Lab Skillz 101

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GENERAL LABORATORY RULES

Appropriate Laboratory Clothing

Required Apparel – Required laboratory apparel includes long pants, closed-toed shoes and, when appropriate, lab coats, gloves, safety glasses and enhanced PPE (hearing protection, respirators etc.). Lab coats, eye and ear protection, dust masks, etc can be obtained from AFNS Stores (AF 3-51).

- Shorts, skirts and sandals are not considered appropriate laboratory clothing.
- Gloves and lab coats are worn to protect yourself and your experiment from contamination – these are not to be worn in 'Public Areas' (SUB, lounges, etc) or in office environments (some laboratories have 'office' designated spaces). After removing disposable gloves it is important to wash your hands before leaving the workspace. ***Do not touch door handles, instrument controls, keyboards, elevator buttons, etc while wearing gloves to avoid contamination***. Disposable gloves, if contaminated, must be safely decontaminated before being placed in the garbage.
- Ensure that those working around you are wearing PPE appropriate for the hazards presented by your work.
- Contact lenses and regular glasses are not a substitute for safety glasses. You should make it known that you wear contacts in case of an eye injury.
- Be selective in the personal clothing you wear. Spills can ruin expensive clothing.
- Refrain from wearing loose fitting clothing and dangly jewellery. Ensure that long hair is tied back.

Food or Drink in the Laboratory

Food or drink must never be consumed in laboratories. Eating and drinking is allowed in offices and 'office' spaces (this is a primary reason why lab coats, gloves etc are not allowed). Do not store food in laboratory refrigerators, use only designated areas. Never dispose of food wastes (pop cans, food wrappers) in lab garbage cans.

Unauthorized Assistance with Work

Unauthorized persons may ***not enter*** a laboratory and may ***not assist*** you in your work. Examples are family members or friends who are not University employees. All persons working in laboratories are required to undergo the same orientation and training as you prior to beginning work. When necessary consult the department's volunteer policies.

- Children are especially not allowed in restricted areas or labs. Concerns have been expressed regarding work and safety issues surrounding children in restricted areas and labs during office hours, evenings and on weekends. The University of Alberta Risk Management policy is that ***children of staff, teaching assistants, graduate students, volunteers or students are not allowed in restricted areas or in laboratories at any time***. The liability issues for the University and the potential dangers for children are such that the University requires everyone to adhere to this policy in all circumstances.

Uncluttered laboratories & other work areas

Cluttered bench and table tops are an invitation to contamination and other hazards. Chemicals, *clean* glassware and equipment must be returned to proper storage when no longer in use.

- Aisles and passageways must be free of obstruction to avoid trips and spills and to allow a ready means of evacuation.
- Hand-held emergency eye wash and showers must be readily accessible, not blocked by items nearby. They should be *flushed weekly* to remove rusty water.
- No unlabelled containers. Proper labelling makes those around you aware of potential hazards.
- Workspace and fume hoods must be kept clean. Chemical storage in fume hoods is prohibited unless absolutely necessary.
- No chemicals should be stored on the floor, especially below or en route to safety showers and eyewashes
- Do not store personal items (backpacks, purses, textbooks, etc) in your laboratory work area. Keep these items in your office space or lockers to avoid having them contaminated and to avoid contaminating the work around you. Space in AFNS labs is very limited, therefore, keeping these items out will reduce the clutter and open up space for laboratory work.
- Bicycles and pets are not allowed inside buildings.

Transporting Chemicals

When transporting chemicals within laboratories, or between laboratories, an appropriate secondary containment vessel is required. This assists in reducing breakage and containing spilled products. Transportation of Dangerous Goods (TDG) regulations may apply and training may be necessary when transporting chemicals via vehicles.

Labelling Chemicals, Solutions, Mixtures, Samples

Proper labelling of laboratory materials is important for several reasons. A proper label allows identification of owners, possibly avoiding inadvertent disposal. All chemical and hazardous wastes are disposed of through the University's ***CHEMATIX*** system and

unidentified materials are not accepted. On new reagents, chemicals and materials print the *month and year received* and your *supervisor's name*. On containers of transferred reagents, solutions, mixtures or samples you must include your name, your supervisor's name, chemical composition or a description of the material (i.e. canola extract, feces etc), and the date prepared.

Storage of Chemicals and other Hazardous Materials in the Laboratory

Maintenance of an inventory of all chemicals is required by the Occupational Health & Safety Act. Date products on arrival and, especially for peroxide-forming products, date when opened.

Chemicals must be segregated by classification (acids, bases, flammable etc) to ensure that incompatible chemicals are not stored next to each other. Also ensure compatibility of chemical with container, ***especially if it is a prepared solution***.

Ensure that chemicals are stored according to instructions (e.g. refrigerate, freeze, away from sunlight, stored under N2). Check MSDSs for additional storage information.

Use storage and shelving appropriate to the chemicals. ***Do not*** store chemicals above eye level. ***Do not*** store liquids or chemicals in glass containers above countertop level. ***Do not*** store chemicals on the floor. ***Do not*** use fume hoods for general storage, except when absolutely necessary and only if the hood is not actively used.

- *Flammable and combustible liquids* must be stored in approved cabinets (which should not be near room exits or in main aisles). Ordinary refrigerators and cold rooms *must not* be used for storage of flammable liquids. *Refrigerators rated for storage of flammables are in AF 1-20K, 4-35 and 4-60*.
- *Routine reagent chemicals* should be stored on shelves (best with glass sliders or doors) or in cabinets designed for this purpose.
- Noxious chemicals should be stored in well ventilated cupboards, or in designated fume hoods or areas in fume hoods (note: fume hoods used routinely for lab work should *not* be major storage places for chemicals).
- *Biohazardous infectious materials* must be clearly marked and in secure storage. Incubators, refrigerators, cold rooms, etc. containing potential pathogens must have clearly visible *biohazard* labels; those containing *radioisotopes* must also be properly marked.

Send obsolete products for waste disposal or recycling.

Disposal of Waste Materials

Containers are provided for the segregation of waste (chemicals, glass, regular garbage etc) in each laboratory. Do Not put any chemicals or hazardous wastes down the sink or in the garbage. All properly identified chemicals and hazardous wastes must be disposed of by creating a ***CHEMATIX*** label for each container and submitting a waste pickup worksheet online.

- No chemicals or waste products in glass can be stored on the laboratory floor.

- All containers must be capped (do not leave funnel in neck of container), i.e. no open waste containers.
- All waste containers must be labelled prior to waste products being added and the contents *specifically listed* when adding to a container, including approximate percentages if a mixture (note: if water is present, list it as well). ***Unknown wastes will not be accepted by EH&S and disposal can be very costly - remember to appropriately label wastes.***
- Do not fill containers to the top; leave 5-10% free ("headspace").
- Segregate waste into separate containers.
- Rinse out empty containers and deface labels prior to disposal in the non-contaminated glass waste containers (yellow pail). Contaminated glassware (e.g. pasteur pipettes) must be disposed of separately.

Use of Equipment

A variety of equipment is available in the AFNS laboratories. Most of it is supported by individual research groups. It is, therefore, ***absolutely important that you obtain proper permissions and training prior to utilizing any equipment***, no matter how basic.

- Use equipment only for its intended purpose and do *not* use defective equipment. Report all damaged and inoperative equipment. ***Do not just leave it for the next person to deal with.***
- Leave or return equipment clean and in good working order. If there is a problem with the equipment, or if you have damaged it, advise the appropriate personnel. ***Do not just leave it for the next person to deal with***.
- Respond to all equipment alarms immediately, even if you are not using the item. Alarms are intended to alert users to issues (timers, problems, etc), ignoring them can have consequences for the instrument and, potentially, the safety of those around it.

Safety

In Alberta the Occupational Health & Safety Act, Regulation and Code (documentation and important information found at http://employment.alberta.ca/SFW/53.html) set out minimum requirements for health and safety in the workplace. In accordance with this, The University of Alberta Health & Safety Policy (see U of A Policies and Procedures Online: UAPPOL) reinforces the provincial legislation, outlining expectations of faculty, staff and students with respect to safety on campus. At present, the department provides a safety information seminar however; ***each Supervisor is responsible for the development and implementation of their own safety program***. The University and Government of Alberta recognize that safety in the workplace is a shared responsibility. Just as your supervisor has a responsibility to do everything that can reasonably be done to protect your health and safety, you must also do everything that can reasonably be done to protect your own health and safety and that of others around you. Accordingly, the following are applicable rules outlined under the Alberta Occupational Health & Safety Act:

- You must work in a safe manner and be safety conscious on the job.
- You must co-operate with your supervisor in the health and safety measures they have established.
- You must participate in training and apply it to your work.
- You must use PPE, such as footwear, eyewear, headwear, hearing protection, and protective clothing.
- You must bring your concerns regarding anything you feel is unsafe to your supervisor.
- You must refuse to perform unsafe work.

Courtesy and Respect

Laboratories in AFNS are busy places and space is at a premium. Obeying rules, particularly those related to reducing clutter, will help to make the workspace more comfortable for yourself and others.

Aside from that there are several common sense rules that come under the heading of 'courtesy and respect':

- ***Adhere to appropriate cell phone etiquette***. Cell phones have become very useful tools in our everyday lives; however, they can also be very obtrusive. Cell phones should be turned off when you are entering meetings or training sessions. You should never answer a cell phone in the middle of a conversation, especially if you are receiving assistance or advice from someone. Doing so gives the impression that you place little value on their time. If you are expecting an important call, inform those around before you start so that they will also expect it.
- ***Prepare for things well in advance***. Get training for procedures and equipment ahead of time, when it is more convenient for both the trainer and yourself. Waiting until the last minute can put your experiment or analysis at risk. Remember, it is you that needs the help and courtesy should always be shown to those whose help you seek. When receiving help and training, listen and/or read instructions very carefully – take notes if necessary. We are all very busy and it is often difficult to make time to do training the first time, let alone a second or third time.
- ***Obtain all necessary supplies in advance of your experiment or analysis***. The middle of an experiment or analysis is not the time to be looking for a critical chemical. Never assume somebody else is going to provide chemicals and supplies for you; your supplies are the responsibility of your supervisor. If you do need to borrow something; ***ASK first***, don't just take. If you borrow something return it or replace it as soon as possible – in the best condition possible. To avoid contamination of chemicals, especially those that you have borrowed, do not return unused chemicals to their original container – they are to be disposed of.
- ***Clean your glassware immediately*** after use; do not leave it in common areas, especially sinks, for someone else to deal with. Clean your workspace, especially those in common areas immediately after use.

- ***Dispose of old chemicals and samples*** in a timely manner; do not leave them on benches, in fume hoods, in refrigerators, or in freezers. Make sure everything is appropriately labelled.

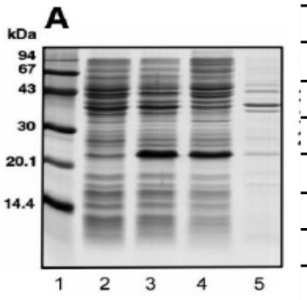
Lab Note Books

A well kept notebook provides a reliable reference for writing up experiments for publication and enables others to reproduce an experiment accurately.

Guidelines for keeping a notebook:

- Use permanently bound notebooks
- All entries should be written in ink
- Enter all information directly into your notebook
- Blank spaces should be avoided but if they occur a line should be drawn through the area and initialed
- Staple or glue items such as computer printouts into the notebook. Alternatively, if there is a large amount of data use binders and reference them in your notebook
- Use the past tense
- Provide detail on items such as chemicals, equipment and protocols
- Set aside the first 4 pages of your lab book as a table of contents, and keep it up to date.
- Set aside 1-2 pages immediately after your table of contents as a consolidated space for defining all abbreviations, and keep it up to date.
- All numeric values should have the appropriate units associated with them.
- You must include sufficient detail in your lab book that someone unfamiliar with your project can understand why and how you conducted the experiments you did, and what conclusions you drew from them.
- Centrifugation steps – if you report speeds in rpm, you must also identify the centrifuge and the rotor. Ideally, all speeds should be converted to x g.
- Buffers/media compositions – provide recipes for all buffers/media at least once per lab book (for subsequent mention of the same buffer refer to the page number where the composition is defined). If the composition is proprietary, list the manufacturer and the catalog number of the item you are using.
- Kits – if you are using a kit according to the manufacturer's instructions, include at minimum, the name of the kit, the catalog number and the manufacturer. If you can reasonably do so in the space provided, past in the manufacturer's protocol or briefly summarize. Identify any options or deviations from the standard protocol.

Date	*Title of Experiment* **5**
	Purpose of Experiment: Briefly describe the purpose and objectives of your
	experiment. If the current experiment is a continuation of a previous one,
	refer to the page number of the experiment you are continuing.
	Protocol: Give all details of the protocol that are required for someone to
	reproduce your work. Include description of the samples used and their source,
	make and model of all equipment used, and step-by-step details of the
	procedure. Define all abbreviations.
	If you are referring to a published method, give the full citation. If you
	obtain a method from someone else, identify who you got it from. If you
	have already given details of the procedure elsewhere in the CURRENT
	lab book, reference the page number.
	Fully explain any modifications you made to the protocol you are referencing
	regardless of its source.
	Results: Show the data required for you to come to your conclusion.
	If you paste data or photos into your book, ensure that the figures/tables are
	fully annotated (lanes labelled, bands of interest identified, etc.)
	All data should be taped securely onto a
	numbered, permanently bound page – do not
	"add" pages to your lab book by taping the
	figures into the margin. Clear tape preferred.
	Never remove a page from a lab book. If a mistake has been made, strike
	through the text with pen with two lines, such that the mistake remains legible.
	Conclusion: In a sentence or two, sum up the results of your experiment and
	any conclusions that you were able to draw from it. You may wish to identify
	future experiments that need to be done to confirm or expand upon these
	conclusions.
	Leave THREE LINES at the bottom of each page for the witness stamp.



AFNS

Stores

Storeroom Manager:	Laura Smith 780-492-7663
Location:	Room 3-51 of the Agriculture/ Forestry Centre
Hours of Operation:	**Monday - Friday** 9:00 am - 3:00 pm
Office Hours:	**Monday – Friday** 10:30 am – 11:30 am 1:00 pm – 3:00 pm

Purchasing Inventory Items

AFNS Stores is located on the main campus of the University of Alberta in Room 3-51 Agriculture/Forestry Centre. We provide a variety of lab supplies and gases as well as a limited number of chemicals.

Department members needing to make inventory purchases must present an active grant speed code. All graduate students and technical staff must complete a "Delegation of Signing Authority" form before purchases may be made. (These are available from the Department General Office, Room 4-10 Agriculture/Forestry Centre.) Most inventory items are located in Rooms 3-53 and 3-50 AF. After choosing your items the transaction is then entered on the AFNS Stores computer located in Room 3-50 AF.

Solvent chemicals and ethanol are located in the flammable storage room (restricted access) and the gas cylinders are in 1-40A. Liquid nitrogen is located in Room 3-49. When purchasing liquid nitrogen or other gases complete the sheet hanging on the wall next to the door. Enter the amount taken, in liters for liquid nitrogen, or name the gas and include the date, your name and your supervisor's name.

Purchasing Dry Ice

On Monday and Wednesday mornings 69 lbs. of dry ice is delivered to 3-49 Agriculture/Forestry Centre. This dry ice is meant for use in procedures and you must sign out indicating how many scoops of dry ice you have taken. If you are shipping an item or require large amounts, the dry ice must be special ordered and you will be charged accordingly. Please let Stores staff know 3 days ahead of when you require extra dry ice.

Purchases of Non-Inventory Items

AFNS Stores will place orders with contract and non-contract suppliers. Please complete an AFNS Stores requisition, available from AFNS Stores. For such orders there is a surcharge based on a percentage of the original value of the order, up to but not exceeding a total of $30.00. If there are any problems with orders or returns to suppliers, Stores will handle this for any orders they have placed.

Orders with Fisher Scientific and Sigma/Aldrich/Supelco Chemicals are placed on Tuesday and Thursday (~ 11:30 am). All other orders are placed on Fridays. Should you request an order be placed outside of these days a $20.00 surcharge will be assessed. If you want e-mail notification of delivery time please indicate this on the Stores Requisition. Stores Requisitions are available from the Stores Manager, as well as in the bookcase outside Stores (3-51AF).

ORDERS PLACED THROUGH AFNS STORES MUST BE DELIVERED TO THE MAIN CAMPUS, AGRICULTURE/FORESTRY CENTRE. Delivery to other locations will not be handled by AFNS Stores. AFNS Stores can provide information on how this type of order may best be placed.

ORDERS PLACED BY INDIVIDUALS will be delivered to 3-51 AF. Ensure your name appears on all orders you place. When the items arrive the requisitioner is notified by e-mail and all packages are placed in 3-49.

Receiving

AFNS Stores receives packages daily, usually between 1:00 pm and 2:00 pm. When items are received the requisitioner is notified, usually by e-mail. If you are ordering item(s) that require special handling instructions (i.e. dry ice packaging) please leave a telephone number you may be reached at and the name of an alternate person (and their telephone number) in case Stores is unable to contact the originator. Dry ice cannot be placed in a freezer, therefore, all orders received on dry ice will be held at room temperature. If the package is not picked up in a timely manner AFNS Stores will not be held responsible for any spoilage or loss of product.

If the item(s) require refrigeration or freezing they will be placed in the cold room/freezer room, 3-48, 3-48A AF. All other items will be left in 3-49AF. If 3-49 AF is closed an ABA1 key will open the lock.

Please remember we are not a storage area and have very limited space, so pick up your items as soon as possible.

Shipping

For help in shipping a package please see Stores Manager. If the packages do not contain Dangerous Goods then you can access the Shipping Form at http://www.financial.ualberta.ca/formscabinet.cfm; Supply Management Services, Distribution, Shipping Form. If the package is small it can go in Campus Mail; if larger then call Dispatcher at 2-4122 to make arrangements for pick- up. To ship an item containing a dangerous good (dry ice, chemical) the shipper must have a Transportation of Dangerous Goods Certificate. AFNS Stores manager has current certification and will handle requests for shipping. If the only item in the package classed as a Dangerous Good is dry ice then shipping can usually be arranged in a day. For other Dangerous Goods items up to a week may be required for Stores to prepare the appropriate shipping forms, packaging and placards.

The researcher or designate must ensure that any items leaving Canada have the necessary import or export permits, provided in triplicate when the package is presented for shipping. Also, a letter stating that the samples/specimens are for research purposes only, the total number of containers and the volume/weight in each container and a brief description of the contents should be included in the letter and signed by the researcher. Dry Ice is considered a Dangerous Good and must be processed by an individual holding a valid Transportation of Dangerous Goods Certificate.

Statements and Invoices

Once a month a Statement and all Invoices for transactions through Stores will be sent to the Finance Office and the Finance Office processes the charges. If you require copies of invoices, please see the Finance Office in 4-10C Agriculture/Forestry Centre.

QIAGEN BIOBAR

AFNS Stores operates a Qiagen BioBar. Stock items are limited but any item from Qiagen may be ordered through the BioBar, providing savings on some items over regular University contract pricing and there are no shipping charges. Orders are placed on Friday mornings.

Lab Coats

Lab coats may be laundered through AFNS Stores. The dirty lab coats are picked up on Monday morning and returned the following Monday.

Complete an AFNS Stores – Lab Coat/Linen Cleaning form, available in 3-50, indicating the speedcode and return to the Stores Manager. Dirty lab coats go in the bin. Clean lab coats are placed on the shelf in 3-53 – Aisle 1, under the supervisor's last name. Return the enclosed sheet to the Stores Manager.

UNIVERSITY OF ALBERTA
Research Services Office

DELEGATION OF SIGNING AUTHORITY
Sponsored Research Funds

Project Holder			Position		
Faculty			Department/Division/Centre		
Project Title			Project Sponsor		
SpeedCode	Fund	Dept ID	Program	Class	Project Number

Signing Authority is: (√ Check one of the following)

☐ **Delegated to the person named below for this specific Project (complete sections 1, 2 & 3)**

☐ **Rescinded for the person named below for this specific Project (complete sections 1 & 3)**

☐ **Not delegated to anyone for this Project (optional - complete section 3 only)**

1. Delegate

Name of Delegate	Position
Term of Delegation: Start Date	End Date
Conditions of Delegation (e.g. type of expenditure, expenditure dollar limit)	

2. Declaration of Delegate Accepting Signing Authority

1) I hereby accept responsibility as delegated signing authority for the above Project, in accordance with University of Alberta policies and procedures, including the *Delegation of Signing Authority Procedure*.
2) I have the skill and knowledge necessary for the effective exercise of this authority.
3) I will use the funds only for the purposes for which they were awarded.
4) I will not authorize disbursements for which I am the recipient.
5) I will ensure that all expenditures:
- **a) are supported by appropriate documentation,**
- **b) conform to the terms and conditions of the sponsor,**
- **c) are consistent with the Project budget, where applicable,**
- **d) are processed to the correct expense classification, and**
- **e) occur within the award period.**

Signature of Delegate	Date

3. Declaration of Project Holder

I make this declaration in accordance with the *Delegation of Signing Authority Procedure*. If delegation of signing authority is made, I acknowledge that I will continue to be accountable for all expenditures charged to this Project.

Signature of Project Holder	Date

The personal information requested on this form is collected under the authority of Section 33(c) of the *Alberta Freedom of Information and Protection of Privacy Act* and will be protected under Part 2 of that *Act*. It will be used for the purpose of documenting the delegation of signing authority for the named project. Direct any questions about this collection to: Assistant Director – Agreements, Research Services Office, 222 Campus Tower, 8625 – 112 Street, Edmonton, AB T6G 2E1, 492-9705.

REV. 15 NOV 2005 RETAIN ORIGINAL IN DEPARTMENT/FACULTY

Solutions

Many experiments require that chemicals be used in a solution. A standard solution is a mixture of two or more pure substances. A solute is dissolved in a solvent forming a solution. Solutions are described as molar (M), normal (N), percent solutions (%) or mass per volume (ug/mL).

Definitions

Molarity (M) A 1 Molar solution contains 1 mole of a solute dissolved in 1 Liter of solvent. A mole is the mass (g) divided by the molecular weight (formula weight) (g/mole).

To prepare a molar solution you must know:

- the volume of solution required
- the molecular weight (formula weight) of the solute.
- the molecular weight of the solute can be found on the bottle the solute comes in purity of standard

Formula for molarity (M) is moles of solute per liter of solution.

Example:

Prepare 500 ml of a 0.25 M $Na_2B_4O_7$ solution

Purchased Sodium Tetraborate Decahydrate (99.5%)

The formula weight of $Na_2B_4O_7 \cdot 10\ H_2O$ is 381.4 grams per mole.

To determine what mass (m) of $Na_2B_4O_7$ is required to prepare 500 ml (0.5L) of 0.25 molar $Na_2B_4O_7$ multiply the molarity (M) by the volume (L) required and by the molecular weight (MW).

m=M x volume x MW

m= 0.25mol/L x 0.5L x 381.4 g/mol

=47.68 g

However in this case the purity of the standard is only 99.5%, therefore you must actually weigh out more than 47.68 g to get a 0.25 M solution.

47.68 g / .995 = 47.92 g should be weighed out.

Normal Solutions

Normality (N) A 1 Normal solution contains 1 equivalent weight of solute dissolved in 1 Liter of solvent. An equivalent weight is the mass (g) divided by the equivalent molecular weight. The equivalent molecular weight is the mass of the solute that will react with one mole of hydrogen.

In the case of Sodium Tetraborate 2 hydrogen atoms can be combined with $Na_2B_4O_7$ (or 2 electrons can be transferred).

Example:

Prepare 500 ml of a 0.25 N $Na_2B_4O_7$ solution

Purchased Sodium Tetraborate Decahydrate (99.5%)

The molecular weight of $Na_2B_4O_7 \cdot 10\ H_2O$ is 381.4 grams per mole.

The equivalent of $Na_2B_4O_7 \cdot 10\ H_2O$ is 381.4 divide by 2 = 190.7 grams per eqivalent.

To determine what mass (m) of $Na_2B_4O_7$ is required to prepare 500 ml (0.5L) of 0.25 N $Na_2B_4O_7$ multiply the normality (N)by the volume (L) required and by the equivalent weight.

m=N x volume x MW

m= 0.25 equivalents /L x 0.5L x 190.7 g/equivalent

=23.84 g

However in this case the purity of the standard is only 99.5%, therefore you must actually weigh out more than 23.84 g to get a 0.25 N solution.

23.84 g / .995 = 23.96 g should be weighed out.

A 2M solution may or may not be equivalent to a 2N solution:

A solution of HCl with a concentration of 1 molar is the same as a solution with a concentration of 1 normal.

A 1 molar solution of H_2SO_4 would have a normality of 2.

N=M x #electrons transferred

Percent Solutions

To prepare a percent solution you will need to know how the concentration is expressed. Percent concentrations may be expressed in three ways:

- Percent weight by volume (w/v) – grams of solute per 100 ml of solution
- Percent by volume (v/v) – ml of solute per 100 ml of solution
- Percent by weight (w/w) – grams of solute per 100 grams of solvent

To get a 10 % (w/v) of a $Na_2B_4O_7$ solution you do not weigh out 10 g of $Na_2B_4O_7 \cdot 10\ H_2O$ and dissolve it in 100 mL of solvent. If you did this you would actually get a 5.5% solution. Remember that only 2/3 of that weight is $Na_2B_4O_7$ the rest is water (201.2/381.4)

Mass per volume (g/mL) The weight of solute divided by the solvent volume.

Units

Milli = 10^{-3}

Micro = 10^{-6}

Nano = 10^{-9}

Pico = 10^{-12}

Femta = 10^{-15}

Steps in Solution Preparation

- Refer to a laboratory reference manual for any specific instructions on preparation of the particular solution and the bottle label for any specific precautions in handling the chemical. You may also refer to the material safety data sheet for the chemical.
- Weigh out the desired amount of chemical(s). Use an analytical balance if the amount is less than 0.1 g. Calibrate and level the balance prior to use.
- Add less than the required amount of water into appropriate size beaker containing a stir bar.
- Place chemical(s) in the beaker and mix on a magnetic stirrer.
- Prepare all solutions with MilliQ water
- When the chemical is dissolved, transfer to a volumetric flask and add the required amount of MilliQ water to achieve the final volume.

Store the solution in a bottle that has been labeled with the name of the solution, its concentration, the date and your name

Dilution of Stock Solutions

A stock solution is a concentrated solution. To determine how to dilute stock solutions to give the desired concentration use the following equation

$$C_I \text{ X } V_I = C_F \text{ X } V_F$$

C_I = intial concentration before dilution (concentration of stock solution)

V_I = initial volume before dilution (volume of stock solution required)

C_F = final concentration after dilution (desired concentration)

V_F = final volume after dilution (desired volume)

Example

You need 200 ml of a 0.1 M solution and have a 5 M stock solution

$5M \text{ X } V_I = 0.1M \text{ X } 200ml$

$V_I = 4$ ml

Add 4 ml of the stock solution to a volumetric flask and bring up to volume with distilled water.

Standard solutions should always be made up in volumetric flasks

Beakers $\pm$ 5% error

Erlenmeyer flasks $\pm$ 5% error

Graduated cylinders $\pm$ 1-2% error

Volumetric flasks $\pm$ 0.1-1% error

Dilution Factor

Solutions used in a lab are often much more concentrated than are desired or can be managed for a given protocol. It is frequently necessary to dilute these solutions to a desired level.

Definitions:
Aliquot: a measured sub-volume of original sample.
Diluent: material with which the sample is diluted

Dilution factor (DF)**:** ratio of final volume/aliquot volume (final volume = aliquot + diluent)

To calculate a dilution factor:

The dilution factor is the final volume/aliquot volume.

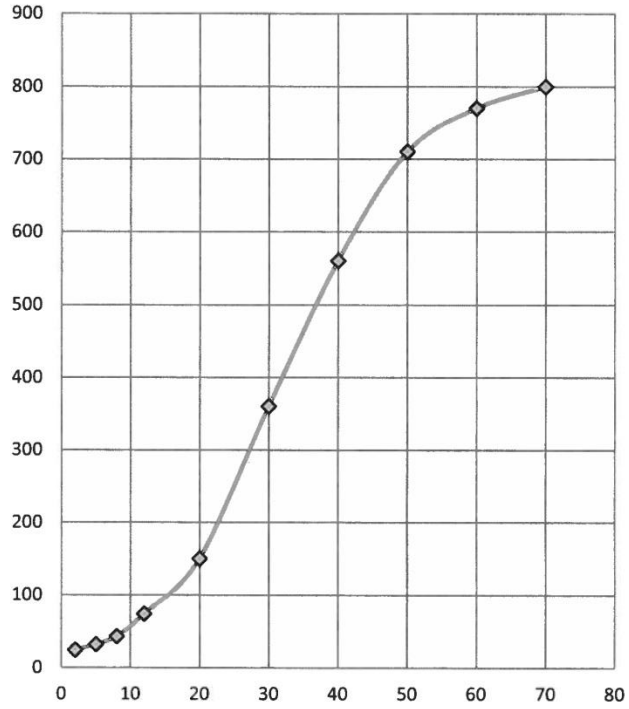
What is the dilution factor if you add 0.1 mL aliquot of a specimen to 9.9 mL of diluent?

1. The final volume is equal to the aliquot volume plus the diluent volume:
   0.1 mL + 9.9 mL = 10 mL

2. The dilution factor is equal to the final volume divided by the aliquot volume: 10 mL/0.1 mL = 1:100 dilution (10^2)

Standard Curves

- Incorrect Standard Curves can lead to erroneous results
- This curve exhibits linear, exponential and polynomial responses



- Depending upon the type of response, time, cost and range of samples will dictate the number of data points required.

- If you are only interested in qualitative response (is the compound present or not) then 1 standard amount is alright.

- Any other circumstance you should have at least 3 different amounts.

- In my opinion you should have 5 different amounts and duplicates of each amount.

1 Point Curve

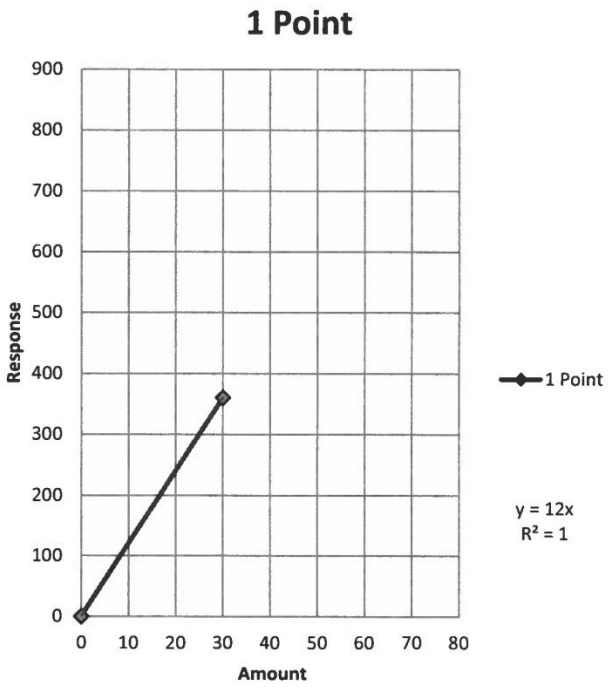
- Using 1 data point with of zero will yield a linear curve and a R2 of 1.00

Data points

Amount	Response
0	0
30	360

Unknown Samples

Response	Curve	Calculated
100	9	8.3
350	30	29.2
600	50	50.0



2 Point Curve

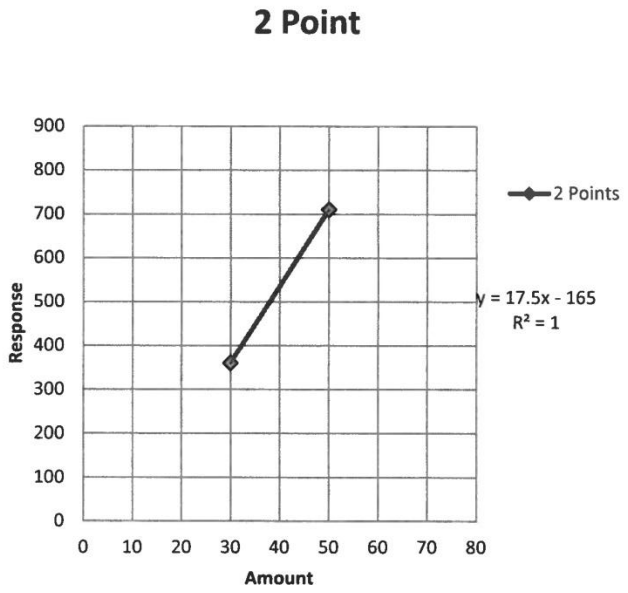
- Using 2 data points will yield a linear curve and a R2 of 1.00

Data points

Amount	Response
30	360
50	710

Unknown Samples

Response	Curve	Calculated
100	16	15.1
350	30	29.2
600	43	43.7



3 Point Curve

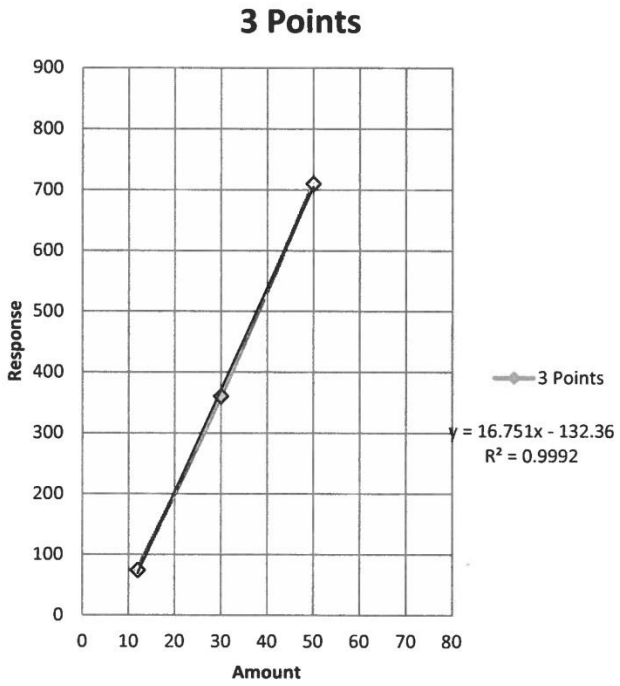
- Using 3 data points will yield a linear curve and a R^2 of 0.9992

Data points

Amount	Response
12	74
30	360
50	710

Unknown Samples

Response	Curve	Calculated
100	9	8.3
350	30	29.2
600	50	50.0



5 Point Curve

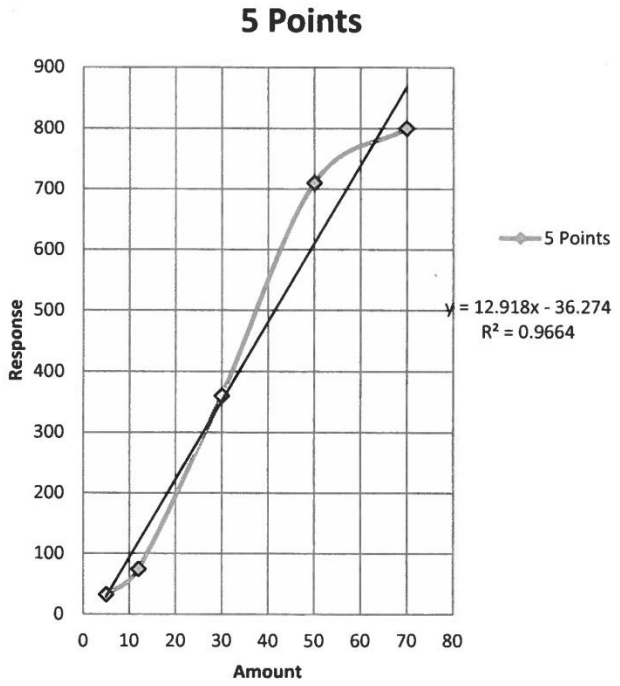
- Using 5 data points will yield a linear curve and a R^2 of 0.9664

Data points

Amount	Response
5	32
12	74
30	360
50	710
70	800

Unknown Samples

Response	Curve	Calculated
100	10	10.5
350	30	29.9
600	42	49.3



10 Point Curve

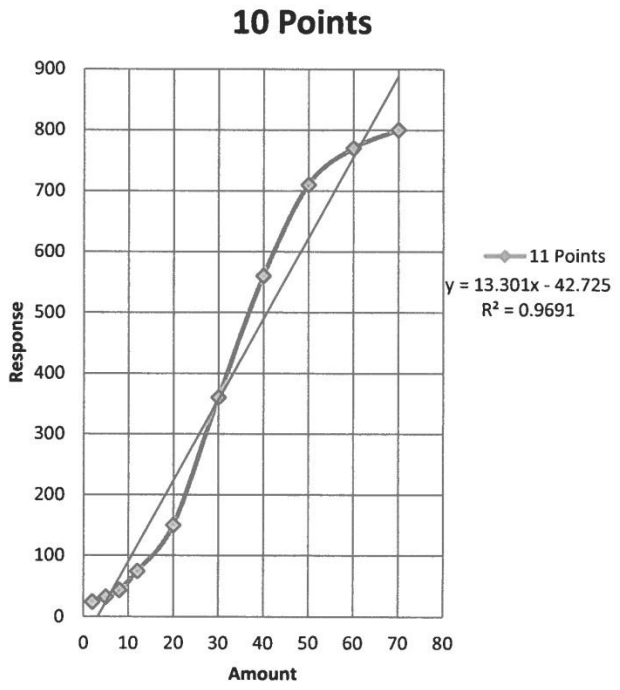
- Using 10 data points will yield a polynomial curve and a R^2 of 0.9691

Data points

Amount	Response
2	24
5	32
8	43
12	74
20	150
30	360
40	560
50	710
60	770
70	800

Unknown Samples

Response	Curve	Calculated
100	10	10.7
350	30	29.5
600	42	48.3



Balances

The choice of a top loading balance versus an analytical balance depends on the degree of precision required by the experiment. If decimal precision accuracy is not a major requirement a top loading balance is appropriate. If a test result is adversely affected by even minor changes in the weight an analytical balance would be a better choice.

An **analytical balance** is used to determine mass to a high degree of accuracy (0.001g). Care must be taken to ensure accurate weighing when an analytical balance is used. Analytical balances are enclosed to minimize dust contamination, to stop air currents and vibrations from affecting the operation of the balance.

- Ensure the balance is level by checking the leveling bubble. It should appear centered. If off center, use the leveling screws to level.
- Turn the balance on by pressing the control bar. The display lights up for several seconds and then resets to 0.000
- Close the doors, press the zero/control bar and when all zeros appear, the balance is tared.

- Center creased weighing paper on the balance pan. Always use a container when liquids, granules or powders are weighed. Do not weigh directly on the pan.
- Close the sliding doors. Wait for the green dot on the left to go out. This is the stability indicator light, indicating that the weight is stable. Press the control bar to cancel out the weight of the container or paper. The display will again read 0.0000.

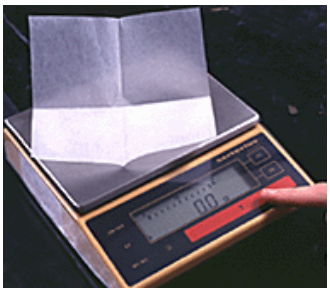
- Carefully weigh out the desired mass of the substance.
- Close the doors and allow the balance to stabilize.

- Before recording the mass, close the glass doors and wait until the stability detector lamp goes out. Record mass of solid.

Use the brush provided to clean spills in the weighing chamber. Discard any disposable tare containers, weighing paper, or Kimwipes in the proper waste disposal in the lab.

A **top loading balance** is used to weigh solids when a precision of 0.1 grams is adequate.
Check the level indicator to ensure the balance is level. Make adjustments as required to level the balance before use.

Check if the balance is turned on. If not, press the on/off button and wait until the display reads 0.0 g.



Place a container or large, creased weighing paper on the balance pan. Push tare button to zero the balance.

Carefully add substance to the container or paper. Allow the balance to stabilize. Record mass.

Use the brush provided to clean any spills. Discard any disposable tare containers, weighing paper, or Kimwipes in the nearest wastebasket.

A double pan balance is a scale that has two pans that are balanced against each other. Two pan balances are used for comparative weighing to determine the difference in mass between two objects.

- Move the sliding weight all the way to the left. The balance will be at or very near zero
- To set the balance to zero turn the adjust knob located at the right end of the beam
- Double pan balances can be used to balance tubes of samples prior to putting in a centrifuge

Some general notes for balances

1. Never use the balance for weighing an object of higher mass then is the capacity of the balance! Information about the capacity of the balance is usually given in the front panel of the balance.
2. If you are doing several weighings in a row, be sure to follow the same exact process every time.
3. If you are weighing hygroscopic materials, be prepared to work quickly. These materials tend to absorb water during the process, which alters results.
4. Always level and zero the balance. After you have done this, be very careful not to place objects on your table or bench, as this can affect the results. Also, watch your step. A good bump can vary results, too.

Demonstration: Weighing your signature

Try this experiment with two balances, the top loader and the analytical. You can start with either one.

1. Zero the balance by pressing the zero bar on the front face. This should give a digital display of zero.

2. Place a piece of paper near the center of the balance pan and record the mass directly from the digital display.

3. Remove the paper and write your signature on it with a sharpie.

4. Reweigh the paper with signature. Record the result on data page. Find out how much your signature weighs by subtracting the mass of the plain paper from that of the signed paper.

5. Repeat with the other kind of balance.

Weight of signature data sheet

	Analytical balance	Top loading balance
Weight of weigh paper		
Weight of weigh paper and signature		
Weight of signature		

Pipetting

The most common pipets used are air displacement pipets. The most common brand of pipets are Gilson. Other brands of pipets are operated in a similar manner; consult the manual that came with the pipette you are using for brand-specific instructions.

Air displacement pipets are laboratory tools used to transfer specific sample fluid volumes from one container to another. If done correctly, this transfer will be accurate and reproducible. Several factors contribute to maintaining an accurate and reproducible volume transfer of fluid. These include proper care of the pipet, routine calibrations and development of a skill level that is associated with correct technique. A pipet will only perform as well as the operator's technique. Despite the quality of the instrument and its calibration status variations in technique can alter delivery volumes.

Objectives of this exercise are the following:
1. Select the appropriate pipet and set the dial for the correct volume.
2. Distinguish the first and second stops and dispense the correct volume of liquid.
3. Draw up liquids without contaminating the pipet barrel.

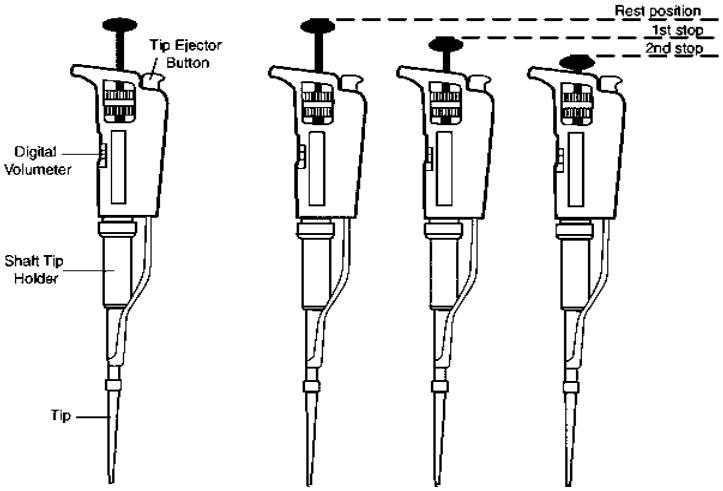
Setting the Volume
The volume of liquid to be aspirated is set using the volumeter. The dials are colored black or red to indicate the position of the decimal point. Turning the thumbwheel or the push-button sets the volume. The pushbutton makes it easier and quicker to set volumes, especially when wearing gloves. The thumbwheel may be turned to slowly reach the required setting. To obtain maximum accuracy when setting the volume, proceed as follows: when **decreasing** the volume setting, slowly reach the required setting, making sure not to overshoot the mark. When **increasing** the volume setting, pass the required value by 1/3 of a turn and then slowly decrease to reach the volume, making sure not to overshoot the mark.

The scales on pipets are in microliters (1000µl = 1 ml). They come in three sizes which are capable of pipetting three ranges of volumes: **P20** = 0.5- 20 µl, **P200** = 20-200 µl, and **P1000** = 200-1000 µl.
Never exceed the upper or lower limits of the pipettor.

IMPORTANT: Use common sense - if the dial is becoming hard to turn, don't continue to turn it. Normally, the dials are not difficult to turn. If you feel resistance, check to make sure that you have selected the correct pipettor.

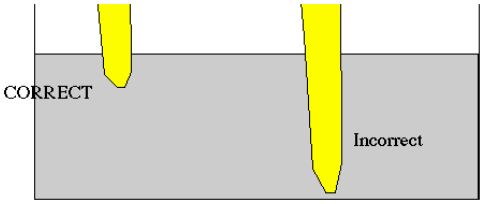
Add a tip.

Pipets use disposable tips. Tips are packed in plastic boxes. Make sure you are using tips that are the correct size/volume for your pipet (brands differ). Add a tip to the pipet by pushing down on the tip with a slight twisting motion. This will ensure a good seal between the tip and the pipet. Do not repetitively bang the pipette shaft into the tip. This may cause damage to the shaft over time. Tips are a single use item and should be discarded after use. REMEMBER TO CHANGE TIPS BETWEEN SOLUTIONS TO AVOID MIXING OR CONTAMINATING THE SOLUTIONS USED!! **DOUBLE DIPPING IS NOT ACCEPTABLE!**



The plunger will stop at two different positions when it is depressed. Be sure you can distinguish the two stop positions. The first of these stopping points is the point of initial resistance and is the level of depression that will result in the desired volume of solution being transferred. PUSHING BEYOND THE FIRST STOP IS THE MOST COMMON PIPETTING ERROR. Because this first stopping point is dependent on the volume that is being transferred, the distance you have to push the plunger to reach the point of initial resistance will change depending on the volume being pipetted. The second stopping point can be found when the plunger is depressed beyond the initial resistance until it is in contact with the body of the pipet. At this point the plunger cannot be further depressed. This second stopping point is used for the complete discharging of solutions from the plastic tip. You should not reach this second stop when drawing liquid into the pipet, only when expelling the last drop. **Before continuing, practice depressing the plunger to each of these stopping points until you can easily distinguish between these points**

Aspirate - Press the push-button to the **first stop**. Hold the pipette vertically and immerse the tip in the liquid.



Release the push-button slowly and smoothly (to **top** position) to aspirate the set volume of liquid. Wait one second then withdraw the pipet-tip from the liquid. You may wipe any droplets away from the outside of the tip using a kimwipe, however avoid touching the tip's orifice. Be sure there are no bubbles in the tip. Never let the operating button snap back to the ready position while aspirating the liquid. If you release the plunger too quickly, the liquid will shoot up to the top of the tip and into the barrel of the pipet. If you press the plunger past the first stop, and *then* draw up a liquid, you will have drawn up a volume that is greater than what you wanted.

Dispense - Place the end of the tip against the inside wall of the recipient vessel (at an angle of 10° to 40°). Press the push-button slowly and smoothly to the **first stop**. Wait for at least a second, press the push- button to the **second stop** to expel any residual liquid from the tip. Keep the push-button pressed fully down and (while removing the pipette) draw the tip along the inside surface of the vessel. Release the push-button, smoothly. Eject the tip by pressing firmly on the tip-ejector button.

General Cautions:

Be careful not to drop the pipet. The best way to avoid dropping the pipet is to keep it away from the edge of the lab bench.

When not in use the pipet should be rested upright on the pipette stand in the vertical position with the tip cone pointing downwards to prevent liquids entering the inside of the pipet.

Never invert or lay the pipet on its side while there is liquid in the tip.

Pipetting Hints:

When pipetting into microcentrifuge tubes, dispense the liquid near the bottom of the tube and hold the tube at a slight angle. Touch the tip to the bottom of sides of the tube while pipetting.

When adding several different reagents to one tube, pipet the largest volume into the tube first, then pipet the other reagents into that liquid. This is especially important when pipetting small volumes (<2 µl). Capillary pressure tends to keep the liquid on the end of the tip yet the small drop comes off easily when placed into the larger drop of liquid. If you hold the tip suspended over the tube, some of the liquid will stick to the tip. If you touch the tip to the side of the tube while pipetting, then all the liquid will be released from the tip.

PIPETTING EXERCISE

Use the tubes with colored water to practice pipetting. Try to develop a sense for the volume of liquid you are pipetting by looking at the amount of liquid in the tip. It is easy to grab the wrong pipet or to set it incorrectly, however you should be able to tell the difference between 1 µl in a tip and 10 µl in a tip just by looking at the amount of liquid in the tip. Often the yellow tips will be graduated (with marks at 10 µl, 50 µl, and 100µl).

Exercise 1.

Which pipette would you use to pipette the following volumes:

35 µl **100 µl** **250 µl** **630 µl**

Exercise 2.

Check your pipetting technique by weighing 100 µl of colored liquid using the appropriate pipette, tip and balance. You can assume the colored water has a density of 1mg/µl (e.g. 100 µl = 0.100g).

Volume	**100 µl**
Measure 1	
Measure 2	
Measure 3	
Average	

pH Meter

Please note: The pH meter in your lab may be different than the one we are using today. Please check the manual and/or ask one of you lab mates for help.

Basic information about pH

- pH measures the acidity or alkalinity of a solution.
- The pH scale runs from 0-14. pH 7 is considered neutral and pH values below 7 are acidic, while pH values above 7 are basic or alkaline i.e. lemon juice has a pH of 2, laundry detergent has a pH of ~8.0, distilled water has a pH of around 5.5
- pH is a term used to translate the amount of hydrogen ions (H+) in solution into a number between 0 and 14. The pH meter uses an electrode to measure the concentration of H+ in the solution and reports this as a pH value
- If there are many H+ the pH value will be closer to 0, or more acidic and if the pH value is larger that means there are less H+ in solution.
- The pH scale is a logarithmic scale. That means that pH 2 is 10 times more acidic than pH 3 and is 100 times more acidic that pH4.

How to standardize the pH meter

- It is important to standardize the pH meter daily
- Remove the electrode from the electrode buffer and rinse with distilled water and then blot dry with a Kimwipe
- Put the electrode into the first standard buffer solution (pH 4, pH7, or pH10) and then standardize the meter to the pH of this solution. Note: choose the two standards closest to your desired pH. For example, if you need a pH 6 you would want to standardize using pH 4 and pH7 buffers.
- Remove the electrode and rinse with distilled water and blot dry with a Kimwipe.
- Put the electrode into the second standard buffer solution and then standardize the meter to the pH of the second solution.
- Again, rinse the electrode with distilled water and blot dry.
- In order to make sure that you have calibrated the pH meter correctly measure the pH of the standard solution. For the pH 10 standard you should get a pH reading of 10.

How to measure the pH of a solution

- It is easiest to measure the pH if your solution is in a beaker.

- Place your beaker onto a magnetic stir plate. Place a magnetic stir bar into the beaker and make sure that there is clearance for the electrode to be submersed in the solution while not coming into contact with the stir bar. Turn the stir plate on low to gently stir the solution
- Again, rinse the electrode in distilled water and blot dry. Immerse the electrode into the solution.
- Change the pH meter from standby to measure.
- Then you will then see the pH reading on the screen.
- If the pH is too low (acidic) you will need to add NaOH to make the solution more basic. If the pH of the solution in too high you will need to add HCl to make the solution more acidic.
- If the pH is off by a whole unit, for example – the pH reads 3 and you need a value of pH 4, you will use more concentrated NaOH (5 M). If you needed a pH of 2 you would add concentrated HCl (12 M). As you approach your desired pH you would switch to 0.1 M NaOH or 1 M HCl, respectively.
- Using a dropper add the acid or base dropwise, waiting for the pH of the solution to stabilize before adding the next drop
- When your solution reaches the desired pH remove the electrode from the solution and rinse with distilled water
- Return the electrode to the buffer solution and set the meter to standby
- When you have finished using the pH meter empty the rinse water container, properly dispose of your dropper and wipe off the lab bench so the area is clean for the next user.

Before making solutions check to see if components need a special pH to dissolve (eg. EDTA needs pH 8.0) and always set the pH (and remove the stir bar) before autoclaving.

Centrifugation

Centrifugation is a process that involves the use of centrifugal force to separate the components of a mixture. The process used may be differential or density gradient centrifugation. Differential centrifugation entails spinning samples at a given speed resulting in a supernatant and a pellet. Density gradient centrifugation separates mixture components based on their relative density. In a molecular lab, we usually use differential centrifugation. Centrifuges are generally divided into three classes: Low Speed including benchtop microcentrifuges (up to 15,000 rpm), high Speed (15,000 rpm to 25,000 rpm) and ultracentrifuge (25,000 rpm or higher).

The fixed-angle and swinging-bucket are the standard laboratory rotors. High-speed and ultracentrifuges are equipped with refrigeration units, which is necessary as high-speed spins result in the generation of heat. Selection of centrifuge and its rotor is based on several factors including the sample size, the number of samples, the need for refrigeration, or the centrifugal force necessary to separate the mixture components.

Basic steps of centrifugation

- Prior to operation of any centrifuge, the user is expected to review the Manufacturer's manual for the specific unit.
- Rotors should be used only with the correct centrifuge and the speed limitation of the rotor is stamped on the top of the rotor.
- Ensure that the rotors are fastened securely into the rotor shaft to avoid damage of the centrifuge.
- For selecting the appropriate tubes and rotor – consider sample volume, speed of centrifugation, and the composition of the tubes.
- The composition and style of the tube will vary with the composition of the sample.
- Not all tubes can withstand all speeds. For centrifugation above 5000g, ensure that the tube is appropriate for that speed.
- Microwell plates may be spun in a swing-out rotor in microplate carriers, or in a rotor that has been adapted for microplates.
- Fill the tubes to within 1-2 cm of the top.
- Balance the tubes – for benchtop and high-speed centrifuges, pan balance should be used to balance sample tubes against each other; microcentrifuge tubes should be balanced by volume.
- Cover the tubes.
- Put tubes in the centrifuge always in the same orientation; by employing this technique it will be easy to look for the sample pellet.
- Ensure that each tube has a balance tube directly across from it.

- Sample loads must be balanced and swinging bucket rotors must not be run with missing buckets.
- Improper loading and balancing of the rotors can cause the rotors to break loose while spinning, leading to damage.
- Make sure you record your name and necessary information in the sign-up book.
- Put the lid on the rotor, screw it down and adjust settings.
- Start the centrifuge; the user shall not leave the centrifuge until full operating speed is attained and machine appears to be running safely without vibration.
- If vibration occurs, immediately stop the centrifuge and check that the load is balanced.
- Remove samples from the centrifuge as soon as it stops spinning – pellets can become dispersed if the samples are left to sit.
- Immediately clean up any spills inside the centrifuge
- Clean rotors using rotor cleaning solution and a rotor cleaning brush.
- Close the lid on refrigerated centrifuges between runs to avoid condensation.

How to determine the centrifuge speed

Speed in any protocol is described as either gravitational force (g) or revolutions per minute (rpm). G-force is also called RCF or relative centrifugal force and is the force exerted during centrifugation. Protocols usually provide centrifuge speed in g, which is a constant. The rpm is dependent on the force exerted during centrifugation as well as on the type and size of the rotor and the centrifuge model.

The relationship between RPM and RCF is as follows:

$g = (1.118 \times 10^{-5}) R S^2$

Where *g* is the relative centrifugal force, R is the radius of the rotor in centimeters, and S is the speed of the centrifuge in revolutions per minute. For example, centrifugation of a sample at 5,000 RPM in a microcentrifuge that has a rotor with a radius of 7 cm will deliver a centrifugal force of 1,957 × *g*.

Fume hood

- Any work that involves the use of toxic, hazardous or odourous materials must be performed in the fume hood.
- The fume hood works by creating a negative pressure environment; it pulls air into the hood and out the exhaust system. That way fumes and vapours from the materials you are working with will be vented rather than inhaled by yourself or others.
- Fume hoods in our building are functional 24 hours a day, i.e. they are always on. Notice will be given if the fume hoods are to be shut down.
- If you have concerns about whether or not the hood is functioning hold a Kim wipe or tissue at the sash opening.
- Do not use the fume hood for storage of chemicals or other materials. The reason for this is that air flows into the hood and up the back of the hood into the exhaust system. Obstruction of this area prevents the hood from properly venting and presents an opportunity for fumes to enter the lab.
- Do not leave the hood wide open. The sash must be open 30cm/12 inches from the base when not in use. When working in the hood lower the sash so that there is enough rooms to fit your arms in and perform the procedure. Keep in mind that the sash acts as a barrier for fumes as well as splashes.
- Perform your work at least 25 cm from the front of the hood.
- Use slow direct movements while working in the hood. Minimize side to side movements as well as movements in and out of the hood. Such movements disrupt the flow of air and cause fumes to enter the lab rather than be exhausted.
- The fume hoods are equipped with lighting.
- When you have finished your work remove all the equipment that you introduced into the hood, clean the area and turn off the light. Return the sash to 30 cm/12 inches from the base.

Before beginning a project check the MSDS to see if any of the chemicals you will be working with should be used in a fume hood

Waste Disposal

Waste disposal is the responsibility of each lab group. The University of Alberta has a Waste Management System that provides the means for the disposal of hazardous and non-hazardous wastes. You will need an understanding of the laboratory rules for waste disposal prior to starting work in a laboratory setting.

Waste is categorized for disposal purposes. Categories include hazardous chemicals, nonhazardous chemicals, radioactive solids, radioactive liquids, biohazards and sharps.

Waste disposal is handled through a web-based system called Chematix. The Chematix system is designed to streamline the waste handling process and enable users as well as Environmental Services staff to process the removal and disposal of hazardous materials in a timely and efficient manner. The Office of Environmental Health and Safety provides passwords to access the Chematix system.

Hazardous Waste:

Hazardous wastes are those wastes which, due to their nature and quantity, are potentially hazardous to human health and/or the environment and which require special disposal techniques to eliminate or reduce the hazard.

- Hazardous chemical waste must be disposed of in accordance with Environmental Health and Safety regulations
- Label all waste – label must include the full chemical name, and the percentage of the mixture of any contents, must be specifically listed on the label
- Liquid waste containers must be capped
    - Use a tight fitting lid that will not leak if the bottle is tipped
    - Do not store bottles of liquid waste on the floor
    - Do not over fill bottles – leave about 10% airspace
- Do not flush hazardous chemical waste down the drain
- Do not put hazardous waste in the garbage

Non-Hazardous Waste :

Non-hazardous waste is defined as all waste that does not meet the definition of hazardous waste or hazardous recyclable material.

- Flush non-hazardous liquid waste down the drain using large amounts of water
- Collect non-hazardous syringes in a separate waste container and send to the Office of Environmental Health and Safety for disposal
- Non-hazardous solid waste may be put in the regular garbage
- Rinse and deface empty chemical bottles before discarding

Radioactive Waste:

- Prior to working with radioisotopes users must complete the Radiation Safety Course offered by the Office of Environmental Health and Safety
- Package liquid and solid waste for disposal through the Office of Environmental Health and Safety
    - Do not mix isotopes
- Specify the type and amount of isotope

Biohazardous Waste:

- Biohazardous waste includes human or animal tissues, body fluids, and microbial waste (including infectious agents)
- Transgenic materials, such as leaves and seeds, are also treated as biohazardous
- Biohazardous waste must be rendered non-viable and noninfectious before disposal – autoclave solids, add bleach to liquids
- Biohazardous waste transported to an autoclave outside the immediate laboratory must be on a cart, double bagged, inside a leak proof tray

Sharps Waste:

- A sharp is any device that can cut or puncture the skin
- Examples of sharps include needles, needles with syringes, scalpel blades, and razor blades
- Sharps must be disposed of in a self-locking, sealable, single use container made for that purpose
- Do not remove needles from syringes before disposal
- Do not cap needles before disposal

Equipment Maintenance and Repairs

Using Equipment

"If you don't understand the equipment you are using for an experiment, you don't fully understand the experiment."*

For each piece of equipment you use, you should know:

-how to operate it – read the instruction manual or protocol prior to using the equipment
-get a demonstration of its use from a qualified person in the lab
-is the equipment left on all the time or is it turned off after each use
-does the equipment require a warm-up
-is there a sign-up sheet

When you are finished using a piece of equipment you must wash or clean it and if you have moved it, return it to its proper location.

If there is ever a problem with a piece of equipment, determine the following:

-if you can easily and properly fix the equipment, then do so
-if the repair is beyond your means, arrange to get the equipment fixed by informing the person in your lab who is in charge of this, or by contacting:

Will Cornet -equipment maintenance technologist
3-05 Ag/For; ph 780-499-5853 (do not leave a message; rather call back if there is no answer)
-complete an instrument repair request form
-be sure to leave a note on the equipment indicating that it is not working

If a piece of equipment is in alarm you MUST respond, whether or not you are using the equipment:

-identify the source of the alarm
-notify the person in charge of the equipment, OR
-find someone who knows more about the equipment
-if you must deal with the alarm, determine first that there is no safety issue or lab emergency and only then can you proceed to deal with the situation
-be sure to let the pertinent person(s) know there has been a problem

*Barker, K. 2005. *At the Bench: A Laboratory Navigator*. Cold Spring Harbour Press, Cold Spring Harbour, New York.