School of
Civil Engineering

UNIVERSITY OF LEEDS

CIVE3750

Individual Research Project Dissertation- Aims and Objectives

A Comparison of Different Air Samplers for Evaluating Indoor and Outdoor Air Quality

By

[redacted]

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List of Abbreviations

AGI- All Glass Impinger
SKC- SKC Bio-sampler
1SA- Single Stage Anderson
6SA- Six Stage Anderson

1. An introduction to Air Quality

An aerosol is a solid particle which is suspended within a gaseous substance such as air, a bioaerosol is an aerosol which has a biological source such as plants, animals, soil or water. Bioaerosols can come in many forms including bacteria, fungi, viruses and pollen and have a size range of 20nm- 100μm (Heo, Kim, & Lee, 2014) (Mandal & Brandl, 2011). Bioaerosols are present in in all environments but indoor they can account for up to 34% of the air pollution and outside it can be as high as 50% of the airborne particles. The particles found in certain areas may not have originated from that place but could have been transported from another area due to their lightweight nature, therefore many bioaerosols which have outside sources are found inside (Mandal & Brandl, 2011).

Variety	Diameter (μm)
Bacteria	0.3-20
Fungi	1-100
Virus	0.003-0.3

Table 1- Bioaerosol Diameter
(Chen, Du, Xu, Liu, & Yan, 2017)

As shown in **Table 1** the largest bioaerosols are fungi and the smallest are viruses.

1.1 Factors that Affect Air Quality

There are many factors which affect air quality, some of these occur when artificially altering an environment while with others it is a more natural process.

Services within a building such as heating, air-conditioning and ventilation can cause changes in temperature and humidity which can lead to significant fluctuations in bioaerosol concentrations (Mandal & Brandl, 2011). This is due to temperatures affect on microorganism growth, when the temperature in a room is more than 18°C but less than 180°C, microorganism growth such as bacteria has been shown to increase. An increase in moisture in the air caused by an increased humidity also increases bacteria growth (Arfalk, 2015). Areas which have poor ventilation but are thoroughly insulated have been known to have elevated bioaerosals especially fungal spores such as mould (Douwes, Thorne, Pearce, & Heederik, 2003).

Human beings are also some of the largest sources of airborne bacteria, this is due to many daily actions such as talking, sneezing, coughing and toilet flushing (Mandal & Brandl, 2011), these actions expel particles which are then suspended within the air. The amount of bacteria being generated is therefore increased when the occupancy of a room is greater (Hospodsky, et al., 2012).

The materials used within buildings such as plants and textiles can release fungal spores into the air from the natural materials they are made out of, or from transference such as from someone's shoes being rubbed onto a carpet (Mandal & Brandl, 2011).

1.2 Health Risks of Bioaerosols

Indoor air quality can greatly effect the quality of life of the people who regularly inhale the air (Hospodsky, et al., 2012). The bioaerosols which are small enough to be inhaled are what cause the most concern, especially particles which have a diameter of less than 2.5μm as they are able to reach further within the respiratory system (Hinds, 1999)(Chung, et al., 2001). **Figure 1** is a visual representation of the respiratory system and the particle sizes which reach certain levels, the six stages refer to the stages of an Anderson sampler which will be discussed in section **2.1.1** of this report. The higher the concentration of airborne particles the more likely inhalation will occur, and particles such as allergens and bacteria are usually the most common to be inhaled (Douwes, Thorne, Pearce, & Heederik, 2003). Typically, bioaerosols which have sizes less than 1μm such as viruses, pose less of a health risk as natural processes such as sneezing mean they do not remain within the body for long (Kesavan, Schepers, & McFarland, 2010).

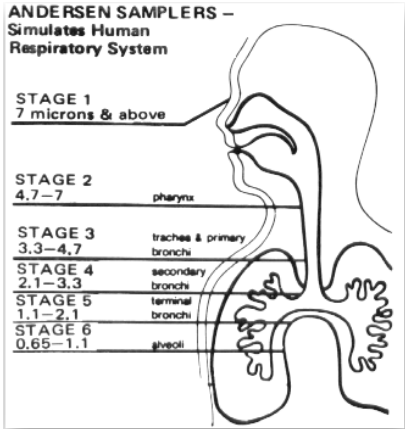


Figure 1- Aerosol Size Distribution within Respiratory System
(Fletcher, 2018)

The length of exposure to bioaerosols has been linked to many negative health imapcts (Mandal & Brandl, 2011). In general, bioaerosols can cause health problems such as respiratory issues, allergic reactions, asthma and infection, and fungal spores can cause headaches, eye irritation, coughs and building related disease as well as nasal and sinus congestion (Mirhoseini, Nikaeen , Satoh, & Makimura, 2016) (Xu & Yao, 2011).

It has been found that areas which extremely high concentrations of bioaerosols such as waste management sites have an increased rate of adverse health effects (Yao & Mainelis, 2006). The exposure to high levels of microorganisms causes a high chance of developing respiratory symptoms and airway inflammation (Douwes, Thorne, Pearce, & Heederik, 2003).

2. Air Samplers for Determining the Concentration of Bioaerosols

Bioaerosol concentration can be detemined in indoor and outdoor air by a range of samplers which use techniques such as impaction, impingement, filtration and electrostatic precipetation (Mainelis & Tabayoyong, 2010). The two most common air samplers are impingers and Anderson impactors (Mirhoseini, Nikaeen , Satoh, & Makimura, 2016).

There are some issues when it comes to bioaerosol sampling. When taking a sample it can be difficult to ensure than the sample is representative of the entire environment, this is due to some particle concentrations being extremely low. There is also risk of the microorganisms a being killed during the collection process due to their delicate nature (Fletcher, 2018).

2.1 Impactors

Impactors use a solid media such as agar to collect the samples (Mandal & Brandl, 2011). A fan draws air into the sampler through the perforations and directly onto the plates within, this is achieved by using the particles inertia to separate it from the gas (Mandal & Brandl, 2011) (Mainelis & Tabayoyong, 2010), an overview of this process this is shown in **Figure 2**. After a set period of time an aqequate volume of air to be sampled is reached, the sample can then be incubated to develop colonies which can later be counted and used to determine estimate numbers of mircoorgaisms in the air (Mandal & Brandl, 2011). Due to the samples being collected onto agar plates there is no process after sampling in order to prepare for culture (Main elis & Tabayoyong, 2010).

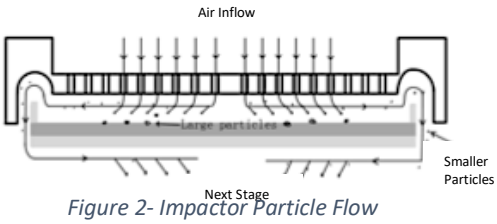


Figure 2- Impactor Particle Flow
(Chen, Du, Xu, Liu, & Yan, 2017)

The advantages of impactors are that they are portable (Fletcher, 2018), as they can be battery powered while still maintaining high flow rates (Mainelis & Tabayoyong, 2010). They are also easy to use, as once the plates have been prepared before hand they just need to be fitted into the sampler (Fletcher, 2018).

Some disadvantages are that the force from the impaction can cause particle damage to 99% of the sample due to the amount of stress the particles encounter when impacting onto the agar plate, as well as only one species is able to be determined at a time due to having to use different culturing media. After the sample is collected culturing is required to gain results and this can take numerous days (Fletcher, 2018). If sampling occurs over extended periods of time there can be a sample overload, this is most common in high concentration areas (Mainelis & Tabayoyong, 2010). Particles may rebound from the collection media if the impact force is too high or if there is already a particle impacted in that location (Kesavan, Schepers, & McFarland, 2010), this is shown in **Figure 3**.

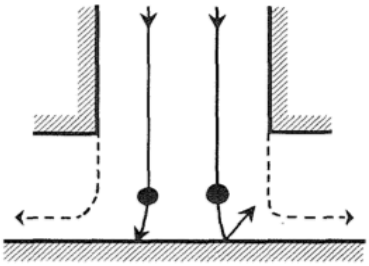


Figure 3- Particle Impaction and Rebound
(Kesavan, Schepers, & McFarland, 2010)

2.1.1 Single-Stage and Six-Stage Anderson Samplers

The Anderson sampler is one of the most widely used samplers. Both the single-stage and six-stage samplers have a flow rate of 28.3 litres/second (Chen, Du, Xu, Liu, & Yan, 2017). Examples of two samplers can be found in **Figure 4** and **Figure 5**.

Figure 4- Single-Stage Anderson Sampler
(EMLab, 2010)

Figure 5- Six-Stage Anderson Sampler
(Staplex, 2019)

Advantages of both types of Anderson sampler are that is has been shown that only low percentages ges of particles are lost to impacting on the walls of the sampler, therefore sterilization of the sampler between use if in quick succession is not necessary (Chen, Du, Xu, Liu, & Yan, 2017).

The key difference between the two impactors is that the single stage Anderson sample uses one agar plate and cannot determine size distribution, whereas the six-stage sampler uses six agar plates.

The six stage Anderson sampler allows particle size distributions to be determined (Fletcher, 2018) as shown in **Figure 1**, this is due to the multi-stage layout of the unit. The perforated plates use progressively smaller holes meaning the larger particles impact higher up the unit (Mandal & Brandl, 2011). This mimics the respiratory system, as not all particles reach the deepest parts of the lungs. At each stage there is a plate with perforations, in each plate the perforations are the same, however, further down the sampler the perforations are smaller (Chen, Du, Xu, Liu, & Yan, 2017). Large particles have a relatively high inertia and therefore impact further up the sampler than smaller particles (Chen, Du, Xu, Liu, & Yan, 2017).

2.1.2 Handheld, battery operated impactors

Handheld, battery operated impactors operate in the same way as standard impactors in that they draw air through a series of perforations, causing particles to impact on the culturing media (Cantium Scientific, 2015). They are portable, lightweight and still use standard petri dish sizes. Two types of handheld impactors as the Micro-Bio MB2 shown in **Figure 6** and the SAS sampler shown in **Figure 7**, they are best suited for use in controlled environments such as cleanrooms, operating theatres and in food manufacturing (Cherwell Laboratories, 2018). (Cherwell Laboratories, 2018)

Figure 7- Micro-Bio MB2
(Cantium Scientific, 2018)

Figure 6- SAS Sampler
(Cherwell Laboratories, 2018)

2.1.3 Positive-Hole Correction

Macher (1989) said that impactors with which the bioaerosols impact directly onto the agar plate must account for the probability that more than one particle was collected through the same hole. Due to only being able to count one colony per hole this skews the data collected. Macher (1989) devised a table which accounts for error at each stage of a six-stage Anderson sampler using jet diameter and velocity, this then leads to a table which determines how to adjust the colony counts for a 200-hole impactor.

2.2 Impingers

Impingers collect particles using a liquid media as opposed to the solid media used by the impactor. Within the impinger there is a vacuum which draws air through an inlet tube, the diameter of which determines the flow rate (Mandal & Brandl, 2011) (Fletcher, 2018). The air is then drawn into a flask which contains the collection medium and accelerated towards the medium's surface. Once the air hits the liquid it either suspended in the liquid or continues to the bottom of the flask where it rebounds and is then impinged into the collection liquid (Clauß, Clauss, & Hartung, 2011), **Figure 8** shows this occuring. For greater efficiency the jet of air enters the vacuum at a higher point than the flask and at an angle (Fletcher, 2018). After the collection process is complete, the liquid can be cultivated in multiple types of growth media (Mandal & Brandl, 2011).

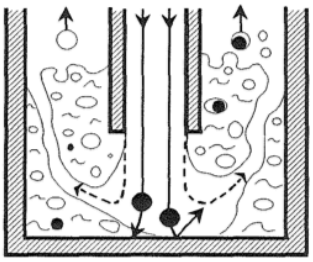


Figure 8- Particles Leaving Airflow in Impingers
(Kesavan, Schepers, & McFarland, 2010)

The advantages of this type of sampling are that a wide range of species can be determined in each sample due to the liquid having to be spread onto cultivation medium after collection is complete. Unlike with impactors there is no limit to sample time. It is also more efficient as more samples can be made with one test than with the impactor (Fletcher, 2018). During sampling, air is continuously drawn through the liquid, and in highly contaminated environments they are not overloaded as impactors are (Kesavan, Schepers, & McFarland, 2010).

Some disadvantages are that no size distribution can be determined as all of the sample is collected in one flask and that both a pump and power supply are required to collect the sample (Fletcher, 2018). The airflow requires to pull the air through the liquid can cause the liquid to bubble, this in turn can cause the particles already impinged within the liquid to be forced from the media and become aerosols once more. Due to the collecting media being similar in viscosity and surface tension to water this means that the liquid is prone to evaporation, therefore sampling time should be kept to a minimum to ensure the smallest amount of evaporation occurs (Kesavan, Schepers, & McFarland, 2010).

Both types of impinger discussed below uses an airflow rate of 12.5 litres/minute (Clauß, Clauss, & Hartung, 2011).

2.2.1 All Glass Impinger (AGI-30)

The AGI-30 was developed almost ninety years ago (Clauß, Clauss, & Hartung, 2011) and is typically the sampler used in bioaerosol sampling (Kesavan, Schepers, & McFarland, 2010). The sampler has a liquid capacity of 30 millilitres and a 30 mm distance from the bottom of the nozzle to the bottom of the flask (Clauß, Clauss, & Hartung, 2011). An example of the AGI-30 can be seen in the form of a sketch in **Figure 9.**

The high airflow velocity and low pressure within the flask can cause the particles collected to leave the liquid once they have already been collected (Clauß, Clauss, & Hartung, 2011).

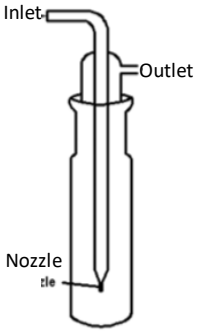


Figure 9- AGI-30
(Kesavan, Schepers, & McFarland, 2010)

2.2.2 SKC Bio-Sampler

The Bio-sampler was designed to slow down the airflow velocity and use an un-vaporised collection liquid. These improvements were to improve the sampling and retention efficiencies of the AGI-30. The volume of collection liquid is typically 20 millilitres for this sampler (Chen, Du, Xu, Liu, & Yan, 2017). **Figure 10** shows an example sketch of an SKC Bio-sampler.

Unlike the AGI-30 with which the singular nozzle is perpendicular to the collecting liquid, the SKC has three nozzles with curvatures causing the particles to impinge tangentially into the collecting liquid. Experiments have shown that the bio-sampler has better collection efficiency than the AGI-30 (Chen, Du, Xu, Liu, & Yan, 2017).

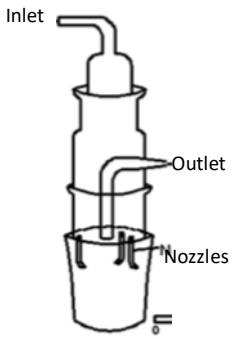


Figure 10- SKC Bio-Sampler
(Kesavan, Schepers, & McFarland, 2010)

The curved nozzles causes jets which simulate the upper respiratory system (Kesavan, Schepers, & McFarland, 2010), however these curvatures can cause the particles to impact against the walls of the flask, therefore leading to some losses of the smaller sized particles. These losses are however less than the losses caused by the airflow hitting the collecting liquid perpendicularly (Clauß, Clauss, & Hartung, 2011).

2.3 Filter samplers

Filter samplers pull air through filters which then intercept particles, the particles are then removed from the air and the efficiency of this process is determined by the face velocity of the filter, **Figure 11** is an example of a filter sampler. The sample is then washed from the surface, diluted and cultured (Casella, 2014) (Fletcher, 2018).

Filter samplers are easy to use, and one sample can be plated onto many media types, therefore many species can be determined from a single sample. Also, particles smaller than pore size are able to be collected. However, the more the filter is used the more contaminated it gets which reduces the air flow rate through the filter and therefore makes determining actual air sample volume difficult. The size distribution of the particles can also not be determined. There is also some error in sampling and from recovering the sample (Fletcher, 2018).

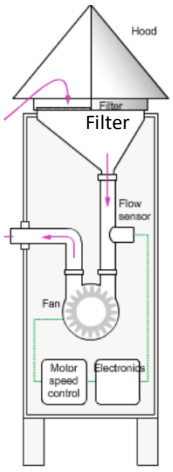
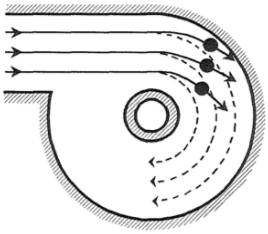


Figure 11- Filter Sampler
(Queensland Government, 2017)

2.4 Centrifugal samplers

There are three types of centrifugal sampler, the type depends on how the sample is collected, the three types are dry samples, liquid samples or samples onto a semi solid media (Fletcher, 2018). Typically, centrifugal samplers are used as pre-samplers before other samplers are used (Kesavan, Schepers, & McFarland, 2010).



With liquid samples a cyclone sampler is used. The air is drawn in at a tangent into the top of the chamber, the air then spirals down the cylindrical chamber. As the air is drawn through the chamber the particles impact on the inner walls (Kesavan, Schepers, & McFarland, 2010) (Fletcher, 2018), this is shown in **Figure 12.**

Figure 12- Centrifugal Cyclone Sampler
(Kesavan, Schepers, & McFarland, 2010)

The samples from semi solid media are collected using a centrifuge, where the air is drawn in using an impeller into a shallow drum and the air is forced onto the inner wall which is covered in agar where the particles then impact (Fletcher, 2018), **Figure 13** shows a representation of this occurring.

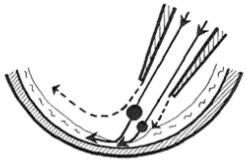


Figure 13- Centrifugal Centrifuge Sampler (Kesavan, Schepers, & McFarland, 2010)

In order to get a size distribution with these samplers a range of samplers with different chamber dimensions must be used. However, due to how the particles travel through the chamber it is less likely for particles to rebound from the walls or collection media than with other samplers (Kesavan, Schepers, & McFarland, 2010).

3. Aims and Objectives

The overall aim of this study is to compare the performance of different types of air sampler for determining the concentration of bacteria and fungi in indoor and outdoor environments. By comparing the various air samplers, it will determine the optimum sampler for different conditions.

A series of SMART objectives have been identified to achieve the overall aim, they are as follows:

1. ***Determine what research has already been done comparing different air samplers.***
   This will be carried out by conducting a comprehensive literature review.
2. ***Select the type and number of air samplers to be compared.***
   This has been done through discussions with Louise Fletcher, four air samplers have been selected two impactors and two impingers. Using the variation in the qualities of the samplers stated in section **2** as well as the range of samplers available for use differences in results will be determined.
3. ***Identify the locations for the air samplers to be tested.***
   This will be done by selecting a range of locations which have varying environmental conditions. The indoor areas such as offices, laboratories and lecture theatres within the School of Civil Engineering have been selected. These locations have varying degrees of occupation and sampling at various times throughout the day will give an accurate measurement of occupancy affecting concentration of bacteria and fungi. Outdoor locations such a green space, a roadside location and a waste management facility will also test the effect of concentration on the device's performances.
4. ***Carry out sampling for bioaerosol in indoor and outdoor environments***
   Using the three air samplers which have been chosen the concentration of bioaerosols at the selected locations will be determined. The methodology as stated in section **4** will be followed to collect this data. The samples will be taken simultaneously in order to make the results more comparable.
5. ***Determine the impact of bioaerosol concentration on performance***
   This is partly done during the location selection as this will give the varying concentrations. After carrying out the sampling the results will be collated and compared to see if the air samplers perform differently depending on the bioaerosol concentrations.
6. ***Determine the impact of bioaerosol type on performance***
   The total viable count detected from the four air samplers will be compared, but bacteria and fungi will also be counted separately. This will determine if the type of bioaerosol effects what is detected by the samplers.

4. Proposed Methodology

4.1 Sampling

4.1.1 Locations

The samples will be taken at twenty-one locations. These locations will be a wide variety of indoor and outdoor environments with a range of bioaerosol concentration levels. One of these locations will include going onsite at a materials recovery facility as this is likely to have high bioaerosol concentrations, other locations will include offices both of single and multiple occupancy and they will most likely range between low and medium bioaerosol concentrations, there will also be various outdoor locations.

4.1.2 Samplers in Use

The four samplers being tested in this project are the single-stage Anderson sampler, the six-stage Anderson sampler, the all glass impinger (AGI-30) and the SKC Bio-Sampler. Both impactors have a 28.3 litre/minute flow rate and both impingers have a 12.5 litre/minute flow rate. Two pumps, two filters and two flow meters will be used in order to run two samplers at a time, these pieces of equipment will be connected by tubing. The performances of the different impactors and different impingers will be tested as well as comparing the performances of impactors to impingers.

4.1.3 Preparation

On a Monday all the preparations will be made to streamline the sample taking later in the week.

1. First all of the equipment will be sterilised before use.
    - This is to ensure that no contamination of the samples occurs an autoclave will be used.
2. Next the agar will be made.
3. Then this agar will be poured into plates and stored until needed for sampling.

4.1.4 Sampling Process

All the samplers will be tested at the same time within the same environments. Three samples each will be taken with the six-stage Anderson sampler, the AGI-30 and the SKC Bio-Sampler, the single-stage Anderson sampler will have twelve samples taken with it on four types of agar. The agar will be sampling for total viable count, total fungi, gramma negative bacteria and total bacteria, for the six-stage Anderson sampler only total viable count will be sampled.
The set up for the impingers and impactors are shown in **Figure 14** below.

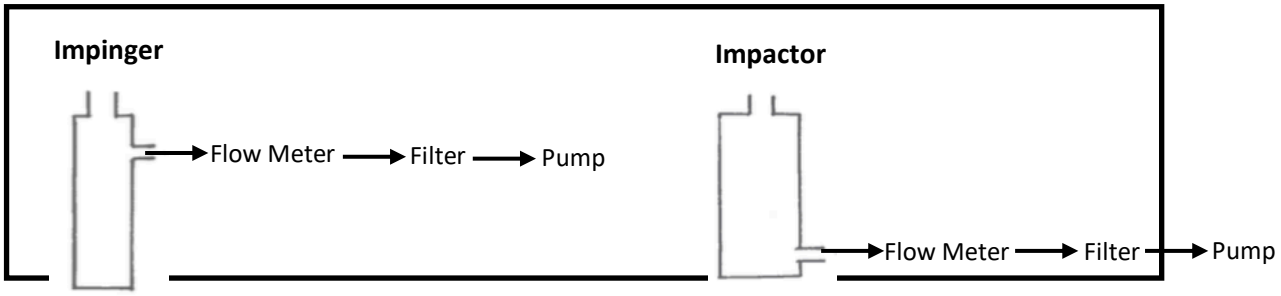


Figure 14- Sampler Set Up

Between 1-3 days a week (typically Tuesday, Wednesday and Thursday) will be dedicated to taking the samples, and three locations will be sampled per week.

1. When arriving at the location notes will be made on the conditions, such as if a window is open, if it is a multi-occupancy room or if outside whether it has rained recently.
2. First the single stage Anderson sampler and the all glass impinger (AGI) are set up, this entails connecting the samplers to filters, flowmeters and pumps.
    - Both samplers will be run at the same time to ensure the reliability of the samplers as they will be subject to the same conditions.
    - The AGI will be sampling for 15 minutes.
    - The single-stage Anderson sampler will be run for 5 minutes, therefore for each AGI sample three 1SA samples will be taken.
3. After the single stage Anderson and AGI the same test will be run for SKC, six stage Anderson sampler and the single stage Anderson sampler.
    - The SKC will run for 15 minutes
    - The six-stage Anderson sampler will run for 5 minutes
    - After the completion of the single stage Anderson sampler another sample will be taken from the six stage Anderson sampler.
4. Stages 2 and 3 will be repeated 3 times per location, there will be a total of 21 locations.

This process is displayed further in a flowchart in Appendix 8.1.

4.1.5 Culturing

Culturing is the next stage after collecting the samples.

1. First the samples will be taken to the lab.
2. Then the collection fluid used for the impingers will need to be diluted
    - This will ensure the results are able to be analysed without too many colonies to count.
3. Next the impingers diluted collection fluid will have to be plated on agar plates, each sample will be plated onto 4 types of agar.
    - There will be four plates per sample, there will be four types of agar in use.
    - Each plate will have 0.1 millilitre of solution spread onto it.
4. All of the samples for all samplers will then be incubated for 24 hours.

4.1.6 Analysis

At the end of the week on a Friday the samples will be analysed by counting the number of colonies on each agar plate.

4.2 Literature Review

Using SCOPUS:

The SCOPUS database will be used to carry out the literature review, therefore a range pf search terms needed to be determined.

- Terms such as “bioaerosol” and “Sampler” as well as terms related to specific samplers for example “impinger” “All glass impinger” and “AGI-30” etc.
- The search was then restricted to reports which had “Open Access”

5. Proposed Dissertation Structure

Chapter 1

An introduction to indoor and outdoor air quality as well as to the samplers available.

Chapter 2

The final aims and objectives for this project. It will also contain a literature review covering what is already known about air samplers and what the different types of culturing media used will give results of.

Chapter 3

The final materials, equipment and methodology used to obtain the experimental data.

Chapter 4

Analysis of the data collected accompanied by visual representation of the data.

Chapter 5

Conclusions and recommendations about the results obtained and the process of the project.

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7. Appendix

7.1 Sampling Methodology Flow Chart

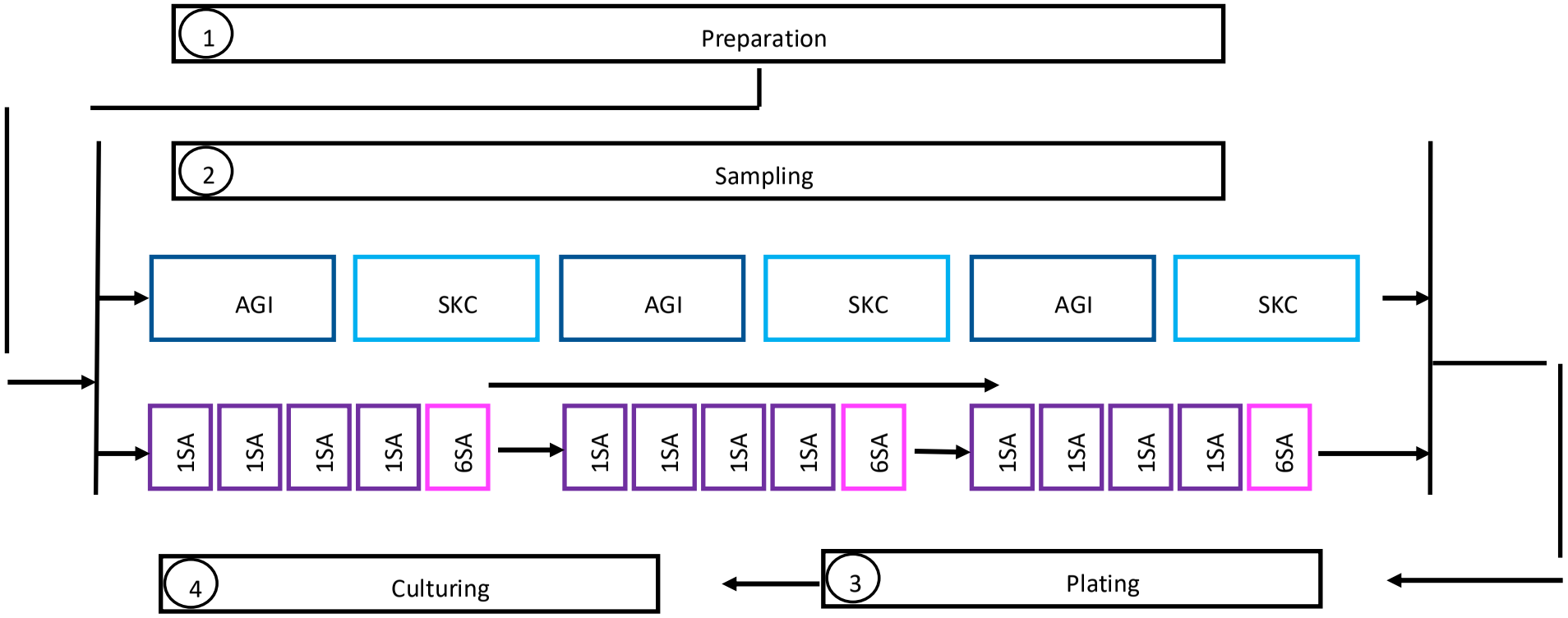
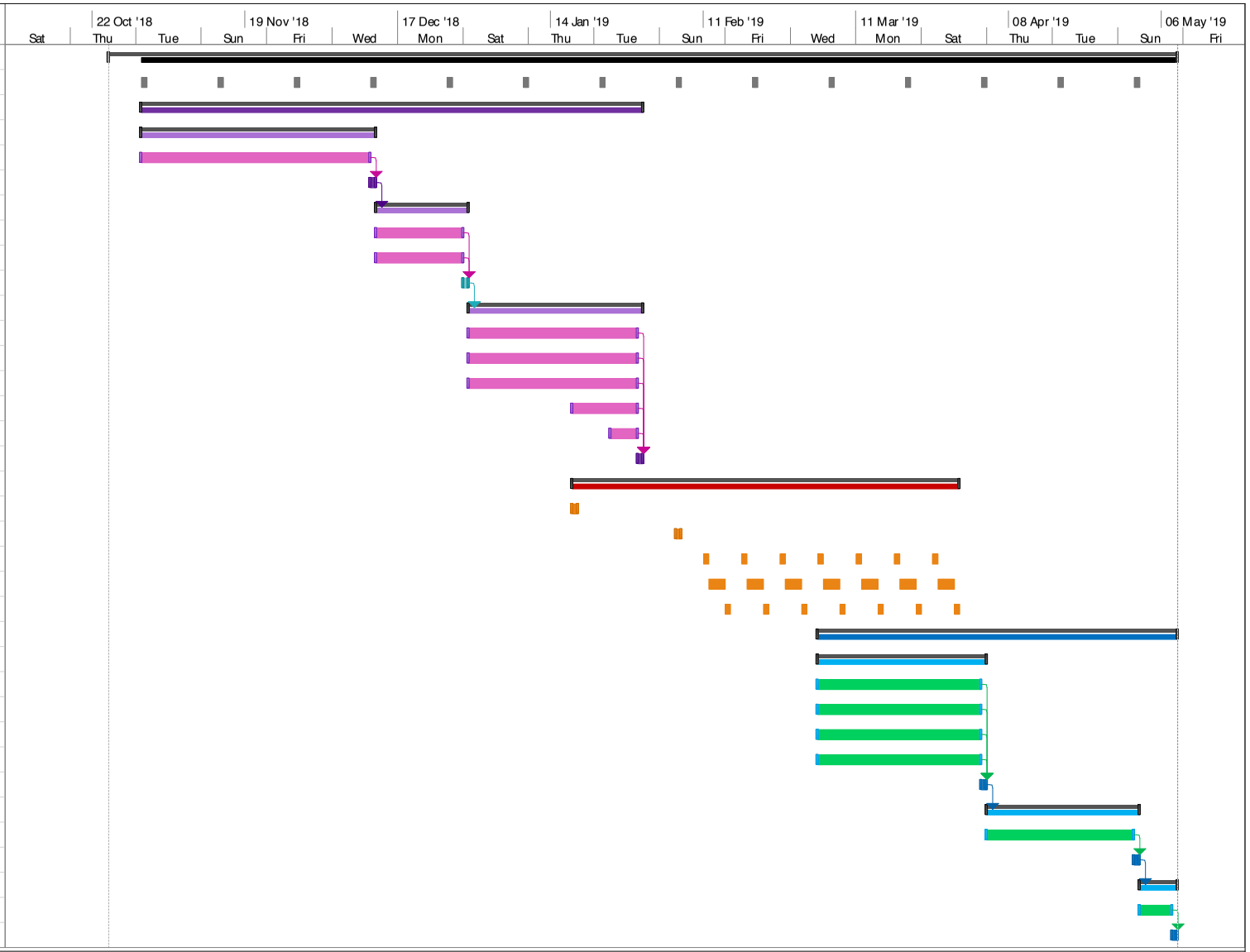


Figure 15- Sampling Methodology Flowchart

7.2 Project Gantt Chart

ID	Task Name	Start
1	Individual Research Project	Thu 25/10/18
2	Fortnightly Supervisor Meeting	Wed 31/10/18
17	Stage 1- Aims and Objectives	Wed 31/10/18
18	First Draft	Wed 31/10/18
19	Write Aims and Objectives	Wed 31/10/18
20	Submitted For Review	Wed 12/12/18
21	Second Draft	Thu 13/12/18
22	Reviewed Aims and Objectives	Thu 13/12/18
23	Literature Review	Thu 13/12/18
24	Submitted For Review	Sat 29/12/18
25	Third and Final Draft	Sun 30/12/18
26	Reviewed Aims and Objectives	Sun 30/12/18
27	Reviewed Literature Review	Sun 30/12/18
28	Ethical Approval	Sun 30/12/18
29	Methodology	Fri 18/01/19
30	Risk Assessment	Fri 25/01/19
31	Submitted For Marking	Wed 30/01/19
32	Stage 2- Sampling and Laboratory Work	Fri 18/01/19
33	Laboratory Induction	Fri 18/01/19
34	Laboratory Training	Wed 06/02/19
35	Preparatory Work	Mon 11/02/19
43	Sampling at Location	Tue 12/02/19
65	Analysing Samples	Fri 15/02/19
73	Stage 3- Dissertation	Mon 04/03/19
74	First Draft	Mon 04/03/19
75	Reviewed Literature Review	Mon 04/03/19
76	Reviewed Aims and Objectives	Mon 04/03/19
77	Reviewed Methodology	Mon 04/03/19
78	Written Analysis of Samples	Mon 04/03/19
79	Submitted For Review	Wed 03/04/19
80	Second Draft	Thu 04/04/19
81	Written Analysis of Samples	Thu 04/04/19
82	Submitted For Review	Wed 01/05/19
83	Third and Final Draft	Thu 02/05/19
84	Written Analysis of Samples	Thu 02/05/19
85	Submitted For Review	Wed 08/05/19



7.3 Risk Assessment

FACULTY OF ENGINEERING – RISK ASSESSMENT		UNIVERSITY OF LEEDS
A comparison of different air samplers for evaluating indoor and outdoor air quality	Assessment Nº 1	

Preliminary notes
● Risk assessment must be conducted by Academic supervisor/Principal Investigator/Manager. ● Other team members and/or people involved or affected by the activities must be consulted in the construction of the risk assessment. ● Academic supervisor/PIs/Managers and other team members must ensure that the control arrangements and safe systems of work are followed. ● Academic supervisor/PIs/Managers must review this risk assessment at least annually, or in the event of incident, accident or changes to operating/maintenance procedures/personnel. ● Review of risk assessment must be conducted by the Academic supervisor/PI/Manager and initialled and dated in the space provided.

Reference information
● Health and Safety at Work etc Act 1974 ● Management of Health and Safety at Work Regulations 1999

Copies
● Original signed risk assessment must be retained by the Academic supervisor/PI/Manager in their office. ● A copy (and any appendices or cross-referenced documents) must be retained in the health and safety file in the workplace. ● The Academic Supervisor/PI/Manager must send a copy by email to the Faculty Safety Team before work begins.

Academic supervisor/PI/Manager's approval				
ACADEMIC SUP/PI/MANAGER	PRINT NAME	SIGNATURE	EMAIL/TELEPHONE	DATE
Dr Louise Fletcher	Dr Louise Fletcher	[redacted]	l.a.fletcher@leeds.ac.uk	29.01.2019

RA review [to be dated initialled by Academic supervisor/PI/MANAGER]							
Date of next review due:	29.01.2020						

Countersignatures (other members of risk assessment team) [Only if applicable – see Faculty RA Procedures]				
ROLE	**PRINT NAME**	**SIGNATURE**	**EMAIL/TELEPHONE**	**DATE**
[Student]	[redacted]	[redacted]	[ll15mlt@leeds.ac.uk]	[29.01.2019]

IDENTIFICATION OF RISKS, CONTROLS & ACTIONS

Consequence/Severity of Harm (C) x Likelihood of harm being realised (L) = Risk Rating [see table following]

HAZARD TYPE	WHO MAY BE HARMED?	RISK RATING WIITHOUT CONTROLS C x L = E,H,M,L,T	CURRENT CONTROL MEASURES (IF ANY)	NEW RISK RATING C x L = E,H,M,L,T	ADDITIONAL CONTROL MEASURES IDENTIFIED	FINAL RISK RATING C x L = E,H,M,L,T	ACTION BY (& DEADLINE)
[Slips/trips and falls]	[Leeds staff and students and others in same environment]	[L]	[Make sure any cables and equipment are sensibly located when sampling. Ensure no trailing cables or tubing]	[T]	[None]	[T]	[ ]
[Electric shock]	[Leeds staff and students and others in same environment]	[H]	[Visual inspection of electrical equipment, correct operation. Only equipment that has a valid PAT label will be used. Only Faculty Electrical services approved extension cables will be used.]	[L]	[None]	[L]	[ ]
[General safety and conduct when sampling around the School of Civil Engineering]	[Leeds staff and students and others in same environment/public]	[L]	[Observe all local rules and hygiene procedures including any access restrictions. Be aware of others in conduct and moving equip etc.]	[T]	[None]	[T]	[ ]
[Manual handling of equipment	[Staff/Student]	[M]	[The only potentially heavy item is the pump for the Andersen sampler and this will be moved using a small trolley when moving around the School of Civil Engineering.]	[T]	[None	[T	[
[Equipment security]	[Staff/Student]	[L]	[When sampling in public areas in the School equipment will not be left unattended. All equipment will be returned to the laboratory overnight.]	[T]	None]	T]	]
[Biological - Potential for a person to	[Staff/Student]	[L]	[Samples will be taken from the air in the test rooms. No artificial aerosol will be used and	[L]	[None]	[L]	[ ]

become exposed to microbial contamination			exposure will be no more than would be experienced under normal conditions. After sampling onto agar plates the plates will be double packed for safe transport back to the laboratory for incubation and subsequent enumeration After sampling into liquid the samplers will be sealed and double packed for safe transport back to the laboratory for subsequent enumeration and incubation				

Methods used in this experiment are also covered by the following risk assessments on RIVO:
199367 - General Risk Assessment - Handling and Culturing of Microbiological Materials
257769 - Enumeration of microbial cultures - Total viable count procedure
350100 - Inoculation of Plates and Broths LAF
350101 - Preparation of Agar Plates LAF
350103 - Preparation of Serial Dilution Bottles LAF
350104 - Preparation and Operation of the Andersen Sampler LAF

COMMUNICATION OF RISK ASSESSMENT FINDINGS TO THOSE INVOLVED				
	METHOD	**YES**	**DATE**	**COMMENTS**
METHODS OF COMMUNICATION USED	Local induction	✓	TBC	
	Details of risk assessment discussed and agreed	✓	29.01.2019	
	Copy of risk assessment available	✓	29.01.2019	
	Controls covered by local protocols & procedures			
	Safety Handbook location notified			
	Toolbox talk			
	Team meeting			
	Email circulation	✓	29.01.2019	
	Other -			

Consequence/Severity of Harm (C)	Likelihood of harm being realised (L)				
	Remote Possibility	Possible	Likely	Highly probable	Virtual Certainty
Minor injury or illness	Trivial	Trivial	Low	Low	Low
Injury/illness requiring medical attention	Trivial	Low	Medium	Medium	High
Injury/illness involving more than 3 days off work	Low	Medium	Medium	High	High
Major injury or long term illness	Low	Medium	High	Extreme	Extreme
Fatal injury/illness	Low	High	High	Extreme	Extreme

RISK RATING = (C x L/S)		ACTION & TIMESCALES
Extreme	E	Work must not be started or continued until the risk level has been reduced. While the control measures should be cost-effective, the legal duty to reduce the risk is absolute. This means that if it is not possible to reduce the risk, even with unlimited resources, then the work must not be started or must remain prohibited.
High	H	Work must not be started until the risk has been reduced. Considerable resources may have to be allocated to reduce the risk. Where the risk involves work in progress, the problem should be remedied as quickly as possible. (Action within 1 Week)
Medium	M	Efforts should be made to reduce the risk, but the costs of prevention should be carefully measured and limited. Depending on the number of people exposed to the hazard risk reduction measures should normally be implemented (Action within 1 Month)
Low	L	Consideration should be given to cost-effective solutions, or improvements that impose minimal operating standards which will maintain Low level of risk. Monitoring is required to ensure that the controls are maintained. (Review Assessment Annually)
Trivial	T	No action is required to deal with trivial risks, and no documentary records need be kept (insignificant risk)(Review Assessment Annually)

7.4 Ethics Approval Form - Students

This form should be completed by the student and passed to the supervisor prior to a review of the possible ethical implications of the proposed dissertation or project.

No primary data collection can be undertaken before the supervisor has approved the plan.

If, following review of this form, amendments to the proposals are agreed to be necessary, the student should provide the supervisor with an amended version for endorsement.

The final signed and dated version of this form must be handed in with the dissertation. Failure to provide a signed and dated form on hand-in will be treated as if the dissertation itself was not submitted.

1. What are the objectives of the dissertation / research project?

   The overall aim of this study is to compare the performance of different types of air sampler for determining the concentration of bacteria and fungi in indoor and outdoor environments. By comparing the various air samplers, it will determine the optimum sampler for different conditions.

2. Does the research involve *NHS patients, resources or staff*? YES / (NO) (please circle).

   If YES, it is likely that full ethical review must be obtained from the NHS process before the research can start.

3. Do you intend to collect *primary data* from human subjects or data that are identifiable with individuals? (This includes, for example, questionnaires and interviews.) YES / (NO) (please circle)

   If you do not intend to collect such primary data then please go to question 14.

   If you do intend to collect such primary data then please respond to ALL the questions 4 through 13. If you feel a question does not apply then please respond with N/A (for not applicable).

4. What is the *purpose* of the primary data in the dissertation / research project?

5. What is/are the *survey population(s)*?

6. How big is the *sample* for each of the survey populations and how was this sample arrived at?

7. How will respondents be *selected and recruited*?

8. What steps are proposed to ensure that the requirements of *informed consent* will be met for those taking part in the research? If an Information Sheet for participants is to be used, please attach it to this form. If not, please explain how you will be able to demonstrate that informed consent has been gained from participants.

9. How will *data* be *collected* from each of the sample groups?

10. How will *data* be *stored* and what will happen to the data at the end of the research?

11. How will *confidentiality* be assured for respondents?

12. What steps are proposed to safeguard the *anonymity* of the respondents?

13. Are there any *risks* (physical or other, including reputational) *to respondents* that may result from taking part in this research? YES / NO (please circle).

    If YES, please specify and state what measures are proposed to deal with these risks.

14. Are there any *risks* (physical or other, including reputational) *to the researcher or to the University* that may result from conducting this research? YES / NO (please circle).

    If YES, please specify and state what measures are proposed to manage these risks.

    See section 7.3

15. Will any *data* be *obtained from a company or* other *organisation*? YES / NO (please circle) For example, information provided by an employer or its employees.

    If NO, then please go to question 18.

16. What steps are proposed to ensure that the requirements of *informed consent* will be met for that organisation? How will *confidentiality* be assured for the organisation?

17. Does the organisation have its own ethics procedure relating to the research you intend to carry out? YES / NO (please circle).

    If YES, the University will require written evidence from the organisation that they have approved the research.

18. Will the proposed research involve any of the following (please put a √ next to 'yes' or 'no'; consult your supervisor if you are unsure):

• Vulnerable groups (e.g. children)?	YES		NO	√
• Particularly sensitive topics?	YES		NO	√
• Access to respondents via 'gatekeepers'?	YES		NO	√
• Use of deception?	YES		NO	√
• Access to confidential personal data?	YES		NO	√
• Psychological stress, anxiety etc?	YES		NO	√
• Intrusive interventions?	YES		NO	√

19. Are there any other ethical issues that may arise from the proposed research?

Please print the name of: I/We grant Ethical Approval

Student [redacted] Supervisor [redacted]

Signed:

(Student) [redacted] (Supervisor) [redacted]

Date 29/01/2019 Date