves/tongs
- beaker
- dry ice
- warm water

Method

1. Place some dry ice in a beaker of warm water. Immediately, the dry ice will start to sublimate and a cool white cloud is formed which is perfectly safe.
2. When the “smoking” starts to slow down, replace the cold water with some warm water.
3. For fog effect use boiling water, clouds of white fogs are created which consist of condensed water vapour mixed in the invisible carbon dioxide gas.
4. The fog can be used to extinguish a lit match or candle.

Candles experiment

This activity involves **sublimation, fog effect** and can also be used to demonstrate the **flame extinguishing** process.

You will need:

- gloves/tongs
- beaker
- candles
- dry ice
- warm water

Method

1. Place some dry ice in a beaker of warm water and allow the dry ice to sublimate into a white cloud.
2. Secure 3 different lengths of candles into a large beaker or container and light them.
3. Slowly pour the white smoke/fog down the side of the beaker and watch the candles extinguish one-by-one from the bottom to the top.

Making a big ice bubble

This activity involves **sublimation, fog effect** processes.

You will need:

- gloves/tongs
- bowl
- water
- detergent
- dry ice

Method

1. Place 5–10 pieces of dry ice into a bowl, which has a lip around the top.
2. Add 250 mL of warm water to the dry ice.
3. Soak a piece of cloth in some dishwashing detergent and run it around the lip of the bowl before dragging it across the top of the bowl to form a bubble layer over the dry ice.

The carbon dioxide vapour is trapped under the soapy layer and forms a big bubble.

The bubble will grow bigger until the pressure becomes too much and it will explode, spilling fog over the edge of the bowl.

Dry ice bubbles

This activity involves **sublimation, fog effect** processes.

You will need:

- gloves/tongs
- 1L or 2L Büchner flask (or vacuum pressure flask)
- rubber stopper
- Morning Fresh Original® detergent
- glycerine
- rubber tubing
- dry ice
- plastic funnel or plastic cup with a hole in the end to allow the tubing to fit through

Method

1. Prepare a bubble solution by mixing 70 mL of water, 30 mL of detergent (Morning Fresh Original®) and 10 mL of glycerine. Stir the mixture slowly and thoroughly. Allow the bubble mix to sit for at least 1–2 hours.
2. Firmly attach the rubber tubing to the side arm of a 1L or 2L Büchner flask or vacuum pressure flask.

3. Connect the other end of the rubber tubing to a funnel, or slide it into a hole in a small cup.
4. Place 100–200 mL of warm water into the flask.
5. Place 2–3 pieces of dry ice into the flask.
6. Dip the free end of the rubber tubing into the bubble solution to wet the end of the tube.
7. Remove the tubing from the bubble solution with one hand and then place the rubber stopper into the neck of the flask with the other hand. The aim is to blow a bubble filled with fog due to the production of carbon dioxide gas and the build-up of pressure.

8. When the bubble reaches the desired size, gently shake it off of the tubing.

The bubbles formed are quite long-lived. Students enjoy taking part in this activity.

For more information and instructions for how to make your own 'Boo Bubbles generator', see http://www.stevespanglerscience.com/lab/experiments/boo-bubbles-dry-ice-science/

Acid properties of dry ice in water

You will need:

- gloves/tongs
- glass measuring cylinder
- water
- universal indicator
- dry ice

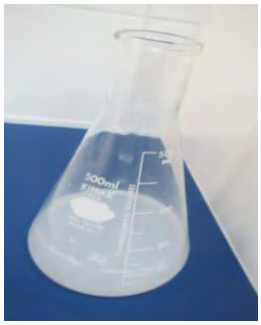
Method

1. Half fill a 100mL glass-measuring cylinder with warm water.
2. Add 20 drops of universal indicator followed by 1 piece of dry ice.

The initial green colour changes to yellow and finally to orange-red as the water becomes more acidic.

Carbon dioxide being an acidic oxide dissolves in water to give carbonic acid.

Making calcium carbonate (acidic properties of carbon dioxide)



You will need:

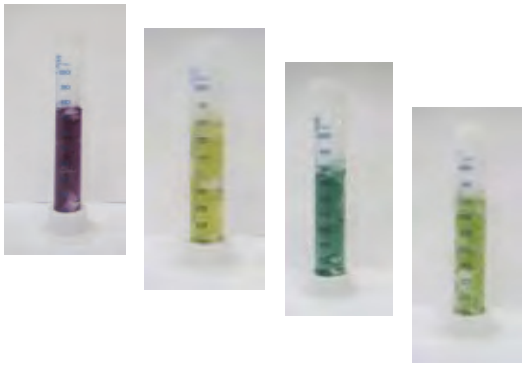
- gloves/tongs
- conical flask
- hotplate
- lime water
- dry ice

Method

1. Pour 200 mL of clear limewater into a 500 mL conical flask followed by 5 pieces of dry ice.

A white precipitate of calcium carbonate is formed which eventually dissolves to give a clear solution. The clear solution can be heated on a hotplate and stirred to once again generate the white precipitate. The resulting mixture can be filtered to obtain the solid calcium carbonate.

Neutralisation reaction



You will need:

- gloves/tongs
- glass measuring cylinder
- water
- universal indicator
- 0.1 M sodium hydroxide
- sodium bicarbonate
- dry ice

Method

1. Half fill a 100 mL glass measuring cylinder with warm water and add 20 drops of universal indicator.
2. Then add 0.1 M sodium hydroxide drop wise to the measuring cylinder until a purple colour is obtained.
3. Shake the measuring cylinder gently to get a uniform purple colour.
4. Add 3 pieces of dry ice to the measuring cylinder. The solution colour will change from purple to blue through to green and finally to a yellowish orange.
5. Once the bubbling stops, gradually add sodium bicarbonate into the measuring cylinder until a green neutral colour is obtained.
6. The resulting solution is then safe to pour down the sink.

Super freeze

This activity involves the **cryogenics** process.

You will need:

- gloves/tongs
- beaker
- methylated spirit
- flower
- dry ice

Method

1. Place 12–15 pieces of dry ice in a 500 mL beaker.
2. Then slowly pour 150mL of methylated spirit over the dry ice. The methylated spirit will turn into a viscous super cooling (cryo) liquid when all the dry ice is used up, fog has cleared and dry ice barely bubbling.

The super cooling liquid can be as cold as -72 ^{0}C. Flowers and green leaves will freeze in just a few minutes.

3. Using tongs, hold the flower in the super cooling liquid for approximately 1–2 minutes. The flower will become brittle and can be snapped apart.
4. At the end of this activity, place the beaker containing the cryo fluid in the fume cupboard with the fan on. This will allow the methylated spirit to evaporate over time.
5. Once the solution has reached room temperature, it can be discarded down the sink whilst running cold water to dilute it further.

Waste disposal

Allow any unused dry ice to sublime in a well-ventilated area.

References

'Awesome dry ice experiments', Steve Spangler Science website http://www.stevespanglerscience.com/lab/experiments/awesome-dry-ice-experiments (Accessed December 2015)

'Dry ice fog and special effects', dryiceInfo.com website, http://www.dryiceinfo.com/fog.htm (Accessed December 2015)

'Experimenting with Dry Ice', Brian Wesley Rich's science website, http://http://www.west.net/~science/co2.htm (Accessed December 2015)

All images courtesy of P Hosany and V Ward.

STANDARD OPERATING PROCEDURE:

Handling liquid nitrogen (LN_2)

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment. ***Check with your school jurisdiction for any restrictions or policies regarding the use of liquid nitrogen.***

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1. Introduction

Liquid nitrogen (LN_2) is a colourless, odourless, non-toxic, non-flammable liquid form of nitrogen. It is a cryogenic liquid with a boiling point of -196 °C, and it is this property that affords it a range of industrial and medical applications and allows for many engaging school science demonstrations.

At room temperature, small amounts of liquid nitrogen rapidly vaporise to produce large volumes of gas, which is impossible to detect with the human senses. In confined spaces this can be an asphyxiation risk. Due to this and its other associated hazards, great care is required when using liquid nitrogen.

2. Context

- These instructions are only for the use of experienced science teachers and technicians, who are trained in the handling of liquid nitrogen.
- The use of liquid nitrogen is for demonstration purposes only.

3. Safety notes

Hazards:

The main hazards associated with using liquid nitrogen are the potential for cryogenic (cold) burns and injuries, asphyxiation, pressure build up in closed vessels, the embrittlement of materials not compatible with cryogenic liquids and fire due to oxygen enrichment.

Extremely low temperatures – cryogenic (cold) burns

- Contact with the extremely cold liquid or vapours on the skin or in the eyes, even for a short time, may result in cold burns, frostbite, tissue damage or permanent eye damage.
- Unprotected skin coming into contact with items that have been exposed to liquid nitrogen may stick to the cold items and possibly tear on removal.
- Wear appropriate protective clothing that does not 'trap' pools of liquid nitrogen close to the skin.
- Ensure access to a safety shower and eye wash facilities.

Asphyxiation due to oxygen depletion

- When it boils, liquid nitrogen becomes gaseous nitrogen that can displace oxygen from the air. The volume expansion ratio is about 1:700, meaning that 1 litre of liquid nitrogen will vaporise to produce about 700 litres of nitrogen gas.

- Liquid nitrogen has a vapour density of 0.97 i.e. marginally less dense than air at the same temperature; thus, nitrogen gas at ambient temperature will mix very evenly in air. However, because of its very low temperature, the vapours generated by boiling liquid nitrogen are denser than the surrounding warmer air, and therefore may pool at and below ground level in confined spaces. Such accumulations pose an asphyxiation hazard due to the depletion of oxygen in these spaces.
- Rapid release of liquid nitrogen can create vapour fog clouds which can also pose an asphyxiation hazard.
- Liquid nitrogen must therefore be used and stored in a well-ventilated area to prevent nitrogen gas build up and the risk of asphyxiation. This is unlikely to be an issue in school science if demonstrations are carried out with small volumes of liquid nitrogen in large well-ventilated rooms.

Pressure build up in closed vessels

- When they vaporise, all cryogenic liquids produce large volumes of gas. If stored in a sealed container (closed system) this can produce huge pressures that could lead to the vessel rupturing.
- Specialised portable vessels called Dewar flasks are used to store cryogenic liquids as they have insulated walls and special vented lids that allow the vapours to escape.

Embrittlement

- Many materials such as plastics, glass and rubber can become brittle and shatter or crack when exposed to liquid nitrogen.
- Ensure suitable vessels are used for the containment of liquid nitrogen: stainless steel bowls, small polystyrene containers or polystyrene cups.
- Avoid spills of liquid nitrogen onto floors/benchtops as surfaces may be damaged.

Fire in an oxygen enriched environment

- Oxygen will condense when in contact with a surface cooled below 191°C forming an oxygen-enriched condensate and atmosphere. The flammability of combustible materials is increased in an oxygen-enriched atmosphere. This is unlikely to be a hazard in school science where liquid nitrogen is used infrequently and only in small volumes.
- Keep combustible materials away from containers of liquid nitrogen and do not return unused liquid nitrogen to the storage Dewar flask.

Storage of liquid nitrogen:

Liquid nitrogen must be transported and stored in vessels designed and approved for cryogenic fluids. Suppliers use cryogenic tankers and deliver to schools in Dewar flasks.

Dewar flasks:

- Non-pressurised, double-walled insulating vessels with loose fitting insulated caps for the venting of vapours.
- Should not be filled to more than 80% capacity.
- Expensive to purchase and maintain, however supply companies can provide a Dewar flask to schools at a nominal hiring cost.
- Available in various sizes. To reduce manual handling issues and to limit the quantity, a size of five (5) litres is recommended for school use.

- Store in a secure, dry, cool (below 45•C), well-ventilated place away from heavy traffic and combustible materials. Dewar flasks should be stored upright on a firm level floor and secured to prevent tipping or falling. Ensure appropriate hazard warning signs are displayed. If stored in a small room, ensure that a second person is on standby when retrieving.

Liquid nitrogen must not be stored in domestic vacuum flasks. These have tight fitting lids that do not allow the gas from the boiling liquid to escape resulting in pressure build up and the risk of an explosion.

Transport of liquid nitrogen:

- Schools should arrange delivery of liquid nitrogen by the gas supply company. Schools are strongly advised against transporting liquid nitrogen in motor vehicles. **Under no circumstances should liquid nitrogen be transported in an enclosed vehicle.**
  IF transported in a motor vehicle (not recommended); it must be secured and transported in an open vehicle such as an open trailer or utility, in an air space that is separate from the driver and passengers.)
- Clear procedures should be established for the movement of a Dewar flask at the school. The transport route should be assessed for all potential hazards such as movement through people, obstructions, uneven ground and stairs. A suitable trolley which can secure the Dewar flask can be used if necessary.
- If a lift is used to transport the Dewar flask then the lift should be closed to passengers. It should **never** be accompanied in the lift due to the risk of asphyxiation. A sign shall be displayed forbidding entry and someone should be waiting at the destination floor. The potential risk of oxygen deficiency in the event of the lift being stopped between floors should also be considered and if a lift travels past a floor, signs shall be displayed.

Personal Protective Equipment (PPE):

When handling liquid nitrogen, the following protective equipment and clothing should be worn:

- A full face-shield or safety goggles to protect the eyes when handling liquid nitrogen e.g. when transferring from one vessel to another and when immersing objects (in case items shatter or implode when cooled by liquid nitrogen).
- Loose fitting, dry, cold insulating or leather gloves that can be **easily removed** in the event of splashes entering a glove. These gloves are not designed for immersion into the liquid nitrogen. Do not wear rubber gloves.

- Laboratory coat with long sleeves and no cuffs to protect the arms should be worn over the top of the gloves to reduce the risk of splashes entering a glove.
- Closed leather boots or shoes that are **easily removed** in the event of a splash entering a boot or shoe. Shoes made of absorbent materials should not be worn.
- Long pants without cuffs worn **over the top of the shoes/boots** to reduce the risk of splashes entering a boot or shoe.

Handling and use of liquid nitrogen:

- Follow the instructions in section 6.
- Liquid nitrogen is transferred from a Dewar flask by careful pouring to a secondary container that can withstand the cold temperatures (-196•C).
- Activities involving boiling liquid expanding gas vapour explosions (BLEVEs) should not be conducted by school staff because of the potential risk of explosions showering nearby people with liquid nitrogen splashes and other very cold materials.

4. Regulations, licences and permits

Not applicable.

5. Equipment

- Personal protective equipment (PPE): as above.
- Safety screen for use in class demonstrations.
- Tongs (long and high quality) for placing objects into liquid nitrogen.
- Suitable vessels for containment of the cryogen for demonstration purposes: stainless steel bowls, polystyrene containers, such as a cooler box or polystyrene cups. Do not use glass vessels.
- Other equipment as applicable, depending on the specific activities being undertaken.
- Access to a safety shower and eye wash facility.
- Signage: e.g.

OR

(*These signs, both portrait and landscape, have been included at A4 size at the end of this SOP for you to print and laminate. You could also change your printer settings to print A3 size*.)

6. Operating procedure

1. Wear Personal Protective Equipment (PPE).
2. Work in a well-ventilated area.
3. Do not work alone; a second person should be on standby at all times when liquid nitrogen is being used or transported.
4. Avoid direct contact of liquid nitrogen and its vapours with the skin and eyes. Avoid inhalation of vapours.
5. Use containers and tools designed for use with cryogenic liquids. A stainless-steel bowl or small polystyrene container can be used for the immersion of large items such as an inflated balloon. A polystyrene cup can be used where small quantities of liquid nitrogen are involved. The containers and tools should be clean and dry. Do not use glass vessels.
6. Use tongs to place and remove objects into or from liquid nitrogen.
7. Pouring liquid nitrogen or immersing objects should be done slowly and carefully to minimise boil off and splashing. Never pour from a height above eye level.
8. No activities using liquid nitrogen should be brought close to the face or ears.
9. Liquid nitrogen must not be put into a vessel and sealed due to the risk of an explosion.

7. Trouble shooting/emergencies

- First Aid: See latest SDS for more detailed information
    - Cold burns: Remove contaminated clothing if not stuck to the skin. Flush affected area with tepid water for 15 minutes. Seek medical assistance.
    - In case of frostbite, spray with tepid water for at least 15 minutes. Apply a sterile dressing and seek medical assistance. Do not apply hot water or radiant heat.
    - In case of eye exposure, irrigate eye with tepid water for 15 minutes. Seek medical attention immediately.
    - Inhalation in high concentrations may cause asphyxiation. Symptoms may include dizziness, drowsiness, weakness, fatigue and unconsciousness. Victim may not be aware of asphyxiation. **Rescuers should not put themselves at risk** and should only enter a potentially contaminated area if safe to do so. Remove the person to fresh air and keep them warm and rested. Apply artificial respiration if breathing has stopped. Obtain immediate medical assistance.

8. Waste disposal

- Small amounts of surplus liquid nitrogen may be allowed to boil off as a gas in an operating fume cupboard or a well-ventilated area.
- Liquid nitrogen must not be poured down sinks or drains.
- In case of a spill, evacuate and ventilate the area and allow gas to dissipate.

9. Related material

- SDS
- Risk Assessment
- Suggested activities

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Activities using liquid nitrogen

Many scientific principles can be demonstrated by using liquid nitrogen. These activities are for the use of experienced science teachers and technicians as demonstrations only.

For all activities the following are required:

- **Wear appropriate PPE:** Full face-shield or safety goggles, cold insulating or leather gloves, enclosed shoes, laboratory coat,
- Safety shield as required.
- Liquid nitrogen in a wide mouthed Dewar flask.
- Polystyrene container, long tongs and additional items as noted.
- Access to a safety shower and eye wash facility.

Liquid nitrogen must not be put into a vessel and sealed due to the risk of an explosion.

None of the activities should be brought close to the face or ears.

Note: *Many online videos do not demonstrate safe activities or safe procedures and we advise caution in performing similar demonstrations or showing of these videos. We have included links to some videos that can be shown in place of or in addition to the suggested activities.*

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1. Cryogenic freezing of organic materials

The properties of many common organic materials change irreversibly when immersed into liquid nitrogen. Organic materials are high in water content and they become hard and brittle when all the water freezes, forms crystals and their cells burst.

1.1 Freezing and crushing a flower

Additional item: Flower (carnation, rose or daisy work well)

Method:

1. Using tongs immerse a flower into the liquid nitrogen and wait until it stops boiling.
2. Remove the flower and observe that it looks normal.
3. The flower however has become brittle and will crumble when squeezed with gloved hands.

Video:

'The rose crush experiment', YouTube, https://youtu.be/TFUNt0Byyxw (1.58 min)

1.2 Freezing and brittle nature of banana

Additional items: Banana, hammer, nail, piece of chipboard or other soft timber

Method:

1. Using tongs, immerse the unpeeled banana in the liquid nitrogen until the liquid nitrogen stops boiling.
2. Remove the frozen banana with the tongs, hold it with a gloved hand and use it to hammer a nail into a piece of chipboard.
3. Observe the banana when it thaws (it becomes very mushy).

Video:

'The banana hammer experiment', YouTube, https://youtu.be/KrHm0LHc078 (1.43 min)

1.3 Making ice cream and freezing food

The use of liquid nitrogen for making ice cream for human consumption should be ONLY conducted under the following circumstances:

- Using food grade liquid nitrogen and a Dewar flask dedicated for this use to ensure that the liquid nitrogen is not contaminated with foreign matter or hazardous substances
- In a non-science area, using facilities and utensils suitable for use with food for human consumption.

Freezing food items for human consumption is advised against due to a high risk of injury, where liquid nitrogen has not completely evaporated. Injuries may include cryogenic burns to the mouth, respiratory system and stomach.

Additional items: milk, cream, sugar, vanilla, large stainless-steel bowl, whisk, wooden spoon.

Method:

1. Mix all ingredients in bowl until the sugar has dissolved.
2. Slowly add small quantities of liquid nitrogen to the mixture until it begins to harden. The whisk may need to be replaced with a wooden spoon to make mixing easier.
3. All the liquid nitrogen needs to be allowed to evaporate off before it is safe to eat.

Video and resources:

'The ice cream experiment', YouTube, https://youtu.be/mUVyCKxfGd8 (2.41 min)

Liquid nitrogen ice cream recipe
https://www.acs.org/content/dam/acsorg/education/resources/highschool/chemmatters/liquid-nitrogen-ice-cream-recipe.pdf

2. Cryogenic freezing of other objects

Exposure of some materials at very low temperatures can change their properties. Cooling a solid will not change its state but will make it more rigid and brittle.

2.1 Changes in elasticity of rubber materials

Additional items: Small rubber objects such as rubber tubing, a rubber glove or rubber band and a hammer.

Method:

1. Demonstrate that the rubber object is pliable and will stretch at room temperature.
2. Using tongs, immerse the rubber object into the liquid nitrogen until it stops boiling.
3. Remove the rubber object with the tongs and use it to hit the table to show that it now is not flexible or stretchy.
4. Hit the rubber object into pieces with the hammer in a tub to contain broken pieces.
5. When the pieces have warmed up they will return to their original pliable material.

Note: rubber bouncy balls are likely to shatter when attempting to bounce them and rubber tubing may act as a conduit to spray liquid nitrogen, so ensure that this will not impact nearby people. It is not recommended to freeze rubber stoppers as the outside cools quicker than the internal part of the stopper, which can create an explosion.

Explanation: The rubber can be snapped whilst frozen but will regain its elasticity when it is warmed to room temperature.

Video:

'Effects of Liquid Nitrogen on Rubber', YouTube, https://youtu.be/BPaNEtlQ8sg (2.11min)

2.2 Other materials

Other materials such as marshmallows could be demonstrated, however it is advised against eating frozen marshmallows due to the risk of severe burns.

Note: Soft drink cans must not be frozen due to the risk of an explosion.

3. Contraction and expansion

Gases will contract and liquefy when they are cooled and turn back into a gas when heated. Charles's law states that the volume of gas decreases when the temperature is decreased.

3.1 Shrinking balloon

Additional item: Balloon

Method:

1. Half fill the polystyrene container or stainless-steel bowl with liquid nitrogen
2. Inflate a balloon and tie off the end.
3. Using tongs carefully push the inflated balloon into the liquid nitrogen. The balloon will shrink and become rigid in the liquid nitrogen.
4. Once the balloon has shrunk, remove it with the tongs. Liquified air may be observed in the balloon as it is removed from the liquid nitrogen.
5. Allow the balloon to warm to room temperature.

Explanation: When the balloon is cooled the air-pressure is reduced allowing it to shrink, but when it returns to room temperature, the air-pressure and volume inside the balloon is restored bringing it back to its original shape.

Videos:

'Freezing Balloons!', Frostbite Theater, http://education.jlab.org/frost/balloon.html (2.39 min)

'MIT Physics Demo – Balloons in Liquid Nitrogen', YouTube, https://youtu.be/ZvrJgGhnmJo (1.18 min)

3.2 Whistling tea kettle

Additional items: Whistling kettle, Perspex safety shield

Method:

1. Carefully transfer a very small amount of liquid nitrogen into a whistling kettle.
2. Place the whistle cap over the opening (when required)
3. The liquid nitrogen boils and the kettle will begin to whistle.

Explanation: As the liquid nitrogen boils, nitrogen gas is produced which escapes from the kettle activating the whistle

Video:

'Liquid nitrogen and the Tea Kettle Mystery', Frostbite Theater, http://education.jlab.org/frost/live_tea_kettle.html (5.58 mins)

3.3 Spinning ping pong ball

Additional items: Ping pong ball, pin, large plastic Petri dish or small hoop

Method

1. Using the pin, poke a hole in a ping pong ball.
2. Using tongs, submerge the ping pong ball into the liquid nitrogen and hold it firmly until the liquid nitrogen stops boiling and the ball contains some liquid nitrogen (about 30 seconds).

3. Remove the ball and place it on a flat surface such as a table inside a large plastic Petri dish or small hoop.
4. The ball will spin.

Explanation:

The liquid nitrogen in the ball vaporises to nitrogen gas at room temperature. The gas expands and as it leaves through the hole in the ball, the force pushes the ball into a circular motion—like a sprinkler effect.

Video:

'The spinning ping pong ball experiment', YouTube, https://youtu.be/a16AaNWeOU4 (2.12 min)

3.4 Thermal contraction and expansion of metal

Additional items: metal ring and ball

Method

1. Demonstrate how the ball can fit through the ring
2. Place only the metal ring into the liquid nitrogen for around 15 seconds
3. Show how the ball no longer fits through the ring
4. After the ring has warmed to ambient temperature, the ball will now again fit through the ring.

Explanation:

When the metal ring is cooled the metal contracts, so that the metal ball no longer fits through it. When the metal ring is warmed the metal expands, enabling the metal ball to again fit through the ring,

Video:

'The thermal contraction experiment', YouTube, https://youtu.be/o02sYBJZBUI (2.04 min)

4. Effects of boiling liquid nitrogen

When liquid nitrogen is poured onto a smooth surface, beads of liquid nitrogen travel quickly and smoothly along the surface due to the Leidenfrost effect. The liquid floats on a small layer or cushion of gas produced by the fast boiling liquid. We recommend that a surface such as a metal tray is used, with an insulating layer between it and the floor or bench. Direct contact with floor or bench surfaces may result in damage to the surfaces.

Video:

'Let's Pour Liquid Nitrogen on the Floor!', Frostbite Theater, http://education.jlab.org/frost/lets_pour_liquid_nitrogen_on_the_floor.html (2.26 min)

4.1 Floating chalk

Additional items: Piece of chalk

Method:

1. Using tongs pick up the piece of chalk and soak it in liquid nitrogen until it stops boiling.
2. Remove the piece of chalk and place it on a smooth flat surface such as a tabletop. The chalk should float.

Explanation:

The nitrogen gas within the chalk evaporates and has sufficient force to lift the chalk off the surface of the table like a hovercraft.

4.2 Extinguishing a flame

Additional items: a suitably sized clear plastic container, 3 candles of varying heights (make sure they all are within the height of the container), matches

Method:

1. Secure the candles to the base of the plastic container using plasticine or melted wax
2. Light the 3 candles
3. Carefully pour a small amount of liquid nitrogen into the bottom of the container.
4. As the liquid nitrogen boils it produces nitrogen gas which fills the container. As the gas rises and fills the container, the shortest candle is extinguished first followed by the middle and then the tallest candle.

Explanation: When the liquid nitrogen boils, the nitrogen gas produced mixes with the air and displaces the oxygen gas that's available and slowly the flames go out. Without oxygen a flame cannot be sustained.

5. Testing of different insulating materials

Additional items: 2 cups, one clear plastic, the other polystyrene; water; 50mL measuring cylinder; 2 small stainless-steel bowls.

Method:

1. Add 50mL of water to each of the cups
2. Add liquid nitrogen to the top of each bowl
3. Place each cup into a bowl of liquid nitrogen for about a minute, holding the container down with a gloved hand or tongs.
4. Remove the cups, observe if any ice has formed and measure the amount of liquid remaining in each
5. Observe the amount of liquid nitrogen in the corresponding bowl

Explanation:

The plastic cup lost a lot of heat through its wall that went into the liquid nitrogen. Therefore, the water lost heat, got colder and started to freeze and the nitrogen gained heat and boiled faster. The polystyrene cup is the better insulator

Video:

'Fair test of insulating properties of different plastic cups', Frostbite Theater, http://education.jlab.org/frost/insulators.html (5.04 min)

6. Effect on electrical resistance

When metals that conduct electricity are cooled, their electrical resistance is greatly reduced and they may be known as super conductors, which also affects their magnetic field. The following videos may be helpful in demonstrating some of these effects.

Videos:

'The floating magnet experiment', YouTube, https://youtu.be/tItB5-TdOA8 (2.00 min)

'The flying ring', Frostbite Theater, http://education.jlab.org/frost/ring_fling.html (3.11 min)

'What happens to circuits in liquid nitrogen', YouTube, https://youtu.be/p8bN2PwlbR0 (2.51 min)

Additional resources

'Liquid Nitrogen Experiments', Frostbite Theater, https://education.jlab.org/frost/ – collection of science videos produced by Jefferson Lab.

LIQUID NITROGEN

EXTREME COLD

DO NOT TOUCH

USE HAND/FACE/EYE PROTECTION

ENSURE ADEQUATE VENTILATION

LIQUID NITROGEN

EXTREME COLD
DO NOT TOUCH

USE HAND/FACE/EYE PROTECTION

ENSURE ADEQUATE VENTILATION

STANDARD OPERATING PROCEDURE:

Demonstrating the flame test using a PET bottle

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

A flame test is a routine practical performed across various year levels and areas of science. It demonstrates the specific emission spectrum of a variety of cations. A sample of a cation is introduced into the blue flame of a Bunsen burner and the flame is then observed for any colour change. The flame test identifies a cation by the characteristic colour that it turns the flame of a Bunsen burner.

The emission spectrum of an element is the colour emitted when the heat of the Bunsen flame causes its electrons to absorb energy from the flame and make a transition from a low-energy state to a high-energy state. As these excited electrons then naturally fall back to their low energy state, they emit energy in the form of a particle of light called a *photon*. The colour of the light produced in the flame is determined by the energy of the emitted photon and displays a characteristic wavelength[1]. Each substance has a specific emission spectrum based on their different electron configuration, which allows them to be differentiated from each other. There have been a number of versions of doing this over the years, from using wire loops, to soaking toothpicks (or similar) in various solutions or solid chemicals, to spraying cation solutions directly into a Bunsen flame. We advise against methods using flammable liquids[2]. Here we describe a method of spraying a cation solution into a Bunsen flame utilising a 2 L PET bottle. The advantage of this method is that the majority of the sprayed cation solution (many of which are hazardous and/or toxic) is contained within the set-up, minimising the spread and inhalation of aerosols in the laboratory environment. This method also allows for the containment and collection of any over spray that can be collected and reused. It also produces a longer lasting colour in the flame for observation with the naked eye or with a spectroscope.

2. Context

- Part A is the method for the construction of the setup for the technician.
- Part B is for the operation of the setup in the classroom to be supervised by experienced teachers and technicians. It can be set up for a teacher/technician demonstration or as stations around the room for students to rotate around

3. Safety notes

Construction

- When using a cutting blade/knife to cut the PET bottle use caution as the knife can slice through quickly once it has started and may not follow the desired path. Ensure that the direction of the knife is away from the person or fingers. Scissors can be used instead once the initial cut has been made.

- Ensure that the bottle is held fairly securely, e.g. tape to a cutting board when drilling the holes as the drill bit may slide off the bottle particularly if the bottle moves.
- Use a drill bit that is smaller than the hole required and then it is easy to enlarge the hole by holding the drill bit in the hole and using the side of the bit to gradually increase it to the required size.
- Label the spray bottle and PET bottle with the cation being tested. Keep them as a pair to avoid the risk of any cross contamination from the other cations interfering with the flame colour.

Classroom

- Ensure that the bottle is held securely with a retort stand and clamp when in use to prevent it from falling over.
- Ensure that the work area around the Bunsen burner is clear of combustible materials.
- Students should be closely supervised when carrying out this activity and appropriate facilities should be available i.e. running water, adequate ventilation and eyewash station.
- Provide relevant Personal Protective Equipment (PPE), i.e. safety glasses.
- Avoid contact with skin and eyes, and avoid breathing in any of the spray.
- For safety, first aid, accident and spill procedures refer to the SDS for each chemical being tested. In all cases, if the chemical gets into the eye, rinse well with water for at least 15 minutes.
- Ensure everybody is aware of when the Bunsen burner is alight as it will be on the blue flame rather than the safety yellow flame.
- Wash hands with soap and water at the completion of the activity

4. Regulations, licences and permits

- Not applicable

5. Equipment

- Bunsen burner (with sufficient length of tubing to comfortably reach the gas tap)
- 15 cm piece of wire (to enable adjustment of the collar of the Bunsen burner from a safety flame to a blue flame once it is in position within the PET bottle)
- 2 L PET bottle (remove original labels and replace with appropriate chemical information)
- 500 mL garden spray bottle with a round nozzle (labelled with the appropriate chemical information)
- 2 cm of plastic tubing (to fit over the nozzle of the spray bottle to allow it to fit into the hole of the PET bottle)
- Piece of copper pipe lagging or silicone bakeware (to wrap around the barrel of the Bunsen burner to allow it to fit snuggly into the neck of the PET bottle)
- Cutting blade/knife such as a box cutter or Stanley knife (to cut an opening in the bottle to fit a Bunsen burner through), scissors can be used after initial cut.
- Electric drill with approximately 10 mm bit (to drill holes in the PET bottle for the Bunsen burner tubing and spray bottle connection)
- Retort stand, boss head and clamp.
- Marking pen
- 1M cation solutions (chloride salts are best) made up with distilled or deionised water
- Heat resistant tape
- Spectroscope (optional)
- PPE: Gloves for cleaning up

6. Operating procedure

See diagram below for a visual of the end product.

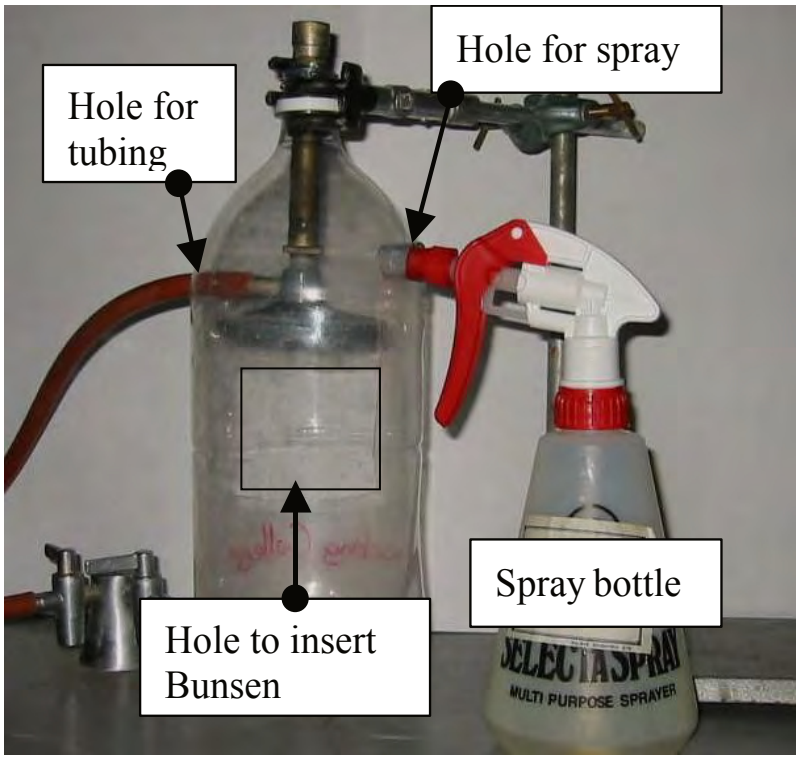
Part A: construction

1. Mark and cut an opening in the PET bottle – about 7 x 6 cm and 10 cm from the bottom of the bottle.
2. Place the garden spray bottle beside the drink bottle.
3. Mark and drill (or cut) a hole at the height of the spray bottle nozzle in the PET bottle to connect the spray bottle nozzle. The hole should be made on the RHS of the Bunsen insertion hole when it is facing you. Attach a short 2 cm piece of plastic tubing that fits on the nozzle as a connector. (NB: do not use a longer piece of connection tubing, as it will interfere with the spray and produce a stream rather than a mist which will not enter the Bunsen flame.)
4. Mark and drill (or cut) a hole opposite the one made for the spray bottle so the Bunsen tubing can be inserted. This should be at a height so the level of the spray bottle nozzle is at the same level as the air intake of the Bunsen burner.
5. Wrap some packing (lagging from copper hot water pipes or some silicon from bakeware) around the barrel of the Bunsen burner and tape it into position.
6. Push the Bunsen burner tubing through the small hole and attach it to the Bunsen burner. Push the Bunsen through the rectangular hole and poke the barrel into the neck of the bottle. Check that the Bunsen holds steady, if not add more lagging or tape.
7. Label the PET bottle and spray bottle with the particular cation being tested. Keep them as a pair.
8. Repeat steps 1–8 for the number of sets required (this is based on the number of cations being tested plus one for an unknown).
9. Fill the spray bottle with 200 mL of the labelled aqueous solution. Use 1M solutions of chloride salts made up with deionised or distilled water.

Part B: Set up and operation in the laboratory

1. Set up a retort stand and clamp the neck of the PET bottle to hold it and the Bunsen assembly stable.
2. Adjust the spray of the matching spray bottle to a fine mist, and attach it to the corresponding hole of the PET bottle with the 2 cm plastic tubing connector piece. Keep this tubing to the smallest length possible or the mist spray may catch it and come through as a stream not a mist.
3. Adjust the air intake valve of the Bunsen with the wire (if necessary) to the fully open position to obtain a blue flame. Connect the tubing to the gas supply and light the flame.
4. Squeeze the trigger of the spray bottle once or twice allowing the fine mist to enter the air intake hole of the Bunsen. The flame will display the corresponding colour for the cation for about 10–20 seconds. This is long enough for students to use spectroscopes to observe the emission. Dim the lights in the laboratory for a clearer result.
5. When the activity is complete, any over spray that is contained in the PET bottle, can be collected and poured back into the spray bottle to be reused.
6. Rinse the PET bottle with tap water. The washings can be emptied down the sink, as only trace amounts of the salts are present. Include rinsing the air intake and gas jet of the Bunsen burner in this process to prevent clogging up of the gas jet.
7. It is also recommended to remove and rinse the spray mechanism from the spray bottle to avoid blockages prior to storage.

Image of the finished product showing the location of the holes to insert the Bunsen and its tubing and the plastic tubing connecting the spray bottle. (Image courtesy of Dale Carroll)



To watch a video demonstrating the set-up in operation see:

'Demonstrating metal ion flame tests using a PET bottle laboratory rig', YouTube (7:26 min) https://youtu.be/kzWbIcpZUi8

There are many cations that can be tested. The following list is a suggestion. Chloride salts are generally used. Permanganates, nitrates and chlorates should be avoided due to hazardous products when burned[3]. All solutions are made with distilled or deionised water to reduce any contamination.

Table of colours for chemicals[1]

Dominant colour	Approximate wavelength (in nm*)	Compound
Red	701	Lithium chloride (LiCl)
Crimson-Red	700	Strontium chloride ($SrCl_2$)
Orange	609	Calcium chloride ($CaCl_2$)
Orange-Yellow	597	Sodium chloride (NaCl)
Yellow-Green	577	Barium chloride ($BaCl_2$)
Green-Blue	492	Copper chloride ($CuCl_2$)
Violet	423	Potassium chloride (KCl)

*Wavelength values here are given for the mid-range of the colour indicated.

7. Trouble shooting/emergencies

First aid: See latest SDS for more detailed information on chemicals used

- **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek medical attention if symptoms persist
- **If in eyes:** Hold open and irrigate with a copious quantity of water for at least 15 minutes. Seek medical attention if symptoms persist
- **If on skin/clothes:** Wash affected area with copious quantities of water immediately. Remove contaminated clothes and wash before reuse. If swelling, redness, blistering or irritation occurs seek medical attention.
- **If inhaled:** Remove to fresh air and seek medical attention if symptoms persist.
- **If toxic to the environment:** Avoid release to the environment.
- For further advice contact the Poisons Information Centre on 131126
- **For burns**: In the event of a burn, hold the burnt area under cold running water for 20 minutes.

Any health concerns should be referred to the school first aid officer for assessment, accompanied by the relevant latest SDS if applicable. Follow your school's accident and incident policy and reporting procedures.

Set up:

- Check that the nozzle of the spray bottle is not blocked. It is best removed and cleaned at the end of each activity.
- Make sure that the plastic tubing connecting the spray bottle to the PET bottle is not too long, as the spray needs to be a 'mist' going into the PET bottle and not a stream.
- Cleaning of the Bunsen burner is recommended at the end of the activity to prevent it from becoming blocked. Periodic, thorough cleaning may be required.

Limitations of flame tests[4,5]

- They are a qualitative, not a quantitative technique. They only detect the presence of a certain element and not how much is present.
- They cannot detect low concentrations of most ions.
- Contaminating substances can mask the flame colour and affect the results.
- Sodium is a common contaminant and its orange-yellow spectrum can dominate over others. Looking through blue glass can filter out this impurity.
- The test cannot differentiate all elements. Some can produce the same colour in the flame and some will not change the colour of the flame.
- Accuracy can be improved by viewing though a spectroscope.
- The colour of the flame is subjective.

8. Waste disposal

- Any unused chemical contained within the set-up can be returned to the spray bottle for future use. The water used to rinse the bottle at the completion can be washed down the sink with water, as it will only contain trace amounts of the salts tested.

9. Related material

- A site specific Risk Assessment should be conducted.

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STANDARD OPERATING PROCEDURE:

Demonstrating the reaction of alkali metals (lithium and sodium) with water

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

In the school science curriculum, lithium and sodium are relatively safe alkali metals for demonstrating the properties of the Group 1 elements of the periodic table. The reaction of these alkali metals with water is one of the most engaging and popular demonstrations carried out in secondary schools.

The alkali metal atoms have one electron in their outer shell and will readily lose that electron to a non-metal to give them a more stable full outer shell. This process releases much energy, which is what makes these elements so very reactive. The reactivity of the Group 1 alkali metals increases down the group. When in contact with water the alkali metal is oxidised, with each atom losing an electron to become a metal ion, while water is reduced to hydrogen gas and hydroxide ion. This reaction can be explosive as the heat generated can ignite the hydrogen gas produced.

Lithium is a rare element, accounting for only 0.0007% of the Earth's crust in its mineral form. Lithium is the lightest of the alkali metals and reacts the least violently when compared with other Group 1 elements. In contact with water, lithium reacts exothermically to form lithium hydroxide and hydrogen gas, with the following chemical equation:

$$2Li(s) + 2H_2O(l) \rightarrow 2LiOH(aq) + H_2(g) + \text{heat}$$

Sodium is the sixth most abundant element in the Earth's crust and probably the most well-known of the alkali metals. Sodium is more reactive than lithium but less reactive than potassium and other Group 1 elements appearing below it in the group. When exposed to air, sodium is oxidised to a mixture of the oxide (Na_2O) and the peroxide (Na_2O_2). In contact with water, sodium reacts exothermically, forming sodium hydroxide and hydrogen gas with the following chemical equation:

$$2Na(s) + 2H_2O(l) \rightarrow 2NaOH(aq) + H_2(g) + \text{heat}$$

2. Context

- These instructions are for the use of experienced science teachers and technicians only.
- **The use of lithium and sodium in their reaction with water is for small-scale demonstration purposes only**.

Science ASSIST strongly advises against schools conducting demonstrations using pieces of lithium or sodium, which are larger than a 3mm-side cube. The use of large pieces of these metals for this activity has the potential to cause explosions and cause injury to people and damage to property. Limiting the size of the piece of metal minimises the risk of a dangerous explosion occurring.

Note: Larger scale demonstrations inside or outside are not recommended due to the uncontrolled release of energy and unpredictable consequences of the reaction. The risk of serious injuries or damage outweighs the educational benefits.

3. Safety notes

- Because they are so reactive with air and water, lithium and sodium require special storage conditions. They should be stored submerged under mineral oil such as paraffin oil. Lithium will float on the paraffin oil, but a film of oil over the lithium will protect it from oxidation, which is also a slower and less vigorous process than for sodium.
- Both lithium and sodium metals are flammable and corrosive. They can cause severe skin burns and eye damage,
- **Minimal quantities only should be taken into the classroom for demonstrations**
- Make sure all implements for handling these metals e.g. spatulas, forceps/tweezers, cutting surface (petri dish or ceramic tile) and scalpel/knife are clean and **dry**.
- Use a separate set of implements and cutting surface for each metal to avoid any cross contamination between them.
- A large volume of water should be used in the reaction vessel to reduce the risk of the reaction going out of control due to the heat generated.
- Both metals release hydrogen gas when reacting with water. Hydrogen is a flammable gas and it should not be allowed to accumulate in a contained volume of space or be handled near any ignition sources. The vessel should be filled with water to within 1cm from the top so there is minimal space for the air/hydrogen mixture to accumulate.
- Caustic fumes can also be produced and should not be inhaled. Therefore this activity should be conducted in a well-ventilated area.
- Pieces of the alkali metal can sometimes be ejected from the water, therefore the use of safety screens is recommended and observers should watch from a safe distance away (at least 2m). Some references suggest covering the trough with wire gauze to prevent the sodium ejecting. **Never cover with a solid piece of glass or Perspex.**
- **Do not wash any unreacted pieces of metal down the sink. Ensure that all remnants of alkali metal are fully reacted at the completion of the demonstration.**

4. Regulations, licences and permits

- Should adhere to storage and disposal requirements as described in current SDS.
- No licences or permits required.

5. Equipment

- PPE for demonstrator: lab coat, nitrile gloves and safety glasses or face shield
- PPE for observers: safety glasses for observers standing at least 2 metres away
- Container with sodium or lithium metal (submerged in paraffin oil)
- Spatula
- Tweezers or forceps
- Petri dish or ceramic tile to use as a cutting surface
- Dry paper towel (to absorb excess oil)
- Scalpel or sharp knife to cut the metal
- Reaction vessel: wide glass trough or large beaker made of quality borosilicate glass.
- 0.1%w/v Phenolphthalein solution in dropper bottle
- Perspex safety screen/s (note: (i) some references suggest the use of an operating fume cupboard; (ii) Science ASSIST recommends using a safety screen, however if the pieces of alkali metal are smaller than a 3mm-side cube, a screen may not be warranted, based upon a local risk assessment)
- Wire gauze to cover trough (optional)
- Overhead projector or a camera (optional)

6. Operating procedure

1. Fill a wide glass trough or large glass beaker (made of quality borosilicate glass) with water to within 1cm from the top so there is minimal space for the air/hydrogen mixture to accumulate.
2. Add a few drops of the 0.1% phenolphthalein solution into the water. The resulting solution from the reaction is basic and will be illustrated with a colour change to pink when phenolphthalein is present in water.
3. Place safety screen/s as close to the trough as possible to protect all observers and demonstrator. Ensure the correct wearing of PPE and that observers are at least 2 metres away.
4. Using a dry and clean spatula, remove one piece of the alkali metal from the storage jar.
5. Place it on the cutting surface, ensuring conditions are dry. Using a dry scalpel/sharp knife, cut a small piece of the metal, no larger than 3mm–side cube.
6. Remove excess oil from the small piece of metal by patting it on dry paper towel; oil can interfere with the reaction. Alternatively the sodium can be cut on a piece of dry paper towel in the petri dish, provided that it is not left on the paper towel for too long.
7. Whilst cutting the metal, students can be shown the shiny silver-white surface of the alkali metal which rapidly darkens due to its oxidation in air. (Sodium reacts faster with air compared to lithium). Do not pass the alkali metal around the room.
8. Return the unused metal piece to the storage container as soon as possible. Ensure metal pieces are covered in oil and that the container is securely closed.
9. Using dry forceps/tweezers, pick up the small piece of metal and carefully place it onto the water surface in the trough or beaker. A piece of wire gauze could also be used to cover the trough (optional).
10. Observe the reaction. The alkali metal should float, fizz and move leaving a pink trail behind as it reacts with the water to form its hydroxide. (The phenolphthalein indicator turns pink in the presence of the hydroxide.) Lithium will react less vigorously than sodium. Never try to constrain the metals; allow them to roam freely over the surface of the water.
11. Ensure that the forceps are dry for handling the next piece of sample of metal.
12. Use a fresh trough (or fresh water) and new **dry** implements for the next metal and repeat steps 1-10.
13. When the demonstration is completed, ensure that no traces of the metals remain on the tweezers/forceps, spatula, petri dish etc. The simplest way is to place them in one of the troughs of water to allow any residual metal to react. Gloved hands should also be rinsed in the water to remove any traces of metal before disposal of the gloves into the bin. Allow the metal to react completely with the water and the remaining hydroxide solution can then be flushed down the sink with plenty of water.
14. An overhead projector or a camera can be used to project the view of the reaction onto a screen. (Optional).

7. Trouble shooting/emergencies

- First Aid: See latest SDS for more detailed information
  - **If swallowed**: Rinse mouth thoroughly with water immediately.Give plenty of water to drink. Give repeated drinks of water one cupful every 10 minutes. Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention.
  - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek medical attention.
  - **If on skin/clothes**: Wash affected area thoroughly with soap and water. Remove contaminated clothing and wash before reuse or discard. Seek medical attention.
- For further advice contact the Poisons Information Centre on 131126

8. Waste disposal

- If there are any small pieces of lithium and/or sodium to be disposed, allow them to react completely with water. Do not wash them directly down the sink.
- The dilute hydroxide solution formed in the reaction vessel after the demonstration can be disposed of down the sink with plenty of water.

9. Related material

- Risk assessment for demonstrating the reaction of alkali metals (lithium and sodium) with water
- Current SDSs for lithium metal, sodium metal and 1% phenolphthalein solution.

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STANDARD OPERATING PROCEDURE:

The Thermite Reaction

Note: **The Thermite Reaction is potentially a very hazardous activity; however the hazards it presents can be safely managed with suitable controls in place. It should only be performed as a demonstration by experienced teachers and technicians in conjunction with current Safety Data Sheets (SDSs) and a site-specific Risk Assessment. Intense heat, brilliant light and molten metal are produced by this reaction. The demonstration should be performed outside on a fire resistant solid surface such as concrete in a windless area well away from any combustible or flammable materials.**

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1. Introduction

The Thermite Reaction is an excellent example of a highly exothermic (heat evolving) and redox reaction involving the oxidation of a metal powder and reduction of a metal oxide. The most commonly performed reaction involves igniting a mixture of aluminium powder and iron (III) oxide to yield molten iron and alumina (aluminium oxide) according to the equation:

$$2Al + Fe_2O_3 \rightarrow Al_2O_3 + 2Fe$$

The Thermite Reaction is an example of a more reactive metal displacing a less reactive metal from a compound. In this reaction, aluminium being more reactive displaces iron from the iron (III) oxide and removes the oxygen. The iron is reduced and the aluminium is oxidised. The iron generated is molten due to the amount of heat released during the reaction. The reaction can reach up to a temperature of about 3000°C.

Industrially, the Thermite Reaction is used for welding metal parts such as railway rails, for underwater welding and in metal refining.

2. Context

- These instructions are for the use of highly experienced chemistry teachers and technicians only.
- Do not attempt this experiment without assistance from an experienced colleague.

3. Safety notes

This activity has many potential hazards which need to be evaluated:

- For all chemicals consult current SDSs and conduct a site-specific risk assessment for the Thermite Reaction. Science ASSIST has developed an example risk assessment for this activity.
- Note that preparation for this demonstration may take several days to allow time for conducting a risk assessment, developing risk-mitigation measures and for the purchase and preparation of materials. A trial of the reaction on a smaller scale should be considered if staff have no prior experience with the reaction. The performance of the

activity is also subject to weather conditions: a day of mild weather without strong breezes should be chosen for this activity. Do not conduct the activity on a day when a total fire ban is in force.

- Select a suitable location for the reaction. As this reaction is highly exothermic and produces molten metal and sparks, select an outside area on a fire resistant solid surface such as concrete, protected from wind and well away from any flammable or combustible materials.
- The reaction produces brilliant light, intense heat, smoke, molten metal, flying sparks and shrapnel which may be thrown a great distance, therefore students are to be observers only and must stand at least 10 metres from the reaction. A Perspex safety screen should be used to help protect from any sparks.
- Check that the flowerpots are free of cracks, and that the bucket and support stand are sturdy and securely placed.
- Do **not** pre-prepare and store the iron oxide/aluminium powder mixture. Only prepare it just before use. Use chemicals which are of good quality and not out of date.
- It is important to ensure that there is no trace of moisture in the reactants, flowerpots, glassware, or sand as this could lead to a violent explosive reaction.
- Note that the Thermite Reaction is self-sustaining and cannot be extinguished by removing oxygen either by smothering with sand or using a fire extinguisher.
- Water should **not** be used to attempt to extinguish the reaction, since addition of water to hot iron produces potentially explosive hydrogen gas.
- A fire extinguisher should be on hand, not for extinguishing the reaction, but in case of flying sparks igniting spot fires.
- Once ignition is initiated (by lighting the magnesium fuse or sparkler or by addition of glycerol to potassium permanganate), the demonstrator must retreat quickly to at least 10 metres from the reaction.
- The demonstrator and observers should not look directly at the burning magnesium ribbon, if this ignition method is used.
- Since the reaction products will remain red hot for some time, the apparatus must not be moved until cooled. Cordon off the area until cooled sufficiently and then wearing safety glasses and heat-protective gloves dismantle the apparatus. Retrieve the iron with tongs.
- Students, wearing gloves, may handle/inspect the cast iron produced when it is cold.

4. Regulations, licences and permits

Not applicable

5. Equipment

- PPE for demonstrator: face shield, heat protective gloves, lab coat, closed in shoes and long trousers
- PPE for observers: Safety glasses for student and other observers standing at least 10 metres away
- Metal trash can or metal bucket ¾ filled with **dry** sand
- Sturdy metal retort stand (base may need weighting)
- Perspex safety screen

- Metal clamp to hold flowerpots (a ring clamp is recommended over a four-pronged clamp)
- Two **new** dry 10cm clay flowerpots with a large central drainage hole in their base. *During the experiment the inner pot will crack or shatter but the outer pot will remain intact. Do not reuse this outer pot as it may contain unseen cracks. During the experiment the molten iron will drop through the holes of the pots and onto the sand.*
- Small filter paper to fit in the base of one flowerpot. *This prevents the powder from falling out the bottom and will burn away during the experiment to allow the molten iron to flow through.*
- Plastic beaker and glass rod for mixing the chemicals
- Large dry glass test tube for packing down the reactants in the flowerpots
- Metal tongs
- Gas lighter (if using magnesium ribbon or a sparkler)
- Fire extinguisher on hand for spot fires
- 6 g aluminium metal, fine granular or aluminium powder
- 18 g iron (III) oxide powder (Fe_2O_3)
- Choice of ignition:
    - Method A: 18cm strip of magnesium ribbon
    - Method B: a sparkler
    - Method C: 8 g potassium permanganate and 3-4mL glycerol. The glycerol is added **last** to react with the potassium permanganate to initiate ignition. The reaction of permanganate with glycerol can take up to a few minutes to start. Fine permanganate crystals will react faster than coarser crystals.

6. Operating procedure

1. Do not rush the set-up of this activity. Plan carefully considering the weather conditions. Do not conduct on days when a total fire ban is in force.
2. Ensure that this demonstration is conducted outside, on a fire resistant solid surface, in a windless area, and that all observers are wearing safety glasses and positioned at least 10 metres from the reaction
3. Remove any flammable or combustible materials from the demonstration area.
4. Instruct observers not to directly look at the burning magnesium ribbon, if this ignition method is used.
5. Stack one clay flowerpot inside the other.
6. Place the small filter paper in the bottom of the inner flowerpot (see picture).

(Image by G. Ward, 2015)

7. Just prior to conducting the reaction, combine the aluminium powder and iron (III) oxide gently in a dry plastic beaker. Ensure that the reactants are well-mixed by stirring with a glass rod. Transfer the mixture to the inner flowerpot.
8. Using the large test tube gently pack down the mixed powders.
9. Place both flowerpots inside the clamp and suspend them over the centre of the metal bucket of dry sand to catch the molten iron (see picture).

(Image by G. Ward, 2015)

10. Select your ignition method - A, B OR C:
    A. Stand an18cm strip of magnesium ribbon in the iron oxide/aluminium powder mixture by inserting at least two-thirds of the depth of the reactant mixture.
    B. Push the metal handle of a sparkler down through both flowerpot holes including the filter paper so the sparkler stands upright and the grey sparkler material just touches the chemicals.
    C. Using the test tube, make a depression in the iron oxide/aluminium powder mixture and transfer the potassium permanganate into this. Then make a smaller depression in the potassium permanganate for the glycerol.
11. Initiating ignition:
    A. Light the magnesium ribbon with a gas lighter.
    B. Light the sparkler with a gas lighter.
    C. Pour the glycerol onto the potassium permanganate. In cold weather, the glycerol should be pre-warmed for easier pouring.
12. Once ignition has been initiated, quickly retreat to at least 10 metres away from the reaction.
13. When the reaction is complete, allow it to cool completely before using tongs to remove the piece of iron for examination. At this point you could demonstrate the magnetic properties of the iron produced.

7. Trouble shooting/emergencies:

- First aid for chemicals: See current SDSs for more detailed information
  - **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention.
  - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek urgent medical attention.
  - **Skin/clothes:** Remove contaminated clothing and flush skin and hair with running water. Seek medical attention.
  - **If inhaled:** Remove to fresh air and seek medical attention if symptoms persist. For further advice contact the Poisons Information Centre on 131 126.
- **Burns:** Hold burnt area under cool running water for 20 minutes. Only remove clothing if it is not stuck to burn. Cover with non-adherent burns dressing, plastic wrap or loosely applied aluminium foil. Do not apply lotions, ointments, or adhesive coverings. Seek urgent medical attention.

8. Waste disposal

- The cast iron produced can be used in the classroom to show magnetic properties.
- If ignition was by method A or B, the cast iron product can be disposed of in the general waste. If ignition was by method C, the product should be disposed of via a licenced waste disposal contractor.
- Dispose of both flowerpots in the general waste. Do not reuse outer flowerpot for the next Thermite Reaction demonstration as it has been stressed and possibly contains unseen cracks.

9. Related material

- Example risk assessment
- Videos are available on YouTube. The teacher should review these carefully before showing to students, as there are various Thermite methods shown using different chemicals and also links to other dubious, explosive experiments, which may not be appropriate for students.

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History of reviews

Date	Version Number	Notes
Oct 2014	Version 1.0	Draft version
Sept 2015	Version 2.0	This replaces earlier draft version Includes updated photos and information

STANDARD OPERATING PROCEDURE:

Performing a brain dissection

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

Sheep or pig brain dissections are conducted to explore the structure and function of the different parts of the brain. Sheep or pig brains are similar in their composition but have a simpler structure than a human brain.

Sheep or pig brains suitable for dissection can be supplied as fresh, frozen or preserved specimens. They can be obtained from the meat sections of some supermarkets, butcher shops, abattoirs or a reputable biological supplier that has passed relevant health inspections.

Brains for dissection can be obtained some weeks beforehand and stored in the freezer, but they are best dissected semi-frozen as they hold their shape better making it easier to identify the different structures. If recently defrosted brains are being used, and the dissection is interrupted, they can be kept for a short time (no longer than 24 hours) in the coldest part of the laboratory fridge or placed in a freezer. There is potential for bacterial growth.

Preserved brains are fixed to prevent tissue breakdown and to render them firm to allow for easy dissection. No refrigeration is required. If using preserved brains, obtain and read the safety data sheet (SDS) from the supplier and prepare a site-specific risk assessment.

2. Context

- These instructions are for the use of experienced science teachers, technicians and students under close supervision.
- When planning a class dissection activity, it is best to discuss beforehand the type of dissection to be undertaken, and warn of the possibility that there may be some blood and odours present during the dissection.
- Demonstrating the dissection to students before they begin is helpful, not only for correct procedure but allows them to adjust to the appearance of the material, and any blood that may be present after dissection material has been washed.
- Let students know they don't have to participate in the dissection and can be excused from the class. Alternative arrangements can be made for students who don't wish to participate, by giving them worksheets to complete and relocating them to a private study area.

3. Safety notes

- A site-specific risk assessment should take into consideration the maturity of students carrying out the dissection and address risks associated with students using scalpels and other dissection equipment.
- Before the dissection it is recommended the teacher or laboratory technician trial the dissecting instruments (scalpels, scissors and pointed forceps) to establish that they are sufficiently sharp enough and to determine the most appropriate equipment for the task considering the student behaviour.

Fainting: signs and symptoms:

- Fainting may occur during this type of activity. Please read the first aid information in section 7 before conducting the dissection.
- Fainting is caused by a sudden drop in blood pressure. Common causes include heat, pain or distress and the sight of blood.
- The possible symptoms include the following.
    - 'Dizziness
    - Light-headedness
    - A pale face
    - Perspiration
    - Heightened anxiety and restlessness
    - Nausea
    - Collapse
    - Unconsciousness, for a few seconds
    - Full recovery after a few minutes'[i]

Handling specimens:

- If using preserved brains, it is important to take note of the preservative solution that the brains are in and the recommended precautions.
    - Rinse the preserved brains under running water immediately upon removal from the preservative solution.
    - Work in a well-ventilated area.
    - It is recommended that people wearing contact lenses should not dissect brains that are in preservative solution. The fumes from the solution can penetrate between the eye and contact lens causing irritation to eyes. It is recommended to wear prescription glasses instead with safety glasses over them OR prescription safety glasses.
- Consider issues such as allergies and chemical sensitivities from handling recently defrosted or preserved brains.
- If using frozen brains, partially defrost overnight in a refrigerator and use within 24 hours. Consistent with safe food handling procedures, all meat products should be stored below 5ºC prior to performing any dissections.
- Good hygiene practices should be observed at all times: Keep hands away from the mouth, nose, eyes and face during and after dissection and wash hands immediately after handling dissection material.

Safety with scalpels and dissecting instruments:

- Store all dissecting instruments securely.
- Care should be taken with sharps such as scalpel blades and scissors. Some school science departments restrict the use of scalpels unless specifically requested by a teacher, and prefer to only issue scissors, probes and forceps to students for dissections.
- Ensure students demonstrate responsible behaviour while using scalpels and other dissecting instruments.
- Scalpels should be provided in and returned to a lined container, blade end down
- Students should not walk around the lab with the dissecting instruments, in particular with a scalpel or pointed scissors, forceps or probes.

- To reduce the possibility of stab wounds or cuts from slippage always point sharp instruments such as scalpels and scissors away from yourself and others.
- Hold the instruments so that any sharp points or exposed sharp edges point down onto the dissection board or tray. If there is any slippage when using the instrument, the point/exposed edge will be absorbed by the board/foam or wax tray.

Scalpel blades:

- Only staff should carefully attach and remove scalpel blades using pliers, forceps or a commercial blade remover.
- The scalpel blade size and handle must be compatible e.g. number 4 handle and number 23 blades.
- Keep the blade in the foil wrapper and attach to the handle with the sharp side of the blade pointing away from the body.
- An alternative is to use disposable scalpels.

4. Regulations, licences and permits

Offal that has passed a health inspection by a meat inspector or produced from a butcher's shop, abattoir or biological supplier is suitable for dissection. In some jurisdictions all dissections need to be reported to the school animal ethics committee.

5. Equipment

- PPE – Lab coat/apron (it is recommended to use plastic disposable aprons), safety glasses and gloves.
- Scalpels (optional subject to a site specific risk assessment)
- Scissors, forceps, probes
- Dissecting board covered in newspaper or disposable foam tray
- Newspaper to protect bench and for wrapping biological materials after dissection
- Paper towel
- Disinfectant—hospital grade general-purpose disinfectant (the label on the front of the pack must state 'hospital grade', which is a general purpose hard-surface disinfectant which will kill micro-organisms).
- 70% v/v ethanol
- Optional: model of human brain

6. Operating procedure

Preparation:

- If any blood is associated with the brains rinse them in cold running water.
- Prepare disinfectant solution according to manufacturer's instructions. Place disinfectant in a container ready for instruments to be placed at the end of the dissection.
- Ensure students have appropriate PPE.
- Distribute the instruments to students. Scalpels, scissors, forceps and probes should be counted out, and counted in when returned.

(Operating procedure cont.)

Examining and dissecting the brain:

1. Examine the outside of the brain by carefully placing the brain on the dissecting board flat side down so the white spinal cord at one end rests on the board.
2. If still attached, observe the dura mater which is the outer layer of the meninges membrane covering the brain. See figure 1.
3. Use forceps to gently peel away and remove the layers of the membrane. Identify the two hemispheres of the brain, the spinal cord, the cerebellum and the cerebrum. See figure 2.
4. Turn the brain over and using forceps gently peel away and remove any remaining membrane. Identify the medulla and pons. See figure 3.
5. Try to identify the olfactory bulb, which lies below the frontal lobe of the cerebrum and the optic chiasma, the x shaped-structure formed by the crossover of the right and left optic nerves. Note the optic nerves have been removed, but portions of the optic chiasma are visible. See figure 3.
6. Turn the brain back over and observe the surface of the cerebrum, notice the folds - the grooves are known as sulci and the ridges are called gyri. Identify the medial longitudinal fissure, which separates the right and left hemispheres of the cerebral cortex. See Figure 4.
7. Try to locate the 4 lobes of the cerebrum; the frontal lobe which controls motor functions, the parietal lobe which receives and processes sensory information, the temporal lobe which is located in the region near the ears which receives and processes sounds and smells and the occipital lobe at the back of the brain which is responsible for vision. See Figure 5.
8. Locate the cerebellum, which is just below the occipital lobe of the cerebrum. The cerebellum has an outer cortex, which is folded and it is incompletely divided at the top by a central ridge called the vermis. The cerebellum controls muscle coordination.
9. Use a scalpel to carefully slice through the brain along the centre line (longitudinal fissure), starting at the cerebrum, and down through the cerebellum and brain stem. Separate the two hemispheres of the brain. See figure 6.
10. With the cut side facing up try to locate the following: the corpus callosum, third and fourth ventricles, thalamus, hypothalamus, pineal body, pituitary gland, pons and medulla. See figure 7.
11. Observe the cut surface of the cerebellum and identify the white matter of the cerebellum forming a branched treelike pattern called the arbor vitae as shown in figure 7.
12. Cut one of the hemispheres in half and identify the inner white matter and the outer grey matter. See figure 8.

(Operating procedure cont.)

Clean up:

- Make sure all instruments are returned.
- All parts of the brain, as well as the disposable foam tray (if used), must be wrapped in newspaper and placed in a dedicated plastic garbage bag along with gloves and disposable aprons (if used). When all waste material is collected, double bag for disposal. Freeze material if unable to dispose of immediately.
- If blood is present on dissecting boards, scissors, forceps, probes, dissecting pins and scalpels they be immediately soaked in disinfectant. Otherwise wash equipment in hot soapy water and rinse or place in a dishwasher to minimize handling.
- After washing, dissecting instruments can be soaked in 70% v/v ethanol for 20 minutes as an optional additional disinfectant and to avoid rusting
- Dry all equipment thoroughly.
- Disinfect workplace and wash hands thoroughly.

7. Trouble shooting/emergencies

- **If fainting occurs:** If students start to feel faint, dizzy or nauseous during the dissection lie them down (if possible) and elevate their feet. They can get up slowly after ten minutes. Sending them outside for some fresh air can also help. If they don't recover quickly, always seek urgent medical attention. '***Do not sit the patient on a chair with head between knees***[ii]
- First Aid: See latest SDS of any chemicals used for more detailed information.
    - **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention
    - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek medical attention.
    - **If on skin/clothes:** If spilt on skin or clothes quickly wipe off with a dry cloth to absorb as much liquid as possible. Remove contaminated clothes and drench the area with excess water under a safety shower. Seek medical attention.
    - **If inhaled:** Remove to fresh air and seek medical attention if symptoms persist.
    - For further advice contact the Poisons Information Centre on 131126.
- First aid: cuts and lacerations should be washed under running water, then patted dry and covered with a clean paper towel or tissue in the first instance and referred to the school first aid officer for assessment.
- Any health concerns should be referred to the school first aid officer for assessment, accompanied by the relevant latest SDS if applicable. Follow your school's accident and incident policy and reporting procedures.
- See safety notes if it is necessary to remove broken or used scalpel blades.

8. Waste disposal

- Used and damaged scalpel blades must be placed in an approved sharps container after use.
- Biological material must be wrapped in newspaper, placed in a double plastic garbage bag and sealed for immediate disposal in the industrial bins.

9. Related material

- Risk Assessment.
- Manufacturer's Safety Data Sheet for disinfectant
- Manufacturer's Safety Data Sheet for preserved specimens

References:

[i] 'Fainting', Better Health Channel website, State Government of Victoria, https://www.betterhealth.vic.gov.au/health/conditionsandtreatments/fainting (August 2014)

[ii] St John Ambulance Australia. 2011. *Australian First Aid. Barton, ACT*

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Figures

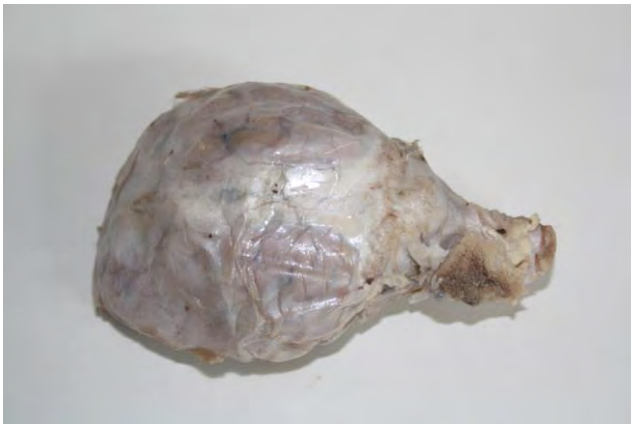

Figure 1: The connective tissue layer of the dura mater covering the sheep brain. (Image by K Szalai)

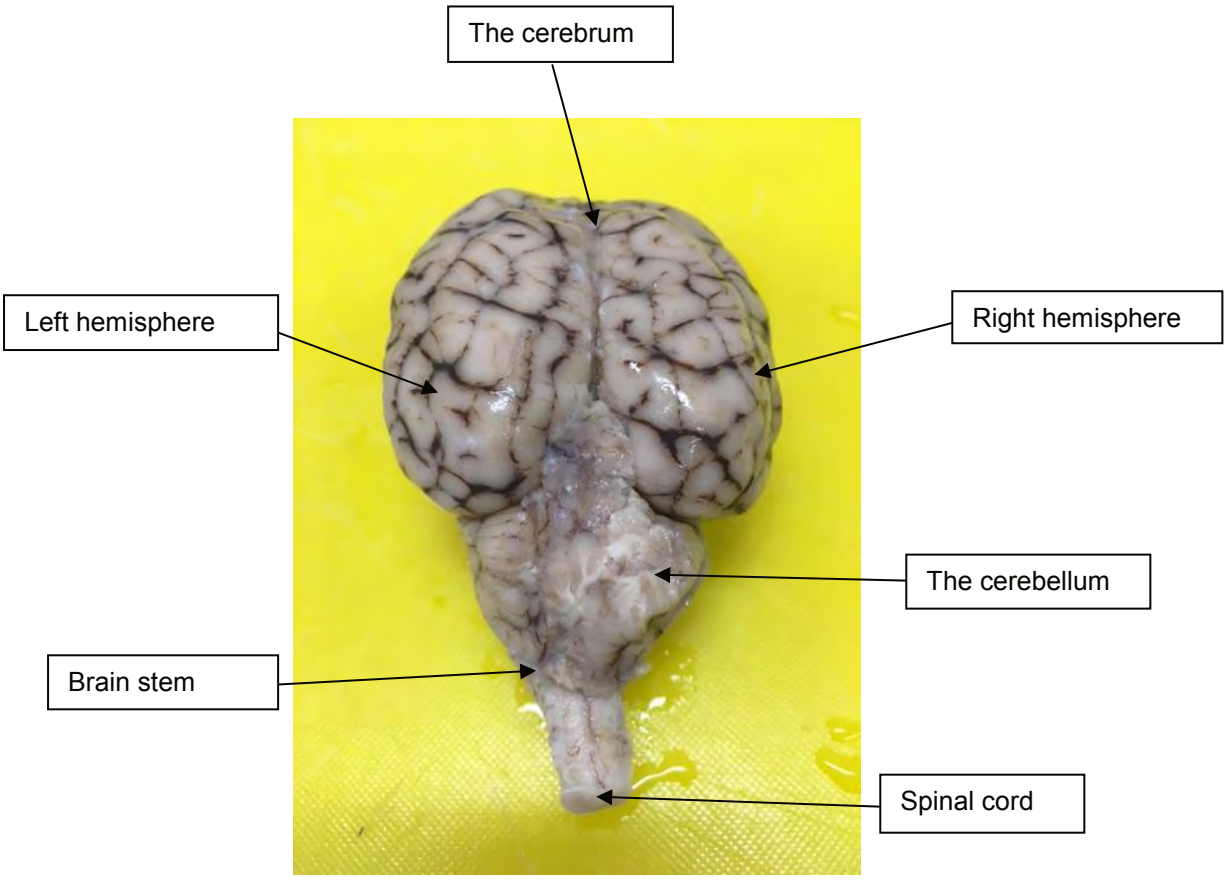

Figure 2: Dura mater removed showing the external structures of the sheep brain.
(Image by K. Szalai, 2017)

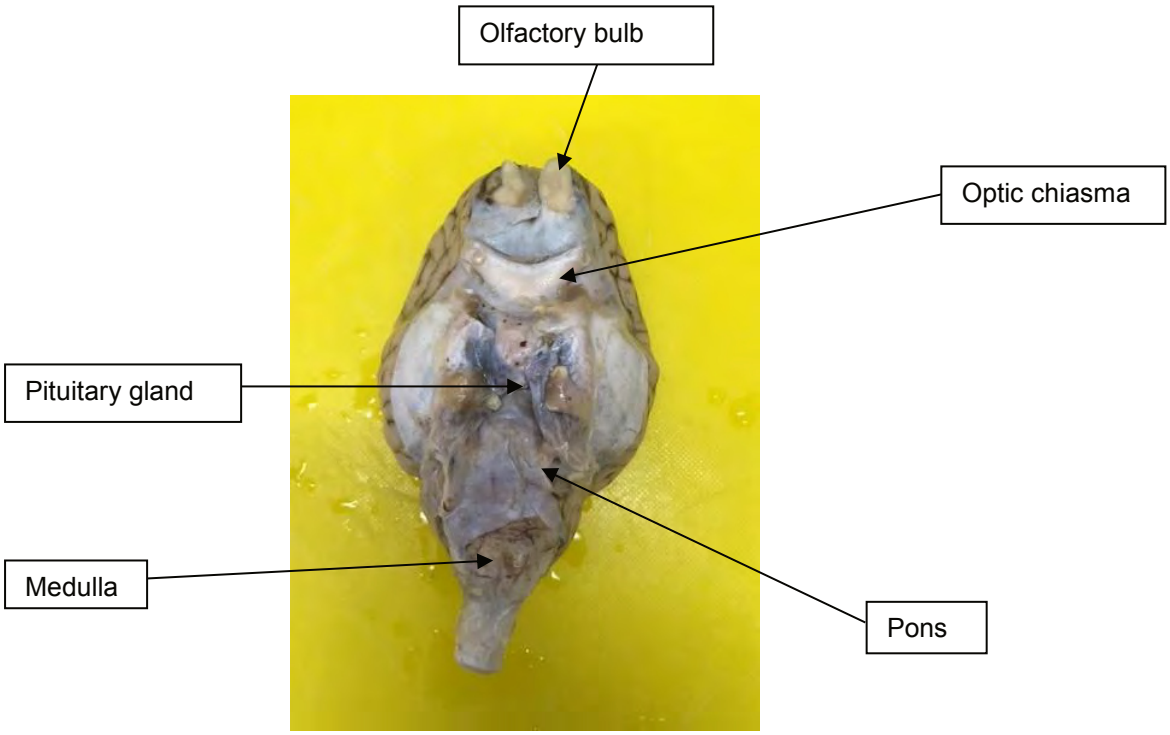

Figure 3: Underside of sheep brain showing external structures. (Image by K. Szalai 2017)

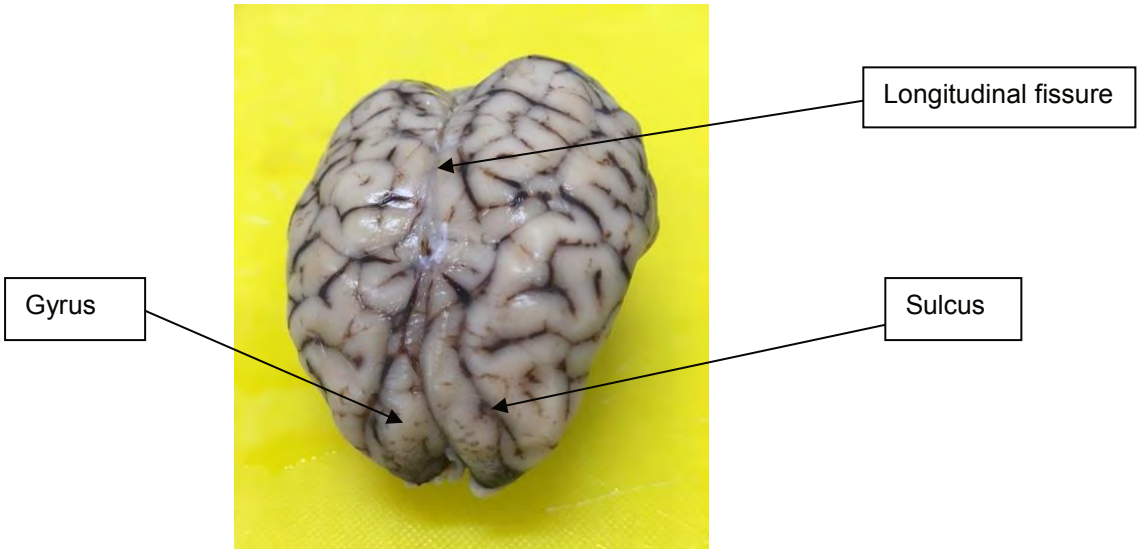

Figure 4: Shows; the longitudinal fissure separating the two hemispheres of the brain, grooves and ridges on the surface of the cerebrum. (Image by K. Szalai 2017)

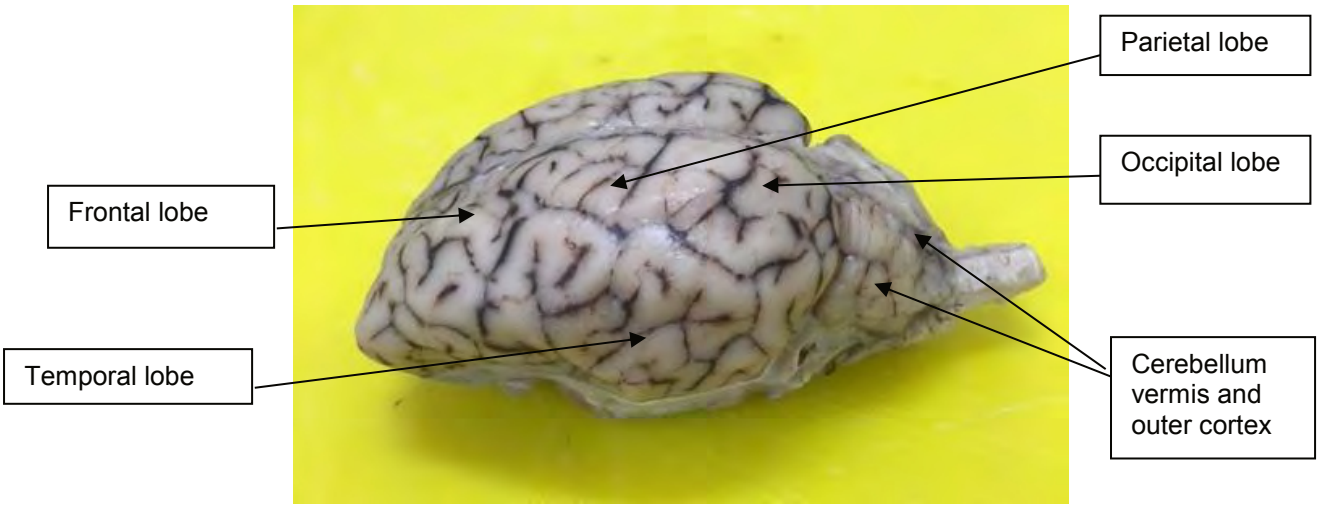

Figure 5: The external structure of the sheep brain showing the four lobes of the cerebrum. (Image by K. Szalai 2017)

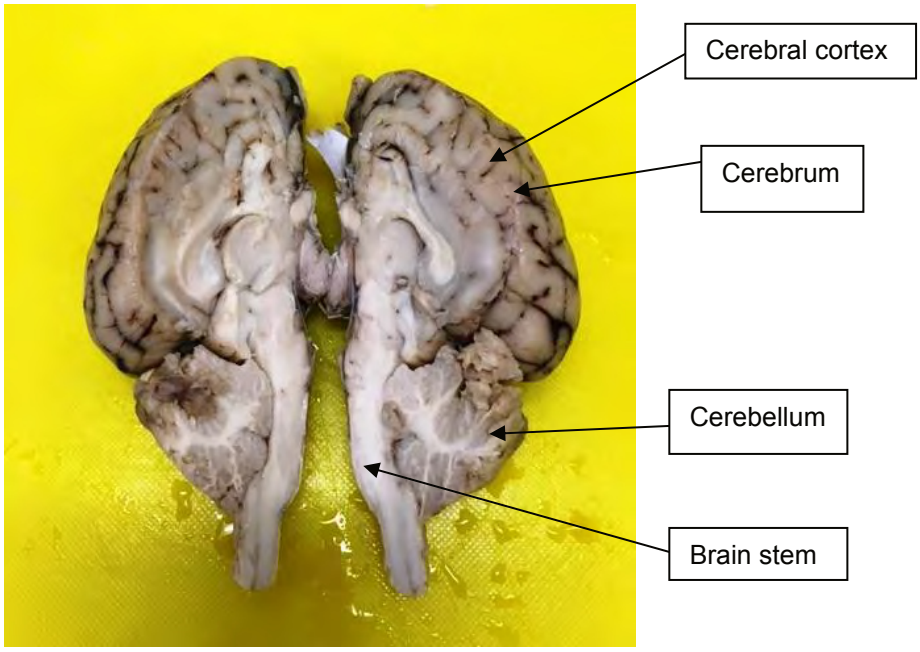

Figure 6: The two hemispheres of the sheep brain separated, dividing the cerebrum, cerebellum and brain stem into two longitudinal halves. (Image by K. Szalai 2017)

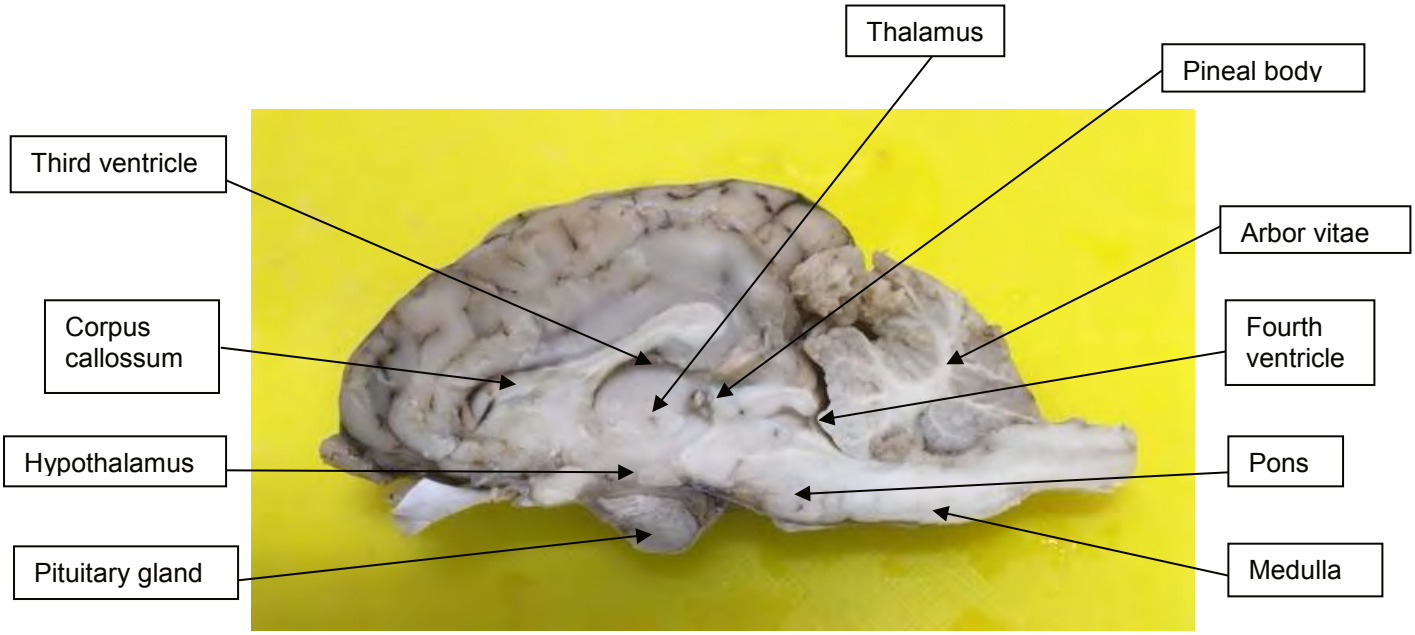

Figure 7: Sheep brain section showing the internal structures.
(Image by K. Szalai 2017)

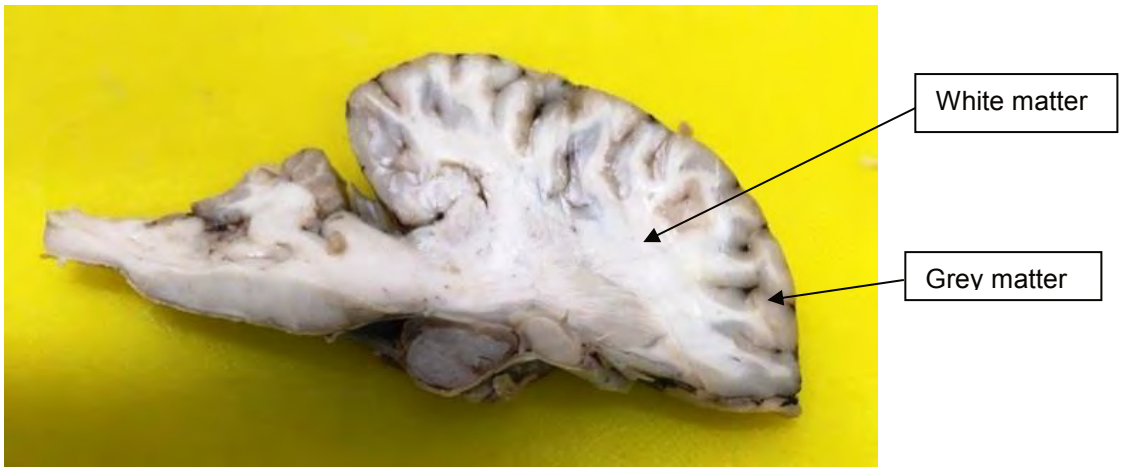

Figure 8: Cross section through the hemisphere of the sheep brain showing the inner white matter and the outer grey matter. (Image by K. Szalai 2017)

Glossary

Arbor vitae – the white matter of the cerebellum

Cerebellum – the part of the brain that controls balance and muscle coordination

Cerebrum – is the largest portion of the mammalian brain, divided into two symmetrical halves the cerebral hemispheres.

Corpus callosum – the large band of nervous tissue that connects the two cerebral hemispheres

Cortex – outer portion of the cerebrum

Dura mater – the tough outermost membrane of the three meninges that cover the brain

Frontal lobe – is the largest of the four major lobes of the cerebral cortex in the mammalian brain. It is located at the front of each cerebral hemisphere and controls motor functions.

Grey matter – the brownish-grey nerve tissue consisting mainly of nerve cell bodies within the brain and spinal cord.

Gyri – the folds of the cerebral cortex

Hemisphere – the brain is divided into left and right hemispheres. Each hemisphere provides a different set of functions

Hypothalamus – central area on the underside of the brain, controlling involuntary functions such as body temperature and the release of hormones

Medulla – part of the brain stem that controls autonomic functions such as breathing, digestion, and heart rate.

Mid brain – the short part of the brainstem just above the pons; it contains the nerve pathways between the cerebral hemispheres and the medulla. The centre for visual reflexes, such as moving the head and eyes

Occipital lobe - is one of the four major lobes of the cerebral cortex at the back of the brain and is the visual processing centre of the mammalian brain.

Olfactory bulb – the structure located in the forebrain of vertebrates that receives neural input about odours detected by cells in the nasal cavity. The axons of olfactory receptor smell cells extend directly into the highly organized olfactory bulb, where information about odours is processed.

Optic chiasma – the crossing point of the optic nerves

Parietal lobe – is one of the four lobes of the cerebral cortex in mammalian brain and processes sensory information; taste, temperature, pain and touch

Pineal body – endocrine gland located in the roof of the third ventricle that secretes the hormone melatonin into the bloodstream.

Pituitary gland – a small oval gland at the base of the brain in vertebrates, producing hormones that control other glands and influence growth of the bone structure, sexual maturing, and general metabolism

Pons – a whitish band of nerve fibres on the surface of the brain stem between the medulla oblongata and midbrain

Sulci – shallow grooves or depressions, separating the folded surface of the cerebral cortex of the brain

Thalamus – a small structure within the brain located just above the brain stem between the cerebral cortex and the midbrain and serves as a sensory relay centre.

Temporal lobe - is one of the four major lobes of the cerebral cortex of the mammalian brain and is located in the region near the ears. It is involved in processing sensory information; sounds, smells and memory formation.

Ventricle – one of the four cavities in the brain filled with cerebral spinal fluid

White matter – is the whitish nerve tissue of the brain and spinal cord, consisting mostly of nerve fibres.

STANDARD OPERATING PROCEDURE:

Performing a chicken wing dissection

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

---

1. Introduction

Chicken wing dissections are conducted to explore the structure and function of muscles, bones and joints, which are comparable to that of a human arm, they have many of the same structures due to their shared evolutionary history as vertebrates.

Skeletal muscles are attached to bones, give shape to the body, generate heat, and make movement possible. Skeletal muscles cannot function without the bones of the skeletal system. Muscles pull on the bones in specific ways and with the guidance of ligaments allow joints to flex or extend in a specific direction. The skeletal system is a network of various living tissues, which provide protection for organs and give the human body its structure. It is also the site of blood formation.

Whole chicken wings suitable for dissection can be purchased fresh from most supermarkets and butchers or poultry suppliers that have passed relevant health inspections. The chicken wings can be obtained some weeks beforehand and stored in a freezer.

2. Context

- These instructions are for the use of experienced science teachers and technicians only and students under close supervision.
- When planning a class dissection activity, it is best to discuss beforehand the type of dissection to be undertaken, and warn of the possibility that there may be some blood and odours present during the dissection.
- Demonstrating the dissection to students before they begin is helpful, not only for correct procedure but allows them to adjust to the appearance of the material, and any blood that may be present after the dissection material has been washed.
- Let students know they don't have to participate in the dissection and can be excused from the class. Alternative arrangements can be made for students who don't wish to participate, by giving them worksheets to complete and relocating them to a private study area.

3. Safety notes

- A site-specific risk assessment should take into consideration the maturity of students carrying out the dissection and address risks associated with students using scalpels and other dissection equipment.
- Before the dissection it is recommended the teacher or laboratory technician trial the dissecting instruments (scalpels, scissors and pointed forceps) to establish that they are sufficiently sharp enough and to determine the most appropriate equipment for the task considering the student behaviour.

Fainting: signs and symptoms:

- Fainting may occur during this type of activity. Please read the first aid information in section 7 before conducting the dissection.
- Fainting is caused by a sudden drop in blood pressure. Common causes include heat, pain or distress and the sight of blood.
- The possible symptoms include the following.
    - 'Dizziness
    - Light-headedness
    - A pale face
    - Perspiration
    - Heightened anxiety and restlessness
    - Nausea
    - Collapse
    - Unconsciousness, for a few seconds
    - Full recovery after a few minutes'[i]

Handling specimens:

- If using frozen chicken wings defrost overnight in a refrigerator and use within 24 hours. Consistent with safe food handling procedures, all meat products should be stored below 5°C prior to dissections.
- Good hygiene practices should be observed at all times: Keep hands away from the mouth, nose, eyes and face during and after dissection and wash hands immediately after handling dissection material.

Safety with scalpels and dissecting instruments:

- Store all dissecting instruments securely.
- Care should be taken with sharps such as scalpel blades and scissors. Some school science departments restrict the use of scalpels unless specifically requested by a teacher, and prefer to only issue scissors, probes and forceps to students for dissections.
- Ensure students demonstrate responsible behaviour while using scalpels and other dissecting instruments.
- Scalpels should be provided in and returned to a lined container, blade end down
- Students should not walk around the lab with the dissecting instruments, in particular with a scalpel or pointed scissors, forceps or probes.
- To reduce the possibility of stab wounds or cuts from slippage always point sharp instruments such as scalpels and scissors away from yourself and others.
- Hold the instruments so that any sharp points or exposed sharp edges point down onto the dissection board or tray. If there is any slippage when using the instrument, the point/exposed edge will be absorbed by the board/foam or wax tray.

Scalpel blades:

- Only staff should carefully attach and remove scalpel blades using pliers, forceps or a commercial blade remover.
- The scalpel blade size and handle must be compatible e.g. number 4 handle and number 23 blades.
- Keep the blade in the foil wrapper and attach to the handle with the sharp side of the blade pointing away from the body.
- An alternative is to use disposable scalpels.

4. Regulations, licences and permits

Offal and animal body parts that have passed a health inspection by a meat inspector or obtained from a butcher's shop, abattoir or biological supplier is suitable for dissection. In some jurisdictions, all dissections need to be reported to the school animal ethics committee.

5. Equipment

- PPE – lab coat/apron (it is recommended to use plastic disposable aprons), safety glasses and gloves)
- Scalpels (optional subject to a site specific risk assessment)
- Scissors, forceps, probes
- Dissecting boards covered in newspaper or disposable foam tray
- Newspaper to protect bench and for wrapping biological materials after dissection
- Paper towel
- Disinfectant – hospital grade general-purpose disinfectant (the label on the front of the pack must state 'hospital grade', which is a general purpose hard surface disinfectant which will kill micro-organisms).
- 70% v/v ethanol

6. Operating procedure

Preparation

- If any blood is associated with the chicken wings rinse them in cold running water.
- Prepare disinfectant solution according to manufacturer's instructions. Place disinfectant in a container ready for instruments to be placed at the end of the dissection.
- Ensure students have appropriate PPE.
- Distribute the instruments to students. Scalpels, scissors, forceps and probes should be counted out, and counted in when returned.

(Operating procedure cont.)

Examining and dissecting the chicken wing:

1. Place the chicken wing on the dissecting board or tray. Study the external appearance and structure of the wing. Feel the skin that is covering the bones and look for places where the feathers were attached.
2. Identify the upper wing, the lower wing, and the wing tip. See Figure 1.
3. Feel for the bones through the flesh, the upper wing consists of one long bone called the humerus; the lower wing consists of two bones, the radius and the ulna. The wing tip consists of modified hand bones, the metacarpals and phalanges. The phalanges are fused in birds to allow for the attachment of feathers.
4. Feel for the muscle and tendons, there are two big muscles on the front, and on the back, of the upper wing that bend and straighten, known as the biceps muscle and triceps muscle. Tendons attach these muscles to the bone in the shoulder and the bones in the forearm of the lower wing.
5. Examine the wing at the shoulder joint where it was removed from the body. You should be able to see the slippery shiny white cartilage covering the end of the bone, and the tough, shiny white ligaments that connected the bone to the joint.
6. Carefully cut the skin of the wing down its length using dissecting scissors.
7. Use the forceps to gently peel back the skin without damaging the underlying tissues, starting with the cut end and working down towards the wing tip. Cut and gently scrape the skin free from the muscle underneath using the dissecting scissors. See Figure 2.
8. Look for the fatty tissue on the underside of the skin. The fat is yellow in colour and feels greasy. Notice the capillaries and muscles that are surrounded by connective tissue, which appears as a thin film or membrane.
9. Examine the exposed skeletal muscles of the wing. They appear as pink bundles of fibre. These muscles are attached to the bones, and cause movement of the bones when they contract and relax.
10. Flex the wing and observe what happens when you pull on the triceps muscle and the biceps muscle. Observe how the muscles work in opposing pairs to move bones.
11. Look for the shiny white strands of tissue that attaches the muscles to the bones. These tissues are called tendons. See figure 3a and 3b.
12. Move the wing again and explore how the muscles, tendons, ligaments, and cartilage play roles in the wings movement.
13. Cut the muscle and fat off the wing to expose the bone. Observe how the different bones of the wing work together. See figures 4a and 4b
14. Observe the cartilage that covers the bones where they meet forming the joints, and locate the ligaments binding the joints together. See figure 5
15. Remove the cartilage from the surface of the bone OR try to break one of the bones in the middle, you will see it is hollow and filled with a pinkish red jelly-like material known as marrow. See figure 6.

(Operating procedure cont.)

Clean up:

- Make sure all instruments are returned.
- All parts of the chicken wing as well as the disposable foam tray (if used) must be wrapped in newspaper and placed in a dedicated plastic garbage bag along with gloves and disposable aprons (if used). When all waste material has been collected, double bag for disposal. Freeze material if unable to dispose of immediately.
- If blood is present on dissecting boards, scissors, forceps, probes and scalpels they must be immediately soaked in disinfectant. Otherwise wash equipment in hot soapy water, and rinse or place in a dishwasher to minimise handling.
- After washing, dissecting instruments can be soaked in 70% v/v alcohol for 20 minutes as an optional additional disinfectant and to avoid rusting.
- Dry all equipment thoroughly.
- Disinfect workplace and wash hands thoroughly.

7. Trouble shooting/emergencies

- **If fainting occurs:** If students start to feel faint, dizzy or nauseous during the dissection lie them down (if possible) and elevate their feet. They can get up slowly after ten minutes. Sending them outside for some fresh air can also help. If they don't recover quickly, always seek urgent medical attention. '***Do not sit the patient on a chair with head between knees***[ii]
- First Aid: See latest SDS of any chemicals used for more detailed information.
    - **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention.
    - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek medical attention.
    - **If on skin/clothes:** If spilt on skin or clothes quickly wipe off with a dry cloth to absorb as much liquid as possible. Remove contaminated clothes and drench the area with excess water under a safety shower. Seek medical attention.
    - **If inhaled:** Remove to fresh air and seek medical attention if symptoms persist.
    - For further advice contact the Poisons Information Centre on 131126.
- First aid: cuts and lacerations should be washed under running water, in the first instance and referred to the school first aid officer for assessment.
- Any health concerns should be referred to the school first aid officer for assessment, accompanied by the relevant latest SDS if applicable. Follow your school's accident and incident policy and reporting procedures.
- See safety notes if it is necessary to remove broken or used scalpel blades

8. Waste disposal

- Used and damaged scalpel blades must be placed in an approved sharps container after use.
- Biological material must be wrapped in newspaper, placed in a double plastic garbage bag and sealed for immediate disposal in the industrial bins.

9. Related material

- Risk Assessment.
- Manufacturer's Safety Data Sheet for disinfectant

References:

[i] 'Fainting', Better Health Channel website, State Government of Victoria https://www.betterhealth.vic.gov.au/health/conditionsandtreatments/fainting (August 2014)

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Figures

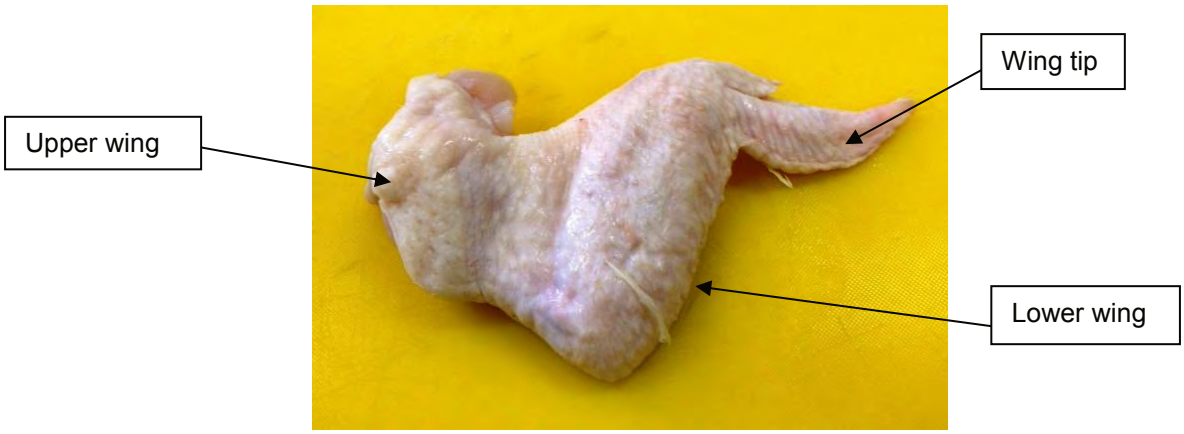

Figure 1: The external structures visible of the chicken wing. (Image by K. Szalai, 2017)

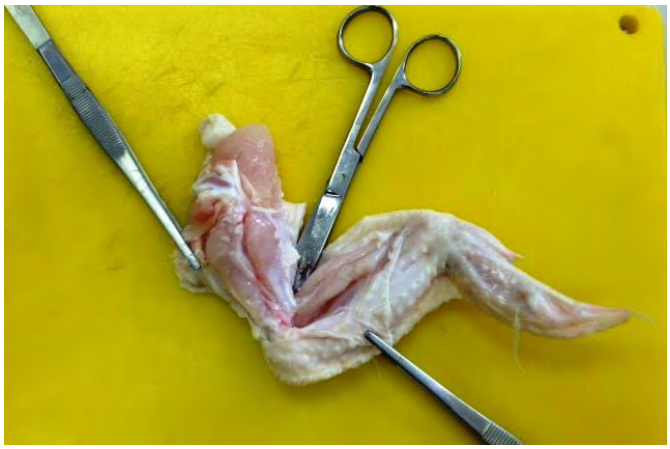

Figure 2: Shows the skin being carefully cut and peeled back from the wing to expose the muscles and underlying tissues. (Image by K. Szalai, 2017)

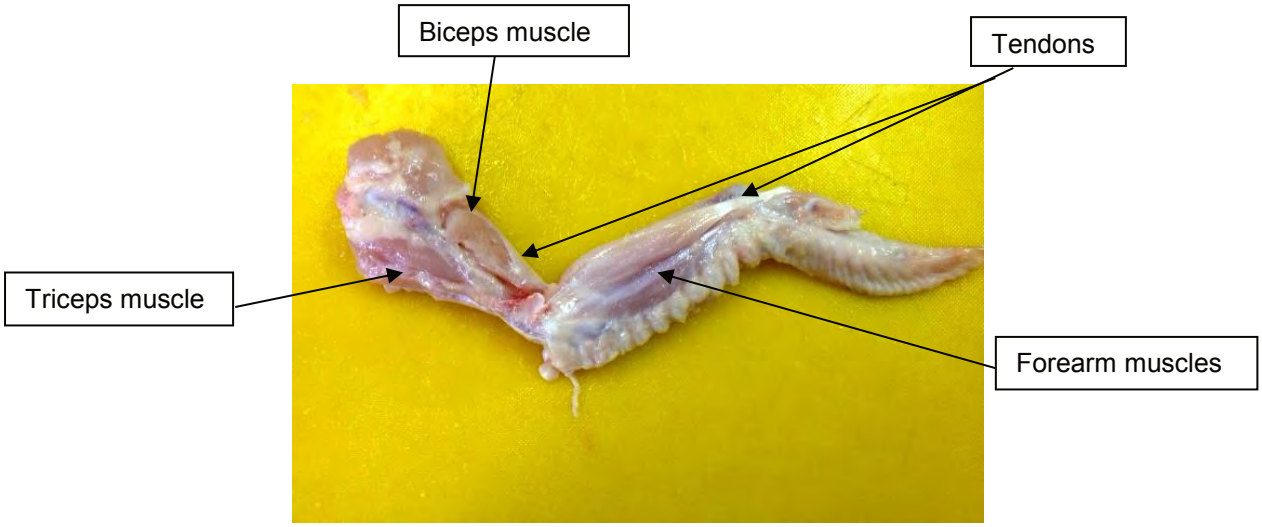

Figure 3a: Shows the muscle groups of the chicken wing. (Image by K. Szalai, 2017)

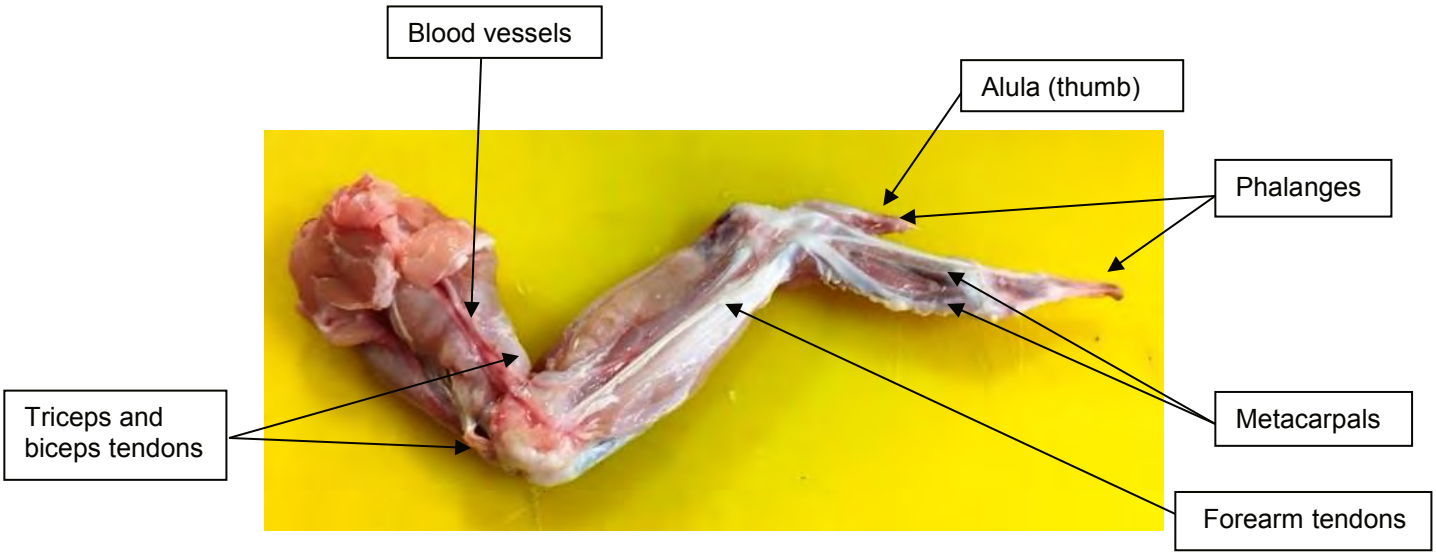

Figure 3b: Shows the tendons connecting the muscle to the bone in several muscle groups, and the rest of the wing tip which composed of modified hand bones. (Image by K. Szalai, 2017)

Figure 4a: The muscles dissected away from the bones showing tendons attached (Image by K. Szalai, 2017)

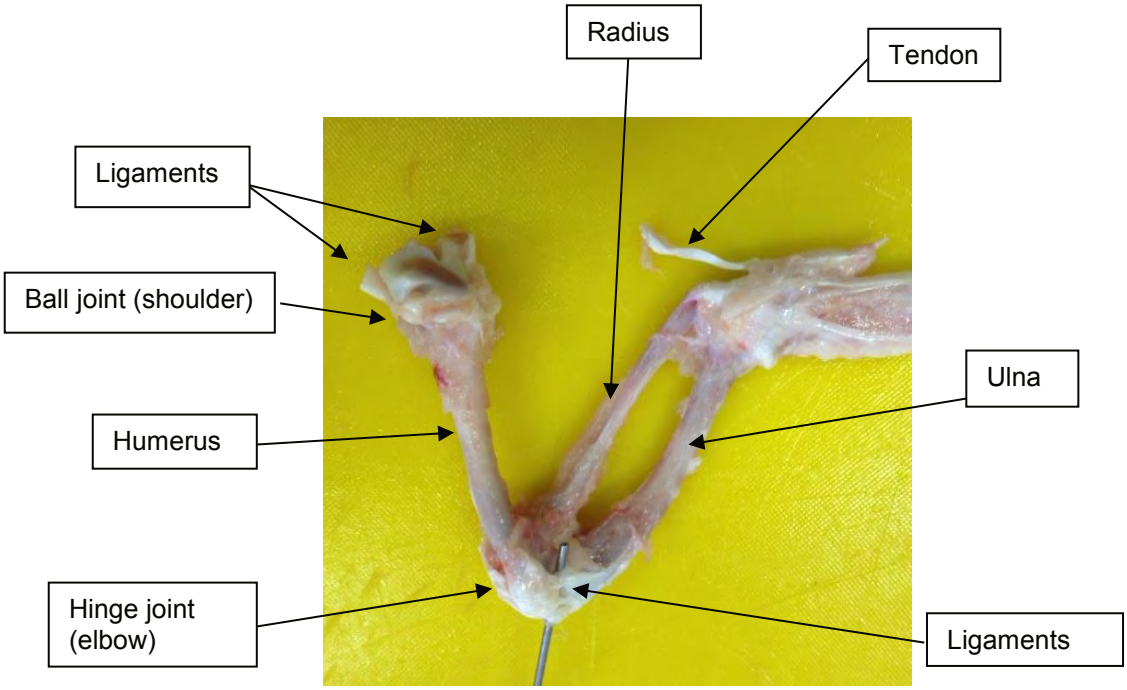

Figure 4b: The muscles removed to expose the bones, joints and ligaments of the wing. (Image by K. Szalai, 2017)

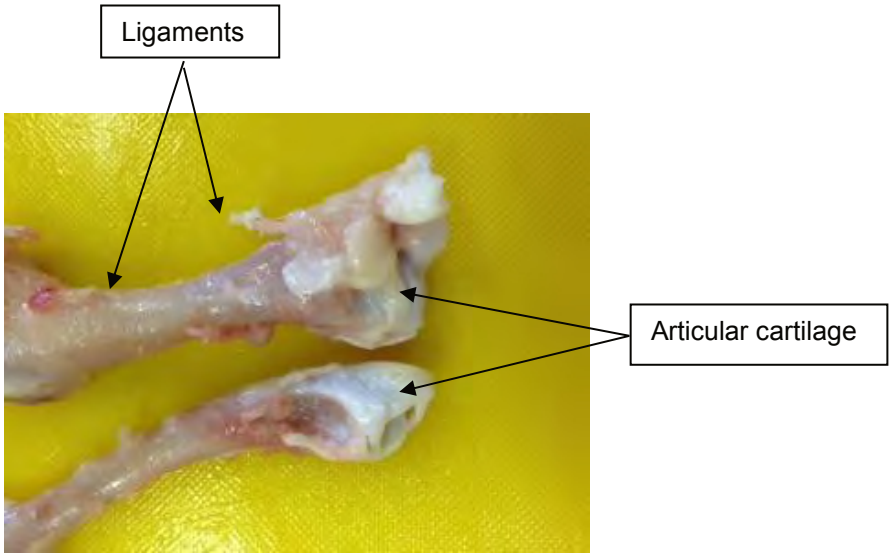

Figure 5: Shows protective cartilage at the surface of the bones forming the elbow joint. (Image by K. Szalai, 2017)

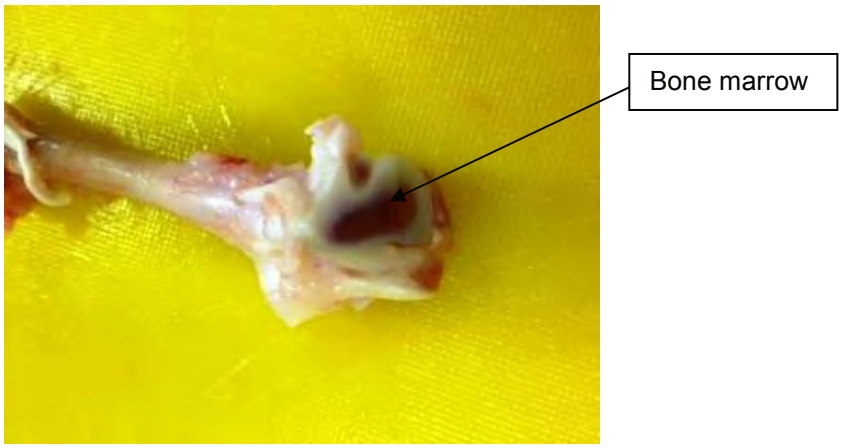

Figure 6: Cartilage removed from the surface of the bone showing pink bone marrow. (Image by K. Szalai, 2017)

Glossary

Biceps muscle – a two-headed muscle which is one of the chief flexors of the forearm that lies on the upper arm between the shoulder and the elbow.

Bone marrow – the soft blood-forming tissue that fills the cavities of large bones containing fat and blood cells

Bones – the main material that forms a vertebrate skeleton, principally collagen fibre and calcium phosphate.

Capillaries – small blood vessels located within the body tissues.

Cartilage – thick, slippery tissue that coats the ends of long bones where they meet to form a joint.

Connective tissue - supports and binds other tissues of the body

Humerus – the long bone of the human upper arm or of a forelimb in other animals

Joints – part of the body where bones are connected

Ligaments – bands of fibrous tissue that hold bones together in a joint.

Metacarpals – bones of the hand can be grouped into three categories: Carpal Bones, Metacarpals and Phalanges. The metacarpals and phalanges of birds are very heavily modified

Muscles – tissue that can undergo repeated contraction and relaxation, so that it is able to produce movement of body parts

Phalanges – bones of the fingers and toes, phalanges of birds are very heavily modified

Radius – one of the two large bones of the forearm, the other being the ulna

Tendons – elastic tissue that attaches muscles to the bones

Triceps muscle – large muscle on the back of the upper limb of many vertebrates responsible for extension of the elbow joint (straightening of the arm).

Ulna – the long bone found in the forearm that stretches from the elbow to the smallest finger or phalanges

Vertebrates – an animal that has a backbone or spinal column.

STANDARD OPERATING PROCEDURE:

Performing a heart dissection

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

Heart dissections are conducted to explore the function of a heart by examining the internal and external structures. Good hygiene practices should be observed at all times. Hearts suitable for dissections include fresh sheep, cow, and ox or pig hearts purchased from a butcher, abattoir or a supplier that has passed relevant health inspections. It is best to ask for as great a length of blood vessels as possible to be left attached and to consider obtaining at least one of them as part of a pluck (heart and lungs) for demonstration purposes.

The hearts can be obtained some weeks beforehand and stored in a freezer.

2. Context

- These instructions are for the use of experienced science teachers and technicians and students under close supervision.
- When planning a class dissection activity it is best to discuss beforehand, the type of dissection to be undertaken, and warn of the possibility that there may be some blood and odours present during the dissection.
- Demonstrating the dissection to students before they begin is helpful, not only for correct procedure but allows the students to adjust to the appearance of the material, and any blood that may be present after the dissection material has been washed.
- Let students know they don't have to participate in the dissection and can be excused from the class. Alternative arrangements can be made for students who don't wish to participate, by giving them worksheets to complete and relocating them to a private study area.

3. Safety notes

- A site specific risk assessment should take into consideration the maturity of students carrying out the dissection and address risks associated with students using scalpels and other dissection equipment
- Before the dissection it is recommended the teacher or laboratory technician trial the dissecting instruments (scalpels, scissors and pointed forceps) to establish that they are sufficiently sharp enough and to determine the most appropriate equipment for the task considering the student behaviour.

Fainting: signs and symptoms:

- Fainting may occur during this type of activity. Please read the first aid information in section 7 before conducting the dissection.

- Fainting is caused by a sudden drop in blood pressure. Common causes include heat, pain or distress and the sight of blood.
- The possible symptoms include the following:
    - Dizziness
    - Light-headedness
    - A pale face
    - Perspiration
    - Heightened anxiety and restlessness
    - Nausea
    - Collapse
    - Unconsciousness, for a few seconds
    - Full recovery after a few minutes [i]

Handling specimens:

- If using frozen hearts defrost overnight in a refrigerator and use within 24 hours. Consistent with safe food handling procedures, all meat products should be stored below 5^0 C prior to dissections.
- Good hygiene practices should be observed at all times.
- Keep hands away from mouth, nose, eyes and face during and after dissection and wash hands immediately after handling dissection material.

Safety with scalpels and dissecting instruments:

- Store all dissecting instruments securely.
- Care should be taken with sharps such as scalpel blades and scissors. Some school science departments restrict the use of scalpels unless specifically requested by a teacher, and prefer to only issue scissors, probes and forceps to students for dissections.
- Ensure students demonstrate responsible behaviour while using scalpels and other dissecting instruments.
- Scalpels should be provided in and returned to a lined container, blade end down
- Students should not walk around the lab with the dissecting instruments, in particular with a scalpel or pointed scissors, forceps or probes.
- To reduce the possibility of stab wounds or cuts from slippage always point sharp instruments such as scalpels and scissors away from yourself and others
- Hold the instruments so that any sharp points or exposed sharp edges point down onto the dissection board or tray. If there is any slippage when using the instrument, the point/exposed edge will be absorbed by the board/foam or wax tray.

Scalpel blades:

- ~~Only staff~~ should carefully attach and remove scalpel blades using pliers, forceps or a commercial blade remover.
- The scalpel blade size and handle must be compatible e.g. number 4 handle and number 23 blades.
- Keep the blade in the foil wrapper and attach to the handle with the sharp side of the blade pointing away from the body.
- An alternative is to use disposable scalpels.

4. Regulations, licences and permits

Offal that has passed a health inspection by a meat inspector or produced from a butchers shop, abattoir or biological supplier is suitable for dissection. In some jurisdictions, all dissections need to be reported to the school animal ethics committee.

5. Equipment

- PPE (Lab coat/apron [it is recommended to use plastic disposable aprons], safety glasses and gloves)
- Scalpels (optional subject to a site specific risk assessment)
- Scissors, Forceps, Probes
- Dissecting board covered in newspaper or disposable foam tray.
- Newspaper to protect bench and for wrapping biological materials after dissection
- Paper towel
- Disinfectant – hospital grade general purpose disinfectant (the label on the front of the pack must state 'hospital grade', which is a general purpose hard surface disinfectant which will kill micro-organisms).
- 70% v/v ethanol

6. Operating procedure

Preparation

- If any blood is associated with the hearts rinse them in cold running water.
- Prepare disinfectant solution according to manufacturer's instructions. Place disinfectant in a container ready for instruments to be placed at the end of the dissection.
- Ensure students have appropriate PPE.
- Distribute the instruments to students. Scalpels, scissors, forceps and probes should be counted out, and counted in when returned.

Examining and dissecting the heart

1. Place the heart on the dissecting board or tray. Examine the outside of the heart, note the coronary arteries (vessels that supply the heart muscle with blood), and identify the left and right sides of the heart. A diagonal furrow on the surface of the heart indicates the divisions between the right and left side.
2. Use your fingers to feel the right side of the heart. Compare the thickness of the right and left sides. The muscles in the wall of the left ventricle feel firm, while those of the right ventricle feel soft and flabby. Feel the muscle wall in the centre of the heart. This is called the septum and separates the left and right side of the heart.
3. Locate the right and left atria on top of the ventricles and compare their thickness. The walls of the atria look quite different from those of the ventricles. Note the fat surrounding the atria.
4. Locate the large blood vessels attached to the atria. The right atrium is connected to the body by the large vein the vena cava and to the lungs by the pulmonary artery. The left atrium is connected to the lungs by the pulmonary vein and to the body by the aorta. Figure 1. Shows photo of sheep heart before dissection

(Operating procedure cont….)

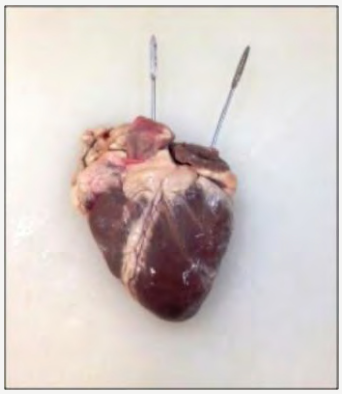

Figure 1. External view of the sheep heart with probes showing the opening to the vena cava and pulmonary vein. (**Image by K. Szalai, 2015)**

5. The main chambers of the heart can be cut open using dissecting scissors. Locate either the vena cava, or if these are missing, the opening of the right atrium and carefully push a dissecting probe through the atrium into the ventricle. Using the probe as a guide insert the rounded end of the dissecting scissors and cut down through the wall of the atrium and right ventricle to the pointed end of the heart. Keeping your cut about 1cm away from the furrow marking the division between the right and left ventricle of the heart.
6. Open out the heart to expose the right atrium and the opening between the atrium and ventricle.
7. Make a similar cut from the pulmonary veins or opening of the left atrium down into the left ventricle. Again make your cut parallel to the furrow on the outside of the heart. Note the ventricles will not fall open until the strong fibrous cords linking the opposing walls are cut. Cut these and open the ventricles.
8. Compare the two sides of the heart. The right side of the heart pumps deoxygenated blood to the lungs. The left side of the heart receives oxygenated blood from the lungs and pumps it to the rest of the body. The walls on the right side are not as thick as the left side because the blood does not have to be pumped as far.
9. Between the ventricles and the atria observe the valves, made up of parachute-like flaps. Each is anchored to the ventricle walls by white tendons. The right atrioventricular valve is a tricuspid valve (it has three cusps). The left atrioventricular valve is a bicuspid valve (it has two cusps). These valves prevent blood flowing back into the atria
10. Locate the openings of the aorta and the pulmonary arteries, high in the ventricles using a probe or your fingers.
11. Identify the valves at these openings. These are called semi-lunar valves and prevent blood from flowing backwards from the arteries. Figure 2. Shows the completed dissection of the sheep heart.

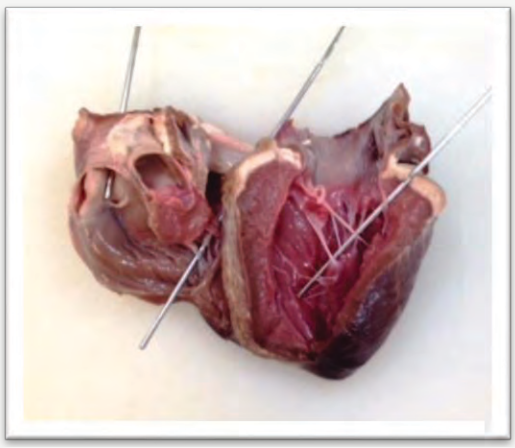

Figure 2. Dissected heart with probes showing the heart valves: The bicuspid in the left ventricle and tricuspid in the right ventricle. The semi-lunar valves can be seen high in the right ventricle in the opening of the pulmonary artery. (**Image by K. Szalai, 2015)**

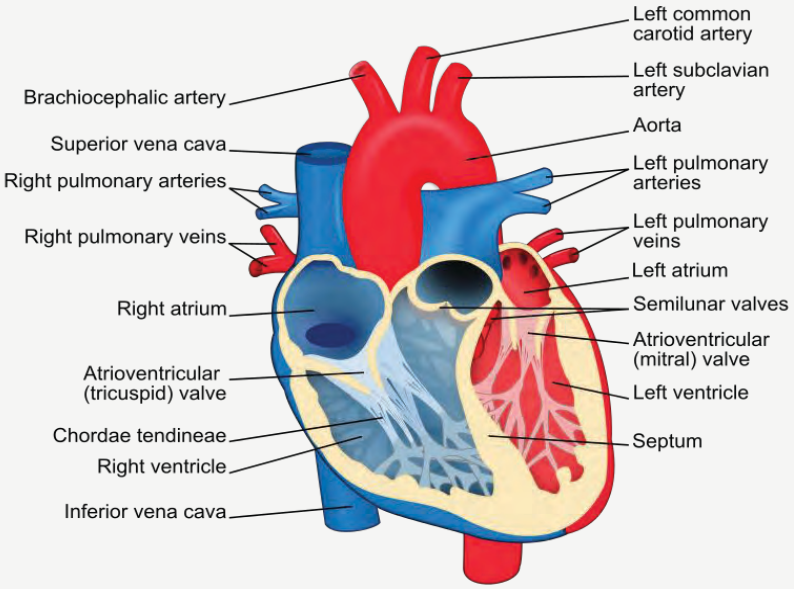

Figure 3. Labelled heart.
Image by Zoofari, Heart diagram, CC BY SA 3.0 https://commons.wikimedia.org/wiki/File:Heart_diagram-en.svg
By ZooFari [CC BY-SA 3.0

Clean up

- Make sure all instruments are returned.
- All parts of the heart as well as the disposable foam tray (if used) must be wrapped in newspaper and placed in a dedicated plastic garbage bag along with gloves and disposable aprons (if used). When all waste material has been collected, double bag for disposal. Freeze material if unable to dispose of immediately.
- If blood is present on dissecting boards, scissors, forceps, probes and scalpels they must be immediately soaked in disinfectant. Otherwise wash equipment in hot soapy water, and rinse or place in a dishwasher to minimise handling.
- After washing, dissecting instruments can be soaked in 70% v/v alcohol for 20 minutes as an optional additional disinfectant and to avoid rusting.
- Dry all equipment thoroughly.
- Disinfect workplace and wash hands thoroughly.

7. Trouble shooting/emergencies

- **If fainting occurs:** If students start to feel faint, dizzy or nauseous during the dissection lie them down (if possible) and elevate their feet. They can get up slowly after ten minutes. Sending them outside for some fresh air can also help. If they don't recover quickly, always seek urgent medical attention. '***Do not sit the patient on a chair with head between knees***'[ii]
- First aid: See latest SDS of any chemicals used for more detailed information
  - **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention.
  - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek medical attention.

- **If on skin/clothes:** If spilt on skin or clothes quickly wipe off with a dry cloth to absorb as much liquid as possible. Remove contaminated clothes and drench the area with excess water under a safety shower. Seek medical attention.
- **If inhaled:** Remove to fresh air and seek medical attention if symptoms persist.
- For further advice contact the Poisons Information Centre on 131 126.

- First aid: cuts and lacerations should be washed under running water, in the first instance and referred to the school first aid officer for assessment.
- Any health concerns should be referred to the school first aid officer for assessment, accompanied by the relevant latest SDS if applicable. Follow your school's accident and incident policy and reporting procedures.
- See safety notes if it is necessary to remove broken or used scalpel blades

8. Waste disposal

- Used and damaged scalpel blades must be placed in an approved sharps container after use.
- Biological material must be wrapped in newspaper, placed in a double plastic garbage bag and sealed for immediate disposal in the industrial bins.

9. Related material

- Risk Assessment
- Manufacturer's Safety Data Sheet for disinfectant
- More detailed information on a heart dissection can be found on the following website: Nuffield Foundation. 'Looking at the heart' http://www.nuffieldfoundation.org/practical-biology/looking-heart (December 2008)

References:

[i] 'Fainting', Better Health Channel website, State Government of Victoria: http://www.betterhealth.vic.gov.au/bhcv2/bhcarticles.nsf/pages/Fainting (August 2014)

[ii]St John Ambulance Australia. 2011. *Australian First Aid. Barton, ACT*

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Dissection Safety Policy and Procedures' Flinn Scientific website' http://www.flinnsci.com/media/948812/sf10490.pdf (2013)

Glossary

Aorta – main artery that carries blood from the left ventricle of the heart to all the branch arteries in the body except those in the lungs.

Atria – (plural of atrium) are the blood collection chambers of the heart.

Atrioventricular (tricuspid) valve – or right atrioventricular valve, is on the right dorsal side of the mammalian heart, between the right atrium and the right ventricle.

Atrioventricular (mitral) valve – or left atrioventricular valve, is a dual-flap valve in the heart that lies between the left atrium and the left ventricle.

Atrium – is one of the two blood collection chambers of the heart.

Bicuspid – is an aortic valve that only has two leaflets, instead of three.

Blood – is living tissue made up of liquid and solids (cells). The liquid part, called plasma, is made of water, salts, and protein. It performs two major functions: transport through the body of oxygen and carbon dioxide; food molecules (glucose, lipids and amino acids). The cell component includes red blood cells, white blood cells and platelets.

Brachiocephalic artery – artery that supplies oxygenated blood from the aorta to the head, neck and arm regions of the body.

Carotid artery – the large artery on each side of the neck that supplies blood to the head.

Chordae tendineae – or heart strings, are cord-like tendons that connect the papillary muscles to the tricuspid valve and the mitral valve in the heart.

Coronary arteries – vessels that supply the heart muscle with blood.

Deoxygenated blood – blood that is rich in carbon dioxide rather than oxygen.

Pulmonary artery – artery carrying deoxygenated blood from the right side of the heart to the lungs.

Pulmonary vein – one of the four veins that carry oxygen-rich blood from the lungs to the left side of the heart.

Semi-lunar valves – heart valves with cusps shaped like half-moons; preventing blood flowing from the aorta back into the heart.

Septum – the muscle wall in the centre of the heart.

Subclavian artery – the major arteries of the upper thorax (chest).

Tendons – a tough band of fibrous connective tissue.

Tricuspid – a valve having three leaflets preventing back flow of blood from the right ventricle into the right atrium.

Vena cava (superior) – the superior of the two venae cavae that return deoxygenated blood from the upper part of the body above the diaphragm to the right atrium of the heart.

Vena cava (inferior) – is a large vein that carries deoxygenated blood from the lower and middle body into the right atrium of the heart.

Ventricle – a ventricle is one of two large chambers of the heart that collect and expel blood received from an atrium pumping it into the arteries.

History of reviews

Date	Version Number	Notes
April 2015	Version 1.0	
Sept 2016	Version 2.0	Additional safety information included • Fainting information • Safe food handling procedures • the use of scalpels and dissecting instruments Glossary included

STANDARD OPERATING PROCEDURE:

Performing a kidney dissection

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site specific risk assessment.

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1. Introduction

Kidney dissections are conducted to explore the function of a kidney by examining the external and internal structures. The kidney filters out metabolic wastes such as urea and some salts and regulates the water and salt balance in the body. Kidneys suitable for dissection include fresh sheep or bullock kidneys purchased from a butcher, abattoir or a supplier that has passed relevant health inspections. It is best to get them left in fat if possible with some of the ureter and blood vessels left attached. The kidneys can be obtained some weeks beforehand and stored in a freezer.

2. Context

- These instructions are for the use of experienced science teachers and technicians and students under close supervision.
- When planning a class dissection activity it is best to discuss beforehand, the type of dissection to be undertaken, and warn of the possibility that there may be some blood and odours present during the dissection.
- Demonstrating the dissection to students before they begin is helpful, not only for correct procedure but allows the students to adjust to the appearance of the material, and any blood that may be present after the dissection material has been washed.
- Let students know they don't have to participate in the dissection and can be excused from the class. Alternative arrangements can be made for students who don't wish to participate, by giving them worksheets to complete and relocating them to a private study area

3. Safety notes

- A site-specific risk assessment should take into consideration the maturity of students carrying out the dissection and address risks associated with students using scalpels and other dissection equipment.
- Before the dissection it is recommended the teacher or laboratory technician trial the dissecting instruments (scalpels, scissors and pointed forceps) to establish that they are sufficiently sharp enough and to determine the most appropriate equipment for the task considering the student behaviour.

Fainting: signs and symptoms:

- Fainting may occur during this type of activity. Please read the first aid information section 7 before conducting the dissection.

in

- Fainting is caused by a sudden drop in blood pressure. Common causes include heat, pain or distress and the sight of blood.
- The possible symptoms include the following.
  - 'Dizziness
  - Light-headedness
  - A pale face
  - Perspiration
  - Heightened anxiety and restlessness
  - Nausea
  - Collapse
  - Unconsciousness, for a few seconds
  - Full recovery after a few minutes'[i]

Handling specimens:

- If using frozen kidneys defrost overnight in a refrigerator and use within 24 hours. Consistent with safe food handling procedures, all meat products should be stored below 5^0 C prior to dissections.
- Good hygiene practices should be observed at all times.
- Keep hands away from mouth, nose, eyes and face during and after dissection and wash hands immediately after handling dissection material.

Safety with scalpels and dissecting instruments:

- Store all dissecting instruments securely.
- Care should be taken with sharps such as scalpel blades and scissors. Some school science departments restrict the use of scalpels unless specifically requested by a teacher, and prefer to only issue scissors, probes and forceps to students for dissections.
- Ensure students demonstrate responsible behaviour while using scalpels and other dissecting instruments.
- Scalpels should be provided in and returned to a lined container, blade end down
- Students should not walk around the lab with the dissecting instruments, in particular with a scalpel or pointed scissors, forceps or probes.
- To reduce the possibility of stab wounds or cuts from slippage always point sharp instruments such as scalpels and scissors away from yourself and others
- Hold the instruments so that any sharp points or exposed sharp edges point down onto the dissection board or tray. If there is any slippage when using the instrument, the point/exposed edge will be absorbed by the board/foam or wax tray.

Scalpel blades:

- Only staff should carefully attach and remove scalpel blades using pliers, forceps or a commercial blade remover.
- The scalpel blade size and handle must be compatible e.g. number 4 handle and number 23 blades.
- Keep the blade in the foil wrapper and attach to the handle with the sharp side of the blade pointing away from the body.
- An alternative is to use disposable scalpels.

4. Regulations, licences and permits

Offal that has passed a health inspection by a meat inspector or obtained from a butcher's shop, abattoir or biological supplier is suitable for dissection. In some jurisdictions, all dissections need to be reported to the school animal ethics committee.

5. Equipment

- PPE (Lab coat/apron [it is recommended to use plastic disposable aprons], safety glasses and gloves)
- Scalpels (optional subject to a site specific risk assessment)
- Point-end scissors, Forceps, Probes
- Dissecting boards covered in newspaper or disposable foam tray
- Newspaper to protect bench and for wrapping biological materials after dissection
- Paper towel
- Disinfectant – hospital grade general purpose disinfectant (the label on the front of the pack must state 'hospital grade', which is a general purpose hard surface disinfectant which will kill micro-organisms).
- 70% v/v ethanol

6. Operating procedure

Preparation

- If any blood is associated with the kidneys rinse them in cold running water.
- Prepare disinfectant solution according to manufacturer's instructions. Place disinfectant in a container ready for instruments to be placed at the end of the dissection.
- Ensure students have appropriate PPE.
- Distribute the instruments to students. Scalpels, scissors, forceps and probes should be counted out, and counted in when returned.

Dissecting the kidney

1. Place the kidney on the dissecting board or tray. Remove any protective fatty tissue surrounding the kidney using dissecting scissors taking care not to damage any of the tubes emerging from the kidney.
2. Examine the external structure of the kidney.
3. Gently separate, as much as possible, the three tubes emerging from the kidney's concave side (these may have been cut from the kidney).

Figure 1 shows a photo of sheep kidney before dissection.

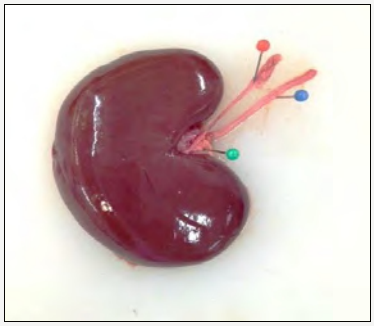

Figure 1. External view of a sheep kidney. The red pin identifies the renal artery and the blue pin shows the renal vein. Below the green pin identifies the ureter which has been cut close to the kidney. (**Image by K. Szalai, 2015**)

Operating procedure cont…..

4. Try to identify the:
   - renal artery, a narrow tube with thick elastic walls which takes blood into the kidney
   - renal vein, a wide tube with limp walls which carries blood out of the kidney
   - ureter, a tough white tube which carries urine from the kidney to the bladder.
5. Cut the kidney in half lengthways using sharp pointed dissecting scissors or scalpel, starting from the concave side just to one side of the centre line. Leave the tubes intact in one side of the dissection.
6. Observe the internal appearance and structure of the kidney.
7. Identify the cortex, medulla and pelvis of the kidney. Note the colour, thickness and texture of these structures.

Figure 2 shows photo of completed dissection of sheep kidney.

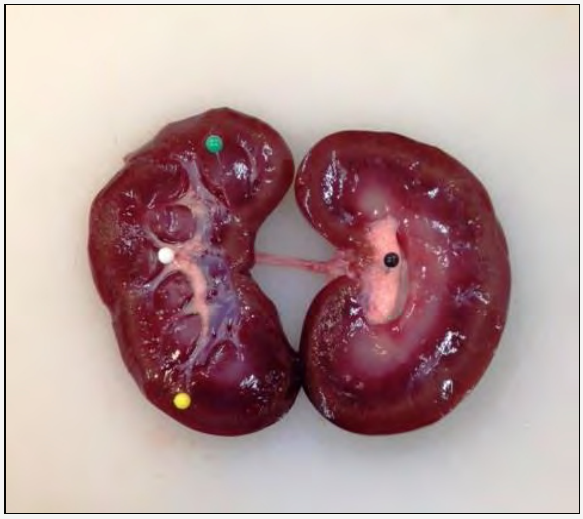

Figure 2. Two halves of the kidney. On the left side the green pin identifies the medulla, the white pin shows the pyramid and the yellow pin identifies the cortex. On the right side the black pin shows the pelvis. (**Image by K. Szalai 2015**)

Figure 3 shows the labelled structure of the kidney.

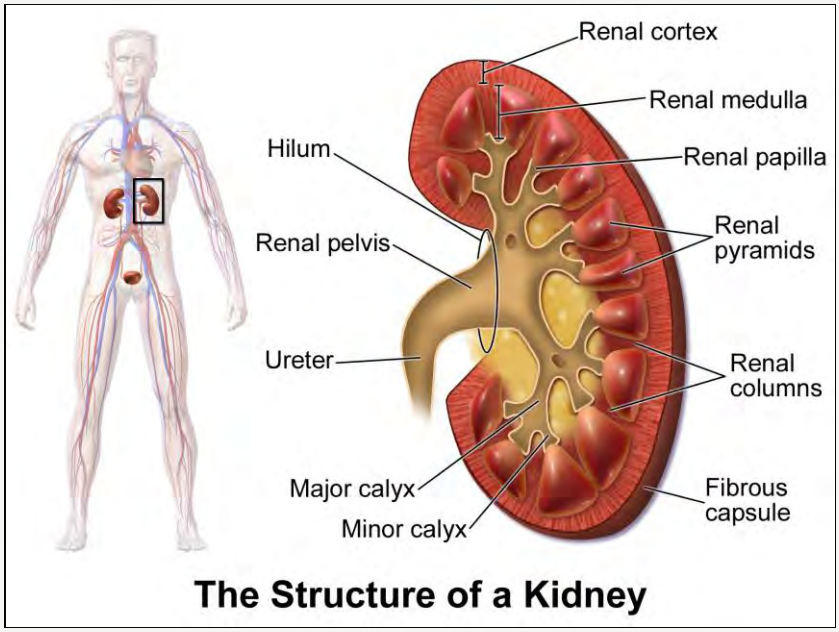

Figure 3. Labelled kidney.

Image by BruceBlaus Kidney Anatomy, CC BY 3.0, http://commons.wikimedia.org/wiki/File:Blausen_0593_KidneyAnatomy_02.png

Clean up

- Make sure all instruments are returned.
- All parts of the kidney as well as the disposable foam tray (if used) must be wrapped in newspaper and placed in a dedicated plastic garbage bag along with gloves and disposable aprons (if used). When all waste material has been collected, double bag for disposal. Freeze material if unable to dispose of immediately.
- If blood is present on dissecting boards, scissors, forceps, probes and scalpels they must be immediately soaked in disinfectant. Otherwise wash equipment in hot soapy water, and rinse or place in a dishwasher to minimise handling.
- After washing, dissecting instruments can be soaked in 70% v/v alcohol for 20 minutes as an optional additional disinfectant and to avoid rusting.
- Dry all equipment thoroughly.
- Disinfect workplace and wash hands thoroughly.

7. Trouble shooting/emergencies

- **If fainting occurs:** If students start to feel faint, dizzy or nauseous during the dissection lie them down (if possible) and elevate their feet. They can get up slowly after ten minutes. Sending them outside for some fresh air can also help. If they don't recover quickly, always seek urgent medical attention. '***Do not sit the patient on a chair with head between knees***'[ii]
- First aid: See latest SDS of any chemicals used for more detailed information
  - **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention.
  - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek medical attention.
  - **If on skin/clothes:** If spilt on skin or clothes quickly wipe off with a dry cloth to absorb as much liquid as possible. Remove contaminated clothes and drench the area with excess water under a safety shower. Seek medical attention.
  - **If inhaled:** Remove to fresh air and seek medical attention if symptoms persist.
  - For further advice contact the Poisons Information Centre on 131 126.
- First aid: cuts and lacerations should be washed under running water, in the first instance and referred to the school first aid officer for assessment.
- Any health concerns should be referred to the school first aid officer for assessment, accompanied by the relevant latest SDS if applicable. Follow your school's accident and incident policy and reporting procedures.
- See safety notes if it is necessary to remove broken or used scalpel blades

8. Waste disposal

- Used and damaged scalpel blades must be placed in an approved sharps container after use.
- Biological material must be wrapped in newspaper, placed in a double plastic garbage bag and sealed for immediate disposal in the industrial bins.

9. Related material

- Risk Assessment
- Manufacturer's Safety Data Sheet for disinfectant

References:

[i] 'Fainting', Better Health Channel website, State Government of Victoria: http://www.betterhealth.vic.gov.au/bhcv2/bhcarticles.nsf/pages/Fainting (August 2014)

[ii] St John Ambulance Australia. 2011. *Australian First Aid. Barton, ACT*

Andrews, C. 2002. Active Science skills and experiments book 2. Oxford University Press

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CLEAPSS. 2014. G268 Dissection: a guide to safe practice. Uxbridge UK http://science.cleapss.org.uk/Resource-Info/G268-Dissection-a-guide-to-safe-practice.aspx (Subscription required.)

'Dissection safety tips', Flinn Scientific website, http://www.flinnsci.com/media/396301/dissectionsafety.pdf (2010)

Dissection Safety Policy and Procedures' Flinn Scientific website' http://www.flinnsci.com/media/948812/sf10490.pdf (2013)

'Kidney', Wikipedia website, https://en.wikipedia.org/wiki/Kidney (Accessed July 2016)

Glossary

Blood – fluid carried by the blood vessels through the body, containing plasma, red and white blood cells, platelets, dissolved gases, nutrients, hormones and salts.

Fibrous capsule – thin collagenous connective tissue envelope that surrounds the outer surface of the kidney.

Hilum – recessed central fissure in the kidney where the vessels, nerves and ureter pass.

Major calyx – chambers of the kidney – two or three minor calyces converge to form a major calyx through which urine passes before continuing through the renal pelvis into the ureter.

Metabolic waste – waste products produced by the body for excretion.

Nephron – tiny structures that produce urine in the process of removing waste substances from the blood.

Renal artery – artery that brings waste-filled blood from the aorta to the kidney for filtering in the nephron.

Renal columns – anchor the renal cortex between the renal pyramids.

Renal cortex – the outer portion of the kidney between the renal capsule and the renal medulla.

Renal medulla – the innermost part of the kidney that is critical for regulating blood pressure.

Renal papilla – part of the kidneys that urine passes through on its way to the renal pelvis and the ureter.

Renal pelvis – area at the centre of the kidney. Urine collects in the renal pelvis and is funnelled into the ureter.

Renal pyramids – located inside the inner kidney. They consist of tubules, which collect urine from the outer kidneys and transfer them to the calyces, which store urine before it passes to the ureter and bladder.

Renal vein – there are two renal veins, a left and a right. They branch off the inferior vena cava and drain oxygen-depleted blood from the kidneys.

Urea – nitrogen-rich waste produced by the body.

Ureter – a tough white tube which carries urine from the kidney to the bladder.

History of reviews

Date	Version Number	Notes
April 2015	Version 1.0	
Sept 2016	Version 2.0	Additional safety information included • Fainting information • Safe food handling procedures • the use of scalpels and dissecting instruments Glossary included

STANDARD OPERATING PROCEDURE:

Performing a lung dissection

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

Lung dissections are conducted to explore and understand the function of the lungs by examining the internal and external structures, as well as the relationship of the lungs to the heart. Good hygiene practices should be observed at all times. Lungs suitable for dissections include fresh sheep, cow, and ox or pig lungs purchased from a butcher, abattoir or a reputable biological supplier that has passed relevant health inspections. It is best to get them as part of a pluck (heart-lung set) as this allows you to better see the connections of the main blood vessels.

A pluck can be obtained some weeks beforehand and stored in a freezer.

2. Context

- These instructions are for the use of experienced science teachers and technicians and students under close supervision.
- When planning a class dissection activity, it is best to discuss beforehand the type of dissection to be undertaken and warn of the possibility that there may be some blood and odours present during the dissection.
- Demonstrating the dissection to students before they begin is helpful, not only for correct procedure but allows the students to adjust to the appearance of the material and any blood that may be present after the dissection material has been washed.
- Let students know they don't have to participate in the dissection and can be excused from the class. Alternative arrangements can be made for students who don't wish to participate, by giving them worksheets to complete and relocating them to a private study area.

3. Safety notes

- A site-specific risk assessment should take into consideration the maturity of students carrying out the dissection and address risks associated with students using scalpels and other dissection equipment.
- Before the dissection it is recommended the teacher or laboratory technician trial the dissecting instruments (scalpels, scissors and pointed forceps) to establish that they are sufficiently sharp enough and to determine the most appropriate equipment for the task considering the student behaviour.

Fainting: signs and symptoms:

- Fainting may occur during this type of activity. Please read the first aid information in section 7 before conducting the dissection.

- Fainting is caused by a sudden drop in blood pressure. Common causes include heat, Pain or distress and the sight of blood.
- The possible symptoms include the following:
    - 'Dizziness
    - Light-headedness
    - A pale face
    - Perspiration
    - Heightened anxiety and restlessness
    - Nausea
    - Collapse
    - Unconsciousness, for a few seconds
    - Full recovery after a few minutes'[1]

Handling specimens:

- If using frozen lungs defrost overnight in a refrigerator and use within 24 hours. Consistent with safe food handling procedures, all meat products should be stored below 5ºC prior to performing any dissections.
- Good hygiene practices should be observed at all times.
- Keep hands away from the mouth, nose, eyes and face during and after dissection and wash hands immediately after handling dissection material.
- For hygiene reasons lungs must not be inflated by mouth. There is a possibility of inhaling aerosols from the lungs as the air is expelled from the lungs. It is best practice to use a pump, such as a bicycle or foot pump, or a small syringe.

Safety with scalpels and dissecting instruments:

- Store all dissecting instruments securely.
- Care should be taken with sharps such as scalpel blades and scissors. Some school science departments restrict the use of scalpels unless specifically requested by a teacher, and prefer to only issue scissors, probes and forceps to students for dissections.
- It is suggested that the teacher and/or laboratory technician use a scalpel to cut through the finer tubes of the bronchi for students. This removes the need for the students to handle scalpels.
- Ensure students demonstrate responsible behaviour while using scalpels and other dissecting instruments.
- Scalpels should be provided in and returned to a lined container, blade end down.
- Students should not walk around the lab with dissecting instruments, in particular with a scalpel or pointed scissors, forceps or probes.
- To reduce the possibility of stab wounds or cuts from slippage always point sharp instruments such as scalpels and scissors away from yourself and others.
- Hold the instruments so that any sharp points or exposed sharp edges point down onto the dissection board or tray. If there is any slippage when using the instrument, the point/exposed edge will be absorbed by the board/foam or wax tray.

Scalpel blades:

- Only staff should carefully attach and remove scalpel blades using pliers, forceps or commercial blade remover.

a

- The scalpel blade size and handle must be compatible, e.g. number 4 handle with number 23 blades.
- Keep the blade in the foil wrapper and attach to the handle with the sharp side of the blade pointing away from the body.
- An alternative is to use disposable scalpels.

4. Regulations, licences and permits

Offal that has passed a health inspection by a meat inspector or produced from a butchers shop, abattoir or biological supplier is suitable for dissection. In some jurisdictions all dissections need to be reported to the school animal ethics committee.

5. Equipment

- PPE (Lab coat/apron [it is recommended to use plastic disposable aprons], safety glasses and gloves
- Scalpels (optional subject to a site specific risk assessment)
- Scissors, forceps, probes
- Dissecting board covered in newspaper or disposable foam tray
- Newspaper to protect bench and for wrapping biological materials after dissection
- Paper towel
- 250 mL glass beaker
- Plastic or rubber tubing to fit cut trachea
- String, for attaching tubing
- Pump (e.g. balloon or bicycle) to inflate the lungs
- 50 mL syringe
- a large, transparent, plastic bag for when inflating the lungs
- Disinfectant—hospital grade general purpose disinfectant (the label on the front of the pack must state ‘hospital grade’, which is a general purpose hard-surface disinfectant which will kill micro-organisms).
- 70% v/v ethanol
- Optional: commercially prepared slides of alveoli and microscopes.

6. Operating procedure

Preparation:

- If any blood is associated with the lungs or pluck rinse them in cold running water.
- Prepare disinfectant solution according to manufacturer’s instructions. Place disinfectant in a container ready for instruments to be placed at the end of the dissection.
- Ensure students have appropriate PPE.
- Distribute the instruments to students. Scalpels, scissors, forceps and probes should be counted out, and counted in when returned.

(Operating procedure cont…)

Examining and dissecting the lungs:

1. If using a pluck (heart lung set) arrange the pluck with heart on top on a dissecting board or tray. If still attached identify the (pleural) membrane surrounding the lungs and the pericardium membrane surrounding the heart. **See Figure 1.**
2. Observe the shape, size and colour of the lungs and attached blood vessels leaving and entering the lungs.
3. Observe the diaphragm which may still be attached. It is the sheet of muscle separating the thoracic or chest cavity from the abdominal cavity.
4. Examine the blood vessels associated with the heart and lungs. Identify the arteries which have thick, rubbery walls and the veins which have much thinner walls. Observe the spongy texture of the lung tissue and identify the trachea or wind pipe and the oesophagus (if still attached) which runs in the groove along the trachea. (Note the hooped cartilage rings in the tracheal wall).
5. Insert a length of plastic/rubber tubing into the cut end of the trachea and tie it tightly with a piece of string.
6. Pump some air into the trachea and inflate the lungs**.** There is a possibility of air escaping from cut surfaces of the lung, so it is best practice to place the lungs inside a large, transparent, plastic bag to stop any aerosols from escaping into the laboratory air. Notice the change in appearance of the lungs before and after inflation. During inflation the lungs become much lighter in colour and almost double in size. **See Figure 2a & 2b.** *Note: the plastic bag is absent for photographic purposes*. Remove the tubing**.**
7. Using dissecting scissors carefully sever the heart from the lungs by cutting through the blood vessels as near to the lungs as possible. The hearts can be frozen for future dissections. Science ASSIST has developed a standard operating procedure (SOP) for performing a heart dissection, see link to SOP: Performing a heart dissection (https://assist.asta.edu.au/resource/2837/sop-performing-heart-dissection?search-id=65fed3c.)
8. With scissors cut down the trachea until it divides into two tubes called the bronchi. Continue cutting down into one of the bronchus tubes which continues to divide into bronchioles. Using sharp pointed scissors carefully cut down one of these branches of the bronchioles until you can no longer distinguish the tube. These tubes end in tiny sacks called ‘alveoli’ or ‘air sacks’ where gas exchange takes place. **See Figure 3a**
9. Observe the structure of the trachea, bronchi and bronchioles and note the size and texture of cartilage rings in the walls of these structures. **See Figures 3b & 3c and Diagram 1**
10. Using scissors cut off the end of one of the lobes. Fill the syringe with air by extending the plunger and insert the end of the syringe into the top of one of the bronchiole tubes. Gently push the plunger down and observe the lung tissue inflate.
11. Cut off a small piece of this spongy lung tissue. Drop in a beaker of water and observe that it floats indicating the tissue still holds a volume of air. Repeat this procedure with a piece of heart tissue or diaphragm muscle and compare the result. **See Figure 4.**
12. Place a commercially prepared microscope slide of alveoli under a microscope and examine its structure.

(Operating procedure cont…

Clean up:

- Make sure all instruments are returned.
- All parts of the lung, as well as the disposable foam tray (if used), must be wrapped in newspaper and placed in a dedicated plastic garbage bag along with gloves and disposable aprons (if used). When all waste material is collected, double bag for disposal. Freeze material if unable to dispose of immediately.
- If blood is present on dissecting boards, scissors, forceps, probes, dissecting pins and scalpels they must be immediately soaked in disinfectant. Otherwise wash equipment in hot soapy water and rinse or place in a dishwasher to minimize handling.
- After washing, dissecting instruments can be soaked in 70% v/v ethanol for 20 minutes as an optional additional disinfectant and to avoid rusting.
- Dry all equipment thoroughly.
- Disinfect workplace and wash hands thoroughly.

7. Trouble shooting/emergencies

- **If fainting occurs:** If students start to feel faint, dizzy or nauseous during the dissection lie them down (if possible) and elevate their feet. They can get up slowly after ten minutes. Sending them outside for some fresh air can also help. If they don't recover quickly, always seek urgent medical attention. '***Do not sit the patient on a chair with head between knees***[ii]
- First Aid: See latest SDS of any chemicals used for more detailed information.
  - **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention.
  - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek medical attention.
  - **If on skin/clothes:** If spilt on skin or clothes quickly wipe off with a dry cloth to absorb as much liquid as possible. Remove contaminated clothes and drench the area with excess water under a safety shower. Seek medical attention.
  - **If inhaled:** Remove to fresh air and seek medical attention if symptoms persist.
  - For further advice contact the Poisons Information Centre on 131126.
- First aid: cuts and lacerations should be washed under running water, in the first instance and referred to the school first aid officer for assessment.
- Any health concerns should be referred to the school first aid officer for assessment, accompanied by the relevant latest SDS if applicable. Follow your school's accident and incident policy and reporting procedures.
- See safety notes if it is necessary to remove broken or used scalpel blades.

8. Waste disposal

- Used and damaged scalpel blades must be placed in an approved sharps container after use.
- Biological material must be wrapped in newspaper, placed in a double plastic garbage bag and sealed for immediate disposal in the industrial bins.

9. Related material

- Risk Assessment.
- Manufacturer's Safety Data Sheet for disinfectant

More detailed information on a lung dissection can be found on the following websites below:

- 'Dissecting lungs', Nuffield Foundation website, http://www.nuffieldfoundation.org/practical-biology/dissecting-lungs (May 2009). Class practical or demonstration.
- 'Pluck dissection' STEM Learning website, (15:11 min) https://www.stem.org.uk/rx33t8 This is a very good instructional video for teachers/demonstrators.

References:

[i] 'Fainting', Better Health Channel website, State Government of Victoria: http://www.betterhealth.vic.gov.au/bhcv2/bhcarticles.nsf/pages/Fainting (August 2014)

[ii] St John Ambulance Australia. 2011. *Australian First Aid. Barton, ACT*

Andrews, C; Naidu, Satya; Laidler, Greg. 2002. *Active science: skills and experiments: book 2.* Oxford University Press: South Melbourne, Vic.

Chemwatch Gold. 2013. *Safety Data Sheet: Hospital grade disinfectant*, Chemwatch website, http://jr.chemwatch.net/chemwatch.web (Subscription required.) (Accessed July 2016).

CLEAPSS. 2014. *G268 Dissection: a guide to safe practice*, CLEAPPS website, http://science.cleapss.org.uk/Resource-Info/G268-Dissection-a-guide-to-safe-practice.aspx (Subscription required.)

'Dissection safety tips', Flinn Scientific website, http://www.flinnsci.com/media/396301/dissectionsafety.pdf (2010)

'Dissecting lungs', Nuffield Foundation website, http://www.nuffieldfoundation.org/practical-biology/dissecting-lungs (Accessed July 2016)

'Dissection Safety Policy and Procedures' Flinn Scientific website' http://www.flinnsci.com/media/948812/sf10490.pdf (2013)

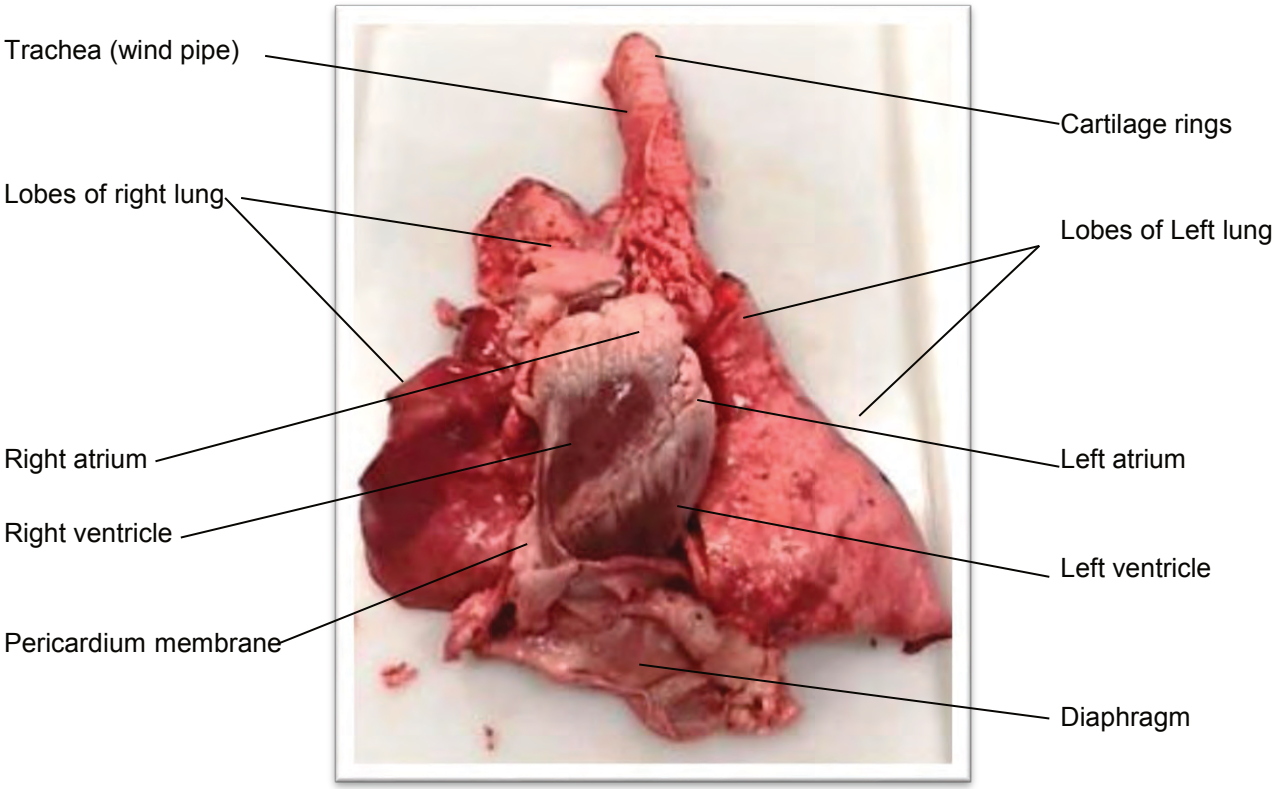
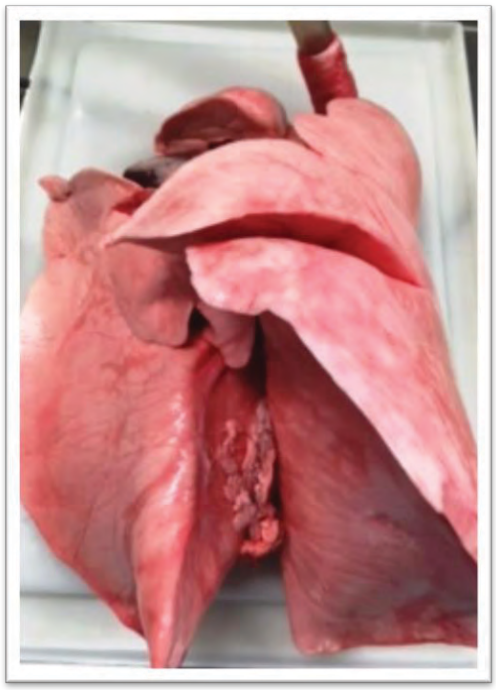


Figure1: The pluck showing the heart-lung arrangement and diaphragm. (Image by K. Szalai 2016)

Figure 2a & 2b: Showing the lungs partially and fully inflated. (Image by K. Szalai 2016)



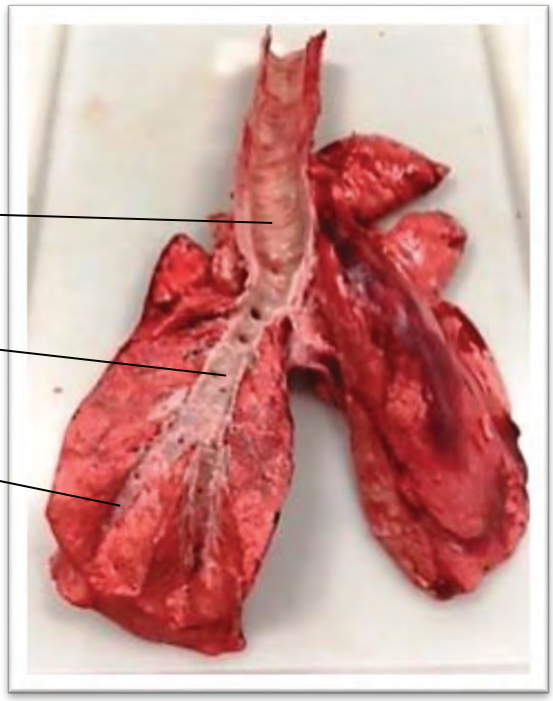


Figure 3a: Shows structure of the dissected right lung and trachea. (Image by K. Szalai 2016)

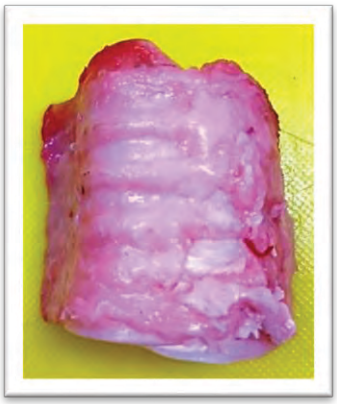


Figure 3b: Sections of the trachea showing cartilage rings. (Image by K. Szalai 2016)

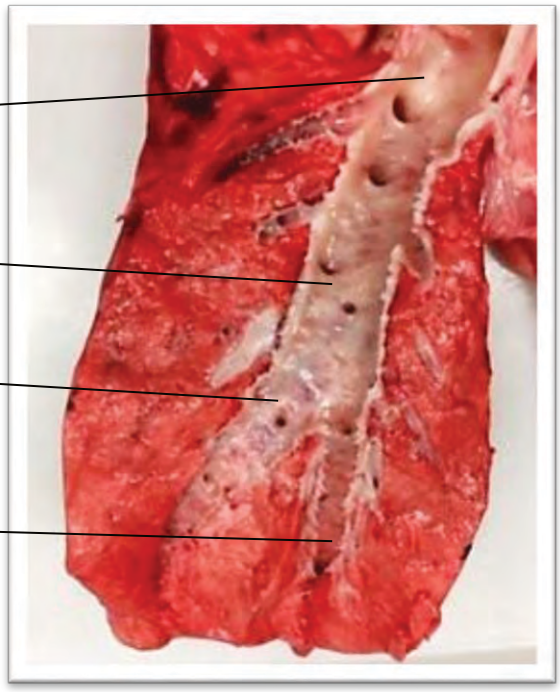


Figure 3c: Shows a close-up of the bronchial tubes and cartilage rings. (Image by K. Szalai 2016)

Figure 4: Shows a comparison between pieces of lung tissue and diaphragm muscle when placed in water. (Image by K. Szalai 2016)

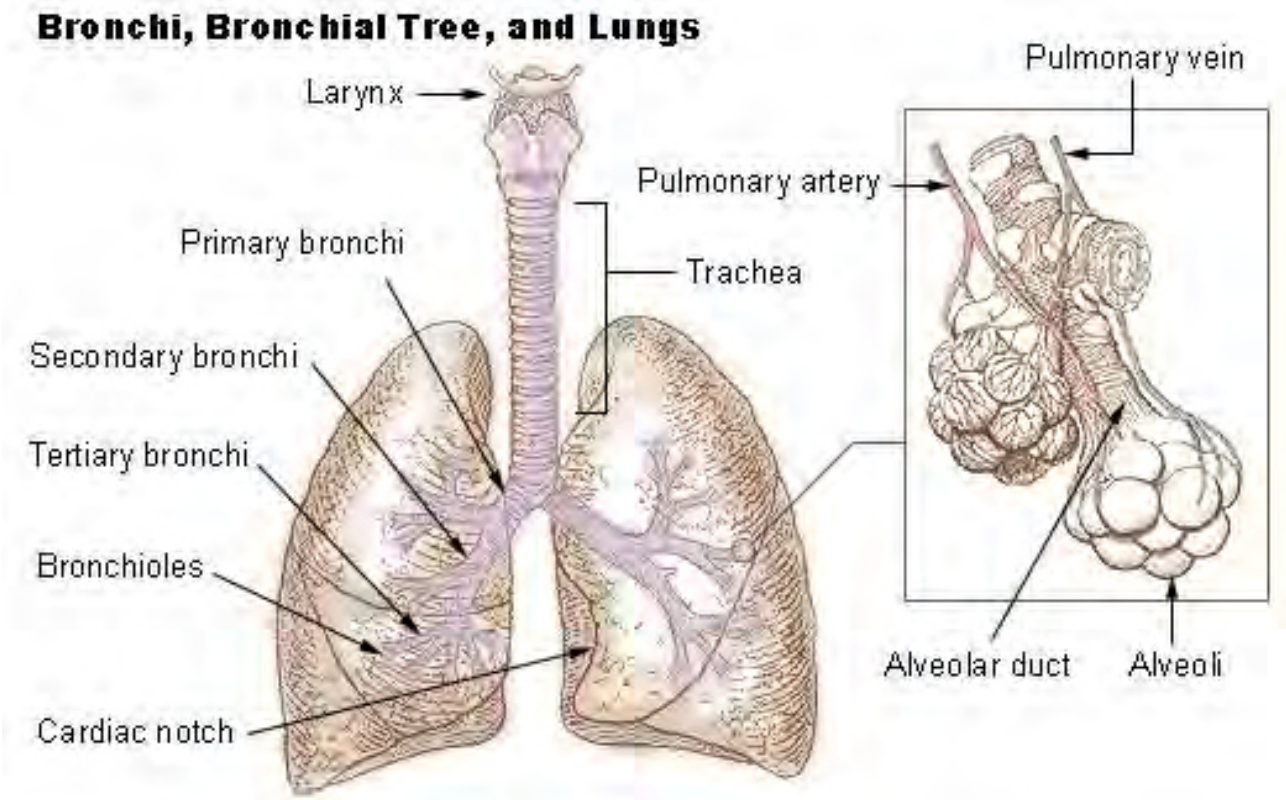


Diagram 1. Labelled structure of the lungs and alveoli.
Wikimedia Commons, Illu_bronchi_lungs.jpg (Accessed July 2016)

Glossary

Alveolar duct – the air passage leading to the alveolar sacs.

Alveoli – tiny air sacs at the end of the bronchioles in the lung where the exchange of oxygen and carbon dioxide takes place.

Arteries – the blood vessels that carry oxygenated blood from the heart to the rest of the body.

Atrium – one of the two blood collection chambers of the heart.

Blood – the liquid that circulates in the blood vessels of many animals.

Blood vessels – the veins, arteries and capillaries through which blood flows around the body.

Bronchi – tubes in the lungs dividing into bronchioles.

Bronchioles – the tubes leading off from the bronchi in the lungs.

Bronchus – one of the two tubes that lead from the trachea to the lungs.

Capillaries – thin-walled blood vessel-like tubes carrying blood to body cells.

Cardiac notch – the deep notch in the alimentary canal at the junction of the oesophagus and the stomach.

Cartilage – flexible connective tissue in animals, including the joints between bones, the rib cage, the ear, the nose, the bronchial tubes and the intervertebral discs.

Diaphragm – the muscle that runs across the base of the chest cavity and separates the thorax and the abdomen. It causes the lungs to expand and contract during breathing.

Heart – the muscular organ that pumps blood around the body.

Larynx – the muscular and cartilaginous structure, lined with mucous membrane, situated at the top of the trachea.

Lungs – inflatable organs used to breathe in oxygen and breathe out carbon dioxide.

Oesophagus – the tube connecting the mouth to the stomach.

Pericardium membrane – the layer of tissue surrounding the heart.

Pleural membrane – the thin layer of connective tissue covering the whole of the lungs.

Pulmonary vein – the large blood vessels that receive oxygenated blood from the lungs and drain into the left atrium of the heart.

Pulmonary artery – the artery that carries deoxygenated blood from the heart to the lungs.

Pluck – heart-lung set from a sheep, pig or cow.

Thoracic – relates to the chest cavity of an animal.

Trachea – the tube through which air enters the lungs from the nose.

Veins – the blood vessels that carry blood in the body or lungs towards the heart**.**

Ventricle – one of two large chambers that collect and expel blood received from an atrium.

History of reviews

Date	Version Number	Notes
Sept 2016	Version 1.0	
Sept 2016	Version 2.0	Correction to first aid information for eyes

STANDARD OPERATING PROCEDURE:

Performing a rat dissection

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

A rat dissection is conducted to explore the internal structure and function of basic mammalian anatomy. The purpose of this exercise is to explore the alimentary canal of the rat and observe the different parts of the digestive system. By looking at the length of the digestive system of the rat, information about their dietary pattern can be determined, and comparisons made between the rat and other mammals such as omnivores, carnivores and herbivores. Because the rat is a placental mammal and an omnivore it has a body structure very similar to that of humans.

Prior to conducting a rat dissection, teachers should ensure that they are able to meet the requirements of the Schools Animal Ethics Committee (SAEC) in their jurisdiction. It is recommended that they consider the educational objectives for this activity, explore the ethical considerations with students and aim to reduce the total number of rats required for this activity. Students should not be forced to participate in a dissection and alternative activities such as videos and virtual dissections can be used for these students instead, as well as to supplement the actual activity.

Rats that have been humanely euthanised and are disease (or infection) free should be sourced from ethical and licenced suppliers. They can be supplied as freshly euthanised, frozen or preserved specimens. If using preserved rats, obtain and read the safety data sheet (SDS) from the supplier and prepare a site-specific risk assessment. If freshly euthanised or recently defrosted rats are being used, and the dissection is interrupted, the rats can be kept for a short time (no longer than 24 hours) in the coldest part of the laboratory fridge or placed in a freezer. There is potential for bacterial growth.

2. Context

- These instructions are for the use of experienced science teachers, technicians and students under close supervision.
- When planning a class dissection activity it is best to discuss beforehand the type of dissection to be undertaken, and warn of the possibility that there may be some blood and odours present during the dissection.
- Demonstrating the dissection to students before they begin is helpful, not only for correct procedure but allows them to adjust to the appearance of the material, and any blood that may be present after dissection material has been washed.
- Let students know they don't have to participate in the dissection and can be excused from the class. Alternative arrangements can be made for students who don't wish to participate, by giving them worksheets to complete and relocating them to a private study area.

3. Safety notes

Fainting: signs and symptoms:

- Fainting may occur during this type of activity. Please read the first aid information in section 7 before conducting the dissection.
- Fainting is caused by a sudden drop in blood pressure. Common causes include heat, pain or distress and the sight of blood.
- The possible symptoms include the following.
    - 'Dizziness
    - Light-headedness
    - A pale face
    - Perspiration
    - Heightened anxiety and restlessness
    - Nausea
    - Collapse
    - Unconsciousness, for a few seconds
    - Full recovery after a few minutes'[i]

Handling specimens:

- If using preserved rats, it is important to take note of the preservative solution that the rats are in and the recommended precautions.
    - Rinse the preserved rats under running water immediately upon removal from the preservative solution.
    - Work in a well-ventilated area.
    - It is recommended that people wearing contact lenses should not dissect rats that are in preservative solution. The fumes from the solution can penetrate between the eye and contact lens causing irritation to eyes. It is recommended to wear prescription glasses instead with safety glasses over them OR prescription safety glasses.
- Consider issues such as allergies and chemical sensitivities from handling freshly euthanised or recently defrosted or preserved rats.
- Be aware of possible microbial aerosols and unpleasant odours released from freshly euthanised or recently defrosted rats if the stomach/intestines are accidently cut.
- If using frozen rats, defrost overnight in a refrigerator and use within 24 hours. Consistent with safe food handling procedures, all meat products should be stored below 5ºC prior to performing any dissections.
- Good hygiene practices should be observed at all times: Keep hands away from the mouth, nose, eyes and face during and after dissection and wash hands immediately after handling dissection material.

Safety with scalpels and dissecting instruments:

- Store all dissecting instruments securely.
- Care should be taken with sharps such as scalpel blades and scissors. Some school science departments restrict the use of scalpels unless specifically requested by a teacher, and prefer to only issue scissors, probes and forceps to students for dissections.
- Ensure students demonstrate responsible behaviour while using scalpels and other dissecting instruments.

- Scalpels should be provided in and returned to a lined container, blade end down
- Students should not walk around the lab with the dissecting instruments, in particular with a scalpel or pointed scissors, forceps or probes.
- To reduce the possibility of stab wounds or cuts from slippage always point sharp instruments such as scalpels and scissors away from yourself and others
- Hold the instruments so that any sharp points or exposed sharp edges point down onto the dissection board or tray. If there is any slippage when using the instrument, the point/exposed edge will be absorbed by the board/foam or wax tray.

Scalpel Blades:

- Only staff should carefully attach and remove scalpel blades using pliers, forceps or a commercial blade remover.
- The scalpel blade size and handle must be compatible e.g. number 4 handle and number 23 blades.
- Keep the blade in the foil wrapper and attach to the handle with the sharp side of the blade pointing away from the body.
- An alternative is to use disposable scalpels.

4. Regulations, licences and permits

Some SAECs may require permission well in advance of the activity being conducted. In some jurisdictions, all dissections need to be reported to the SAEC.

Science ASSIST has developed an information sheet with links to biological safety and jurisdictional SAECs, see AIS: Links — Biological sciences safety

Rats that have been humanely euthanised and are disease (or infection) free should be sourced from ethical and licenced suppliers.

5. Equipment

- PPE – Lab coat/apron (it is recommended to use plastic disposable aprons), safety glasses and gloves.
- Scalpels (optional subject to a site specific risk assessment)
- Scissors
- Forceps
- Probes
- Dissecting pins/needles
- Dissecting boards covered with newspaper. Glazed ceramic tiles, nylon plastic boards, vinyl dissecting pads or disposable foam trays are suitable.
- String
- Paper towel
- Disinfectant – hospital grade, general purpose disinfectant (the label on the front of the pack must state ‘hospital grade’, which is a general purpose hard-surface disinfectant which will kill micro-organisms).
- 70% v/v ethanol

6. Operating procedure

Preparation

- If any blood is associated with the rats rinse them in cold running water.
- Prepare disinfectant solution according to manufacturer's instructions. Place disinfectant in a container ready for instruments to be placed at the end of the dissection.
- Ensure students have appropriate PPE.
- Distribute the instruments to students. Scalpels, scissors, forceps, dissecting pins and probes should be counted out, and counted in when returned.

Examining and dissecting the rat

1. First identify all the external structures visible on the head, thorax and abdomen of the rat. **See Figure 1a.**
2. Observe the mouth cavity and teeth of the rat.
3. Place the rat on the dissecting tray on its back. String is used to fasten the limbs of the rat back. Tie string firmly around one wrist, pass the string under the tray and tie it firmly to the other wrist. Repeat with the hind limbs. **See Figure 1b.**
4. Pick up the skin in the middle of the ventral surface using forceps. Cut (see incision lines in figure 1b) the skin and underlying muscles with the blunt end of the scissors pointing downwards. Cut towards to the bottom of the ribs and then in the other direction to the genital openings. Cut across the bottom of the ribs, and then cut out to both the hind legs.
5. Peel back the flap of skin from the underlying muscle and notice the connective tissue between them. Use the dissecting pins to pin out the skin of the rat to the dissecting board. **See Figure 2.**
6. Open up the abdominal cavity by lifting the abdominal wall with forceps, make an incision with the scissors and follow the same procedure as in step 4 above. Be careful that you do not damage any of the abdominal organs by making the incision too deep.
7. Peel back the abdominal wall and pin out revealing the abdominal organs. **See Figure 3.** Examine the digestive organs in situ and identify all the structures in place. Notice the scattered cream coloured fat deposits. Note the size of the liver in proportion to the abdomen.
8. Remove the digestive system by cutting the oesophagus above its connection with the stomach and by cutting the rectum. Notice the way the small intestine and large intestines are twisted over one another**. See Figure 4.**
9. Using your fingers, carefully pull apart the duodenum and colon from the connective tissue by gently tearing the mesentery and carefully stretch out the alimentary canal. Measure the length of the alimentary canal and the rat's body comparing the ratio of these figures. **See Figure 5.**
10. Note the size of the liver and caecum in relation to the rat's body.

(Operating procedure cont.)

11. Remove the liver carefully and compare the colour of the spleen and liver. Note there is no gall bladder in the rat.
12. Observe the exposed kidneys and associated urinary organs, and the reproductive system left behind in the abdominal cavity**. See Figure 6**
13. If time allows for further investigation, open the thoracic (chest) cavity by making a mid-ventral cut using blunt end of scissors cutting forward through the diaphragm muscle and breast bone up to just under the chin of the rat.
14. The diaphragm, which separates the thoracic from the abdominal cavity, can be cut away from the rib cage.
15. Cut away the ribs to reveal the heart surrounded by a thin membrane called the pericardium, the whitish thymus gland that lies directly over the upper part of the heart and the lungs that lay either side of the heart. **See Figure 7.**
16. Remove the heart and thymus gland to locate the trachea (wind pipe) identifiable by its rings of cartilage. The oesophagus lies just underneath the trachea.

    More detailed information on a rat dissection can be found on the following websites:

    'Digestive System. Rat digestive system'
    https://ratdissection.wikispaces.com/Digestive+System

    'Rat dissection', www.biologycorner.com/myimages/rat-anatomy

Clean up:

- Make sure all instruments are returned.
- All parts of the rat, as well as the disposable foam tray (if used), must be wrapped in newspaper and placed in a dedicated plastic garbage bag along with gloves and disposable aprons (if used). When all waste material is collected, double bag for disposal. Freeze material if unable to dispose of immediately.
- If blood is present on dissecting boards, scissors, forceps, probes, dissecting pins and scalpels they be immediately soaked in disinfectant. Otherwise wash equipment in hot soapy water and rinse or place in dishwasher to minimize handling.
- After washing, dissecting instruments can be soaked in 70% v/v ethanol for 20 minutes as an optional additional disinfectant and to avoid rusting.
- Dry all equipment thoroughly.
- Disinfect workplace and wash hands thoroughly.

7. Trouble shooting/emergencies

- **If fainting occurs:** If students start to feel faint, dizzy or nauseous during the dissection lie them down (if possible) and elevate their feet. They can get up slowly after ten minutes. Sending them outside for some fresh air can also help. If they don't recover quickly, always seek urgent medical attention. '***Do not sit the patient on a chair with head between knees***[ii]
- First Aid: See latest SDS of any chemicals used for more detailed information.

- **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention.
- **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek medical attention.
- **If on skin/clothes:** If spilt on skin or clothes quickly wipe off with a dry cloth to absorb as much liquid as possible. Remove contaminated clothes and drench the area with excess water under a safety shower. Seek medical attention.
- **If inhaled:** Remove to fresh air and seek medical attention if symptoms persist.
- For further advice contact the Poisons Information Centre on 131126.

- First aid: cuts and lacerations should be washed under running water, in the first instance and referred to the school first aid officer for assessment.
- Any health concerns should be referred to the school first aid officer for assessment, accompanied by the relevant latest SDS if applicable. Follow your school's accident and incident policy and reporting procedures.
- See safety notes if it is necessary to remove broken or used scalpel blades.

8. Waste disposal

- Used and damaged scalpel blades must be placed in an approved sharps container after use.
- Biological material must be wrapped in newspaper, placed in a double plastic garbage bag and sealed for immediate disposal in the industrial bins.
- Specimens preserved in Carolina's Perfect Solution ® are not subject to hazardous materials regulations or disposal restrictions in Australia. Refer to SDS and treat as above for biological material.

9. Related material

- Risk Assessment.
- Manufacturer's Safety Data Sheet for disinfectant
- Manufacturer's Safety Data Sheet for preserved specimens

References:

[i] 'Fainting', Better Health Channel website, State Government of Victoria: http://www.betterhealth.vic.gov.au/bhcv2/bhcarticles.nsf/pages/Fainting (August 2014)

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History of reviews

Date	Version Number	Notes
Feb 2016	Version 1.0	
Sept 2016	Version 2.0	Correction to first aid information for eyes and cuts Additional safety information included • Safe food handling procedures • the use of scalpels and dissecting instruments

Figures

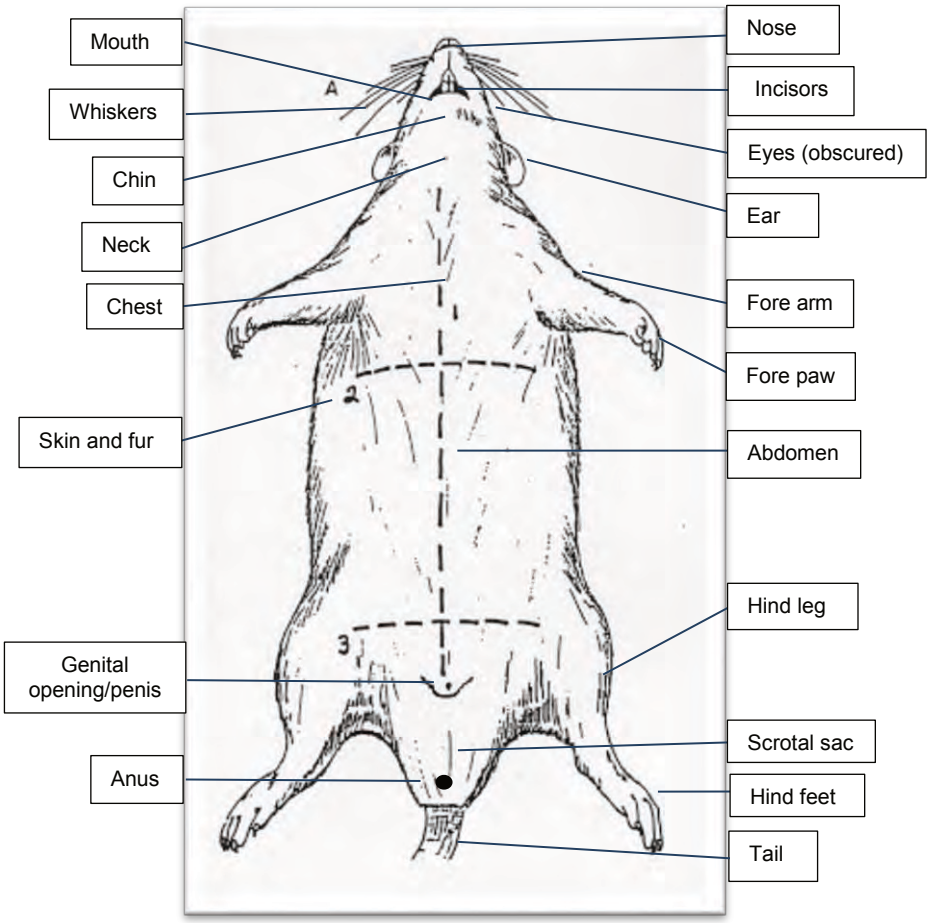

Figure 1a: The external structures visible on the head, thorax (chest) and abdomen including incision lines numbered 1, 2 and 3.

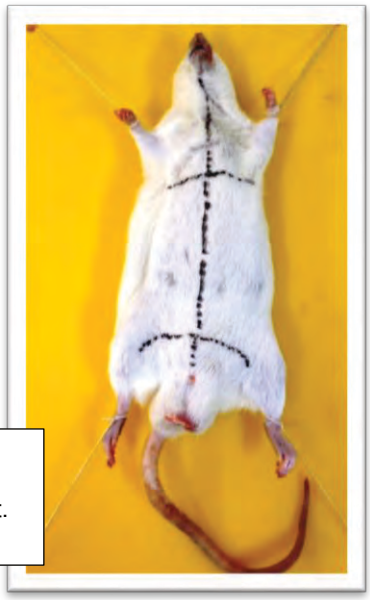

Figure 1b: The dotted lines show the incision lines for dissection of rat. (Image by K. Szalai, 2016)

Figure 2: The abdominal wall exposed. (Image by K. Szalai, 2016)

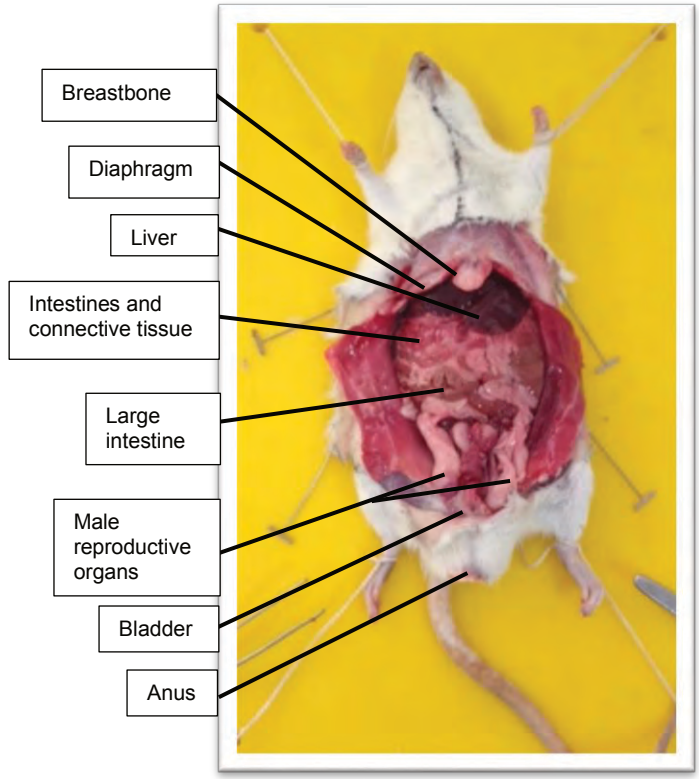

Figure 3: The abdominal organs of the rat in situ. (Image by K. Szalai, 2016)

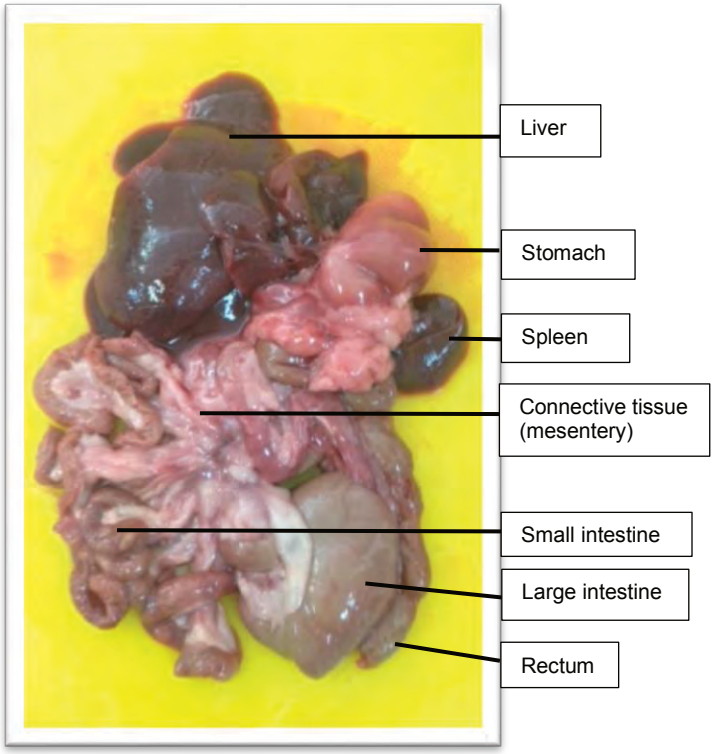

Figure 4: Abdominal organs removed. (Image by K. Szalai, 2016)

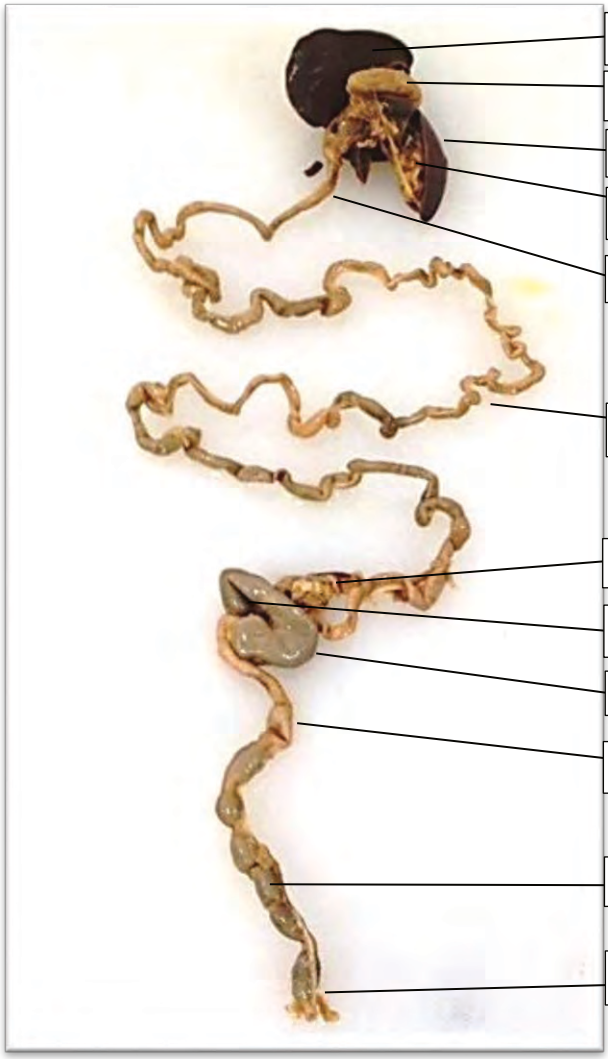

Figure 5: The alimentary canal of the rat with the connective tissue removed. (Image by K. Szalai, 2016)

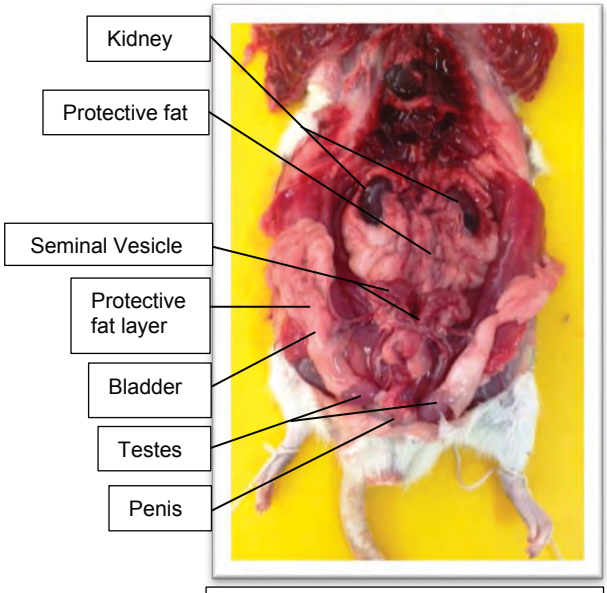

Figure 6: Male Urinogenital system (Image by K. Szalai, 2016)

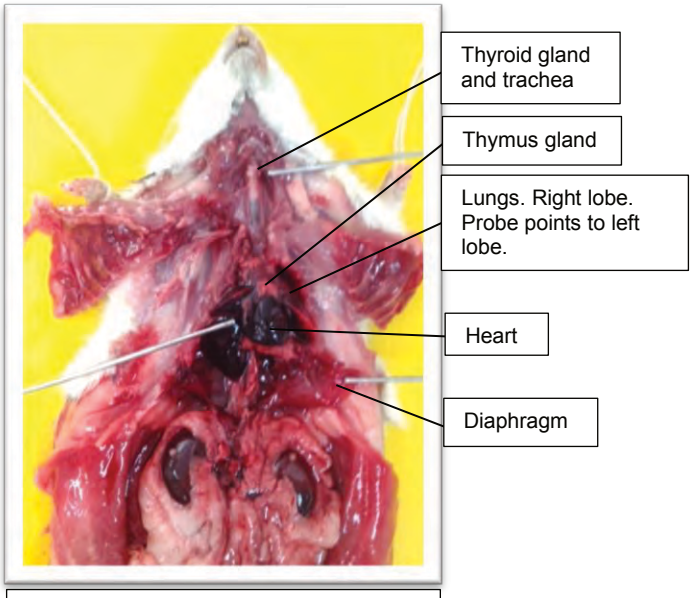

Figure 7: Thorax and neck. (Image by K. Szalai, 2016)

Glossary

Abdominal wall – the lining that encloses the abdominal cavity, consisting mostly of muscle.

Alimentary canal – the tubular passage between the mouth and the anus.

Anus – the last part of the large intestine through which the faeces are excreted.

Appendix – a small tube with only one opening, near the beginning of the large intestine.

Bladder – the organ where urine is stored in the body.

Blood – liquid that circulates in the blood vessels of many animals. It contains plasma, red and white blood cells, platelets, dissolved gases, nutrients, hormones and salts.

Breast bone – the long flat bone shaped in the middle of the chest, forming the front of the rib cage.

Caecum – large organ where the small intestine joins the large intestine.

Carnivore – an animal that eats only meat.

Cartilage – flexible connective tissue in animals, including the joints between bones, the rib cage, the ear, the nose, the bronchial tubes and the intervertebral discs.

Colon – the section of the large intestine that runs from the caecum to the rectum and absorbs water from faeces.

Connective tissue – animal tissue that supports, connects, and surrounds organs and other body parts.

Diaphragm – the muscle that runs across the base of the chest cavity and separates the thorax and the abdomen. It causes the lungs to expand and contract during breathing.

Digestion – the process of converting large, complex organic molecules to smaller, simpler ones that can pass through cell membranes.

Digestive system – the parts of the body where food is digested.

Duodenum – the top part of the small intestine, where bile and pancreatic juice enter the digestive system.

Faeces – undigested food and bacteria that is stored in the rectum and expelled through the anus.

Gall Bladder – small organ that is located beneath the liver and drains bile into the duodenum primarily to break down fat during digestion.

Heart – muscular organ that pumps blood around the body.

Herbivore – an animal that eats only plants.

Incisors – the two front teeth in each jaw of a mammal used for cutting food.

In situ – in its original place.

Kidneys – the major excretory organs that filter the blood, and maintain the concentration of water and salts in the body. They form urine in which excess salts, water urea and waste products are removed from the body.

Large intestine – the end section of the alimentary canal reaching from ileum to anus and consisting of the caecum, colon, and rectum. Its function is to extract water and form faeces.

Liver – a large organ in mammals that controls the level of nutrients and toxic substances in the blood. Has a digestive function, secretes bile, filters blood, and takes part in many metabolic functions such as the conversion of sugars into glycogen.

Lungs – inflatable organs used to breathe in oxygen and breathe out carbon dioxide.

Mammal – class of warm-blooded vertebrate animals.

Mechanical digestion – the breaking down of large pieces of food into smaller pieces.

Mesentery – a supportive membrane surrounding and giving structure to the inner organs.

Oesophagus – the passage down which food moves between the throat and the stomach.

Omnivore – an animal that eats both plants and animals.

Pancreas – an organ in the body that produces pancreatic juice that helps to digest food.

Penis – male sex organ used to transfer semen and expel urine from the body.

Pericardium – membrane that forms a sac surrounding the heart and attached portions of the main blood vessels.

Placental mammal – the embryo develops in the maternal uterus attached to the tissues of the placenta.

Rectum – the last part of the large intestine where faeces is stored.

Scrotal sac – the sac of skin that holds the testicles.

Seminal vesicle – the gland that secretes seminal fluid in semen.

Small intestine – the part of the intestine between the stomach and the large intestine, consisting of the duodenum, jejunum, and ileum, where digestion of food and most absorption of nutrients take place.

Spleen – organ in the left upper abdomen of humans and other vertebrates that helps to destroy old red blood cells, form lymphocytes, and store blood.

Stomach – a hollow muscular organ where mechanical and chemical digestion takes place.

Testes – male sex organs that produce sperm.

Thoracic –relates to the chest and cavity of an animal.

Thorax – the part of the body between the neck and abdomen, enclosed by the ribs and containing lungs, heart and diaphragm.

Thymus – the organ, located at the base of the neck, and is involved in development of cells of the immune system.

Thyroid gland – endocrine gland located in the neck of human beings and other vertebrate animals that secretes the hormones responsible for controlling metabolism and growth.

Trachea – a tube through which air enters the lungs from the nose.

Ventral surface – the surface of the abdomen or lower body.

STANDARD OPERATING PROCEDURE:

Performing an eye dissection

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

Eye dissections are conducted to examine the structure of an eye and to consider how the components of the eye work separately and together. Good hygiene practices should be observed. Eyes suitable for dissections include fresh cow, sheep or pigs eyes purchased from a butcher, abattoir or a supplier that has passed relevant health inspections. Fresh eyes need to be used as soon as possible as the lenses go cloudy after a few days.

2. Context

- These instructions are for the use of experienced science teachers and technicians.
- Students must be closely supervised when performing eye dissections.

3. Safety notes

- A site specific risk assessment should take into consideration the maturity of students carrying out the dissection and should address risks associated with students using scalpels and other dissection equipment.
- Before the dissection it is recommended the teacher or laboratory technician trial the dissecting instruments (scalpels, scissors and pointed forceps) to establish that they are sufficiently sharp enough and to determine the most appropriate equipment for the task considering the student behaviour.

Fainting signs and symptoms:

- Fainting may occur during this type of activity. Please read the first aid information in section 7 before conducting the dissection.
- Fainting is caused by a sudden drop in blood pressure. Common causes include heat, pain or distress and the sight of blood.
- The possible symptoms include the following.
    - Dizziness
    - Light-headedness
    - A pale face
    - Perspiration
    - Heightened anxiety and restlessness
    - Nausea
    - Collapse
    - Unconsciousness, for a few seconds
    - Full recovery after a few minutes'[i]

Handling specimens:

- The eyeballs should never be held in the hand to dissect
- Eyes differ from standard offal because they are not sold as food. Bacteria and viruses could be present.
- If using preserved eyes, it is important to take note of the preservative solution that the eyes are in:
    - Rinse the preserved eyes under running water immediately upon removal from the preservative solution.
    - Work in a well-ventilated area.
    - It is recommended that people wearing contact lenses should not dissect eyes that are in preservative solution. The fumes from the solution can penetrate between the eye and contact lens causing irritation to eyes. It is recommended to wear safety glasses over prescription glasses instead OR prescription safety glasses.
- Good hygiene practices should be observed at all times.
- Keep hands away from mouth, nose, eyes and face during and after dissection and wash hands immediately after handling dissection material.

Safety with scalpels and dissecting instruments:

- Store all dissecting instruments securely.
- Care should be taken with sharps such as scalpel blades and scissors. Some school science departments restrict the use of scalpels unless specifically requested by a teacher, and prefer to only issue scissors, probes and forceps to students for dissections.
- It is suggested that the teacher and/or laboratory technician use a scalpel to make the initial slit in the eyes for students, who are then able to continue the dissection using scissors. This removes the need for the students to handle scalpels.
- Ensure students demonstrate responsible behaviour while using scalpels and other dissecting instruments.
- Scalpels should be provided in and returned to a lined container, blade end down
- Students should not walk around the lab with the dissecting instruments, in particular with a scalpel or pointed scissors, forceps or probes.
- To reduce the possibility of stab wounds or cuts from slippage always point sharp instruments such as scalpels and scissors away from yourself and others
- Hold the instruments so that any sharp points or exposed sharp edges point down onto the dissection board or tray. If there is any slippage when using the instrument, the point/exposed edge will be absorbed by the board/foam or wax tray.

Scalpel Blades:

- Only staff should carefully attach and remove scalpel blades using pliers, forceps or a commercial blade remover.
- The scalpel blade size and handle must be compatible e.g. number 4 handle and number 23 blades.
- Keep the blade in the foil wrapper and attach to the handle with the sharp side of the blade pointing away from the body.
- An alternative is to use disposable scalpels.

4. Regulations, licences and permits

Offal that has passed a health inspection by a meat inspector or procured from a butchers shop, abattoir or biological supplier is suitable for dissection. Eyes can be obtained for educational purposes with safe handling procedures put in place. In some jurisdictions, all dissections need to be reported to the school animal ethics committee

5. Equipment

- PPE (Lab coat/apron, gloves and safety glasses)
- Scalpel (optional subject to a site specific risk assessment)
- Scissors, Forceps
- Dissecting board covered in newspaper, or disposable foam tray
- Newspaper to protect bench and for wrapping biological materials after dissection
- Paper towel
- Disinfectant – hospital grade general purpose disinfectant (the label on the front of the pack must state 'hospital grade', which is a general purpose hard surface disinfectant which will kill micro-organisms).
- 70% v/v ethanol

6. Operating procedure

Preparation

- Prepare disinfectant solution according to manufacturer's instructions. Place disinfectant in a container ready for instruments to be placed at the end of the dissection.
- Ensure students have appropriate PPE.
- Distribute instruments to students. Scalpels and scissors should be counted out, and counted in when returned.

Performing an eye dissection

1. Place the eye on the dissection board or tray. With forceps and the scissors, carefully remove the fatty tissue from around the eyeball.
2. Locate the transparent skin of the cornea and note that the eyeball is protected by a tough protective layer (sclera).
3. Locate the optic nerve. It should look like a thick white tube at the back of the eye. You may have to remove some fat to see it.

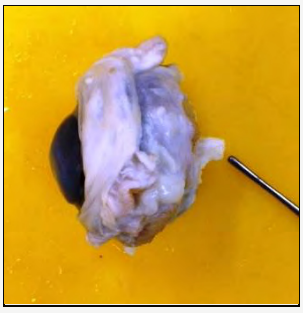

Figure 1. External view of bullock's eye. The probe points to the optic nerve at the back of the eye. At the front of the eye note the sharp curve of the sclera coating the cornea. (**Image by K. Szalai, 2015**)

(Operating procedure cont….)

4. Look at the coloured part of the eye (iris) and the black part in the centre (pupil).
5. The cornea of the eyeball is tough and extra care is required when trying to cut into it. Using a scalpel* or scissors carefully cut a small window in the side of the eyeball, just behind the iris. Some aqueous or vitreous humour fluid may ooze out. Do not squeeze the eyeball too tightly, whilst doing this or the fluid will spurt out due to the pressure. Try looking through this window and record your observations.

   *Safety note: It is suggested that the teacher and/or laboratory technician use a scalpel to make the initial slit in the eyes for students, who are then able to continue the dissection using scissors. This removes the need for the students to handle scalpels.
6. Starting from this window, using scissors cut forward and around the iris so you have cut the eye into two parts.

   Figure 2 shows the two halves of the eye

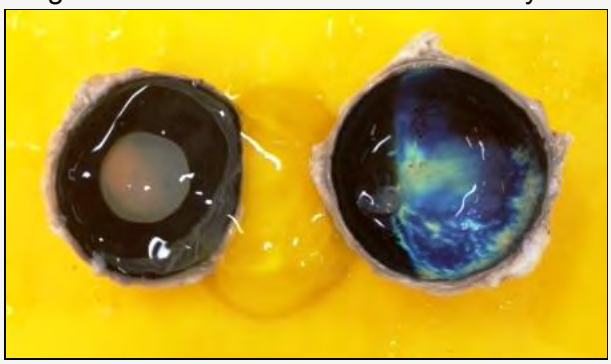

Figure 2. On the left the iris and pupil can be seen with the lens in the centre and jelly-like material of the vitreous humour. On the right the black, pearly-green inside layer the *tapetum lucidum* and the retina can be seen. (**Image by K. Szalai, 2015**)

7. With forceps lift off the top part of the eye; this consists of the cornea, iris and pupil, aqueous humour and lens.
8. Carefully separate the lens from the rest of the eye with forceps. Some of the jelly-like material of the vitreous humour may adhere to the lens. Rinse the lens with water and put it on a piece of newspaper. Try gently squeezing the lens from the side as you look. Note what you observe.
9. Gently remove the rest of the vitreous humour from the eyeball, rinse out the inside of the eye with water and notice the black, pearly-green inside layer. Observe the retina.
10. Examine the back of the front part of the eye from which you took the lens. The muscular ring-like structure of the iris surrounding the pupil is now exposed.

(Operating procedure cont….)

11. With forceps carefully remove the iris exposing the cornea.

Figure 3 shows the labelled structure of an eye.

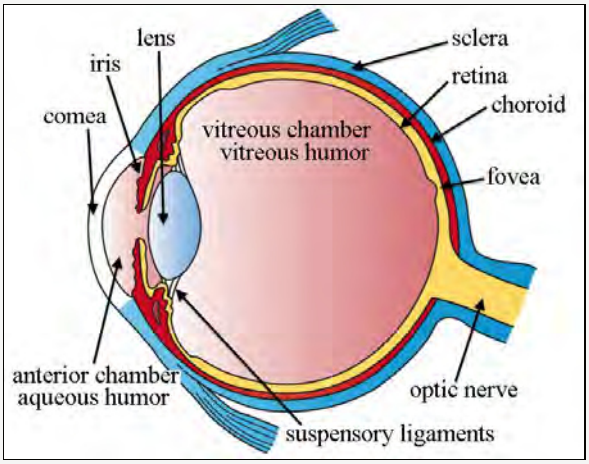

Figure 3. Shows the labelled cross-section of an eye.

Image by Holly Fischer, Three main layers of the eye, CC BY 3.0 http://commons.wikimedia.org/wiki/File:Three_Main_Layers_of_the_Eye.png#/media/File:Three_Main_Layers_of_the_Eye.png

Clean up

- Make sure all instruments are returned.
- All parts of the eye must be wrapped in newspaper, as well as the disposable foam tray (if used) and placed in a dedicated plastic garbage bag. When all waste material is collected, double bag for disposal.
- Dissecting boards, scissors, forceps and scalpels must be immediately soaked in disinfectant, then washed in hot soapy water, then rinsed.
- After washing, dissecting instruments can be soaked in 70% alcohol for 20 minutes as an optional additional disinfectant and to avoid rusting. Dry all equipment thoroughly.
- Disinfect workplace and wash hands thoroughly.

7. Trouble shooting/emergencies

- **If fainting occurs:** If students start to feel faint, dizzy or nauseous during the dissection lie them down (if possible) and elevate their feet. They can get up slowly after ten minutes. Sending them outside for some fresh air can also help. If they don't recover quickly, always seek urgent medical attention. '*Do not sit the patient on a chair with head between knees*[ii]
- Any health concerns should be referred to the school first aid officer for assessment, accompanied by the relevant latest SDS if applicable. Follow your school's accident and incident policy and reporting procedures.
- First aid: See latest disinfectant SDS of any chemicals used for more detailed information
  - **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention.
  - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek medical attention.
  - **If on skin/clothes:** If spilt on skin or clothes quickly wipe off with a dry cloth to absorb as much liquid as possible. Remove contaminated clothes and drench the area with excess water under a safety shower. Seek medical attention.

- **If inhaled:** Remove to fresh air and seek medical attention if symptoms persist.
- For further advice contact the Poisons Information Centre on 131 126.

- See safety notes if it is necessary to remove broken or used scalpel blades.
- First aid: cuts and lacerations should be washed under running water in the first instance and referred to the school first aid officer for assessment.

8. Waste disposal

- Used and damaged scalpel blades must be placed in an approved sharps container after use.
- Biological material must be wrapped in newspaper, placed in a double plastic garbage bag and sealed for immediate disposal in the industrial bins.

9. Related material

- Risk Assessment
- Manufacturer's Safety Data Sheet for disinfectant
- Manufacturer's Safety Data Sheet for preserved specimens

References:

[i] 'Fainting', Better Health Channel website, State Government of Victoria: http://www.betterhealth.vic.gov.au/bhcv2/bhcarticles.nsf/pages/Fainting (August 2014)

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Glossary

Aqueous humour – a transparent, watery fluid filling the front chamber of the eye between the back of the cornea and the front of the iris and pupil

Bacteria – a group of microscopic, single-celled organisms lacking a nucleus

Cornea – the transparent curved front part of the eye that covers the iris and pupil and initially bends light rays into the lens

Choroid – a brownish membrane between the retina and the white of the eye

Fovea – a small depression located in the macula of the retina that provides the clearest vision of all

Iris – the coloured part of the eye

Lens – the part of the eye that focuses light to produce an image on the light-sensitive cells of the retina

Macula – is an oval-shaped pigmented area near the centre of the retina

Optic nerve – nerve fibres transmitting visual light signals from the eye to the brain

Pupil – the dark circular opening at the centre of the iris in the eye, where light enters the eye

Retina – a light-sensitive membrane in the back of the eye containing rods and cones that receive an image from the lens and send it to the brain through the optic nerve

Sclera – a protective layer coating the cornea of the eyeball that forms the white of the eye

Suspensory ligaments – a fibrous membrane that holds the lens of the eye in place

tapetum lucidum – a layer of cells in the wall of the eye of nocturnal and deep-sea animals that reflects light back onto the retina, enhancing visual sensitivity in dim light. Light reflected by this layer is responsible for the shining eyes of cats seen when they are illuminated at night.

Viruses – a sub-microscopic parasitic particle of a nucleic acid surrounded by protein that can only replicate within a host cell

Vitreous humour – a transparent, jelly-like fluid that fills the space in the eye between the lens and the retina

History of reviews

Date	Version number	Notes
July 2014	Version 1.0	
April 2015	Version 2.0	First aid for disinfectant included Photos and diagrams added
April 2016	Version 3.0	Fainting and preservative information included
Aug 2016	Version 4.0	Additional safety information included regarding the use of scalpels and suggestion for staff to make initial cut into eye Detail regarding the tapetum lucidum added in Figure 2 Glossary added

STANDARD OPERATING PROCEDURE:

Use and care of the compound light microscope

Note: To be undertaken only by trained personnel in conjunction with a current site-specific risk assessment.

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1. Introduction

The microscope is a tool that enables us to view things that are too small to be seen with the naked eye. The most common type of microscope used in school science laboratories is the compound light microscope. It uses a system of two or more lenses to collect and focus transmitted visible light through a specimen to the eye. It is the principle tool for the study of biology and is often referred to as *bright field microscopy*. Animal cells, plant cells, protozoa and bacteria can be easily seen with a compound light microscope. The typical compound light microscope is able to magnify from 40x to 1000x, increasing our ability to see detail so that objects as small as 0.2 micrometres (µm) or 200 nanometres (nm) can be seen. Compound light microscopes may be monocular (for viewing using only one eye) or binocular (for viewing using both eyes). Compound light microscopes from various manufacturers may appear different but operate on similar principles. A microscope is a delicate precision instrument and care must always be used when using, transporting and maintaining it.

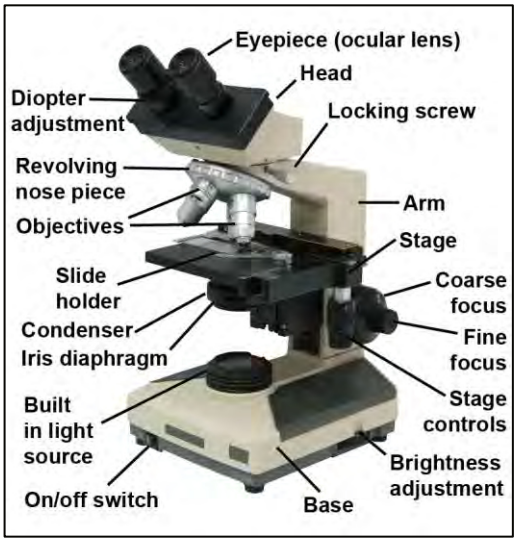
A typical school microscope has three magnifications: Scanning, Low and High. Each objective and eyepiece (ocular lens) will have the magnification written on it. Some microscopes will also have an oil immersion objective.

The total magnification is the ocular magnification multiplied by the objective magnification.

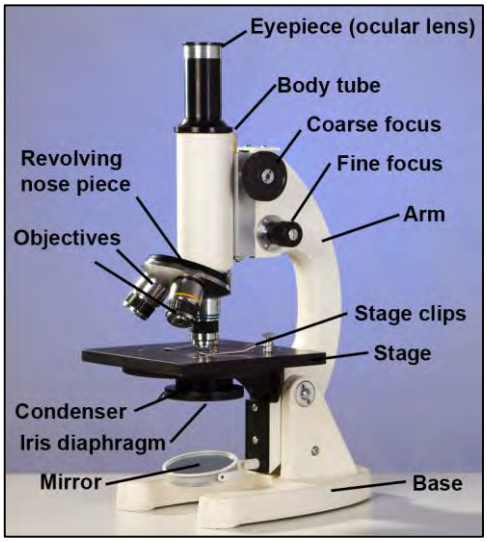
Objective	Magnification	Ocular lens	Total magnification
Scanning	4x	10x	40x
Low power	10x	10x	100x
High power	40x	10x	400x
Oil immersion	100x	10x	1000x

Below are images of typical light microscopes with parts labelled:

a) With built in light source



b) With mirror to direct an external light source



2. Context

- These instructions are for the use of science teachers, technicians and secondary school students who are under the direct supervision of a teacher.
- This SOP contains general guidelines only. Please consult the user manual for your particular microscope.

3. Safety notes

- Always carry the microscope with two hands. It is advisable to place one hand under the base of the box for extra support.
- If the microscope is stored in a box, always ensure the door is locked before picking up the box.
- If using sunlight as the external light source, never face the adjustable mirror directly into the sun.
- Never disassemble the microscope as doing so may damage it.
- Unplug from the power supply before replacing the bulb or moving the microscope.
- Never use coarse focus except with the scanning and low power objective lens. It is very easy to drive an objective through a slide.
- Make sure your workstation is set up ergonomically to use the microscope.
- Avoid prolonged use of the microscope. Take breaks to rest your eyes and make sure that the light intensity is not excessive.
- Make sure the microscope and any external light sources are regularly electrically tested and tagged.
- For all chemicals consult current SDS's.

4. Regulations, licences and permits

NA

5. Equipment

- Compound light microscope

- User manual for your microscope
- PPE: laboratory coat, enclosed shoes, hair tied back
- Microscope cover
- Prepared slides with coverslips
- Immersion oil (if using 100x objective)
- Light source (if required)
- Lens tissue and blower brush
- Lens cleaning fluid or Windex®. Do not use any other solvents on microscope lenses as they can loosen the glue used to hold the lens elements in place and ruin the lens.

6. Operating procedure

Set up and use of a compound light microscope

1. Read and be familiar with the user manual for your model of microscope.
2. Carry the microscope with two hands, one under the base and the other gripping the arm or frame.
3. Gently place the microscope on a flat, level surface and plug into a power source. Some microscopes have a built in light source but others have a mirror to focus natural light or an external light source.
4. With a built-in light source, turn on the light source and adjust the light setting so that it is not too bright by turning or sliding the brightness adjustment knob on the base.
5. 'If using an external light source direct the light via the mirror. Rotate the low power objective into position. Remove the eyepiece, look down the body tube and adjust the mirror and diaphragm setting so light is reflected up the tube and a circle of evenly illuminated light is visible in the field of view. Replace the eyepiece. Use the concave mirror side if the microscope has a fixed condenser lens or the flat mirror side if the microscope has an adjustable condenser'[1].
6. The iris diaphragm is located just above the light source on the bottom side of the stage. Using the lever attached, you can increase or decrease the amount of light reaching the specimen. Look through the eyepiece and adjust the sub-stage iris diaphragm to allow sufficient comfortable light through.
7. Between the stage and the iris diaphragm is the condenser. The condenser further aids in the focusing of the light onto the specimen. In some microscopes it can be moved up and down. To begin with, position it close to the stage. If you have a problem focusing your specimen then adjust the position of the condenser.
8. Adjust the stage down as low as possible with the coarse focus knob.
9. Begin by viewing the specimen with the lowest power objective lens in place and then increase to the higher power objective lenses.
10. Select the 4x scanning objective by rotating the nosepiece, ensuring it clicks into place.

(Operating procedure cont.…)

11. Place a prepared slide onto the stage and hold it in place with the metal clips. Centre it so that the specimen is under the objective lens. Move it with the stage control knobs either left to right or backwards and forwards.
12. After placing the slide on the stage look at the objective lens and the stage from the side and use the coarse focussing knob to bring the slide as close to the objective as possible without touching it.
13. Look in the eyepiece/s and slowly move the stage away from the objective lens with the coarse focusing knob. Stop when the image comes into view.
14. If using a binocular microscope adjust the distance between the eyepieces to suit your eyes by sliding the eyepieces in and out until you see one image. This is called the interocular distance.
15. Use the fine focus to sharpen the image. Scan the slide, select the part of the specimen you are interested in and centre it in your field of view.
16. Adjust the sub-stage iris diaphragm to optimise the lighting.
17. Rotate in the low power 10x objective and refocus with the fine focus. You may need to open the iris diaphragm to let more light in. In general, the higher the power, the more light you require.
18. Repeat with the high power 40x objective, adjusting the iris diaphragm if required. Use only the fine adjustment knob to focus the microscope when using the higher power objective lenses.
19. If you have a 100x oil immersion objective, you will need to first focus on the specimen with the 40x objective. Next rotate the nosepiece so that a midway position is obtained between the 40x objective and the 100x objective. Place a small drop of immersion oil onto the slide coverslip then continue to rotate the nosepiece so that the 100x objective is rotated into the oil. The immersion oil should be used sparingly. **Never** use immersion oil with any of the other objectives. (N.B. It is possible to place the oil directly on a specimen that has been fixed or heat fixed and stained without a coverslip, e.g. bacterial slides. However, it is difficult to remove the oil from the slide without damaging the smear.) Any attempt to re-look at the slide with a low or high power objective may result in contamination of these objectives with the immersion oil. Do not use immersion oil on a wet mount unless you can secure the coverslip well.
20. Sharpen the image with the fine focus only and adjust the light with the iris diaphragm if required.
21. When finished, lower the stage, rotate the low power objective (4x) into position and remove the slide.
22. Clean the oil off the slide and the objective when finished with lens tissue and lens cleaning fluid. In order to return to work at the lower magnifications, the slide must be completely cleaned of any residual oil. Wipe the stage clean with a paper towel.
23. Turn off the light and at the main switch.
24. Report any problems to your teacher.
25. Cover the microscope with its dust cover.

(Operating procedure cont….)

Microscope handling and storage

1. When work is completed, lower the stage, remove the slide, rotate in the lowest power objective, wrap the cord loosely around the base and cover with a dust cover. Take care not to wrap the cord around a hot (built in) light source.
2. Always keep your microscope covered when not in use. Optics and mechanical parts must be protected from dust.
3. Always move the microscope with one hand under the base and the other hand gripping the arm or frame.
4. Keep microscopes away from vibration, moisture, high temperatures and direct sunlight.
5. Never store microscopes in chemical storage areas as corrosive fumes may damage metal and lenses.

Microscope maintenance

1. Treat lenses with great care as they can be easily scratched. Never use anything abrasive.
2. When cleaning lenses, first blow away any dust with a blower brush then use lens tissue and lens cleaning fluid such as Windex® to clean the objectives and eyepieces. Do not use paper towel or regular tissues, as they will scratch the lens. Do not use other solvents.
3. Do not remove eyepieces or objectives from their location but clean only their external surfaces.
4. Remove immersion oil from the 100x objective immediately after use with lens tissue and lens cleaning fluid.
5. Wipe dust off the body of the microscope with a damp cloth.
6. Never attempt to take a microscope apart. This could impair operation, efficiency and accuracy.
7. Have the microscope serviced regularly by a professional, as most microscopes require periodic lubricating and minor adjustment of their mechanical parts.
8. Follow your user's manual for instruction in replacing the bulb. Always allow a bulb to cool before replacing it. When replacing bulbs avoid touching the glass with your hands, use a tissue. Fingerprints can reduce bulb quality and reduce its life.

7. Trouble shooting/emergencies

COMMON FAULTS	POSSIBLE CAUSES
No light	• Power cord is not connected, power switch is off • Wrong bulb is installed • The bulb has burnt out • Light intensity control is turned down too low • Objective is not properly in position • If using the 100x objective immersion oil has not been applied
Image is too dark	• Increase light intensity • Sub-stage iris diaphragm is not open enough • Condenser is too low
Image is too light	• Decrease light intensity • Sub-stage iris diaphragm is open too much
Spot in the field of view that doesn't move when the slide is moved	Lens is dirty. Clean both the objective and eyepiece.
Poor image quality, poor resolution, image not sharp (100x oil objective)	• Clean objective, eyepiece and condenser • Check if immersion oil is contaminated or cloudy or air bubbles are present • Slide is wrong way up
Poor image quality, poor resolution, image not sharp (40x objective)	There is dirt or oil on the lens
Uneven illumination	• Adjust condenser • Make sure objective has clicked into place
Flickering light	• Bulb needs replacement • Loose connection at the outlet • Bulb not properly inserted • Check voltage supply
Half the viewing field is illuminated	Make sure the objective is clicked into place.
Unable to focus the slide	• Coverslip is too thick • Slide is the wrong way up • The stage is slowly dropping, adjust tension of course focus knob • Clean the slide, objective and eyepiece

8. Waste disposal

- Dispose of used lens cleaning tissue into the regular waste.
- Place used coverslips into a sharps container.
- Commercially prepared slides should be returned to the slide box.
- Used microscope slides from wet mounts can be washed and reused OR should be disposed of with broken glass. Slides with heat fixed smears are difficult to clean so are not reused and are disposed of with broken glass: refer to Science ASSIST AIS: Lab glass and porcelain disposal. (http://assist.asta.edu.au/resource/2395/ais-lab-glass-and-porcelain-disposal)

9. Related material

- Microscope User Manual specific to your microscope
- Risk assessment for use of the light microscope
- SDS for immersion oil
- SDS for lens cleaning fluid/Windex

Glossary:

Coarse focus – moves the mechanical stage to give approximate focus of the specimen.

Condenser – focuses light from the light source onto the specimen.

Condenser focus – adjusts the height of the condenser so that it focuses light from the light source onto the specimen.

Eyepieces or oculars – lenses that further magnify the image of the specimen produced by the objective lens (secondary magnification).

Field of view – the circular area of the specimen that you can see through the eyepiece.

Fine focus – moves the mechanical stage to give sharp focus of the specimen.

Immersion oil – an oil medium that has a high refractive index and is used with the 100x oil immersion objective to increase the resolution. Use immersion oil only for the purpose of microscopy.

Interocular adjustment – controls the distance between the two oculars to match the distance between the eyes of the user.

Magnification – is the degree of enlargement of the specimen.

Numerical aperture – a number written on the objectives that expresses the ability of a lens to resolve fine detail.

Objectives – lenses that produce primary magnification of the specimen.

Parfocal – allows the rotation from one objective to another with only fine focus adjustment required to focus the image.

Resolution – ability of a lens to distinguish and separate fine detail.

Stage – platform on which slides are supported for viewing.

Stage controls – allows movement left to right and backwards and forwards.

Sub-stage iris diaphragm – controls the amount of light entering the specimen.

Total magnification – magnification of the objective x magnification of the eyepiece.

Turret or nosepiece – holds the objectives.

Working distance – is the distance between the front lens of the objective and the specimen when it is focused. As magnification increases the working distance decreases.

References:

[1] WA Department of Education, 2010 *Science Laboratory Manual: Biology Techniques*, p 49. ©Department of Education (WA) (with permission)

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STANDARD OPERATING PROCEDURE:

Preparing animal and plant cell slides

Note: To be undertaken only by trained personnel in conjunction with a current Safety Data Sheet (SDS) and site-specific risk assessment.

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1. Introduction

The examination and comparison of plant and animal cells is a hands-on activity suitable for junior and senior secondary science students. Many cells are almost transparent under the microscope and the use of simple stains allows the cells and some of their structures to be easily visible.

A wet mount of an onion membrane is used to represent plant cells and is stained with an iodine stain that reacts with any starch present to produce a blue/black colour. A cell smear prepared from fresh meat purchased from retail stores is used to represent animal cells that are stained with methylene blue stain. Methylene blue is a basic dye that is used to stain animal cells making the nucleus more visible.

Cell type	Specific features	Some common features
Animal Cell	No cell wall Irregular shape One or more small vacuoles	Cell membrane Nucleus Cytoplasm Mitochondria Ribosomes Endoplasmic reticulum
Plant Cell	Cell wall Fixed shape Chloroplasts Large central vacuole	

2. Context

- These instructions are for the use of science teachers, technicians and secondary school students who are under the direct supervision of a teacher.
- Do not make up staining solutions for the first time without seeking practical advice from an experienced colleague.

3. Safety notes

- Science ASSIST does not recommend the use of cells harvested from humans.
- Only science technicians or teachers should prepare the staining solutions.
- A chemical risk assessment should be performed before preparing solutions of iodine and methylene blue. Avoid breathing dust, vapours or mist. These chemicals are toxic when ingested and can cause skin and eye irritation. Consider if anyone is allergic to iodine.
- Pregnant women should take particular care to avoid exposure to iodine.
- Sensitive individuals may show symptoms of Iodism on exposure to small amounts of iodine.

- Students should take care when using glass slides as breakage may occur and small chips can be sharp and cause cuts.
- Ensure students demonstrate responsible behaviour while using sharp knives and other dissecting instruments.

4. Regulations, licences and permits

None.

5. Equipment

Onion cell slide

- Light microscope
- 0.01M Iodine stain in a dropper bottle – teacher or technician to prepare.
- A fresh onion
- Glass microscope slides and No 1 glass or plastic cover slips
- Paper towel or tissue
- Forceps
- PPE including gloves and safety glasses

Animal cell slide

- Light microscope
- 0.15% Methylene blue stain in a dropper bottle – teacher or technician to prepare.
- Clean sharp knife
- One 2 cm cube of fresh beef or a sheep kidney purchased from a retail store.
- Glass microscope slides and No1 glass or plastic cover slips
- Paper towel or tissue

Personal Protective Equipment (PPE)

- Gloves and safety glasses

6. Operating procedure

Onion cell slide

1. Prepare a 0.01M Iodine stain two days prior to class. In an operating fume cupboard or well-ventilated area, weigh 15 g potassium iodide and dissolve in 100 mL of distilled water. Add 3 g iodine crystals and stir to dissolve. Iodine dissolves quickly in concentrated potassium iodide solution. When dissolved, make up to 1 L with distilled water. Store in a dark bottle in a cool area. Label and aliquot into class sets of amber dropper bottles.
2. A teacher or technician should prepare onion sections by cutting the onion into quarters and distributing to students.
3. Divide the onion into its fleshy layers.
4. Using forceps, gently peel back a section of thin epidermis from the concave underside of the layer as shown in Figure 1. Alternatively break the onion layer toward the shiny side and gently peel the two pieces apart revealing the transparent layer of epidermis.

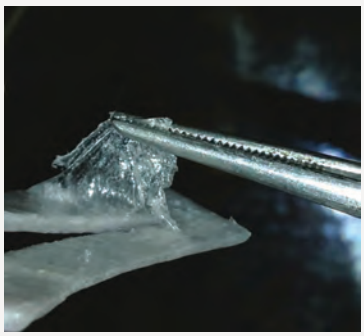


Figure 1: Peeling a section of onion epidermis

5. Carefully place the epidermis in a **single flat layer** on the centre of a microscope slide. Note that the epidermis is very thin, easy to tear, wrinkle and can fold onto itself.
6. Place 1–2 drops of Iodine stain on the epidermis and spread it out carefully with forceps if folded. Put one edge of a cover slip on the slide to one side of the stain and slowly lower it by means of a mounting needle or toothpick. Make sure there are no air bubbles. See the following references below [1,2,3.]
7. Remove excess stain by touching the edge of the cover slip with a tissue or paper towel.
8. Examine cells under the microscope using the 4x objective. When focused, view at high power (x400 magnification) to make observations of cell size, shape and visible cell structures. Students can draw a labelled diagram.
9. Figure 2 shows a typical plant cell structure.
10. Figure 3 shows onion epidermis stained with Iodine stain at 100x magnification. There are regularly shaped cells lying side by side in a pavement-like arrangement. The cells have a distinct cell wall, cell membrane, nucleus, cytoplasm and vacuole present in the centre of each cell.
11. Remove cover slip and dispose in sharps bin. Wash slide in soapy water and rinse well

Figure 2: Labelled plant cell

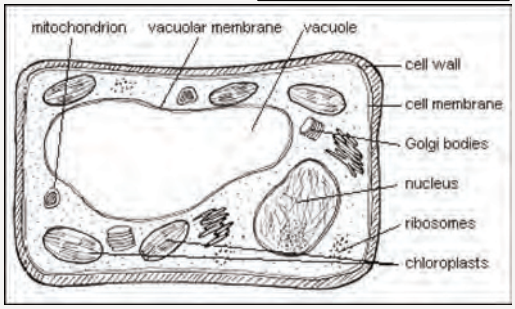
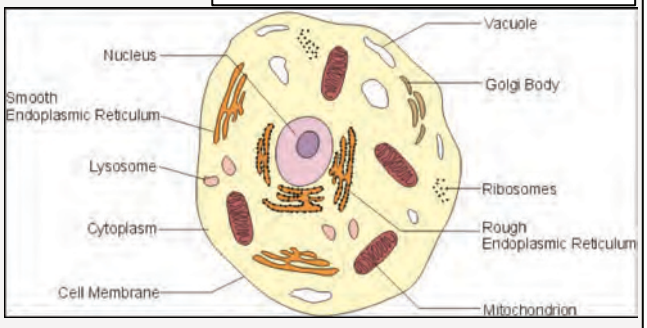


Figure 3: Onion epidermis at 100x magnification. Iodine stain.

Animal cell slide

1. Prepare a 0.15% methylene blue stain prior to class. Weigh 1.5g methylene blue powder and dissolve in 100mL of ethanol and make up to 1L with distilled water while stirring. Methylene blue can also be made up in water, however, it dissolves quickly in ethanol. Label and aliquot into class sets of dropper bottles.
2. If using a kidney cut the organ open with a clean sharp knife. Take a glass microscope slide and touch the surface of the cut area with the microscope slide.
3. Add one drop of the methylene blue stain to the microscope slide.
4. Gently place a cover slip over the stained smear by putting one edge of the cover slip on the slide to one side of the stain and slowly lower it by means of a mounting needle or toothpick. Should be free of air bubbles.
5. Remove excess stain by touching the edge of the cover slip with a tissue or paper towel.
6. Examine under the microscope using the 4x objective. When focused, view at high power (x400 magnification) to make observations of cell size, shape and visible cell structures. Students draw a labelled diagram. Look for epithelial cells.
7. If using a piece of red meat cut along the grain line with a clean sharp knife. Take a glass microscope slide and touch the surface of the cut area with the microscope slide. Repeat steps 3-6. This time look for bundles of skeletal muscle fibres and individual fibrils. They are striated in appearance. There may also be some spindle shaped smooth muscle cells from blood vessels with a distinct central nucleus
8. Figure 4 shows a typical animal cell structure.
9. Figure 5 shows and epithelial cell stained with methylene blue stain at 400x magnification. At this magnification, flat, irregular shaped cells with thin cell membrane and nucleus can be seen. The cells do not have a cell wall
10. Figure 6 shows animal cells from a beef sample stained at 400x magnification. At this magnification, striated muscle fibres and some individual fibrils can be seen along with cell walls, cytoplasm and some nuclei.
11. When finished, remove the glass cover slip and dispose in sharps bin. Wash slide in soapy water and rinse well.

Figure 4: Labelled animal cell. Source: http://www.oum.ox.ac.uk/thezone/animals/life/respire1.htm



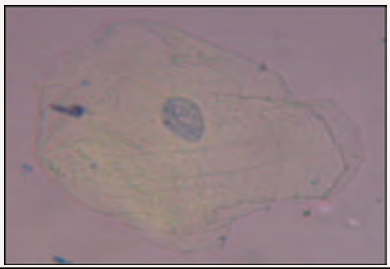


Figure 5: Cell smear showing an epithelial cell at 400x magnification. Methylene blue

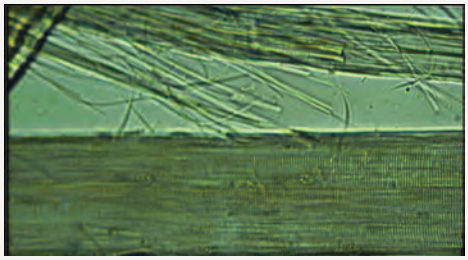


Figure 6: Cell smear showing striated muscle fibres at 400x magnification. **Source:** http://www.aps.uoguelph.ca/~swatland/ch5_0.htm

7. Trouble shooting/emergencies

- First aid: See latest SDS for more detailed information
  - **If swallowed:** Do not induce vomiting. Rinse mouth with water, and then give water to drink. Seek urgent medical attention.
  - **If in eyes:** Hold open and irrigate with copious quantity of water for at least 15 minutes. Seek medical attention.
  - **If on skin/clothes:** If spilt on skin or clothes quickly wipe off with a dry cloth to absorb as much liquid as possible. Remove contaminated clothes and drench the area with excess water under a safety shower. Seek medical attention.
  - **If inhaled:** Remove to fresh air and seek medical attention if symptoms persist.
  - For further advice contact the Poisons Information Centre on 131 126.

8. Waste disposal

- Animal meat should be wrapped and placed in the regular waste for disposal.
- Microscope slides could be washed and reused or disposed of with broken glass, and coverslips should be disposed of with broken glass See Science ASSIST AIS: Lab glass and porcelain disposal

9. Related material

- Manufacturer's Safety Data Sheet
- Science ASSIST SOP: Use and care of the compound light microscope
- Science ASSIST AIS: Lab glass and porcelain disposal
- Risk Assessment.

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STANDARD OPERATING PROCEDURE:

Demonstrating the Van de Graaff generator

Note: To be undertaken only by trained personnel in conjunction with a site-specific risk assessment.

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1. Introduction

The Van de Graaff generator (VDG) is a device, which produces and stores a large electrostatic charge on a metal dome. It consists of a base, an insulating column (usually Perspex), a large metal dome and a smaller discharge sphere on an earthed and insulated handle. A nylon pulley in the base drives a belt (rubber or latex) over a metal pulley within the top dome. Negative ions jump from the belt to the earthed comb at the lower pulley and the positive charge is carried to the top where it is collected by the top metal pulley and comb and transferred to the dome. This charge can reach very high voltages but with low current and will discharge back to earth by sparking to the discharge sphere connected to earth. In ideal conditions, it can produce a voltage of 400,000 volts with a short circuit current of 20 micro-amps. The voltage of a spark is approximately 30,000 volts per centimetre. It can be used to demonstrate the presence and nature of static electricity and electric fields.

2. Context

- The Van de Graaff generator is to be used for demonstration purposes only.

3. Safety notes

- Ensure that any person, who may be pregnant, has a heart condition, metal plates, a pacemaker, cochlear implant(s), electronic insulin pump or similar electronic device is not in the vicinity of the Van de Graaff generator.
- Ensure that people in close vicinity to the unit remove personal metal objects or jewellery.
- Ensure that the earthing sphere remains connected to the unit and is between the operator and the main dome at all times.
- Always hold the discharge sphere support rod by the insulated handle and have the earth cable attached to the base. Do not touch the metal base of the unit while it is running.
- Approach the machine with caution at all times. The spark generated has a high voltage but a low current. A spark discharge to the hand, although harmless, is a little painful.
- Do not operate the generator near flammable liquids. Keep any computer, mobile phones or similar device away from operating unit.
- After switching off, always earth the large dome by touching the dome with the discharging sphere to discharge it before touching it with the hand.
- Avoid using unit on metal-framed tables/benches.
- **Science ASSIST strongly advises against the forming of a human chain in demonstrations using the Van de Graaff generator.**

4. Regulations, licences and permits

The Van de Graaff should have a regular electrical Portable Appliance Test.

5. Equipment

- Van de Graaff generator
- Hair dryer may help to dry the column in humid conditions
- Attachments i.e. lock of hair, small aluminium pie plates, polystyrene cup and beads
- Insulating platform such as block of polystyrene (e.g. a lid of a polystyrene crate), sturdy plastic crate or rubber mat
- Wooden ruler such as a metre rule (without metal tips)

6. Operating procedure

General instructions:

- Be familiar with the safety notes and do not allow students to operate the generator unsupervised.
- Ensure that any person with who may be pregnant, has a heart condition, metal plates, a pacemaker, cochlear implant(s), electronic insulin pump or similar electronic devices is not in the vicinity of the Van de Graaff generator.
- Remove any jewellery or metal objects if in close proximity to the operating unit.
- Place the Van de Graff generator at least 1.5 metres from walls, plumbing and light fittings, away from computers, mobile phones and similar equipment. Best results are obtained in a darkened laboratory.
- Wipe the dome, the sphere and the Perspex column with a clean dry cloth to remove any dust particles. Occasionally polish the dome with metal polish.
- Allow time for the unit to warm up. The unit operates best on a dry day. A hair dryer may help to reduce the humidity of the air in the column and dome.
- Plug in the machine and if it has a variable speed (most new models do), adjust the speed to the maximum.
- Touch the dome with the discharging sphere and build up the capability of the machine in the following manner:
    - Position the discharge sphere just a few millimetres away from the charging dome and wait till there is a regular steady discharge of approximately one per sec.
    - Increase the distance and repeat the procedure.
    - The rate of discharge is a better indicator of the performance of the machine than the optimum voltage, because the optimum voltage is affected so greatly by the conditions of the atmosphere.
    - Leave the machine running, discharging regularly as described above, for 5–10 minutes before doing any demonstrations.
- When finished earth the large dome and hold the sphere on the dome and switch off the Van de Graaff generator before touching it.
- Wipe the dome before storing in a dry cupboard protected from light or cover with a dark plastic bag.

(Operating procedure cont…)

Demonstrations:

When ready for operation there are a range of demonstrations that can be performed. Here are a few examples:

(Note: Ensure that the dome is 'discharged' prior to adding any attachments, attach the item to the top of the dome prior to switching the unit on)

- Flying pies – stack some small aluminium pie plates onto the top and watch them fly off one by one.
- Sit a polystyrene cup full of polystyrene balls on top on the unit and watch them fly off.
- Light a fluorescent tube – hold the tube with a non-conducting holder (glove) and bring one end of the tube towards the dome. The glass surface of the tube nearest the dome acquires a negative charge by induction. The charge builds up on the glass surface to discharge intensity. As discharge occurs negative charges run through the entire tube lighting it up for the duration of the discharge.
- Hair-raising – attach a lock of hair and watch it rise. Discharge it with the sphere and watch the hair fall.

If a student or other volunteer is to be used to see the hair raising demonstration a risk assessment should be performed.

The recommended procedure is:

a) Ask for a volunteer – a person with fine hair gets the best results.
b) Check with the person that they are not pregnant, have a heart condition, metal plates, a pacemaker, cochlear implant(s), electronic insulin pump or other similar electronic devices.
c) Ask the person to take off any jewellery or metal objects.
d) Ensure that the generator is turned off and discharged before approaching.
e) Have the person stand on the insulating platform.
f) Get the person to run their hands through their hair and shake their hair.
g) Have the person place both hands on the large dome BEFORE switching on the Van de Graaff generator.
h) Switch on the generator, being careful to keep below the dome level so that the dome does not discharge.
i) Hair should be seen to rise.
j) When finished, earth the large dome by touching the discharge sphere to the dome on the opposite side to their hands and hold the sphere on the dome and switch off the Van de Graaff generator.
k) Discharge the person by touching them with a wooden ruler.
l) Ask the person to take their hands off the dome and jump from the insulating platform onto the floor, hitting the floor hard. The person can discharge any remaining static by shaking their hands.

Science ASSIST strongly advises against the forming of a human chain in demonstrations using the Van de Graaff generator.

7. Trouble shooting/emergencies

- Do not operate the generator in humid conditions.
- Allow time for the unit to warm up. A light or hair dryer can be used to warm and dry the air inside the unit.

- Wipe dome and insulating tube with a soft cloth before use.
- Avoid getting fingerprints on the insulating tube.
- Polish dome periodically with metal polish (such as Silvo® or similar).
- Remove the belt for storage. The nylon or rubber belt loses its efficiency if it is stretched for long periods of time. (See below for link to manufacturer's instructions).
- Remove and clean the belt with warm soapy water periodically.
- Check belt for wear or fraying.
- Adjust the tension on the belt, if this an option.
- Lubricate pulleys occasionally.
- Clean top metal pulley with alcohol.

8. Waste disposal

- Not applicable

9. Related material

- Manufacturer's instruction manual.
- Risk Assessment
- Van de Graaff information sheet http://www.iecpl.com.au/z_pdfs/em4133-101.pdf
- Van de Graaff attachments EM4144-001 http://www.iecpl.com.au/z_pdfs/em4144-001.pdf
- Van de Graaff belt Information http://iecpl.com.au/z_exp/em4137belt.pdf

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STANDARD OPERATING PROCEDURE:

Handling sealed radioactive sources: alpha, beta and gamma

Note: To be undertaken only by trained personnel in conjunction with a site-specific risk assessment. Contact your state or territory radiation regulatory authority for the interpretation of regulatory matters. For contact details see:
http://www.arpansa.gov.au/Regulation/Regulators/index.cfm

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1. Introduction

It is desirable for students to develop a respect for the safe and proper use of radiation. The most common radioactive materials used in school science practical work are sealed radioactive sources. These sources are provided as small sealed, circular clear plastic discs approx. 2cm in diameter. Sources provided to schools produce ionising radiation and are generally supplied as an alpha-emitting americium-241 (Am-241) or polonium-210 (Po-210), a beta-emitting strontium (Sr-90) and a gamma-emitting cobalt (Co-60). The potential for harm from these sources is very low.

The Australian Radiation Protection and Nuclear Safety Agency (ARPANSA) is the Australian Government's primary authority on radiation protection and nuclear safety. ARPANSA works with state and territory regulators to promote national uniformity of radiation protection. For details of your state/territory regulator see
http://www.arpansa.gov.au/Regulation/regulators/index.cfm

2. Context

- These instructions are for the use of experienced science teachers, technicians and students over the age of 16, who are under the direct supervision of a teacher.

3. Safety notes

- All persons handling radioactive sources must be trained in their use. Class work with radioactive sources for students in year 10 and under is restricted to teacher demonstrations. Students should be kept at least 2 metres away from these sources during demonstrations.
- For detailed information regarding practice-specific guidance on best practice see the ARPANSA *Safety Guide for the Use of Radiation in Schools* (2012) see http://www.arpansa.gov.au/pubs/rps/RPS18.pdf. It is recommended that persons handling sealed radioactive sources be familiar with this document and a **printed hard copy of this resource is recommended for reference.**
- Protection from radioactive sources can be achieved by:
  - reducing the time of exposure
  - increasing the distance from the source
  - increasing the shielding between persons and the source.

- Radioactive sources should therefore:
  - be handled for the shortest time possible (recommended less than 2 minutes)
  - **never** be picked up with fingers, but always with long tongs
  - be stored at least 2 metres (ignoring walls) from a place where anyone spends extended periods of time
  - be stored within lead lining in a metal box in a locked secure area in the science department away from highly flammable material
  - See the supplementary information on storage of radioactive sources (see p5).

4. Regulations, licences and permits

The ARPANSA *Safety Guide for the Use of Radiation in Schools (2012)* sets out the legal requirements for schools to use ionising radiation sources. For detailed information regarding regulations see the ARPANSA *Safety Guide for the Use of Radiation in Schools* (2012). To comply with these regulations your school will need:

- a responsible person who ensures that all legal and safety requirements are met, usually the school principal or the head of your state/territory education department
- a school radiation supervisor who will have responsibility for the safe storage, use and monitoring of radiation sources. See the supplementary information on Radiation supervisor appointment and responsibilities (see p10)
- a set of local rules for your **specific school site** that document where the radiation sources are stored, where they can be used, who can use them and where the use log is kept. See the supplementary information on the use of sealed radioactive sources (see p6)
- to contact your state Radiation Regulatory Authority to ascertain whether you need a radioactive licence. Consult the ARPANSA website (www.arpansa.gov.au) for the most up to date list of regulators.

5. Equipment

- Radioactive sources, eg americium-241/polonium-210 (alpha); strontium-90 (beta); cobalt-60 (gamma)

Disc source – caesium-137

Disk sources in storage box

Images: ARPANSA. 2012. *Safety Guide for the Use of Radiation in Schools (2012)* http://www.arpansa.gov.au/pubs/rps/RPS18.pdf CC BY NC 3.0 AU

- Geiger Müller (GM) Tube and Electronic Counter
- Long forceps or tongs
- Various radioactive absorption materials such as paper, plastic, aluminium and lead in different thicknesses.

6. Operating procedure

1. Follow the local rules for **your specific school site.**
2. Sources must not be handled by persons with wounds on their hands
3. Refer to ARPANSA *Safety Guide for the Use of Radiation in Schools* (2012) page 22 for concerns regarding expectant mothers
4. Sources should not be left unattended
5. Carry each source in its storage container and keep it there until it is required. Do not handle the container for longer than necessary.
6. Use only one source at a time in any one investigation.
7. Handle the source with a long tongs, which keep the fingers at least 10 cm away.
8. Keep the source at least 20cm away from your eyes
9. Plug in GM-tube and electronic counter and operate reset switch to set counter to zero. See manufacturer's instructions.
10. Measure background radiation away from the sources for at least 2 minutes. This is normal everyday radiation which is always present.
11. Pick the source up with the long tongs.
12. Hold the GM-tube 5cm above the source and measure radiation.
13. Place absorption materials between the source and the GM-tube to observe the effects on the radiation count.

    Alpha particles – stopped by sheet of paper or surface layers of skin.

    Beta particles – stopped by a few millimetres of aluminium or 1-2 centimetres of plastic.

    Gamma rays – almost completely stopped by about 1 metre of concrete or about five centimetres of lead. Most will pass through the human body.
14. Complete the investigation in the shortest time possible, consistent with good results.
15. Return the source to its normal container immediately after completing the investigation.
16. The member of staff in charge is to check all sources for signs of damage on return.
17. Immediately report to the member of staff in charge any event in which a source cannot be accounted for, is dropped or may have been damaged.
18. Always wash hands thoroughly immediately after working with any radioactive source.

7. Trouble shooting/emergencies

- Make sure that the Geiger counter you are using is set up and working properly and will detect the radiation being tested. Some GM-tubes used in schools will not detect alpha radiation, as their end-window is too thick. Am-241, which is routinely used in schools, also emits a gamma ray as well as an alpha particle. The gamma ray will penetrate the thick-end window of the GM-tube. The result from the emission of the gamma ray can be interpreted

as the emission of an alpha particle if the GM tube only responds to beta and gamma radiation.

- If any source is dropped and damage to the source is suspected, follow the instructions in the supplementary information on conducting an inspection, wipe test and contamination check (see p8)

8. Waste disposal

- Radioactive sources can last for many years depending upon their half-life. In the unlikely event that a source is dropped and damaged or fails the wipe test, it will need to be disposed of.
- See the supplementary information on disposal of sealed radioactive sources (see p9) •

If you are unsure of how to dispose of your radioactive source, you should contact your radiation regulatory authority for advice about disposal

9. Related material

- Local rules for your specific school site
- Risk assessment.
- *Safety Guide for the Use of Radiation in Schools (2012)*. ARPANSA–Part1,Ionising

  Radiation': in particular see:
  - Model Local Rules: Annex 5, pp 50–54.
  - Model Risk Assessment: Annex 4, pp 37–49.
- Manufacturer's Instructions for Electronic Counter and GM-Tube. (e.g. http://www.iecpl.com.au/z_pdfs/ap1884-001+002.pdf)

References:

ARPANSA. 2012. *Safety Guide for the Use of Radiation in Schools (2012)* http://www.arpansa.gov.au/pubs/rps/RPS18.pdf CC BY NC 3.0 AU http://creativecommons.org/licenses/by-nc/3.0/au/

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SUPPLEMENTARY INFORMATION:

Storage of radioactive sources

Radioactive sources must be stored to minimise the risk of exposure to people in the surrounding area when not in use, as well as to provide a secure location to reduce the risk of theft or vandalism.

In consideration of **minimising the risk of exposure** to people in the surrounding area, the sources need to be shielded in lead to absorb the radiation. Very often they are delivered in a lead lined metal container, which is a suitable one for radioactive sources to be stored. Some of the gamma rays will still penetrate lead and for this reason the location of the store needs to be at least 2 metres in a straight line (ignoring walls) from a place where anyone spends extended periods of time.

In consideration of the **possibility of a fire**, the sources need to be housed in a strong steel container (such as a tool box) that should be recognisable after a fire or other such major incident and stored away from flammable materials, so that firefighters do not have to contend with both radioactive sources and flammables. This means that they should not be stored in the chemical storeroom, which will also reduce the likelihood of corrosion. Often they will be stored in the preparation room giving consideration to relevant distances.

In consideration of the **risk of theft or vandalism**, the sources need to be in a secure store, such as the steel container being kept in a fixed, locked cupboard or drawer, making sure there is no access via an adjacent cupboard or drawer, or in a lockable, steel cupboard that is securely fixed. Some schools use wall safes for this purpose. The key to this locked store should be unique and also kept secure.

In order to **warn others in the area of the presence of radioactive sources**, the outside of the containers should be clearly labelled with the contents and the international radiation symbol (also called a trefoil sign) with the wording ‘radioactive materials’. The outside of the cupboard, drawer, wall safe (and the separate metal container, if used) should also be labelled with the trefoil warning sign. The text is optional, but is useful for those unfamiliar with the symbol. Further signage of room doors is not required as it may draw unwanted attention to their presence, however this can be a school-based decision.

Summary for storage of radioactive sources:


Radioactive Materials

Radioactive sources must be stored

- in a lead lined container
- so that the outside of the container is clearly labelled with
  - the contents
  - a trefoil warning sign and
  - the wording ‘radioactive materials‘
- at least 2 metres in a straight line (ignoring walls) from a place where anyone spends extended periods of time
- in a strong steel container
- away from flammable materials and the chemical store room
- in a secure location, with a unique key for access

Use of sealed radioactive sources

Rules must be established for the use of radioactive sources to minimise the risk of exposure to people using the sources.

Records must be kept to ensure that the type and whereabouts of the sources is known at all times, and that inspections, wipe tests, contamination checks and disposal of sources is documented.

Rules

Local rules should be written for **your specific school site**. They are written to ensure that radiation doses and risks of contamination are minimised.

Annex 4 of the *Safety Guide for the Use of Radiation in Schools (2012)* contains example details of the model local rules that can be adapted to meet the needs of **your specific school site:**

Local rules for the **use** of radioactive sources must contain:

- Name of school
- Name of responsible person
- Name and contact details of radiation supervisor
- Location of secure store for radioactive materials
- Laboratories/rooms where radioactive sources are used
- Location of the following documentation
    - Radioactive source history
    - Use log for radioactive sources
    - Monitoring record for radioactive sources and store
    - Contact details of your Radiation Regulatory Authority

Local rules for **science department staff** which detail the safety procedures and administrative requirements should be developed for your **specific school site**: see example set in *Safety Guide for the Use of Radiation in Schools (2012)* pp 50-51

Local rules for **staff and supervised students in years 11 and 12** see example set in *Safety Guide for the Use of Radiation in Schools (2012)* p 52

Record keeping: radioactive source history

An inventory of all radiation sources should be kept and the following **individual** records need to be kept for **each** radioactive source:

- their purchase.
    - a copy of the purchase order/invoice/receipt
    - the radioactive source history including, where possible, the:
        - unique name or reference number
        - radionuclide or chemical name
        - original activity
        - delivery date, supplier and manufacturer.
- their use, inspections, wipe tests and contamination checks.
- their disposal.

Record keeping: use log for radioactive sources

Each time a radioactive source is accessed it needs to be recorded in a 'use log'. The following events constitute a use:

- **a security check for the presence of the sources** (required at 'appropriate intervals').In most schools where they are used over a period of a few days, and then not for another year, a year would be an appropriate interval.
- **when used in classroom investigations**
- **inspections, wipe tests and contamination checks** (see supplementary information Conducting an inspection, wipe test and contamination check, p8)

Note: Managers and staff in schools and colleges should take all possible steps to ensure that loss of a radiation source cannot happen. However, in the event that a source is missing, the radiation supervisor should check that it has not been:

- returned to the wrong store
- left inside the piece of equipment within which it was last used
- temporarily removed to another area, or
- placed with waste for disposal.

All incidents or accidents involving radiation sources, such as a situation where a radiation source cannot be found, must be reported to the relevant radiation regulatory authority. In the first instance, this will usually be the Principal. If you suspect that someone has removed the source unlawfully from the premises, the Principal, in consultation with the radiation regulatory authority, will need to inform the police.

Record keeping: monitoring record for radioactive sources and store

Regular inspections and wipe tests should be conducted and the results entered into the use log. Instructions for conducting an inspection, wipe test and contamination check are on p8.

Regular inspections, wipe tests and contamination checks ensure that the mechanisms for preventing dispersal of radioactive materials are functioning as intended. You should carry out an inspection and wipe test once a year on each source kept in the radioactive materials store, including stock bottles of radio chemicals if applicable. A simple record such as pass or fail in the use log will be necessary for each source.

Contamination checks should be conducted anywhere that there is a possibility that radioactive materials may have been deposited on surfaces, e.g. containers, radioactive store or when a source has been dropped.

Summary of the use of sealed radioactive sources:

- Local rules should be written for **your specific school site**
- An inventory of all radiation sources should be kept
- Each time a radioactive source is accessed it needs to be recorded in the use log
- Regular inspections, wipe tests and contamination checks should be conducted and the results entered into the use log

Conducting an inspection, wipe test and contamination check

Good light is needed to conduct a regular, visual inspection of a radioactive source which should be held at least 20cm away from your eyes. All scratches and any signs of deterioration particularly to the seals need to be recorded. A routine check and wipe test should be carried out annually. The wipe test is used to check for the unlikely event of leaks of radioactive material from a sealed radioactive source. If there is any damage the source should be withdrawn from use and disposed of appropriately.

A contamination check should also be conducted anywhere that there is a possibility that radioactive materials may have been deposited on surfaces, e.g. containers, radioactive store or when a source has been dropped.

Conducting an inspection, wipe test and contamination check:

An annual set of monitoring checks needs to be conducted using the following procedure:

- Wear PPE: a disposable apron, safety glasses and disposable gloves.
- Carry out the work on at least two sheets of newspaper.
- With all sources at least 2 metres away and using the GM-tube, the background radiation should be measured for 2 minutes. This background count is recorded.
- **Inspect and test one source at a time**, keeping other sources in their normal containers at least 2 metres away.
- Using long tongs pick up the sealed source and carry out a visual inspection keeping the source at least 20cm away from your eyes. A mirror could be placed on the work surface so that the window side can be viewed facing away from the eyes. Record any small blemishes or scratches for future reference.
- **Wipe test:** Using a clean, dry, paper tissue, gently wipe across the window side of the source.
- Move the source at least 2 metres away and measure the radioactivity 2mm from the wipe area on the tissue paper with a GM-Tube for 2 minutes. The count is recorded.
- Repeat this procedure with other sources using new tissue paper and record the results
- Providing the tissue count was less than 1.5 times the background count, the source has passed the wipe test. A record of the annual results needs to be kept for comparison and any noticeable changes need investigating for leaks or expiry.
- **Contamination check:** The GM-tube also needs to be passed over the interior of the lead lined storage box or metal box to check for any contamination. If the count is 1.5 times higher than local background radiation then the storage areas need to be carefully and thoroughly wiped out with tissues soaked in strong detergent. Shelves and cupboards need to be checked also. The origin of the contamination needs to be defined.
- If the source appears to be damaged or fails its wipe test, note the action taken. Keep any such source inside its normal container and place it in a strong plastic bag. Seal and suitably label this bag and keep it in the usual store. Consult the radiation regulatory authority as professional disposal may be required.
- When work is complete, place disposable materials that were used in a strong plastic bag, which is tied for disposal with normal garbage.
- Always wash hands thoroughly immediately after working with any radioactive source.

Disposal of sealed radioactive sources

A sealed radioactive source with an activity below the exemption level as specified in Table 3 in the *Safety Guide for the Use of Radiation in Schools (2012)* may be disposed of without regard to its radioactive properties. However it is important to check the current requirements in your jurisdiction with your radiation regulatory authority.

Radionuclide	Max activity of sealed radioactive sources in NDRP for use in schools (kBq)	Exempt activity in NDRP (kBq)
Cobalt-60	200	100
Strontium-90	80	10
Caesium-137	200	10
Polonium-210	–	10
Radium-226	20	10
Americium-241	40	10

Table 3: Sealed radioactive sources for use in schools and colleges

Exempt radioactive sources do not need to be treated as radioactive for disposal and can therefore go out with general garbage when they are no longer needed. There may however, be requirements for its chemical properties. If you are disposing of a source that has decayed to an activity below the exemption level for regulatory control, you should permanently remove or obscure all markings relating to its previous radioactive status.

If you wish to dispose of a radioactive source with an activity above the exempt level, or if you wish to dispose of any other type of radiation source, such as an X-ray unit or a Crookes tube or if you are unsure of how to dispose of your radioactive source, you should contact your radiation regulatory authority for advice about disposal.

Summary of disposal of sealed radioactive sources:

- A radioactive source with an activity below the exemption level:
  - Should have all markings relating to its previous radioactive status permanently removed or obscured and then
  - Can be disposed of with the general garbage
- A radioactive source with an activity above the exemption level:
  - Contact your radiation regulatory authority for advice about disposal

Radiation supervisor appointment and responsibilities

Each science department should have a named radiation supervisor nominated by the Principal. The radiation supervisor would probably be a member of the teaching staff such as the Head of the science department or the Head of Physics. The Principal/education department should ensure that the Radiation Supervisor is competent and is fully aware of his or her duties. The radiation supervisor should understand the basic principles of radiation protection and the relevant requirements of the local radiation protection legislation. They should be fully aware of the hazards and control measures associated with each radiation source in his or her care.

A school radiation supervisor has responsibility for the safe storage, use and monitoring of radiation sources. The radiation supervisor should liaise with the Principal/education department regarding the development and agreement of written 'local rules' for your specific school site and ensure that all staff and permitted students who handle radioactive materials should be familiar with, and have easy access to, the 'local rules'. The Radiation Supervisor should be satisfied that all persons involved are informed and trained to a level which enables them to carry out procedures safely.

Summary of Radiation supervisor appointment and responsibilities:

The radiation supervisor should ensure that:

- *they are fully aware of the hazards and control measures associated with each radiation source in his or her care*
- *all such work is carried out in accordance with the local rules*
- *regular monitoring is carried out on radioactive sources and their containers*
- *all records required are accurate and kept up to date*
- *they know what to do in an emergency*
- *there are written standard operating procedures for work with radioactive materials*
- *all radiation sources have been replaced in the store after use*
- *relevant checks have been conducted*
- *correct disposal procedures are followed*

The radiation supervisor should provide appropriate instruction and training in:

- *security and storage arrangements*
- *record keeping (inventory of sources and the use log)*
- *safe handling of each type of radiation source*
- *correct use of associated equipment, particularly that used for monitoring purposes*
- *action to take if a radioactive source is dropped or a spill occurs*
- *when to seek help and advice from the radiation supervisor*

Other aspects

This document summarises the main requirements for the safe storage, use and disposal of sealed radioactive sources. There are many other aspects that are addressed in the *Safety Guide for the Use of Radiation in Schools (2012)* such as radiation and its properties, choices of radiation sources; cleaning up a spill and example Risk Assessments.

References

ARPANSA. 2012. *Safety Guide for the Use of Radiation in Schools (2012)* http://www.arpansa.gov.au/pubs/rps/RPS18.pdf CC BY NC 3.0 AU http://creativecommons.org/licenses/by-nc/3.0/au/

History of reviews

Date	Version Number	Notes
Sept 2014	Version 1.0	
Mar 2015	Version 1.1	Corrected incorrect terminology from gamma **particle** to gamma **ray**

ASSIST AUSTRALIAN SCHOOL SCIENCE INFORMATION SUPPORT FOR TEACHERS AND TECHNICIANS

STANDARD OPERATING PROCEDURE:

Use of lasers in schools

Part 1: Laser pointers

Note: To be undertaken only once a site-specific risk assessment has been conducted.

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1. Introduction

The word LASER is an acronym for **L**ight **A**mplification by **S**timulated **E**mission of **R**adiation, which describes the process by which lasers generate light (electromagnetic radiation). There are many applications of the use of lasers in schools, including as a white board pointer, a pointer for astronomy observations, the teaching of optics, ray paths, refraction and diffraction, the modulation of laser beams to carry signals, and the applications in fibre optic communications.

As laser technology is very widely used in industry, medicine and in everyday life, it is desirable that students learn to safely and responsibly use and manage lasers in a supervised school science environment.

The most common laser pointers emit red light (635 nm wavelength). Units emitting green light (523 nm) are also available and sometimes used as pointers in astronomy.

Due to the high portability, ready availability and low cost of pointer lasers, there is greater potential for their misuse than for conventional gas lasers. Consequently, there are stricter controls regarding their permitted power output.

2. Context

- This document summarises the guidelines for the storage and use of laser pointers in Australian primary schools, secondary schools and colleges.
  - **This is <u>Part 1 of a three part document</u> addressing three separate applications:**
    - **Part 1**: **Laser pointers** (solid state diode lasers)
    - **Part 2: Bench lasers** (as used in Physics investigations, usually He-Ne gas lasers)
    - **Part 3: The construction of laser equipment**

3. Safety notes

- Laser pointers should be stored in a secure locked cupboard (e.g. in a storeroom) to prevent unsupervised student access.
- Do not leave a laser unattended.
- Avoid shining a laser beam at or near persons.
- Despite the low power rating i.e. Class 1 or 2 lasers, eye damage is possible if magnifying viewing instruments such as lenses, microscopes, binoculars or telescopes are used. Do not use these to view a laser beam.

- Laser light differs from light emitted from conventional sources. It is monochromatic, coherent and is collimated (refer to definitions below). This last feature is very important in considering the safety of lasers.

  Definitions of properties of laser light:

Monochromatic	The emitted light is concentrated at a single wavelength
Coherent	The light waves remain in phase as they propagate
Collimated	The light rays are parallel, with very little divergence, so that the intensity is maintained over long distances

- Conventional light sources emit light in all directions, and so the intensity of the light rapidly diminishes with distance from the source (the inverse square law). As laser light is collimated into a beam with very little divergence, the intensity is maintained over long distances. There is then a much greater capacity for laser beams to cause eye damage.
- The Class rating, the appropriate laser symbol and hazard warning statement are to be clearly marked on the instrument. A table of the laser classes is given in the 'Supplementary information' section at the end of this document. If the class of any laser is not clearly identified then it should not be used.
- Laser pointers used in schools should not normally exceed Class 2 and a power rating of 1 milliWatt (mW). At this power any possible eye injury is normally avoided through the human aversion responses (blinking and turning away). Exemptions for higher powered laser pointers are available for specific uses, but these would not normally apply to schools. **Science ASSIST strongly recommends that laser pointers for general school use do not exceed Class 2 (1 mW power output).** As a guiding principle, always use the lowest power laser for any particular purpose.
- Laser pointer purchases should be made from a reputable local supplier who can confirm that the laser has been tested according to Australian Standards rather than from overseas or through the internet. Concerns have been raised regarding lasers purchased from overseas, or through the internet, where there can be a misclassification of the laser. It may be labelled as Class 1 or 2, but may in fact be a higher class when tested according to Australian Standards.
- In addition, although it is illegal to import laser pointers of a power greater than 1mW into Australia without special written approval, there are many countries where the import/export of higher-powered lasers are not regulated. Caution is advised because their low price and common availability overseas means that some higher powered lasers do find their way into Australia, and in some cases into schools.

4. Regulations, licences and permits

- The use of lasers in schools is informed by the *Safety Guide: Use of Radiation in Schools (2012), Part 2: Lasers* http://www.arpansa.gov.au/pubs/rps/RPS18.pdf.

  It is recommended that persons handling lasers be familiar with this document and a **printed hard copy of this resource is recommended for reference.**

  For details of your state/territory regulator see:
  http://www.arpansa.gov.au/Regulation/regulators/index.cfm

- It is unlawful (Australian Customs) to import laser pointers of over 1 mW power without written approval. They are classified as prohibited weapons in the ACT, New South Wales, South Australia, and Victoria, and as controlled weapons in the Northern Territory and Western Australia. They are restricted items in Queensland, and in Tasmania they require a lawful excuse. For further information refer to your state/territory regulator.
- In some circumstances exemptions can be sought for the use of higher powered (Class 3R/ 3A) laser pointers e.g. for use in astronomy. This introduces higher levels of hazard that would require risk assessment and controls beyond the scope of this document.

5. Equipment

- Laser pointer Class 1 or Class 2

6. Operating procedure

1. **Storage**

   Laser pointers should be stored in a secure locked cupboard (e.g. in a storeroom) to prevent unsupervised student access.

2. **Steps before use**

   2.1 Check that the laser pointer is rated at no higher than Class 2, and with a power output not exceeding 1 milliWatt (mW).

   2.2 Check that the laser pointer is appropriately labelled as below:

   Class 1 laser

   

   Class 2 laser

   

   

3. **Use**

   The use of Class 1 or 2 laser pointers does not normally require any specialist knowledge on the part of the operator.

7. Trouble shooting/emergencies

- Not applicable

8. Waste disposal

- Not applicable

9. Related material

- Australian Radiation Protection and Nuclear Safety Agency ‘Safety Guide: Use of radiation in schools Part 2: Lasers. Radiation protection series no. 18’, ARPANSA website, June 2012 http://www.arpansa.gov.au/pubs/rps/RPS18.pdf. This includes guidance on developing a Risk Assessment for the use of lasers in schools (see Section 10).
- Manufacturer’s instructions

- Risk Assessment
- Useful links:
  - Macquarie University 'Faculty of Science Occupational Health and Safety – Lasers' Macquarie University website. (Accessed January 2015). http://web.science.mq.edu.au/intranet/ohs/lasers/. This page contains much useful information including information on laser classifications and laser induction.

    Overview of IEC laser classes. http://web.science.mq.edu.au/intranet/ohs/lasers/documents/classoverview.pdf

    Typical accessible emission limits (AEL) for visible CW lasers. http://web.science.mq.edu.au/intranet/ohs/lasers/documents/classifications.pdf

    Laser Induction: with links to watch the MPG free Faculty Laser Induction Module (PPT, 3.3 MB) plus the Eye Effects Video (46 MB, 5 minutes) and the Laser Safety Video (231 MB, 30 minutes)

References:

Australian Radiation Protection and Nuclear Safety Agency (ARPANSA) 2012. *Safety Guide for the Use of Radiation in Schools (2012)* http://www.arpansa.gov.au/pubs/rps/RPS18.pdf CC BY NC 3.0 AU http://creativecommons.org/licenses/by-nc/3.0/au/

Australian/New Zealand Standard AS/NZS IEC 60825.1:2014. Safety of laser products Part 1: Equipment classification and requirements.

SUPPLEMENTARY INFORMATION:

Classification of lasers

Lasers are classified according to their potential degree of hazard, with the higher numbers indicating higher hazard levels. Older units may be classified under a similar early system, with the comparisons set out in the table below. It is important to remember that laser classification is made on the basis of the *entire* laser product. This means it is possible that a laser product could contain a high power laser internally with the engineering design allowing a lower classification for the unit.

Current classification	Former classification	Notes	Labelling and signage
Class 1	Class 1	Considered safe under foreseeable conditions because of low emission levels or through engineering design. Not harmful to eyes or skin.	
Class 1M		Higher emissions than Class 1, but because of the spread of rays, not capable of causing eye damage. This means the beam can be magnified with the help of optics.	
Class 2	Class 2	Low powered visible lasers (to 1mW power). Not harmful to skin. Eye damage avoided through natural aversion responses (blinking, turning away)	 
Class 2M		Higher emissions than Class 2, but because of the spread of rays, normally safe for viewing with unaided eye. This means the beam can be magnified with the help of optics.	 
Class 3R	Class 3A	For visible wavelengths, up to 5 times the emissions of Class 2 (i.e. up to 5 mW power). Risk of injury from accidental exposure is low.	 
Class 3B	Class 3B	Visible or invisible wavelengths where direct viewing is hazardous to eyes. (i.e. between 5mW and 500mW for visible wavelengths)	 
Class 4	Class 4	High powered lasers capable of causing damage to skin and eyes. (i.e. > 500mW)	 

Lasers in schools

It is expected that in almost all cases, lasers used in schools should be restricted to Class 1 and Class 2 (common outputs of 0.5 to 1 mW). At this level, eye damage from accidental exposure is very unlikely due to the human 'aversion responses' such as blinking and turning away. Lasers classified as Class 3R/ 3A, with power outputs in the 1–5 mW range, may be used subject to gaining school permission and following good safety practices as detailed in Part 2 of this SOP. Lasers of Class 3B and above, and any that emit wavelengths not in the visible spectrum, should not be used in schools. If the class of any laser is not clearly identified then it should not be used.

STANDARD OPERATING PROCEDURE:

Use of lasers in schools

Part 2: Bench lasers

Note: To be undertaken only by trained personnel in conjunction with a site-specific risk assessment. Contact your state or territory radiation regulatory authority for the interpretation of regulatory matters. For contact details see:
http://www.arpansa.gov.au/Regulation/Regulators/index.cfm

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1. Introduction

The word LASER is an acronym for **L**ight **A**mplification by **S**timulated **E**mission of **R**adiation, which describes the process by which lasers generate light (electromagnetic radiation). There are many applications of the use of lasers in schools, including as a white board pointer, a pointer for astronomy observations, the teaching of optics, ray paths, refraction and diffraction, the modulation of laser beams to carry signals, and the applications in fibre optic communications.

As laser technology is very widely used in industry, medicine and in everyday life, it is desirable that students learn to safely and responsibly use and manage lasers in a supervised school science environment.

Bench lasers used in school science are likely to be Helium- Neon gas lasers (Class 2 or Class 3R/ 3A), emitting red light (632.8 nm wavelength). It is recommended that the use of bench lasers **be restricted to Class 3R/3A or below, continuous wave (not pulsed), and emitting visible radiation. Lasers that emit wavelengths that are not in the visible spectrum are not recommended for use in schools.**

2. Context

- This document summarises the guidelines for the storage and use of bench lasers in Australian secondary schools and colleges.
- **This is Part 2 of a three part document addressing three separate applications:**
    - **Part 1**: **Laser pointers** (solid state diode lasers)
    - **Part 2: Bench lasers** (as used in Physics investigations, usually He-Ne gas lasers)
    - **Part 3: The construction of laser equipment**

3. Safety notes

- Bench lasers should be stored in a secure locked cupboard (e.g. in a storeroom) when not in use to prevent unsupervised student access.
- Do not leave a laser unattended.
- Avoid shining a laser beam at or near persons.
- Avoid student seating that could bring the laser beam to eye level.

- Remove from the area all exposed shiny objects such as rings, watches, metal bands, tools or glass.
- When a laser is in use, access and movement around the room should be controlled and nobody should stand between the laser beam and its target.
- Despite the low power rating i.e. Class 1 or 2 lasers, eye damage is possible if magnifying viewing instruments such as lenses, microscopes, binoculars or telescopes are used. Do not use these optical aids to view a laser beam.
- Laser light differs from light emitted from conventional sources. It is monochromatic, coherent and is collimated (refer to definitions below). This last feature is very important in considering the safety of lasers.

Definitions of properties of laser light:

Monochromatic	The emitted light is concentrated at a single wavelength
Coherent	The light waves remain in phase as they propagate
Collimated	The light rays are parallel, with very little divergence, the intensity is maintained over long distances.

- Conventional light sources emit light in all directions, and so the intensity of the light rapidly diminishes with distance from the source (the inverse square law). As laser light is collimated into a beam with very little divergence, the intensity is maintained over long distances. There is then a much greater capacity for laser beams to cause eye damage.
- The Class rating, the appropriate laser symbol and hazard warning statement are to be clearly marked on the instrument. A table of the laser classes is given in the Supplementary Information section at the end of this document. If the class of any laser is not clearly identified then it should not be used.
- **It is recommended that bench lasers used in schools should be rated no higher than Class 2 or Class 3R** (or 3A under the old pre-2001 classification), and with a power output not exceeding 5 milliWatts (mW). They should be **continuous wave (not pulsed), and emitting visible radiation. Lasers that emit wavelengths that are not in the visible spectrum should not be used in schools.** Note: the **Class 3B laser** has a significantly higher energy output than a Class 3R/3A laser.
- It is recommended that when purchasing **new** bench lasers, that they be rated no higher than a Class 2. Class 1 and Class 2 are generally considered to be sufficient for use in school science. As a guiding principle, always use the lowest power laser for any particular purpose.
- The decision to use a Class of 3R/3A lasers in a school should be made on a case by case assessment considering state/territory radiation regulations as well as educational jurisdictional policies. **A person knowledgeable in the potential hazards of laser radiation should conduct this.** Specific permission should be sought from the school to use this class of laser for teaching purposes.
- If a decision is made to use Class 3R/3A bench lasers, although they may be considered to present low risks when good safety practices are followed, additional safety procedures need to put into place such as: the use restricted to teacher demonstrations, keeping of log book records and using a laser controlled area with appropriate signage in place. Whilst special laser eye protection is not considered necessary for these laser classes, consideration could be given to the provision of appropriate laser eyewear that has been tested to Australian Standards.

- Where a school uses a number of lasers, particularly including the Class 3R/3A, consideration should be given to appointing a **Laser Safety Officer (LSO)** to support these activities. The LSO would normally be a staff member who is familiar with the ARPANSA Safety Guide, understands the laser classification system and has a good working knowledge in the use of lasers. The LSO would be available to assist other staff with performing risk assessments and may assume the role of ensuring all staff complies with the safe storage requirements that are part of the school's safety policies.

4. Regulations, licences and permits

- The use of lasers in schools is informed by the *Safety Guide: Use of Radiation in Schools (2012), Part 2: Lasers* http://www.arpansa.gov.au/pubs/rps/RPS18.pdf.

  It is recommended that persons handling lasers be familiar with this document and a **printed hard copy of this resource is recommended for reference.**

- It is important to check if permission is required to use Class 3R/3A bench lasers in your state or territory with the relevant radiation regulatory body. In addition, you will need to check if your educational governing body allows their use.
  For details of your state/territory regulator see:
  http://www.arpansa.gov.au/Regulation/regulators/index.cfm

5. Equipment

- Bench Laser Class 1 or Class 2 (or class 3R/3A with school permissions in place)

6. Operating procedure

1. **Storage:**

   Lasers should be stored in a secure locked cupboard (e.g. in a storeroom) when not in use to prevent unsupervised student access.

2. **Steps before use:**

   2.1 Check that the bench laser is rated at no higher than Class 2, or Class 3R (or 3A), and with a power output not exceeding 5 milliWatts (mW).

   2.2 Check that the bench laser is appropriately labelled as below:

   Class 1 laser

   

   Class 2 laser

   

   

   Class 3R laser

   

   

   2.3 For the use of Class 3R or 3A lasers, check that the planned activity is conducted by a person knowledgeable in the potential hazards of laser radiation, and who has been given specific permission by the school to use the laser for teaching purposes.

   2.4 Check that a Risk Assessment has been carried out for the planned activities. (Examples of Risk Assessments for laser use are given in the ARPANSA Safety Guide - Use of Radiation in Schools Part 2: Lasers, Section 10: http://www.arpansa.gov.au/pubs/rps/RPS18.pdf .)

   2.5 Check that the following items are available, particularly for the use of Class 3R/ 3A lasers:

   - Copy of the ARPANSA Safety Guide
   - Manufacturer's instructions for assembly and safe use
   - Information on emitted wavelengths and maximum power output
   - A log book recording the use of the laser

   2.6 For the use of Class 3R or 3A lasers, establish a laser controlled area, with access limited to persons granted permission by the school to use lasers, and persons under their control. Warning signs are to be displayed on the outside of the laser controlled area.

3. **Use:**

   The specific procedures to be applied will partly depend on the particular laser activity being undertaken. These may include the following.

   1. Operate the laser at bench or waist height. Avoid student seating that could bring the laser beam to eye level. Avoid uncontrolled beam movement of the laser.
   2. Ensure that students are aware of the laser beam hazard.
   3. Begin with a diagram of the planned laser system before introducing and aligning the optical components. Fix optical components securely to the optical bench.
   4. Evaluate the need for screens and baffles to eliminate possible hazardous reflections. Baffles should use a minimal aperture.
   5. Fix and align the optical components at the minimum power needed to see the beam. Check for stray beams at low power. Advice, including procedural flow charts, is given in the ARPANSA Safety Guide (see Annex A, p.90).
   6. Turn off the laser or block the beam with the shutter when not in use.
   7. Eye damage is possible if magnifying viewing instruments such as lenses, microscopes, binoculars or telescopes are used. Do not use these to view a laser beam.

7. Trouble shooting/emergencies

- Note: The risk of burns to the eye or skin is very low when using bench lasers of Class 2 or less.
- Burns to eye: Cover damaged closed eye with a clean eye patch, instruct the casualty to keep eye still, summon ambulance.
- Burns to skin: Cool area under cold running water. Cover damaged area with clean bandage; seek urgent medical advice.

8. Waste disposal

- Not applicable

9. Related material

- Australian Radiation Protection and Nuclear Safety Agency 'Safety Guide: Use of radiation in schools Part 2: Lasers. Radiation protection series no. 18', ARPANSA website, June 2012 http://www.arpansa.gov.au/pubs/rps/RPS18.pdf. This includes guidance on developing a Risk Assessment for the use of lasers in schools (see Section 10).
- Manufacturer's instructions
- Risk Assessment
- Useful links:
  - Macquarie University 'Faculty of Science Occupational Health and Safety – Lasers' Macquarie University website. (Accessed January 2015). http://web.science.mq.edu.au/intranet/ohs/lasers/. This page contains much useful information including information on laser classifications and laser induction.

    Overview of IEC laser classes. http://web.science.mq.edu.au/intranet/ohs/lasers/documents/classoverview.pdf

    Typical accessible emission limits (AEL) for visible CW lasers. http://web.science.mq.edu.au/intranet/ohs/lasers/documents/classifications.pdf

    Laser Induction: with links to watch the MPG free Faculty Laser Induction Module (PPT, 3.3 MB) plus the Eye Effects Video (46 MB, 5 minutes) and the Laser Safety Video (231 MB, 30 minutes)

References:

Australian Radiation Protection and Nuclear Safety Agency (ARPANSA) 2012. *Safety Guide for the Use of Radiation in Schools (2012)* http://www.arpansa.gov.au/pubs/rps/RPS18.pdf CC BY NC 3.0 AU http://creativecommons.org/licenses/by-nc/3.0/au/

Australian/New Zealand Standard AS/NZS IEC 60825.1:2014. Safety of laser products Part 1: Equipment classification and requirements.

St John Ambulance. 2010. *First Aid Manual*.

SUPPLEMENTARY INFORMATION:

Classification of lasers

Lasers are classified according to their potential degree of hazard, with the higher numbers indicating higher hazard levels. Older units may be classified under a similar early system, with the comparisons set out in the table below. It is important to remember that laser classification is made on the basis of the *entire* laser product. This means it is possible that a laser product could contain a high power laser internally with the engineering design allowing a lower classification for the unit.

Current classification	Former classification	Notes	Labelling and signage
Class 1	Class 1	Considered safe under foreseeable conditions because of low emission levels or through engineering design. Not harmful to eyes or skin.	
Class 1M		Higher emissions than Class 1, but because of the spread of rays, not capable of causing eye damage. This means the beam can be magnified with the help of optics.	
Class 2	Class 2	Low powered visible lasers (to 1mW power). Not harmful to skin. Eye damage avoided through natural aversion responses (blinking, turning away)	 
Class 2M		Higher emissions than Class 2, but because of the spread of rays, normally safe for viewing with unaided eye. This means the beam can be magnified with the help of optics.	 
Class 3R	Class 3A	For visible wavelengths, up to 5 times the emissions of Class 2 (i.e. up to 5 mW power). Risk of injury from accidental exposure is low.	 
Class 3B	Class 3B	Visible or invisible wavelengths where direct viewing is hazardous to eyes. (i.e. between 5mW and 500mW for visible wavelengths)	 
Class 4	Class 4	High powered lasers capable of causing damage to skin and eyes. (i.e. > 500mW)	 

Lasers in schools

It is expected that in almost all cases, lasers used in schools should be restricted to Class 1 and Class 2 (common outputs of 0.5 to 1 mW). At this level, eye damage from accidental exposure is very unlikely due to the human 'aversion responses' such as blinking and turning away. Lasers classified as Class 3R/ 3A, with power outputs in the 1–5 mW range, may be used subject to gaining school permission and following good safety practices as detailed in Part 2 of this SOP. Lasers of Class 3B and above, and any that emit wavelengths not in the visible spectrum, should not be used in schools. If the class of any laser is not clearly identified then it should not be used.

STANDARD OPERATING PROCEDURE:

Use of lasers in schools

Part 3: The construction of laser equipment

Note: To be undertaken only by trained personnel in conjunction with a site-specific risk assessment. ***No dismantling or modification of laser equipment should be undertaken in schools.*** *Contact your state or territory radiation regulatory authority for the interpretation of regulatory matters. For contact details see:*
http://www.arpansa.gov.au/Regulation/Regulators/index.cfm

---

1. Introduction

The word LASER is an acronym for **L**ight **A**mplification by **S**timulated **E**mission of **R**adiation, which describes the process by which lasers generate light (electromagnetic radiation). There are many applications of the use of lasers in schools, including as a white board pointer, a pointer for astronomy observations, the teaching of optics, ray paths, refraction and diffraction, the modulation of laser beams to carry signals, and the applications in fibre optic communications.

As laser technology is very widely used in industry, medicine and in everyday life, it is desirable that students learn to safely and responsibly use and manage lasers in a supervised school science environment.

Laser classification is made on the basis of the *entire* laser product. This means it is possible that a laser product could contain a high power laser internally with the engineering design allowing a lower classification for the unit. Therefore the **dismantling or modification of laser equipment should not be undertaken in schools**

2. Context

- This document notes some of the risks that may be associated with either constructing laser equipment, or with dismantling equipment that contains lasers. It recommends conditions for the safe construction of laser equipment in Australian schools, and that the dismantling or modification of laser equipment is not undertaken in schools.
- **This is Part 3 of a three part document addressing three separate applications:**
  - **Part 1**: **Laser pointers** (solid state diode lasers)
  - **Part 2: Bench lasers** (as used in Physics investigations, usually He-Ne gas lasers)
  - **Part 3: The construction of laser equipment**

3. Safety notes

- All lasers should be stored in a secure locked cupboard (e.g. in a storeroom) when not in use to prevent unsupervised student access.
- Do not leave the laser unattended.

- Avoid shining a laser beam at or near persons.
- Avoid student seating that could bring the laser beam to eye level.
- Remove from the area all exposed shiny objects such as rings, watches, metal bands, tools or glass.
- When a laser is in use, access and movement around the room should be controlled and nobody should stand between the laser beam and its target.
- Despite the low power rating i.e. Class 1 or 2 lasers, eye damage is possible if magnifying viewing instruments such as lenses, microscopes, binoculars or telescopes are used. Do not use these optical aids to view a laser beam.
- Laser light differs from light emitted from conventional sources. It is monochromatic, coherent and is collimated (refer to definitions below). This last feature is very important in considering the safety of lasers.

  Definitions of properties of laser light:

Monochromatic	The emitted light is concentrated at a single wavelength
Coherent	The light waves remain in phase as they propagate
Collimated	The light rays are parallel, with very little divergence, the intensity is maintained over long distances.

- Conventional light sources emit light in all directions, and so the intensity of the light rapidly diminishes with distance from the source (the inverse square law). As laser light is collimated into a beam with very little divergence, the intensity is maintained over long distances. There is then a much greater capacity for laser beams to cause eye damage.
- The Class rating, the appropriate laser symbol and hazard warning statement are to be clearly marked on the instrument. A table of the laser classes is given in the 'Supplementary Information' section at the end of this document. If the class of any laser is not clearly identified then it should not be used.
- Lasers constructed in schools should be restricted to Class 1 or Class 2 and should use commercially available low powered laser diodes or Helium-Neon gas tubes. Lasers should not be constructed where the power is unknown, or where it may exceed Class 2. For further information see section 9.10 'Lasers constructed at school' in the *Safety Guide: Use of Radiation in Schools (2012), Part 2: Lasers.*
- Equipment containing lasers must not be dismantled or modified in schools. Some equipment rated as Class 1 or Class 2 (and particularly 1M or 2M) contain lasers of higher power, and achieve a lower Class rating because of shielded enclosures or special optical systems that prevent human exposure to higher emissions during normal operation. Dismantling or modifying such equipment could result in these producing unsafe levels of emission. For further information see section 9.9 'General Warnings relating to construction or modifying laser products' in the *Safety Guide: Use of Radiation in Schools (2012), Part 2: Lasers.*
- Where a school uses a number of lasers, consideration should be given to appointing a **Laser Safety Officer (LSO)** to support these activities. The LSO would normally be a staff member who is familiar with this Safety Guide, understands the laser classification system

and has a good working knowledge in the use of lasers. The LSO would be available to assist other staff with performing risk assessments and may assume the role of ensuring all staff complies with the safe storage requirements that are part of the school's safety policies.

4. Regulations, licences and permits

- The use of lasers in schools is informed by the *Safety Guide: Use of Radiation in Schools (2012), Part 2: Lasers* http://www.arpansa.gov.au/pubs/rps/RPS18.pdf.

  It is recommended that persons handling lasers be familiar with this document and a **printed hard copy of this resource is recommended for reference.**

  For details of your state/territory regulator see:
  http://www.arpansa.gov.au/Regulation/regulators/index.cfm

5. Equipment

- Lasers of Class 1 or Class 2

6. Operating Procedure

1. **Steps before use**

   1.1 Ensure that the laser component being used:

   - produces non-pulsed radiation in the visible spectrum
   - has a maximum output not exceeding 1mW
   - is classified as either Class 1 or Class 2

   These components will typically be commercially available low powered laser diodes or He-Ne gas tubes. If the laser power is unknown, or may exceed 1mW, then do not proceed.

   1.2 Ensure that a Risk Assessment is conducted for both the construction phase and for any activities to be undertaken using the laser.

2. **Use**

   2.1 Ensure that the constructed instrument is properly labelled with laser class, hazard symbol and hazard warning

Class 1 Laser		
Class 2 Laser		

   2.2 Ensure that the constructed instrument is stored in a secure locked cupboard to prevent unsupervised student access.

6. Trouble shooting/emergencies

- Not applicable

8. Waste disposal

- Not applicable

9. Related Material

- Australian Radiation Protection and Nuclear Safety Agency 'Safety Guide: Use of radiation in schools Part 2: Lasers. Radiation protection series no. 18', ARPANSA website, June 2012 http://www.arpansa.gov.au/pubs/rps/RPS18.pdf. This includes guidance on developing a Risk Assessment for the use of lasers in schools (see Section 10).
- Risk Assessment
- Useful links:
  - Macquarie University 'Faculty of Science Occupational Health and Safety – Lasers' Macquarie University website. (Accessed January 2015). http://web.science.mq.edu.au/intranet/ohs/lasers/. This page contains much useful information including information on laser classifications and laser induction.

    Overview of IEC laser classes. http://web.science.mq.edu.au/intranet/ohs/lasers/documents/classoverview.pdf

    Typical accessible emission limits (AEL) for visible CW lasers. http://web.science.mq.edu.au/intranet/ohs/lasers/documents/classifications.pdf

    Laser Induction: with links to watch the MPG free Faculty Laser Induction Module (PPT, 3.3 MB) plus the Eye Effects Video (46 MB, 5 minutes) and the Laser Safety Video (231 MB, 30 minutes)

References:

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Australian/New Zealand Standard AS/NZS IEC 60825.1:2014. Safety of laser products Part 1: Equipment classification and requirements.

SUPPLEMENTARY INFORMATION:

Classification of lasers

Lasers are classified according to their potential degree of hazard, with the higher numbers indicating higher hazard levels. Older units may be classified under a similar early system, with the comparisons set out in the table below. It is important to remember that laser classification is made on the basis of the *entire* laser product. This means it is possible that a laser product could contain a high power laser internally with the engineering design allowing a lower classification for the unit.

Current classification	Former classification	Notes	Labelling and signage
Class 1	Class 1	Considered safe under foreseeable conditions because of low emission levels or through engineering design. Not harmful to eyes or skin.	
Class 1M		Higher emissions than Class 1, but because of the spread of rays, not capable of causing eye damage. This means the beam can be magnified with the help of optics.	
Class 2	Class 2	Low powered visible lasers (to 1mW power). Not harmful to skin. Eye damage avoided through natural aversion responses (blinking, turning away)	 
Class 2M		Higher emissions than Class 2, but because of the spread of rays, normally safe for viewing with unaided eye. This means the beam can be magnified with the help of optics.	 
Class 3R	Class 3A	For visible wavelengths, up to 5 times the emissions of Class 2 (i.e. up to 5 mW power). Risk of injury from accidental exposure is low.	 
Class 3B	Class 3B	Visible or invisible wavelengths where direct viewing is hazardous to eyes. (i.e. between 5mW and 500mW for visible wavelengths)	 
Class 4	Class 4	High powered lasers capable of causing damage to skin and eyes. (i.e. > 500mW)	 

Lasers in schools

It is expected that in almost all cases, lasers used in schools should be restricted to Class 1 and Class 2 (common outputs of 0.5 to 1 mW). At this level, eye damage from accidental exposure is very unlikely due to the human 'aversion responses' such as blinking and turning away. Lasers classified as Class 3R/ 3A, with power outputs in the 1–5 mW range, may be used subject to gaining school permission and following good safety practices as detailed in Part 2 of this SOP. Lasers of Class 3B and above, and any that emit wavelengths not in the visible spectrum, should not be used in schools. If the class of any laser is not clearly identified then it should not be used.

STANDARD OPERATING PROCEDURE:

Fire blankets

Note: These instructions are for the use of adults and responsible students in an emergency.

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1. Introduction

Clothing and hair can easily catch fire when students are working in a school laboratory. Also small fires can suddenly flare up and a fire blanket is often a better option for an untrained person than a fire extinguisher. Purchase of a non-flammable fibreglass fire blanket can help save lives and protect from serious burns. They are quick and easy to use with little training required, are inexpensive and readily available from safety equipment suppliers and hardware stores.

2. Context

- Instructions for emergency fire response should be addressed in the Science Safety Policy and also reflected in your school's emergency planning policies.

3. Safety notes

- It is important to purchase a fire blanket that carries the Australian Standards mark AS 3504.
- Choose a fire blanket large enough to cover an adult rather than a smaller student.
- Hang the fire blanket near a fire extinguisher so all fire-fighting items are close together.
- Secure relevant safety signs above or next to the fire blanket as per AS 2444:2001. The signs should be white on a red background and be visible from 20 metres in all directions.
- Place the fire blanket at a height so both adults and students can quickly pull the hanging tags to release the fire blanket.
- Regularly check the condition of the fire blanket and its quick-release PVC container.
- Familiarise yourself with the operating instructions regularly.
- Only use the fire blanket once and replace with a new fire blanket after use.

4. Regulations, licences and permits

- No permit or licence is required.

5. Equipment

- Fire blanket

6. Operating procedure

1. Stay calm.
2. Remove other students from the vicinity of the casualty or small fire.
3. Remove the original source of heat if safe to do so.
4. Release the blanket by pulling the tags hanging beneath the fire blanket cover.
5. Shake out the blanket so that the longest area is hanging downwards.
6. When clothing is on fire, adopt the procedure 'Stop, Drop, Cover and Roll'.

   STOP: the casualty should stop running.

   DROP: the casualty should drop to the floor. Wrap the fire blanket around them. Do not throw the blanket over them.

   COVER: the casualty should cover their face.

   ROLL: the casualty should be rolled back and forth along the ground until flames are extinguished. (See First Aid for burns in section 7 Trouble shooting/emergencies)
7. When using the fire blanket on a small fire, hold the blanket by any handles attached (see diagram below). If there are no handles, then grasp the top of the blanket with your hands upside-down and rotate your wrists inwards so the top of the blanket covers them. This protects your hands from the heat.

8. Approach the small fire by holding the fire blanket up in front of you and place it slowly over the fire. The blanket should completely cover the fire to reduce the oxygen level in the area on fire. The fire retardant chemicals in the blanket will extinguish the flames so there is no need to smother the fire by applying any further pressure with your hands.
9. Leave the fire blanket in place until the fire cools.

7. Trouble shooting/emergencies

- **First aid:** Treat all thermal burns by holding the burnt area under running water for up to 20 minutes until skin returns to normal temperature. Remove clothing from burnt area unless stuck; cover burn with a non-adherent burns dressing, plastic wrap or loosely applied aluminium foil. Seek urgent medical aid.
- **Smoke inhalation:** Remove casualty from area to fresh air. Sit up and loosen tight clothing. Administer oxygen if available and you are trained in its use. Consider an

asthma inhaler if casualty has difficulty in breathing or is wheezing. If breathing stops commence CPR. Seek urgent medical aid.

- **Maintenance:** Check condition of fire blanket and PVC cover regularly. Promptly replace a used fire blanket.

8. Waste disposal

- Place used fire blankets in general garbage once cooled

9. Related material

Websites for emergency services organisations in Australian states and territories:

- **Australian Capital Territory**
  ACT Government, Emergency Services Agency website http://esa.act.gov.au/ (Accessed May 2015)
- **New South Wales**
  NSW Government, Emergency New South Wales website https://www.emergency.nsw.gov.au/ (Accessed May 2015)
- **Northern Territory**
  Northern Territory Government, Police, Fire and Emergency Services website http://www.pfes.nt.gov.au/ (Accessed May 2015)
- **Queensland**
  Queensland Government, Queensland Fire and Emergency Services (QFES) website https://www.qfes.qld.gov.au/ (Accessed May 2015)
- **South Australia**
  Government of South Australia, South Australian Fire and Emergency Services Commission website http://www.safecom.sa.gov.au/site/home.jsp (Accessed May 2015)
- **Tasmania**
  Tasmanian Government, Department of Police and Emergency Management website http://www.dpem.tas.gov.au/ (Accessed May 2015)
- **Victoria**
  Victorian Government, Emergency services website, http://www.vic.gov.au/emergencies-safety/emergency-services.html (Accessed May 2015)
- **Western Australia**
  'DFES Alert and Warnings', Government of Western Australia WA Department of Fire and Emergency Services website http://www.dfes.wa.gov.au/alerts/Pages/default.aspx (Accessed May 2015)

References:

'Fire Blankets and Extinguishers', Government of Western Australia WA Department of Fire and Emergency Services website, http://www.dfes.wa.gov.au/safetyinformation/fire/fireinthehome/pages/fireblanketsandextinguishers.aspx (Accessed May 2015)

'Safety recommendations' Government of Western Australia WA Department of Fire and Emergency Services website http://www.dfes.wa.gov.au/safetyinformation/fire/fireinthehome/Pages/safetyrecommendations.aspx (Accessed May 2015)

Standards Australia. 2001. AS 2444—2001 *Portable fire extinguishers and fire blankets – Selection and location*. Sydney, Australia.

Standards Australia. 2006. AS/NZS 3504:2006 *Fire blankets.* Sydney, Australia.

St John Ambulance Australia 2011, *Australian First Aid*, 4th edition, St John Ambulance Australia: Barton ACT.

STANDARD OPERATING PROCEDURE:

Fire extinguishers

Note: These instructions are for the use of adults and responsible students in an emergency.

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1. Introduction

Experiments conducted in school laboratories often require a source of heat from a naked flame or some form of ignition, which can cause a sudden fire. Occasionally electrical faults can cause sparking. Students are fascinated with matches and have the potential to behave irresponsibility. Any of these occurrences can introduce fire into a room and therefore fire extinguishers must be placed in each laboratory and also outside a chemical store, usually close to an exit door. A fire needs heat (ignition), fuel, and oxygen and can engulf a room exceedingly fast, although most deaths result from inhaling the toxic smoke containing dangerous fumes released by the fire melting various plastics and paint in the room.

2. Context

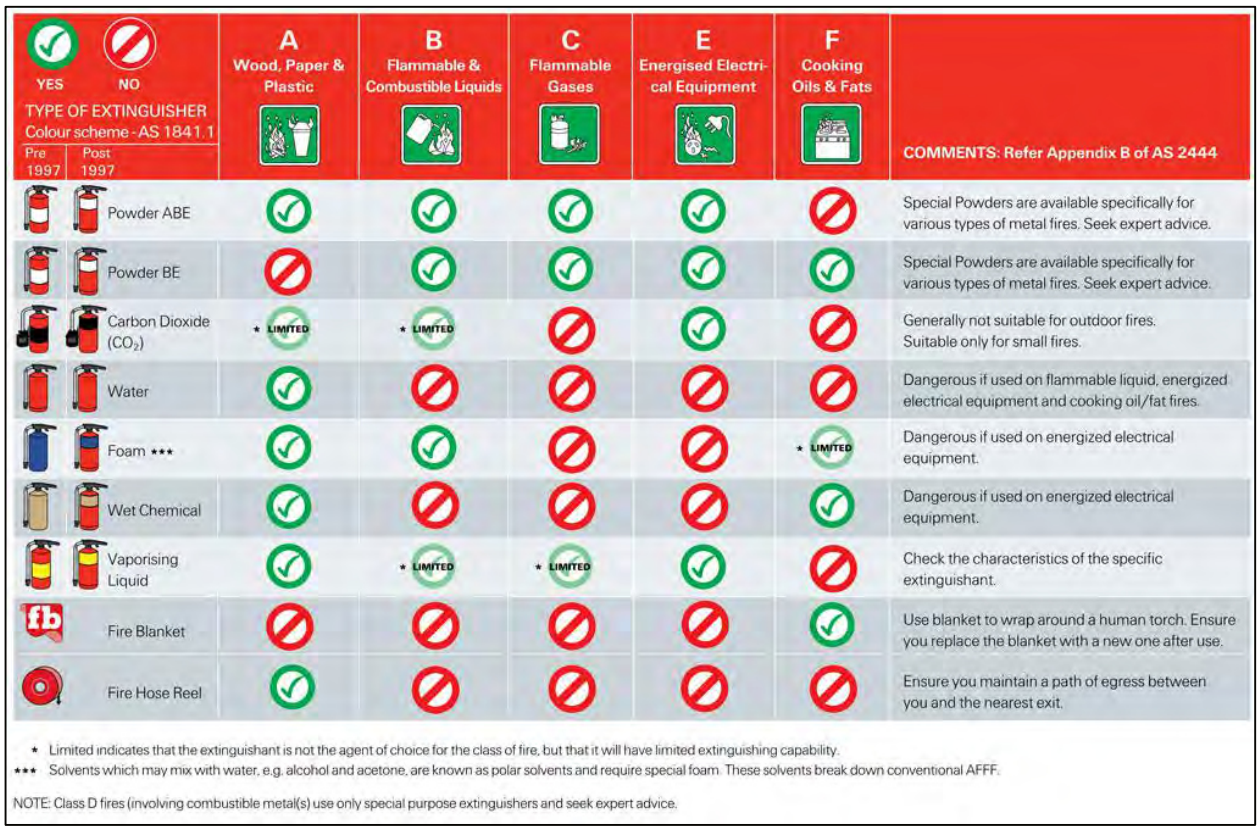
- Instructions for emergency fire response should be addressed in the Science Safety Policy and also reflected in your school's emergency planning policies.

3. Safety notes

- Purchase fire extinguisher/s from a reputable, recognised and certified fire safety company and follow their advice regarding which type of fire extinguisher to purchase.
- Ensure that you purchase a fire extinguisher that has an Australian Standards AS/NZS1841 series label.
- Obtain relevant SDS chemical safety sheets from the manufacturer's web site.
    - Follow the manufacturer's instructions for placement of the fire extinguisher and secure using the correct bracket.
    - Secure the fire extinguisher at a height that can be reached by both adults and students.
    - Secure relevant safety signs above or adjacent to the fire extinguisher as per AS 2444-2001. The signs should be white on a red background and visible from 20 metres in all directions.
    - Reduce combustibles in rooms.
    - Eliminate all combustibles in a chemical store.
    - Install ceiling smoke alarms to provide early warning of a fire.
    - Regularly familiarise yourself with the operating instructions.
    - Professional firefighting hands-on training is recommended for staff in high risk areas.
    - Purchase the correct fire extinguisher to suit the environment in which it will be used. Using the wrong extinguisher type on a fire may have disastrous

consequences. It may feed the fire, causing it to spread or result in the operator being injured. See chart below:

Fire Extinguisher Chart



YES / NO — TYPE OF EXTINGUISHER Colour scheme - AS 1841.1 (Pre 1997 / Post 1997)	A Wood, Paper & Plastic	B Flammable & Combustible Liquids	C Flammable Gases	E Energised Electrical Equipment	F Cooking Oils & Fats	COMMENTS: Refer Appendix B of AS 2444
Powder ABE	✓	✓	✓	✓	✗	Special Powders are available specifically for various types of metal fires. Seek expert advice.
Powder BE	✗	✓	✓	✓	✓	Special Powders are available specifically for various types of metal fires. Seek expert advice.
Carbon Dioxide (CO_2)	* LIMITED	* LIMITED	✗	✓	✗	Generally not suitable for outdoor fires. Suitable only for small fires.
Water	✓	✗	✗	✗	✗	Dangerous if used on flammable liquid, energized electrical equipment and cooking oil/fat fires.
Foam ***	✓	✓	✗	✗	* LIMITED	Dangerous if used on energized electrical equipment.
Wet Chemical	✓	✗	✗	✗	✓	Dangerous if used on energized electrical equipment.
Vaporising Liquid	✓	* LIMITED	* LIMITED	✓	✗	Check the characteristics of the specific extinguishant.
Fire Blanket	✗	✗	✗	✗	✓	Use blanket to wrap around a human torch. Ensure you replace the blanket with a new one after use.
Fire Hose Reel	✓	✗	✗	✗	✗	Ensure you maintain a path of egress between you and the nearest exit.

* Limited indicates that the extinguishant is not the agent of choice for the class of fire, but that it will have limited extinguishing capability.
*** Solvents which may mix with water, e.g. alcohol and acetone, are known as polar solvents and require special foam. These solvents break down conventional AFFF.

NOTE: Class D fires (involving combustible metal(s) use only special purpose extinguishers and seek expert advice.

Source: https://exelgard.com.au/fire_fighting_equipment/extinguishers

4. Regulations, licences and permits

- Fire extinguishers must conform to AS/NZS 1841.6:2008.
- No licence or permit is required.
- Fire extinguisher technicians need to be licensed in Queensland.

5. Equipment

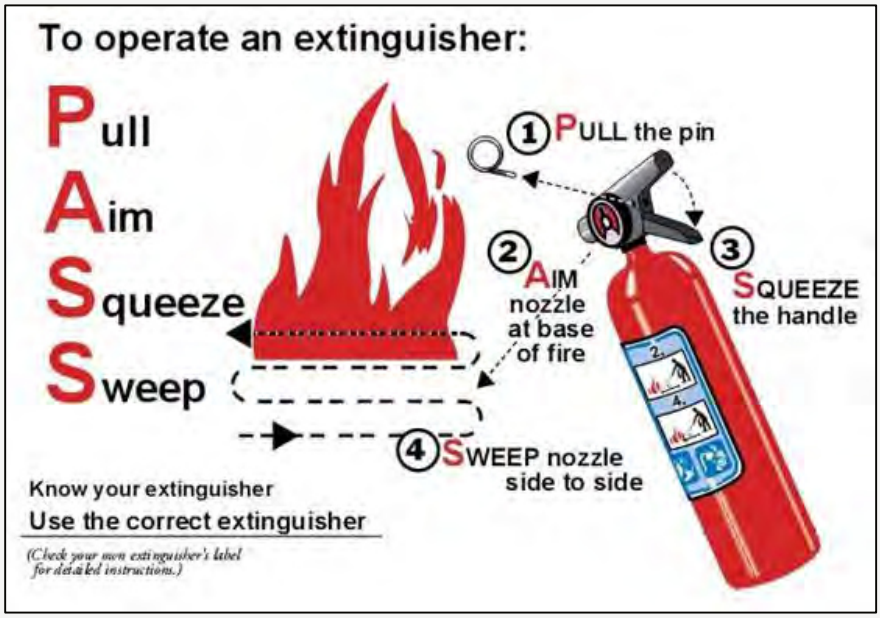
- Fire extinguisher

6. Operating procedure

1. Ignore the fire!
2. Stay calm.
3. Turn off all electrical and gas services to room.
4. Immediately evacuate all staff and students to a safe area.
5. Send responsible students to alert and evacuate rooms either side of the affected area.
6. Send a responsible person to the Administration Office to ask them to call 000.
7. Only return to fight the fire if you are confident that you can bring the fire under control.

(Operating procedure cont….)

8. Make sure the fire is not blocking your exit and ensure you can get out quickly if necessary.
9. If safe to do so, close doors and windows (fans and air conditioners should have ceased operating when electrical power was turned off).
10. Select the correct class of extinguisher for the type of fire.
11. Only stand as close as you can without getting burnt.
12. Point the extinguisher at the base of the fire, operate and use a sweeping motion to extinguish flames. See PASS diagram below:



13. If the fire is not doused by the time the extinguisher is empty, drop the extinguisher and leave the room quickly.
14. Ensure that the Fire Brigade has checked whether any ceiling insulation has been affected as this can smoulder for a considerable time and then reignite

7. Trouble shooting/emergencies

- **First aid:** If clothing is on fire, stop, drop to the floor and wrap around a blanket, coat or rug (not synthetic) and roll along the ground until flames are extinguished.
    - Treat all thermal burns by holding the burnt area under running water for up to twenty minutes until skin returns to normal temperature. Remove clothing from burnt area unless stuck; cover burn with a non-adherent burns dressing, plastic wrap or loosely applied aluminium foil. Seek urgent medical aid.
    - Smoke/Toxic fume inhalation: Remove casualty from area to fresh air. Sit up and loosen tight clothing. Administer oxygen if available and you are trained in its use and consider an asthma inhaler if casualty has difficulty in breathing or is wheezing. If breathing stops commence CPR. Seek urgent medical aid.
- **Maintenance:** A pressure test and service of fire extinguishers is required every six months and must be provided by an experienced person from a recognised and certified fire safety company. Test dates must be recorded, usually on a yellow metal tag

attached to the extinguisher. Extinguishers failing the test must be removed and a temporary one left as a replacement. A partially discharged extinguisher must be replaced with a full extinguisher immediately. Extinguishers need to be emptied, pressure tested and refilled every five years.

8. Waste disposal

- Contact a reputable, recognised and certified fire safety company to remove unwanted, depressurised or used extinguishers: **Note**: a partially used fire extinguished is regarded as an 'empty' extinguisher.

9. Related material

Websites for emergency services organisations in Australian states and territories:

- **Australian Capital Territory**
  ACT Government, Emergency Services Agency website http://esa.act.gov.au/ (Accessed May 2015)
- **New South Wales**
  NSW Government, Emergency New South Wales website https://www.emergency.nsw.gov.au/ (Accessed May 2015)
- **Northern Territory**
  Northern Territory Government, Police, Fire and Emergency Services website http://www.pfes.nt.gov.au/ (Accessed May 2015)
- **Queensland**
  Queensland Government, Queensland Fire and Emergency Services (QFES) website https://www.qfes.qld.gov.au/ (Accessed May 2015)
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- **Victoria**
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- **Western Australia**
  Government of Western Australia WA Department of Fire and Emergency Services website http://www.dfes.wa.gov.au/alerts/Pages/default.aspx (Accessed May 2015)

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'Fire Blankets and Extinguishers', Government of Western Australia. WA Department of Fire and Emergency Services website, http://www.dfes.wa.gov.au/safetyinformation/fire/fireinthehome/pages/fireblanketsandextinguishers.aspx (Accessed May 2015)

'Fire extinguishers' Exelgard website https://exelgard.com.au/fire_fighting_equipment/extinguishers (Accessed May 2015)

Standards Australia. 2001. AS 2444–2001 *Portable fire extinguishers and fire blankets – Selection and location*. Sydney, Australia.

Standards Australia. 2007. AS/NZS 1841 *Portable fire extinguisher series.* Sydney, Australia.

St John Ambulance Australia. 2011. *Australian First Aid 4th Edition*, St John Ambulance Australia. Barton, ACT

On example Risk Assessment

Wormald. 2008. *Pyro-Chem ABC Multipurpose Dry Chemical Powder Material Safety Data Sheet* Fire Systems Services website http://www.firesys.com.au/rs/7/sites/846/user_uploads/File/MSDS%20DCP%20Pyro-Chem%20ABC.pdf

STANDARD OPERATING PROCEDURE:

Gas cylinders in school science areas

Note: Compressed gas cylinders should be handled only by trained personnel in conjunction with a site specific risk assessment and appropriate Safety Data Sheet (SDS) for the gas in use.

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1. Introduction

Many schools use compressed gas cylinders. Some commonly used gases include LPG, O_2, CO_2, He, and compressed air. These gas cylinders are hazardous due to their chemical characteristics, their compressed state and their physical size. The gases within the cylinders vary in chemical properties ranging from inert to explosive. The gas cylinders are made from heavy walled metal that have been manufactured to withstand high pressure. They come in different sizes generally denoted by a letter code. Compressed gases are classified as Class 2 substances in the ADG code and are divided into four sub-classes. The GHS pictogram identifies the hazard class i.e. Gases under Pressure.

Comparison of the GHS Hazard Pictogram and the Corresponding ADG Class Labels

GHS Hazard Pictogram	GHS Hazard	DG Class Labels	DG Sub-Classes
	Gases under Pressure		Class 2.1 Flammable Gases: e.g. LPG
			Class 2.2 Non-flammable, non-toxic and Non-oxidising gases: e.g. CO_2, He and Compressed air.
			Class 2.2/5.1 Oxidising Gases: e.g. O_2
			Class 2.3 Toxic Gases: (**not used in schools**)

Appropriate care in the handling, storage and transportation of gas cylinders is essential.

2. Context

- These instructions are for the use of experienced science teachers and technicians who are properly trained in handling techniques.
- A site-specific risk assessment should be carried out to determine appropriate storage, handling and transport arrangements required by your laboratory.
- Safe operating procedures should be established.
- Information and training in use and hazards of cylinders should be made available.
- Gas equipment operating manuals should be consulted.

3. Safety notes

- Cylinders pose risks relating to the gases they contain:

  A flammable gas may cause an explosion or fast spreading fire. Note: Hydrogen is not recommended for use in schools as it is extremely difficult to store safely due to its fire and explosion hazard. It is a highly flammable gas with the potential to form explosive mixtures with air within the range of 4% and 75%. It is much lighter than air and will collect at the highest point within an enclosed space unless ventilated at a high level, which could create an explosive mixture.

  Oxidizing gases can accelerate combustion and increase the risk of fire in the presence of combustible or flammable materials.

  Some gases are toxic if inhaled. *Note:* Toxic gases are not suitable for use in schools.
- All gas cylinders pose a threat due to the potential for oxygen depletion, making asphyxiation possible. Use in well ventilated areas and never store or use in confined spaces.
- Some gases are heavier than air and collect in low lying areas, while other gases are lighter than air and collect in high points in enclosed spaces. For any gas in use, check safety information as to whether it is heavier or lighter than air, and subject to pooling in high or low spaces.
- Consult appropriate SDSs for information on the chemical ingredients, physical and health hazards, specific handling and storage information, exposure controls and personal protective measures.
- Any gas cylinder that is exposed to fire or extreme heat may rupture resulting in a rapid release of gas and flying shrapnel. Do not use cylinders that have been heated or exposed to a fire.
- A BLEVE or "boiling liquid expanding vapour explosion" can occur when a cylinder containing a pressurised liquid is ruptured. It can occur in a vessel that stores a substance that is usually a gas at atmospheric pressure but is a liquid when pressurised for example, liquefied petroleum gas (LPG).
- Full and/or empty cylinders should not be stored in science teaching areas unless they are in active use.
- Full and empty cylinders should be segregated.
- Large cylinders are heavy and awkward to move, even when empty and unstable due to their slender shape. Incorrect handling may cause injury, such as sprains, strains, falls, bruises, or broken bones.
- Cylinders contain gas under high pressure. There is a possibility of a cylinder becoming a projectile if it falls over and the valve stem is broken so that there is a rapid escape of the compressed gas.

- Cylinders must be properly secured at all times. See Section 6 for further details
- Only use equipment designed for use with gas cylinders and make sure any attachments are compatible with the cylinder and the specific gas in use.
- The cylinder valve is the primary safety mechanism used to contain the contents of a pressurised cylinder.
- ALWAYS turn off the cylinder valve, not just the regulator when not in use
- For transportation, secure the cylinder upright with a chain or belt and leave the valve protection cap in place. Do not drag or slide the cylinder.
- If private vehicle transportation is required an open back utility vehicle is advisable. DO NOT transport in a passenger compartment.
- Be aware of potential hazards and develop emergency response procedures.

4. Regulations, licences and permits

Consult dangerous goods and local government regulations for storage and transportation legislation.

5. Equipment

- Compressed gas cylinder
- Cylinder key if required
- Compatible regulator
- PPE – lab coat, safety shoes, leather gloves, safety glasses
- Cylinder trolley, if transportation is required

6. Operating procedure

Storage of cylinders

1. Full and/or empty cylinders are not to be stored in labs and should be stored separately; preferably in a dedicated cylinder store which is dry, well ventilated, secure and has clear signage in accordance with regulations. Placard the different storage areas of gases with the GHS pictogram to identify the hazard class 'Gases under Pressure', and the DG class label to identify the contents of the cylinder.
2. Avoid below ground storage areas.
3. Only keep cylinders 'in use' in the lab. Minor storage quantities apply in labs. (Refer to AS 4332 *The Storage and Handling of Gases in Cylinders*).
4. Store and segregate different types of gases in accordance with regulations (refer to State Dangerous Goods Legislation and AS 4332).

   Gas cylinders should be segregated from incompatible gases by at least 3 metres. For example Class 2.1 Flammable gases should not be stored with Class 2.2/5.1 oxidising gases and should be stored at least 3 metres from combustible materials.
5. Gases which are denser than air e.g. LPG and CO_2 need to be stored with caution to avoid storage where these gases can collect in low lying areas such as pits, depressions and basements. Ventilation should be provided at floor level.

Operating Procedure continued:

6. Gases much lighter than air e.g. helium, collect at the highest point in any enclosed space. Ventilation should be provided at the highest point of the room.
7. Cylinders must be properly secured to a wall or bench with either a chain or nylon strapping designed for cylinders before any equipment is connected. Restraints should be around the main cylinder body and not the neck. A cylinder trolley is for transport only and should not be used for storage of cylinders
8. Always store cylinders upright, with the valve cap in place on a level floor for easy trolley access and out of traffic areas.
9. Gas cylinders should not be located near heavily travelled areas and any doorway or any other location that could result in the blockage of an exit. Cylinders should be at least 1m away from any opening in a building.
10. Ensure valve guards or caps are fitted when cylinders are not in use to keep the valve clean.
11. Keep all cylinders away from heat sources and any flammable gases away from any ignition sources and electrical outlets.
12. Do not store near combustible materials or flammable liquids.
13. Never hang clothes or equipment on a compressed gas cylinder.

Transporting cylinders

1. Cylinder valves must be closed and regulators and all equipment detached before transportation.
2. Secure cylinder upright to a cylinder trolley when being transported. Seek help if movement requires handling a very large cylinder.
3. Never roll or drag a cylinder on the ground.
4. Once in the lab secure the cylinder to a wall or bench.
5. If private vehicle transport is required, an open back utility vehicle is recommended with cylinders upright and secured. DO NOT transport in a passenger compartment.

Handling cylinders

1. Always check the label on the cylinder first to make sure the correct gas is being used.
2. Attach a compatible regulator i.e. appropriate for the gas, pressure and application. A gas cylinder is designed to supply gas through pressure regulators that meet Australian Standards. Regulators bring down the high pressure to a usable working pressure. Do not over tighten or use excessive force as this can damage the thread.
3. Never open a cylinder valve unless the cylinder is connected to a regulator. Open by turning the hand wheel or cylinder key anticlockwise and close by turning clockwise.
4. The valve should not be fully opened to the point of resistance, but given a half turn back to prevent it locking in an open position.

Operating Procedure continued:

5. Flammable gases have a left hand thread to attach the regulator to distinguish them from non-flammable gases.
6. Never use a faulty or leaking regulator.
7. Do not use thread sealing tape or lubricants on cylinder valves and fittings. Regulators should seal properly without either of these.
8. When not in use or when empty, both the cylinder valve and regulator should **ALWAYS** be closed.
9. Use cylinders in well-ventilated areas.
10. Do not use cylinders that show signs of damage or corrosion or when identification tags/labels are missing.

7. Trouble shooting/emergencies

- First aid: See latest SDSs of individual gases for detailed information
- If a small leak is suspected check by listening for a hissing sound, looking for frosting around the valve or testing with a squeeze bottle of soapy water. Bubbles will form where the gas is escaping. If safe to do so, close cylinder valve and refit the regulator.
- If the leak is significant and unstoppable, evacuate the area and call the fire brigade.
- If any cylinder is involved in a fire, evacuate to at least 100m away and call 000, do not attempt to fight the fire.
- Never attempt to repair any cylinders. Any problems should be referred to the supplier.
- Do not over tighten or use excessive force to attach fittings, or attempt to connect an incompatible regulator or fittings as the threads may be damaged.
- All regulators and hoses should be serviced by a professional according to manufacturer's specifications or at least every 5 years.

8. Waste disposal

- All empty or partially filled cylinders should be labelled as empty, set aside from full cylinders and returned to the supplier.

9. Related material

- Risk assessments for individual gas cylinders.
- SDSs for specific gases.
- Gas equipment operating manuals.

References:

'Australian Dangerous Goods Code', National Transport Commission website http://www.ntc.gov.au/heavy-vehicles/safety/australian-dangerous-goods-code/ (Accessed September 2015)

BOC Limited 'Safety Data Sheet #051: Hydrogen' 20 August 2015. ChemAlert website Msds.chemalert.com/company/5071/download/0009715_001_001.pdf

'Industrial gases Australia' BOC Australia website http://www.boc-gas.com.au/en/index.html (Accessed September 2015). Guidelines for gas cylinder safety.

Safe Work Australia 'Labelling of workplace hazardous chemicals – Code of Practice March 2015', Safe Work Australia website http://www.safeworkaustralia.gov.au/sites/SWA/about/Publications/Documents/643/labelling-workplace-hazardous-chemicals.pdf

Standards Australia. 2004. AS 4332-2004 *The storage and handling of gases in cylinders*. Sydney, Australia.

Standards Australia. 2004. AS/NZS 2243 *Safety in Laboratories, Part 10: 2004 Storage of chemicals*. Sydney, Australia.

Standards Australia. 2006. AS/NZS 2243 *Safety in Laboratories, Part 2: 2006 Chemical Aspects*. Sydney, Australia.

Standards Australia. 2010. AS/NZS 2243 *Safety in Laboratories, Part 6: 2010 Plant and equipment aspects*. Sydney, Australia.

University of Wollongong, WHS Unit. 2012 'Storage and Handling of Gas Cylinders Guidelines' University of Wollongong, Science, Medicine and Health website http://smah.uow.edu.au/content/groups/public/@web/@ohs/documents/doc/uow136686.pdf

STANDARD OPERATING PROCEDURE:

Title

1. **Introduction**

2. **Context**

3. **Safety notes**

4. **Regulations, licences and permits**

5. **Equipment**

6. **Operating procedure**

7. **Trouble shooting/emergencies**

8. **Waste disposal**

9. **Related material**

References: