Impact of Interior Living Walls on Indoor Air Quality: Study in a Dynamic Environment

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Abstract

Interior living walls are promoted partly due to their ability to improve indoor air quality, and possibly reduce energy consumption related to ventilation. However, the studies done to demonstrate this ability are largely conducted in conditions that differs from those of building environments, and they have only focused on a single factor that impacts indoor air quality. This study examined several factors that living walls can affect indoor air quality (volatile organic compounds, carbon dioxide, relative humidity, and bioaerosols) and evaluated how each of these may improve or reduce indoor air quality.

A test chamber was designed to simulate building environment conditions, including temperature, lighting, and ventilation. Three volatile organic compounds and CO_2 were added into the test chamber to simulate occupancy. Samples were taken in the test chamber with and without a living wall to determine differences due to the presence of a living wall.

The interior living wall removed CO_2 and one of the three volatile organic compounds in the test chamber, increased relative humidity, and promoted the increased presence of bioaerosols. While living walls may improve some aspect of indoor air quality, considerations must be taken to mitigate the other impacts on indoor air quality.

Lay Summary

This study looked at some aspects that an interior living wall, a vertical structure that support plant life indoors, may impact indoor air quality. Based on the conclusions drawn from the study, the author aims to help building managers and designers find ways to incorporate living walls into their building that maximizes the positive impacts upon indoor air quality, and reduces the negative impacts upon indoor air quality. The conclusions of the study can assist indoor air quality specialists in determining the causes of inadequate indoor air quality that may arise in buildings with living walls.

Preface

I designed and conducted all of the research as detailed in Chapter 2, based on the concepts originally proposed by Dr. Karen Bartlett and Dr. Maureen Connelly. The sampling equipment involved with the research was either borrowed from the Occupational and Environmental Hygiene Laboratory in the School of Population and Public Health at the University of British Columbia or rented from third parties. The facilities that housed the test chamber and allowed the living walls to grow are courtesy of Dr. Maureen Connelly.

Table of Contents

List of Tables

List of Figures

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1 Introduction

1.1 Indoor Air Quality

Indoor air quality (IAQ) refers to the condition of the air within an indoor space that affects human health. When an indoor space does not have acceptable IAQ, the occupants may experience sick building syndrome (SBS). The symptoms are similar to those of a flu—headache, dizziness, eye, throat, nose, and skin irritation, fatigue, coughing, nausea, and other respiratory symptoms—except these symptoms disappear once a person leaves the indoor space with inadequate IAQ. SBS is caused by many factors, including temperature, humidity, ventilation, and various pollutants. Since people spend a majority of their time indoors, improving IAQ can contribute to improved health and well-being, comfort, and increased productivity (Al Horr et al., 2016; Seppänen, Fisk, & Mendell, 1999; Shendell et al., 2004; Stefan, Gueguen, & Meineri, 2015; Twardella et al., 2012; Vehviläinen et al., 2016).

The control over IAQ factors has traditionally been maintained by the use of mechanical ventilation. Commonly referred to as the heating, ventilation, and air-conditioning (HVAC) system, this involves drawing filtered outside air into the building, tempering the air to an acceptable range of temperature and humidity, and circulating the tempered air inside the building. This is crucial in reducing pollutants from outside sources and diluting the concentration of pollutants from indoor sources. The whole process accounts for a major component of energy use within most buildings.

Due to the intense energy use by HVAC systems and the environmental impacts of energy generation, sustainable building designs and green certification program guidelines directed attention to finding alternatives that operate with considerably less

energy. This usually involves 1) reducing the need to heat or cool the building while reducing ventilation rate, 2) natural ventilation, which uses building openings to direct outside air through the building when environmental conditions facilitate this process, or 3) a combination of mechanical and natural ventilation (Persily, 2014; Junghans & Widerin, 2017). These building designs usually do not consider aspects that affect IAQ. In recent years, green certification programs incorporated IAQ as part of its certification (e.g.: Building Research Establishment Environmental Assessment Method, or BREEAM; Leadership in Energy and Environmental Design, or LEED), but the criteria which IAQ is evaluated against vary greatly among certification programs (Steinemann, Wargocki, & Rismanchi, 2016). While these building designs and guidelines have addressed a building's energy use, the impact of the ventilation scheme on IAQ is not clear until the building is occupied.

1.2 Living Architecture

In the last few decades, green roofs and green walls have been increasingly incorporated into building designs. These structures support vegetative growth across building surfaces. Green roofs are placed on top of a building, while green walls are found on the vertical structures of a building (Peck, 2012).

There are several types of green walls: green facade, living walls, and biowalls. Green facade utilizes a trellis design to allow climbers to grow from the ground upon which the green facade stands and entirely cover the green facade. Due to its installation, most green facade are installed outdoors (Stand & Peck, 2016).

Living walls allow plants to grow from the vertical structure itself. Living walls consists of plants, a growing substrate (e.g.: soil, peat, high porous aggregates) and a

carrier. The carrier is installed onto a wall, with the growing substrate placed securely within the carrier. The plants grow out of the carrier and produces a mural of plants. The carrier facilitates the installation of an irrigation system. The appropriate plants, growing substrate, and irrigation system for each living wall are chosen based on the environmental conditions where the living wall is to be located (Peck, 2012). This allows the living wall to be easily installed both outdoors and indoors.

Biowalls (or air biofilters) consists of the same components as a living wall, and includes a fan to draw air through the substrate. An air biofilter draws air through the growing substrate with a fan, which allows the pollutants to move through the air biofilter. Unlike living walls, the design of an air biofilter must be integrated into a building, with a dedicated space for a mechanical fan that draws the air through the biofilter.

While there are some living walls that have an integrated design to a site, most living walls are modular, made of individual carriers that are easy to install anywhere at relatively low costs (GSky Plant Systems, Inc., c2011; InterCoast Building Solutions, c2012; Modulogreen Vertical Solutions, c2013; Plant Connections Inc., c2013a; Weinmaster, 2009). The advantage of installing interior living walls is that it does not take up much floor area while creating a green space. This retains greater use of the building space while adding the benefits of a green space indoors. This could potentially increase the incentives for installing indoor living walls.

Having plants in the urban environment contributed to reduced negative environmental impacts around the building, such as improved stormwater runoff, reduced urban heat island effect, reduced noise propagation, and reduced energy use within the building (Pérez et al., 2011; Toronto Public Health, 2015). There is also a growing field of research examining biophilia, which posits there are human health

benefits to be achieved from interactions with nature, such as increased positive affect, productivity, and social interactions, and reduced stress (Bringslimark, Hartig, & Patil, 2009; Hartig, Mitchell, de Vries, & Frumkin, 2014; Health Council of the Netherlands, 2004; Lohr, 2009; Söderlund & Newman, 2015; Stefan et al., 2015; Toronto Public Health, 2015).

Due to these factors, the green roof and walls industry has steadily been growing into a multi-billion dollar industry in North America. The area of green roofs installed has more than doubled from 2010 to 2011 (Erlichman, 2012). According to a market report summary, it is projected that green roofs and walls in North America will become a $7.7 billion dollar industry in 2017 through the drive for sustainability and monetary incentives, with 9% coming from green walls (Green Roofs for Healthy Cities, 2013). In a survey completed by eight companies, there were at least 650000 square feet of living walls planted in more than 270 projects by these companies across the United States and Canada 2015 (Stand & Peck, 2016).

One of the many things interior living wall installers advertise about living walls is their benefit to IAQ and acoustics (Plant Connections Inc., c2013b; Weinmaster, 2009). The claims on IAQ benefits are based mostly on the ability of plants to remove various pollutants. These claims do not mention other factors that may positively or negatively affect IAQ (which would be described in the following section). The claims on acoustics benefits are an extension of green roof performance data (Connelly & Hodgson, 2015; Jang, Lee, Jeon, & Kang, 2015). Current research by others is investigating the claim of increased sound absorption of living walls.

The study described below are only examining living walls, and not biowalls. Darlington et al. (2000) found that a biowall can improve IAQ without generating

additional pollutants. Some of the results from Darlington et al. (2000) have been replicated in other studies (Irga, Abdo, Zavattaro, & Torpy, 2017; Russell et al., 2014; Wang & Zhang, 2011).

1.3 Indoor Air Quality Factors

The IAQ factors to be examined include volatile organic compounds (VOCs), carbon dioxide (CO_2), relative humidity (RH), airborne fungi, airborne bacteria, and endotoxin. Based on a literature search, these are the factors that are most likely to be affected by a living wall. While there are studies evaluating the effectiveness of plants removing particulate matter (Lohr & Pearson-Mims, 1996), it was not examined due to the complexity of the experimental setup to do so. In the following, a brief description of each of these factors is provided.

Volatile Organic Compounds

Volatile organic compounds are carbon-based compounds that exist in both gaseous and liquid form at room temperature. While the list of VOCs is extensive, only some of those VOCs (roughly 86) are found in more than 50% of the sampled homes with people experiencing SBS (Kostiainen, 1995). These include aromatic hydrocarbons (e.g.: benzene, toluene, styrene), aliphatic hydrocarbons (e.g.: hexane, octane), halogenated hydrocarbons (e.g.: trichloroethane, tetrachloroethene), terpenes (e.g.: pinene, limonene), aldehydes and ketones (e.g.: formaldehyde, 2-pentanone, benzaldehyde), and alcohols, esters, and organic acids (e.g.: 2-ethoxyethanol, ethylacetate, acetic acid). These VOCs come from a variety of sources, including outdoor air pollution, the building material, the materials used in the furnishings, people,

and human activities within the building (Bernstein et al., 2008; Campagnolo et al., 2017; Hodgson et al., 2000; Jia et al., 2010; Nazaroff & Weschler, 2004; Raw et al., 2004; Zuraimi et al., 2006).

Within residential and commercial buildings, VOCs are associated with some of the symptoms of SBS. Main symptoms include nasopharyngeal and ocular irritation due to inflammation, and dry eyes due to the break-up of the tear film (Chang et al., 2013; EAC-IAQ, 1991; Salonen et al., 2009; Takigawa et al., 2010; Wolkoff, 2013). Some VOCs are associated with unpleasant odours, which may cause comfort issues. In some cases, the odour induced by VOCs triggers sensory irritation, which occurs before the concentration reaches a level that would cause inflammation (Doty et al., 2004; ECA-IAQ, 1991; Wolkoff, 2013). Some VOCs are associated with asthma, most likely exacerbating the symptoms in the presence of the asthmagen (Billionnet et al., 2011; Dale & Raizenne, 2004).

The health effects of VOCs varies among individuals. Asthmatics are more sensitive than the general population to most VOCs in causing the symptoms of SBS. Sensory irritation in the nasal cavity may be caused by activation of the trigeminal nerve without sensing an odour (Hummel, 2000). Due to the interaction of the trigeminal nerve with VOCs with temperature, pH, irritants, and the association with certain odours, not all occupants may become irritated due to the activation of the trigeminal nerve in the nasal cavity (Hummel, 2000; Brüning et al., 2014).

Due to the number of VOCs that may be present at any given time and the additive effect of VOCs upon sensory irritation, all of the VOCs are often sampled and analyzed as a single measurement called total volatile organic compounds (TVOCs). During investigations into IAQ issues, TVOCs are measured more as an indicator of the

presence of pollutants and of the likelihood of complaints related to sensory irritation symptoms, and not for other health effects and comfort issues (ASHRAE, 2010a; Bluyssen et al., 1996; ECA-IAQ, 1997; Mølhave et al., 1997). A guideline limit for TVOCs was set by the European Commission (1997) at 300 $\mu g/m^3$, which is well above the concentrations found in homes sampled in Germany and would not pose an economic burden on homeowners to achieve (ECA-IAQ, 1997).

Living walls have been touted to reduce VOCs. Various species of plants have been found to reduce the concentration from 20 to 90% within 24 hours in sealed test chamber studies, which are summarized in Table 1.1 due to the number of studies on this topic. The studies found were based on a literature search on the Web of Science Core Collection with the keywords “plant” and “volatile organic compound” between the year 1900 and June of 2017. Additional studies were found from the sources referenced in these articles. The VOCs may be adsorbed onto the plant’s surface, absorbed by the leaves and the soil, which are then metabolized (Salisbury & Ross, 1992; Ugrekhelidze, Korte, & Kvesitadze, 1997; Wood et al., 2002). This process is mainly driven by the community of microorganisms growing alongside the roots of the plant and within the soil (Aydogan & Montoya, 2011; Fan & Scow, 1993; Kim et al., 2008; Wood et al., 2002).

While VOCs are usually removed from the building environment by mechanical ventilation through the dilution and displacement of pollutants, the use of interior living walls may potentially allow the building to reduce the ventilation rate and thus the energy used by mechanical systems without impairing IAQ. However, it is currently unclear whether this effect can significantly improve IAQ and what factors determine its efficiency.

Table 1.1: Description of studies examining the removal of VOCs by indoor potted plant or interior living wall

Study	Experiment location	Plant(s) studied	VOCs	Rate of VOC removal
Sealed chamber studies				
Aydogan & Montoya, 2011	Sealed test chamber (61 x 30.5 x 40.6 cm or 75 L)	*Hedera helix*, *Chrysanthemum morifolium*, *Dieffenbachia compacta*, *Epipremnum aureum* grown in hydroponic (in 150- or 160- mm pots)	Formaldehyde (2 mg/m^3), removal from single dose	88-94% reduction in 24 hours, 23-56 min to reach 2/3 of the reduction of each plant
De Kempeneer, Sercu, Vanbrabant, van Langenhove, Verstraete, 2004	Sealed test chamber (23 L)	*Azalea indica* (in 150-mm pots), with or without inoculum containing *Pseudomonas putida* TVA8 on the leaves	Toluene (90 ppm), removal from single dose, repeated with single doses three more times	Time until 95% of toluene removed (mean±SE): No inoculum: 76±14 h No inoculum after the first dose:46±6 h With inoculum: 27±11 h With inoculum after the first dose: 7±4 h
Irga, Torpy, & Burchett 2013	Sealed test chamber (26 x 20 x 30.5 cm or 15.86 L)	*Syngonium podophyllum* 'White Butterfly' in 130-mm pots with potting mix or in hydroculture	Benzene (25 ppm), removal from single dose	Rate at 50% benzene: 1444 $\mu g/m^3/h$/plant in potting mix 739 $\mu g/m^3/h$/plant in hydroculture
Kim et al., 2008	Sealed test chamber (0.9 x 0.9 x 1.23 m) with 6 L/min of air recirculated from outside	*Ficus benjamina* (in 190-mm pots), *Fatsia japonica* (in 150-mm pots) with the whole plant, only the aerial part, or only the root zone, and exposed to VOC in the light or in the dark	Formaldehyde (2 ppm), one initial dose, and continuous addition from recirculated air for 5 hours	*F. japonica,* light: 0.5 $\mu g/m^3/cm^2$ leaf *F. benjamina*, light: 0.55 $\mu g/m^3/cm^2$ leaf *F. japonica*, dark: 0.51 $\mu g/m^3/cm^2$ leaf *F. benjamina*, dark: 0.4 $\mu g/m^3/cm^2$ leaf Shoots, *F. japonica*, light: 0.50 $\mu g/m^3/cm^2$ leaf Shoots, *F. benjamina*, light: 0.32 $\mu g/m^3/cm^2$ leaf Shoots, *F. japonica*, dark: 0.01 $\mu g/m^3/cm^2$ leaf Shoots, *F*. benjamina, dark: 0.05 $\mu g/m^3/cm^2$ leaf Roots, *F. japonica,* light: 0.32 $\mu g/m^3/cm^2$ leaf Roots, *F. benjamina*, light: 0.43 $\mu g/m^3/cm^2$ leaf Roots, *F. japonica*, dark: 0.53 $\mu g/m^3/cm^2$ leaf Roots, *F*. benjamina, dark: 0.45 $\mu g/m^3/cm^2$ leaf 96-160 min to reach half of concentration for the whole plant
Kim et al., 2009	Sealed Test chamber (0.9 x 0.9 x 1.23 m) with 6 L/min of air recirculated from outside	*Epipremnum aureum*, *Rosmarinus officinalis*, or *Gardenia jasminoide*	Formaldehyde (2 ppm), one initial dose, and continuous addition from recirculated air for 5 hours	3.4-6.6 $mg/m^3/h/m^3$ plant volume after 5 hours

Study	Experiment location	Plant(s) studied	VOCs	Rate of VOC removal
Kim et al., 2010	Sealed test chamber (0.9 x 0.9 x 1.23 m) with 6 L/min of air recirculated from outside	86 plant species in 150-mm or 190-mm pots	Formaldehyde (2 ppm), one initial dose, and continuous addition from recirculated air for 5 hours	0.13-6.64 $\mu g/m^3/cm^2$ leaf area after 5 hours
Lim, Kim, Yang, Kim, Lee, & Shin, 2009	Sealed test chamber (0.9 x 0.9 x 1.24 m or 1004 L)	*Fatsia japonica* in 190-mm pot in test chamber	Formaldehyde (2400 $\mu g/m^3$), one initial dose, and continuous addition from recirculated air for 5 hours	Test chamber in 5 h: roughly 700 $\mu g/m^3$ with plant vs. roughly 100 $\mu g/m^3$ without plant
Liu, Mu, Zhu, Ding, & Arens, 2007	Test chamber (0.4 m diameter, 0.6 m height or 75 L) with continuous flow at 2 L/min	73 plant species tested for two hours	150±6.7 ppb benzene, constant flow	23 species had no change on benzene 13 removed 0.1-9.99% benzene 17 removed 10-20% benzene 17 removed 20-40% benzene
Orwell, Wood, Tarran, Torpy, & Burchett, 2004	Sealed test chamber (0.6 x 0.6 x 0.6 m or 216 L)	*Howea forsteriana*, *Spathiphyllum floribundum* 'Petite', *Spathiphyllum floribundum* 'Sensation', *Dracaena deremensis* 'Janet Craig', *Dracaena marginata*, *Epipremnum aureum*, Schefflera actinophylla 'Amate' in 150-mm pots	Benzene (25, 50 ppm), repeated doses given to determine rate after previous doses, and differences in lighting	At 25 ppm in light: 12.6-27.5 ppm/day/plant At 25 ppm in dark: no significant change from 25 ppm in light At 50 ppm in dark: 27.5-49.3 ppm/day/plant
Orwell, Wood, Burchett, Tarran, & Torpy, 2006	Sealed test chamber (0.6 x 0.6 x 0.6 m or 216 L)	*Spathiphyllum* 'Sweet Chico', *Dracaena deremensis* 'Janet Craig' in 150-mm pots	Toluene (0.2, 1, 10, 100 ppm), m-xylene (0.2, 1, 10, 100 ppm), doses were given every day for five days	At 0.2 ppm: 0.08-0.51 mg/day/plant At 1 ppm: 0.57-2.59 mg/day/plant At 10 ppm: 8.2-16.9 mg/day/plant At 100 ppm: 22.6-119 mg/day/plant
Oyabu, Sawada, Onodera, Takenaka, & Wolverton, 2003	Sealed test chamber (300 L)	*Epipremnum aureum* in three different soil types (in 250-mm pots)	Acetone (1, 5, 8 ppm), formaldehyde (5, 10, 20 ppm), single dose	Acetone: 7-15 V/h* Formaldehyde: 20-40 V/h*
Sawada & Oyabu, 2008	Sealed test chamber (300 L)	*Epipremnum aureum* in different growing medium (potting mix, tap water, water with fertilizer)	Formaldehyde (8 ppm), toluene (1.5 ppm), xylene (1.5 ppm), single dose	Greatest from plant growing in pot: Formaldehyde (20 V/h), toluene (9 V/h), xylene (9 V/h)*

Study	Experiment location	Plant(s) studied	VOCs	Rate of VOC removal
Schmitz, Hilger, & Weidner, 2000	Sealed test chamber (320 L) with	*Epipremnum aureum*, and *Ficus benjamina*	Formaldehyde with carbon-14 (0.5 mg/m^3) exposed to the shoots, repeated doses when there is no detectable radioactivity for a total of 48 hours of exposure	0.2-1.6% of carbon-14 recovered from plant surface 127 and 88 ng/g fresh weight/h were taken up from the leaves of *E. aureum* and *F. benjamina* respectively
Tani & Hewitt, 2009	Test chamber with continuous flow of 1.3 L/min	Leaves of *Spathiphyllum clevelandii* exposed to aldehydes, and leaves of *Epipremnum aureum* exposed to aldehydes and ketones	Aldehyde: Propionaldehyde, n-butylaldehyde, iso-butylaldehyde, crotonaldehyde, methacrolein, n-valeraldehyde, iso-valeraldehyde, benzaldehyde Ketone: acetone, methyl ethyl ketone, diethyl ketone, methyl n-propyl ketone, methyl isopropyl ketone, and methyl isobutyl ketone	Aldehyde uptake into *S. clevelandii*: 7.1-19.1 mmol/m^2/s No reduction of acetone by either plant species leaves Ketone uptake into *S. clevelandii*: 2.4-7.0 mmol/m^2/s Ketone uptake into *E. aureum*: 3.5-8.9 mmol/m^2/s
Wolverton, McDonald, & Watkins, 1984	Sealed test chamber (73.7 cm on each edge, 400 L)	*Scindapsus aureus*, *Syngonium podophyllum*, *Chlorophytum elatum* var. *Vittatum* in 3.8 L pots	Formaldehyde (18 ppm) (37 ppm for *C. elatum*)	*S. aureus* and *S podophyllum* reached 50% reduction at 6 hours, 66% at 24 hours *C. elatum* reached nearly 80% reduction at 6 hours and was below limit of detection at 24 hours

Study	Experiment location	Plant(s) studied	VOCs	Rate of VOC removal
Wolverton & Wolverton, 1993	Sealed test chamber (310 L)	*Aechmea fasciata, Aglaonema 'Silver Queen', Aloe barbadensis, Anthurium andraeanum, Calathea ornata, Chamaedorea elegans, Chlorophytum comosum 'Vittatum', Chrysanthemum morifolium, Cissus rhombifolia, Cyclamen persicum, Dendrobium sp., Dieffenbachia camille, Dieffenbachia 'Exotica Compacta', Dieffenbachia maculata, Dracaena deremensis 'Janet Craig', Dracaena deremensis 'Warneckii', Dracaena fragrans, Dracaena marginata, Euphorbia pulcherrima, Ficus benjamina, Ficus sabre, Guzmania 'Cherry', Hedera helix, Homalomena sp., Kalanchoë, Liriope spicata, Neoregelia cv., Nephrolepis exaltata 'Bostoniensis', Nephrolepis obliterata, Phalaenopsis sp., Phoenix roebelenii, Rhapis excelsa, Rhododendron indicum, Sansevieria trifasciata, Senecio cruentus, Spathiphyllum 'Clevelandii', Syngonium podophyllum, Tulip 'Yellow Present'*	Formaldehyde, xylene	47-1863 µg/h
Wood, Orwell, Tarran, Torpy, & Burchett, 2002	Sealed test chamber (0.6 x 0.6 x 0.6 m or 216 L)	*Howea forsteriana*, *Spathiphyllum wallisii* Schott. 'Petite', *Dracaena deremensis* Engl. 'Janet Craig' in 150-mm pots in potting mix or in hydroponic medium	Benzene (25 ppm), n-hexane (100 ppm), single dose	Benzene: 40.8-88.2 mg/m^3/day/plant n-hexane: 53-306 mg/m^3/day/plant
Yoo, Kwon, Son, & Kays, 2006	Sealed test chamber (0.55 x 0.58 x 0.9 m or 287.1 L)	*Hedera helix*, *Spathiphyllum wallisii*, *Syngonium podophyllum*, *Cissus rhombifolia* in 180-mm pots	Benzene (1 ppm) Toluene (1 ppm) Both (0.5 ppm each)	Benzene only: 26.6-174.5 ng/m^3/h/cm^2 leaf area Toluene only: 57.8-220.2 ng/m^3/h/cm^2 leaf area Benzene with both: 18.8-57.5 ng/m^3/h/cm^2 leaf area Toluene with both: 27.1-112.2 ng/m^3/h/cm^2 leaf area

Study	Experiment location	Plant(s) studied	VOCs	Rate of VOC removal
In situ Studies				
Husti et al., 2016	An office room before and two months after plants were placed	*Ficus elastica*, *Dracaena deremensis*, *Sansevieria trifasciata*	Butyraldehyde, formaldehyde, toluene, m,p,o-xylene, limonene, benzene, ethylbenzene, 2-methylpentane, n-hexane, dodecane, methylcyclopentane	23.1-30.5% reduction after two months
Kim et al., 2009	Rooms (7.1 x 3.4 x 2.5 m) from a recently constructed building	One of three plant species filling up 3, 6, and 9% of the room volume (*Epipremnum aureum*, *Rosmarinus officinalis*, or *Gardenia jasminoide*)	Formaldehyde (2 ppm), one initial dose or after closing all room openings	Reduction by 30% with plants filling 3% of room volume to 67% with plants filling 9% of room volume
Kim et al., 2011	2 office rooms with 105 m^2 in recently constructed building and 2 office rooms with 135 m^2 in >20 year old building	One large plant per 10 m^2 of office space (*Howea forsteriana*, *Rhapis excelsa*, *Ficus elastica*, *Dracaena fragrans*), and 12 small pots of plants (*Dieffenbachia camilla*, *Ficus elastica*)	Formaldehyde, benzene, toluene, ethylbenzene, xylene	New building: 10% reduction in formaldehyde, 55% reduction in xylene, little to no reduction in benzene, toluene, and ethylbenzene during summer; 50% reduction in ethylbenzene, and increases in formaldehyde, benzene, toluene, and xylene during winter Aged building (summer and winter): 30-35% reduction in formaldehyde, little to no change in benzene, and toluene, and increases to ethylbenzene and xylene
Kim et al., 2013	4 classroom (9 x 7.5 m), 2 with mechanical ventilation, and 2 with windows for natural ventilation	10 large plants 240- or 300-mm pots (*Rhapis excelsa*, *Ficus elastica*, *Nandina domestica*, *Fatsia japonica*) and 10 small plants in 180-mm pots (*Soleirolia soleirolii*, *Hedera helix*, *Ardisia psilla*, *Epipremnum aureum*)	Formaldehyde, benzene, toluene, ethylbenzene, xylene	No significant difference between the classroom with plants and the one without plants in the school with mechanical ventilation Little difference between classroom with plants and without in naturally-ventilated classroom except for toluene (86.37 to 26.63 $\mu g/m^3$ in the classroom with plants)

Study	Experiment location	Plant(s) studied	VOCs	Rate of VOC removal
Lim, Kim, Yang, Kim, Lee, & Shin, 2009	82 newly constructed households, 40 without plants and 42 with plants	In the first year, two large plants (*Chrysalidocarpus lutescene*, *Ficus elastica*) and two small plants (*Chamaedorea seifrizii* Burret, *Spathiphyllum* spp.) in the living room, *Epipremnum aureum* in the kitchen, and one large plant (*Portulacaria afra*) and two small plants (*Fatsia japonica*, *Rosemarinus officinalis*) in the bedroom In the second year, one large plant (*Citrus unshiu*) and three small plants (*Asplenium nidus, Gardenia jasminoides, Spathiphyllum* spp.) in the living room, *Epipremnum aureum* in the kitchen, and *Rosemarinus officinalis* and *Gardenia jasminoides* in the bedroom	Formaldehyde, toluene, ethylbenzene, xylene sampled in newly constructed households	First year (no plant vs. plant): Formaldehyde during winter (72.0 vs. 33.7 $\mu g/m^3$), formaldehyde during summer (70.6 vs. 10.7 $\mu g/m^3$) No significant difference for toluene, ethylbenzene, xylene Second year winter (no plant vs. plant): Formaldehyde (85.1 vs. 44.7 $\mu g/m^3$), Toluene (168.0 vs. 330.6 $\mu g/m^3$), Ethylbenzene (19.1 vs. 35.2 $\mu g/m^3$), Xylene (1.1 vs. 2.2 $\mu g/m^3$) Second year summer (when windows are opened): Formaldehyde (54.0 vs. 11.9 $\mu g/m^3$), no significant difference in toluene, ethylbenzene and xylene
Pegas et al., 2012	A 52.5 m^2 classroom with approximately 25 students and one teacher during school hours	Comparison between: 1) classroom without plants (from February to March) and 2) classroom with a total of six plants in 300-mm diameter pots containing one of three species: *Dracaena deremensis*, *Dracaena marginata*, or *Spathiphyllum sp.* (from March to May)	No added source Measured 1) TVOC as sum of 21 VOCs each with its own standard, and 2) carbonyls as sum of 15 compounds	TVOC: Decrease from 933±577 $\mu g/m^3$ without plants to 249±74.2 $\mu g/m^3$ with plants (roughly 73%) Carbonyl: Decrease from 81.3-94.3 $\mu g/m^3$ without plants to 57.4-68.7 $\mu g/m^3$ with plants (roughly 40%)
Smith, Fsadni, & Holt, 2017	Ground floor (experiment) and first floor (control) office space arranged in an open floor design in Southern England, mechanical ventilation	12 *Spathiphyllum floribundum* 'Sensation', and 12 *Nephrolepsis* spp. in 180-mm pots, and 20 *Areca* spp., and 10 *Dracaena deremensis* 'Janet Craig' in 430-mm pots Placed for six months	Formaldehyde and TVOC (tested by a testing kit)	Formaldehyde remained at roughly 15 $\mu g/m^3$ on the ground and first floor before and after the plants were placed Greater reduction in TVOC on the ground floor (66 to 42 $\mu g/m^3$) than the first floor (68 to 57 $\mu g/m^3$)

Study	Experiment location	Plant(s) studied	VOCs	Rate of VOC removal
Song, Kim, & Sohn, 2007	Two test rooms (3.5 x 3.5 x 2.4 m) without ventilation. One contains plants of one species and the other does not	Pots of *Aglaonema brevispathum*, *Pachira aquatica*, or *Ficus benjamiana* until they fill 5% or 10% of the room	Formaldehyde, benzene, toluene, ethylbenzene, xylene	Reduction from 10-90% with plants No change in the room without plants
Song, Kim, & Sohn, 2011	Two test rooms (3.5 x 3.5 x 2.4 m) without ventilation. One contains plants and the other does not	Pots of *Aglaonema brevispathum*, *Pachira aquatica, or Ficus benjamiana* until they fill 5% or 10% of the room	Formaldehyde, benzene, toluene, ethylbenzene, xylene, styrene measured for up to 72 hours	Reduction from 10-90% with plants
Wood et al., 2006	Single occupancy offices of roughly 10-12 m^2 and 3-4 m high chosen from three buildings	*Dracaena deremensis* 'Janet Craig' in 300-mm pots (3 offices with no plants, 3 pots, and 6 pots, for a total of 9 offices from the mechanically ventilated buildings) Five *Spathiphyllum* 'Sweet Chico' in 200-mm pot and one *Dracaena deremensis* 'Janet Craig' in 200-mm pot in the naturally ventilated building and one of the mechanically ventilated building	Average weekly TVOC measured by photoionization detector	30% reduction with *D. deremensis* 50% reduction with *D. deremensis* for weekly average TVOC >100 ppb In the naturally ventilated building, if weekly average TVOC > 100 ppb, there is roughly 50-75% reduction in TVOC Average increase in TVOC due S. 'Sweet Chico' were pollinating at time of study

*Purification capability is determined by the quotient of 1) the peak response (in Volts) after a single dose of a chemical is released into the chamber and 2) the time it takes for the plants to reduce the chemical such that the sensor shows a value at half of the peak response.

First, these studies mostly examine the main compounds that have been associated with air pollution, specifically formaldehyde, benzene, toluene, ethylbenzene, and xylene. These five are a few of the many anthropogenic VOCs found within building environments (Bluyssen et al., 1996; Hodgson et al., 2000; Jia et al., 2010; Kostiainen, 1995; Nazaroff & Weschler, 2004). The OFFICAIR study in Europe found that, besides the five compounds mentioned previously, there are other compounds that are highly correlated to outdoor and indoor sources of pollution within office spaces, such as limonene and pinene (from scented consumer products), acetaldehyde (from ozone-reactive compounds in furnishing and many products used indoors), 2-butoxyethanol (from surface coatings and cleaning products), and 2-ethylhexanol and propanal (emitted from vinyl flooring and carpets) (Campagnolo et al., 2017). While the studies on these five compounds allow an estimation of a plant species' VOC removal effectiveness for some species of VOC, the overall effectiveness in the building environment may not be as high as for the other prevalent VOCs found indoors.

Second, the removal of VOC may depend on how many different VOCs are found in the room, along with the plant species. For example, among more than thirty plant species tested, there was a tenfold range in the rate of removal (Wolverton & Wolverton, 1993). The rate of removal can be high among the plant species tested for one VOC and be ranked low for another VOC. Yoo et al. (2006) found that rate of benzene and toluene removal by the plant shoots when both gases were introduced dropped at least by half (except with one species) when comparing with either gas alone. Given the wide variation of VOC species found in the building during the course of a day and throughout seasons, it is likely that a living wall would not be effective in removing all of the VOCs.

Third, the removal of VOCs is mediated in large part by the microorganisms that live in the soil and on the leaves. Wolverton and Wolverton (1993) examined the difference in formaldehyde removal with plants that had sterilized sand over the potting soil and plants that did not. The percent of formaldehyde removed with sterilized sand over the potting soil was less than 40% of plants without sterilized sand. De Kempeneer et al. (2004) found that an inoculum of *Pseudomonas putida* TVA8 was able to achieve the rate of toluene removal in less than half the time without the inoculum. If the plant and soils in the living wall do not contain the appropriate microorganisms, it may not be effective in removing VOCs

Fourth, plants and microbes produce their own VOCs in addition to removing the VOCs commonly found in buildings (Owen, Clark, Pompe, & Semple, 2007; Junker & Tholl, 2013; Peñuelas et al., 2014; Yang et al., 2009). The VOCs produced by bacteria and fungi may negatively impact the inhabitant's health and comfort (Bernstein et al., 2008; Kim et al., 2006; Sahlberg et al., 2013). Recent studies have compiled a list of roughly 800 VOCs produced by bacteria and fungi in the soils (Effmert, Kalderás, Warnke, & Piechulla, 2012; Peñuelas et al., 2014). Given the complexity between the production and removal of specific VOCs in a living wall, investigation into the living wall's influence on the level of various VOCs in a room is recommended.

Finally, most of the earlier studies were done in a highly controlled setting. The plants were placed inside a sealed chamber and under high air change rate, where a single dose of the VOC is given to examine the removal rate (Aydogan & Montoya, 2011; Darlington et al., 2001; Fan & Scow, 1993; Orwell et al., 2004; Orwell et al., 2006; Sawada & Oyabu, 2006; Wolverton et al., 1984; Yoo et al., 2006). The building

environment is not kept in a sealed chamber, where the plant is the only factor in reducing VOC, or kept in a room with high air change rate, which constantly provides the plant with VOC to remove. Even if the living wall is able to significantly reduce VOCs, the effect may not be comparable to a ventilation system.

Carbon Dioxide

CO_2 is a ubiquitous compound found at roughly 300 to 400 ppm in Earth's atmosphere. It is produced as a result of combustion and metabolic activity of living organisms. CO_2 is used as an indicator of ventilation rate during IAQ investigations, and by proxy, the concentration of any pollutant that can cause negative health effects and discomfort. The American Society of Heating, Refrigerating, and Air Conditioning Engineers (ASHRAE) have estimated the amount of ventilation required to adequately ventilate a room based on the level of CO_2. They recommend that the indoor CO_2 concentration be at most around 700 ppm above the ambient outdoor concentration (ASHRAE, 2010a). This equates to having adequate ventilation when the concentration of CO_2 is less than 1000 to 1100 ppm.

Poor ventilation has been correlated to SBS, specifically with headache, dizziness, and fatigue (Apte et al., 2000; Jankovic et al., 1996; Seppänen et al., 1999; Twardella et al., 2012). Recent research showed that exposure to CO_2 concentration between 800 to 1000 ppm while maintaining adequate ventilation are associated with some symptoms of SBS, ability in performing cognitive tasks, and absenteeism despite adequate ventilation (Norbäck & Nordström, 2008; Satish et al., 2012; Shendell et al., 2004; Tsai et al., 2012).

The estimate for adequate ventilation is based on the dilution ventilation model (ACGIH, 2013; ASHRAE, 2010a). By assuming that the air is uniformly generated and the air brought into the room does not contain the gaseous compound, the rate at which a gaseous compound accumulates in a room is determined by the rate of the gaseous compound being generated and the rate at which the gaseous compound is removed, which is represented by the following equations:

$$V\,dC = G\,dt - Q \cdot C\,dt \qquad [1]$$

Where: V is the volume of the room,

C is the concentration of the gaseous compound,

G is the generation rate of the gaseous compound in the room,

Q is the ventilation rate of the room, and

t is the time.

At steady-state equilibrium, when the change in concentration is zero ($dC = 0$), the equation becomes the following:

$$G\,dt = Q \cdot C\,dt$$

$$\int G\,dt = \int Q \cdot C\,dt$$

$$G\,(t_2 - t_1) = Q \cdot C\,(t_2 - t_1)$$

$$C = G / Q \qquad [2]$$

Since the air in most rooms are not perfectly mixed, a mixing factor (K_m) is used to determine the effective flow rate (Q') in the following equation and replaces Q in the dilution ventilation model:

$$Q' = Q / K_m$$

$$\therefore C = G / Q' \qquad [3]$$

By estimating the pollutants generated within a building and applying a maximum limit of pollutant concentration acceptable to occupants, one can determine the ventilation rate required to provide a comfortable environment.

Plants (mostly at their leaves) undergo photosynthesis to generate energy compounds, which occurs in two phases. During the light-dependent reactions, the plant uses light to break down water to produce energy molecules (and oxygen as a byproduct). During the light-independent reactions (which occurs when there is energy molecules and CO_2), the energy generated is used to join CO_2 to form carbohydrates. CO_2 enters the plant through stomata (gated openings underneath the leaf) in response to light levels and high water stress, and usually remain so from sunrise to sunset (Salisbury & Ross, 1992). Therefore, the time period for reduction of CO_2 by plants through photosynthesis may coincide with building occupancy. It is important to know the extent that the living wall acts as a sink or a source to estimate the impact on IAQ from building designs.

In addition, the living walls may interfere with the method of determining outdoor air supply rate using CO_2 and the use of the Wells-Riley equation. The Wells-Riley equation was developed to estimate the number of new cases given an airborne harmful agent's generation rate and effective dose to cause harm in the building environment (Rudnick & Milton, 2003). While infectious agents may not be expected within most building environments, the equation is also useful in determining the outdoor air flow required to remove indoor pollutants. As the method assumes that there are no additional source and sink of CO_2 within the building environment, the living wall would

lead to an overestimation of the actual outdoor air supply rate. Understanding the impact of the living wall on CO_2 can aid in IAQ investigations.

Relative Humidity

Relative humidity refers to the proportion of water vapour in the air to the water vapour at maximum saturation in the air at a specific temperature. It is derived as the ratio of the partial pressure of water vapour in the air to the vapour pressure of water at the temperature which the water vapour is found. RH ranges from 0-100%. When the air reaches water saturation (i.e. RH > 100%), water vapour condenses and becomes water droplets on nearby surfaces and the particles in the air.

Similar to air, all liquids and solids have some capacity to retain water and allow water to condense when the material becomes saturated. The water content of a solid is a proportion of the amount of water (as mass or volume) in the solid to the volume or mass of the solid. Sometimes, the water content is expressed as a percentage of the water saturation level of the material. Water vapour would diffuse between the air and any nearby materials to reach an equilibrium concentration of water vapour such that the partial pressure of water vapour among any nearby material and the air is equal. The water available on materials that can diffuse into the air is often measured as the water activity (A_w), which is the ratio of partial pressure of water vapour to the vapour pressure of pure water at that temperature. A_w ranges between 0-1. As the partial pressure of water vapour of an object cannot be directly measured, it is indirectly computed by finding the RH at equilibrium directly above the surface of the object of interest. The equilibrium RH divided by 100% equates to the A_w of the object (Adan & Samson, 2011).

The main use for the determination of A_w (and thus RH) in building environments is to determine the water available for microbial growth. Building materials can absorb and release water to the building environment. When the amount of water vapour in the air provides a sufficient level of water activity on building material surfaces, bacteria and fungi can use the available water to grow, using building materials and dust gathered on surfaces as nutrients. Given an appropriate temperature for growth, fungi generally require an A_w above 0.8, with the species requiring a lower A_w colonizing the building environment first, while bacteria require an A_w above 0.9 to grow within the building environment (Adan et al., 2011; WHO, 2009).

RH can influence the temperature sensation for a person, as water has a higher specific heat capacity, allowing the air to appear to be of a similar temperature to one's body (ASHRAE, 2010b). Low RH (below 30%) allows static electricity to build up easily, and leads to the loss of the tear film of the eye. The loss of the tear film allows VOCs and other pollutants to cause eye irritation. These consequences of low RH increase prevalence of SBS symptoms, relating to dry skin, nose and throat (Norbäck, Lindgren, & Wieslander, 2006; Nordström, Norbäck, & Akselsson, 1994; Reinikainen, Jaakkola, & Seppänen, 1992). It also relates to negative subjective perception of IAQ (Fang, Clausen, & Fanger, 1998). On the other end, high RH (above 80%) can create an uncomfortable environment when the surrounding temperature is above the recommended comfort range and increase microbial growth. In addition, materials in environments with higher RH and temperature off-gas VOCs from building materials at a faster rate (WHO, 2009). Typically, the desired RH range is between 30% to 60% as it

relates to the comfortable indoor air temperature around 21-25 °C as recommended by ASHRAE (2010b).

Plants mainly release water in order to maintain turgor pressure and carry nutrients from the roots to the leaves, which is achieved by continuously taking water out of the growing media. Some of the water released is used to regulate the temperature of the plant from the surrounding environment and the heat generated from its metabolism. The release of water from plants is done by opening the stomata beneath the leaves, also known as transpiration (Salisbury & Ross, 1992). If a plant releases a significant amount of water into the building environment, there is a potential to increase the relative humidity.

A living wall contains a higher density of plants within the same growing surface area than a potted plant, and requires greater irrigation to maintain adequate moisture content range within the substrate at the furthest parts of a living wall section from the irrigation system. Thus, it may contribute a moderate degree to the RH of the room and the A_w to surfaces in the room, which fosters microbial growth and may increase the possibility of microbial growth within the building environment. On the other hand, the living wall may be useful in buildings with consistently low RH. The living wall would increase RH above the minimum recommended level, making the environment more comfortable for inhabitants. Depending on the outdoor environmental conditions where the living wall is placed, the amount of water vapour may be beneficial or detrimental to the maintenance of the building and the health and comfort of occupants. Understanding the impact of irrigation on RH allows building managers to better control the IAQ of the building.

Bioaerosols

Bioaerosols refer to airborne particulate matter that contains substance from organisms or are made by organisms which are recognized due to the toxicity of the substance, particularly for humans. Bioaerosols associated with IAQ include viruses, bacteria, fungi, allergens, and secondary metabolites of microorganisms (e.g.: VOCs). Some of the bioaerosols may be infectious, especially towards immunocompromised individuals, and some are able to trigger allergic reactions. They can sometimes cause other adverse effects, including effects that impair the functioning of the human immune system. Within the building environment, their presence may lead to symptoms of SBS, such as skin, eye, and nasal irritation, headaches (from VOCs produced by microorganisms), and respiratory symptoms (Burge, 1995; Douwes et al., 2003; Eduard, 2009; Portnoy, 2005).

Since living walls are expected to affect the bacteria and fungi population in the building environment, they will be discussed further below.

Fungi and Bacteria. Fungi are immobile organisms that live by taking nearby materials for nutrients. Within a building when the RH is above 80%, they grow on any surface containing carbonaceous material, such as wooden structural components, gypsum boards, wooden products, some plastic, some insulation material, and organic matter collected within a building. Most of the fungal species that grow indoors are filamentous fungi and yeast, which are brought into the building environment from outdoors. Filamentous fungi and yeast generate spores that can propagate outdoors and within the building environment. If a building remains damp enough, it becomes a

favourable environment for fungi to persist (Burge, 1995; Flannigan, Samson, & Miller, 2011; Stetzenbach, 1996; WHO, 2009).

Bacteria are unicellular organisms that can grow in a variety of environmental conditions, such as standing water, soil, house dust, decaying organic matter, and alongside plants and animals (Burge, 1995; Flannigan, Samson, & Miller, 2011; Stetzenbach, 1996; WHO, 2009). Within the building environment, they are generated from the sloughed skin cells of humans and other organisms that reside in the building, which can lead to an accumulation of airborne bacteria (Burge, 1995; Flannigan, Samson, & Miller, 2011; Tsai & Macher, 2005). Most types of bacteria that can be sampled and cultured in the building environment belong to the morphological groups Gram-positive cocci (which are spherical in form) and rods (which have one dimension greater than another), and to a lesser extent, Gram-negative rods. The greater proportion of Gram-positive bacteria within the building environment is due to the presence of a cell wall containing peptidoglycan, which protects the bacteria from a wide range of environmental conditions, and which Gram-negative bacteria lack.

Despite the presence of bacteria within the building environment, most of these species do not pose a danger to human health, and are commonly found on and within organisms (Wilson, 2005). Rather, specific sources that become favourable to harmful bacteria are of IAQ concern. One example is infectious bacteria transmitted when infected individuals cough and sneeze, and the bioaerosols can remain in the air for other potential hosts to be exposed (Burge, 1995; Wilson, 2005). A great concern within the building environment is due to standing water in an HVAC system (Burge, 1995; Flannigan, Samson, & Miller, 2011; Stetzenbach, 1996). If the system is off, bacteria that

normally cannot establish a stable community in the building environment, such as *Legionella* spp., *Pseudomonas* spp., and *Flavobacterium* spp., can grow and be transported to the rest of the building when the HVAC system is turned on. The infections caused by these bacteria can be fatal, especially to those who are immunocompromised.

In the building environment, bioaerosols are controlled by ventilation. Since most fungal matter comes from the outdoor environment or are brought in when people move into the building environment, mechanical ventilation systems filter the air that enters the building, thus reducing the concentration indoors. In contrast, bacterial matter mostly comes from indoor sources. In this case, mechanical ventilation systems dilute the concentration of bacteria and their associated metabolites within the building environment with filtered outdoor air. A sufficient ventilation rate is required to reduce the concentration of indoor microbial sources to prevent adverse health effects associated with microbes and their metabolites.

Due to the myriad of factors that links environmental concentration to health outcomes, many methods were created to determine the normal airborne fungal concentration within buildings (Rao et al., 1996). Studies attempted to assess fungal concentrations in buildings with varying number of SBS complaints (Pastuszka et al., 2000; Robertson, 1997; Schillinger et al., 1999; Yang et al., 1993). A few conclusions were drawn: One, the concentration of fungi is lower indoors than outdoors in buildings with few SBS complaints despite seasonal variation. Two, the proportion of the species of fungi are found to be similar indoors and outdoors, which indicates the absence of another species growing indoors. IAQ investigations note the change in the proportion to

determine whether there is a source of fungi, and where that source may be. Three, the average concentration of non-pathogenic fungi in mechanically-ventilated buildings with few SBS complaints is within 60-250 CFU/m^3. In order to account for the variability of airborne fungi in the outdoor environment (due to climate) that can infiltrate to the building environment, most government agencies and health and safety organizations regard levels that exceed 500-1000 CFU of fungi/m^3 or a difference in proportion of the outdoor fungal population as an indicator for the existence of indoor fungal sources (Rao et al., 1996).

On the other hand, airborne bacteria levels in buildings fluctuate with occupancy. This includes the presence of pets and animals in the building, which have higher airborne bacteria levels than buildings without pets (Pastuszka et al., 2000). Due to the close association of many bacteria with the human (and animal) skin, a high level of airborne bacteria may simply be a result of a lack of cleaning (Robertson, 1997). Unlike fungi, which can be controlled by the use of filters, airborne bacteria must be removed from the building with proper ventilation. Most mechanically-ventilated buildings would reach equilibrium levels around 200-300 CFU/m^3 (Robertson, 1997; Tsai & Macher, 2005). Schillinger et al. (1999) found that the range of indoor airborne bacterial concentrations within temporary structures (such as tents) did not exceed 350 CFU/m^3. Due to the relationship between occupancy and airborne bacteria, government agencies and health and safety organizations either do not have guidelines, or have opted for a guideline level around 500 CFU/m^3 of non-pathogenic species, which includes fungal species, to indicate a high indoor source of bacteria that requires attention (Rao et al., 1996).

When an interior living wall is installed, bacteria and fungi can grow in the soil and around the plants in the living wall, acting as a microbial source within the building. When provided with a water source (e.g.: run-off from watering the plant), the soil becomes a suitable environment for bacteria and fungi to grow and propagate throughout the building environment. Both the roots and the leaves support a community of microbes that may influence the microbial community in the surrounding area (Junker & Tholl, 2013). Although microbes can bind to the root system of the plant, the binding is species-dependent and affected by environmental factors, with some plant species favouring certain microbial species over others (Bazin et al., 1990; Ortega et al., 2016). It is unknown whether plant-microbe interactions in the living wall may aid in the release of more fungi and bacteria into the building environment, given the different growing substrate used by living walls.

Since the living wall is likely to act as a microbial source within the building, the living wall may influence IAQ and the way it is controlled. In a room with a living wall, the generation rate of airborne microbes and microbial components may become greater. This may require more ventilation to draw the air away from the occupants, or an air purifier nearby to remove the bioaerosols.

Endotoxin. Endotoxins are parts of the bacterial cell wall membrane containing lipopolysaccharides (LPS). Lipopolysaccharides are a group of molecules made of a polysaccharide and a lipid A moiety. The polysaccharide moiety allows cells related to the immune system to recognize and bind to LPS and the lipid A moiety induces the innate immune response. LPS are only found in the membrane of Gram-negative bacteria. When the cell wall membrane is compromised, LPS and associated membrane

compounds are released into the surrounding environment and become attached to dust, creating inhalable endotoxin fragments. Due to the reduced viability of Gram-negative bacteria compared with Gram-positive bacteria in the building environment, endotoxin acts as an indirect measure of Gram-negative bacteria.

The innate immune response occurs when a LPS binding protein binds to the LPS. This allows macrophages and other phagocytic immune cells to be bound to LPS by a toll-like receptor on the cell. Once LPS is broken down inside the macrophage, it signals the cell to release proinflammatory mediators, such as interleukin (IL) 1-beta, tumor necrosis factor alpha, IL-6, and IL-8. LPS appears to have a much stronger effect than most cell wall components in activating the innate immune response (Becker et al., 2002; Douwes et al., 2003), and can induce the production of IL-8 more effectively than other cell wall components. This mediator induces recruitment of other immune cells to the target location. In the bloodstream, the resulting effect from LPS may cause a strong fever, and possibly septic shock. In the lung, phagocytic immune cells can remove all of the LPS such that no LPS travel to the bloodstream. Therefore, inhaled endotoxins do not cause systemic effects, but rather local effects on the lung (Rylander, 2002).

Rylander (2004) suggests that prolonged exposure to low levels (100-200 Endotoxin Unit per cubic metre, or EU/m^3) of endotoxin may mediate SBS through inflammatory response in the respiratory tract. Recent studies have shown that cough, shortness of breath, and wheezing may also occur at this level (Smit et al., 2008; Thorne et al., 2005; Zhang et al., 2011). Several studies have found that endotoxin may exacerbate asthma symptoms in the presence of allergens (Bertelsen et al., 2009; Cho et al., 2013; Michel et al., 1996; Rizzo et al., 1997; Thorne et al., 2005). It is likely that

endotoxin aids in the recruitment of antigen presenting cells, which can induce and increase the allergy-induced antibody-mediated immune response (Schaumann et al., 2008). When building inhabitants are chronically exposed to a low level of endotoxin, they may experience discomfort and reduced productivity.

At the moment, the Health Council of The Netherlands (2010) has proposed an occupational exposure limit of 90 EU/m^3 from inhalable dust exposure for an eight-hour shift, increasing the limit (50 EU/m^3) from the previous report made in 1998. This limit is proposed to reflect the negligible health effect found in recent studies that have examined lung function and endotoxin exposure. However, the buildings in which living walls are installed may have populations that stay longer within the building environment than workers (e.g.: hospitals, apartment buildings) and may be more susceptible to endotoxins, such as infants, and elderly. Rabinovitch et al. (2005) examined the personal endotoxin exposure of a small group of elementary school children in relation to their asthma symptoms and the forced expiratory volume in one second (FEV_1). The study found a decrease of 316 mL in FEV_1 per 1 EU/m^3 for particles at or below an aerodynamic diameter of 10 μm, despite an average exposure (median = 0.37 EU/m^3) well below the limit set by The Netherlands. This suggests that the endotoxin concentrations in community settings may be high enough to induce adverse health effects.

Since Gram-negative bacteria can be found among organic materials, the living wall would likely be a source for endotoxins. It is common to find bacteria in soil, which is used in some living wall designs. It is also common to find bacteria in the growing substrate in the living wall, for which the plant provides nutrient to recruit

microorganisms to harvest other nutrients. The addition of a living wall into the building environment may thus increase airborne endotoxin.

Study Goals

The study outlined in the following section was designed to evaluate the living wall's influence on IAQ by determining the living wall's impact upon these IAQ indicators. Based on the literature, it is expected that the presence of an interior living wall would decrease the concentration of CO_2 and VOCs, and increase the RH and the concentration of bioaerosols.

Given the many existing living wall designs, there may be differences in their impact on IAQ. It is hypothesized that the size of the living wall design is related to the impact on the IAQ factors. It is hypothesized that 1) CO_2 is correlated mostly with the mass of the leaves and shoots, 2) RH is correlated mostly with watering needs, and 3) bioaerosols is correlated mostly with the mass of the plants and substrate.

The results of the study aims to provide recommendations for buildings with interior living walls to make use of the possible IAQ benefits and reduce the possible IAQ issues that may arise. The results can show potential sources of IAQ issues during IAQ investigations, and may be used to estimate the impact upon IAQ if any specific size parameter(s) are correlated with any of the IAQ factors.

2 Methods and Materials

2.1 Living Walls Systems

To determine the variability among different living wall carriers, three living wall systems were examined (Figure 2.1).

1) G-O_2™ (Plant Connections Inc.), is made of stainless steel (61 x 61 x 15.6 cm), and has 24 cells (15 cm by 10 cm) in each panel. Small holes below each cell and a gap at the top of the panel allow water to drain out of each cells.
2) Modulogreen™ (ByNature Design), resembles a black vertical planter box sticking out of the wall. Each of its 20 cells (18.6 cm wide, 18.6 cm tall) is connected by a space in the back of the panel for irrigation purposes and is slanted at an angle from the vertical plane (106.2 x 75.2 x 6.5 cm).
3) Evergreen® Flexipanel 60-50 (Intercoast Building Solutions), resembles a cushion with 30 slits arranged in a rectangular array with 6 slits across the length and 5 slits across the height of the assembly (60 x 51 x 5.2 cm). Each slit is roughly two inches wide, and allow up to 30 two-inch potted plants into the wall. This living wall contains rock wool inside, which acts as the growing substrate, covered with a layer of UV stabilized, water absorptive fleece.

Growing Conditions

Living walls are irrigated from the top of each panel or the top of a group of panels and then gravity fed to the lower sections, therefore the growing substrate must accommodate the flow of water and slightly reduce water retention across the living wall. The growing substrate used in this study was a mix of indoor potting soil (Miracle-Gro®

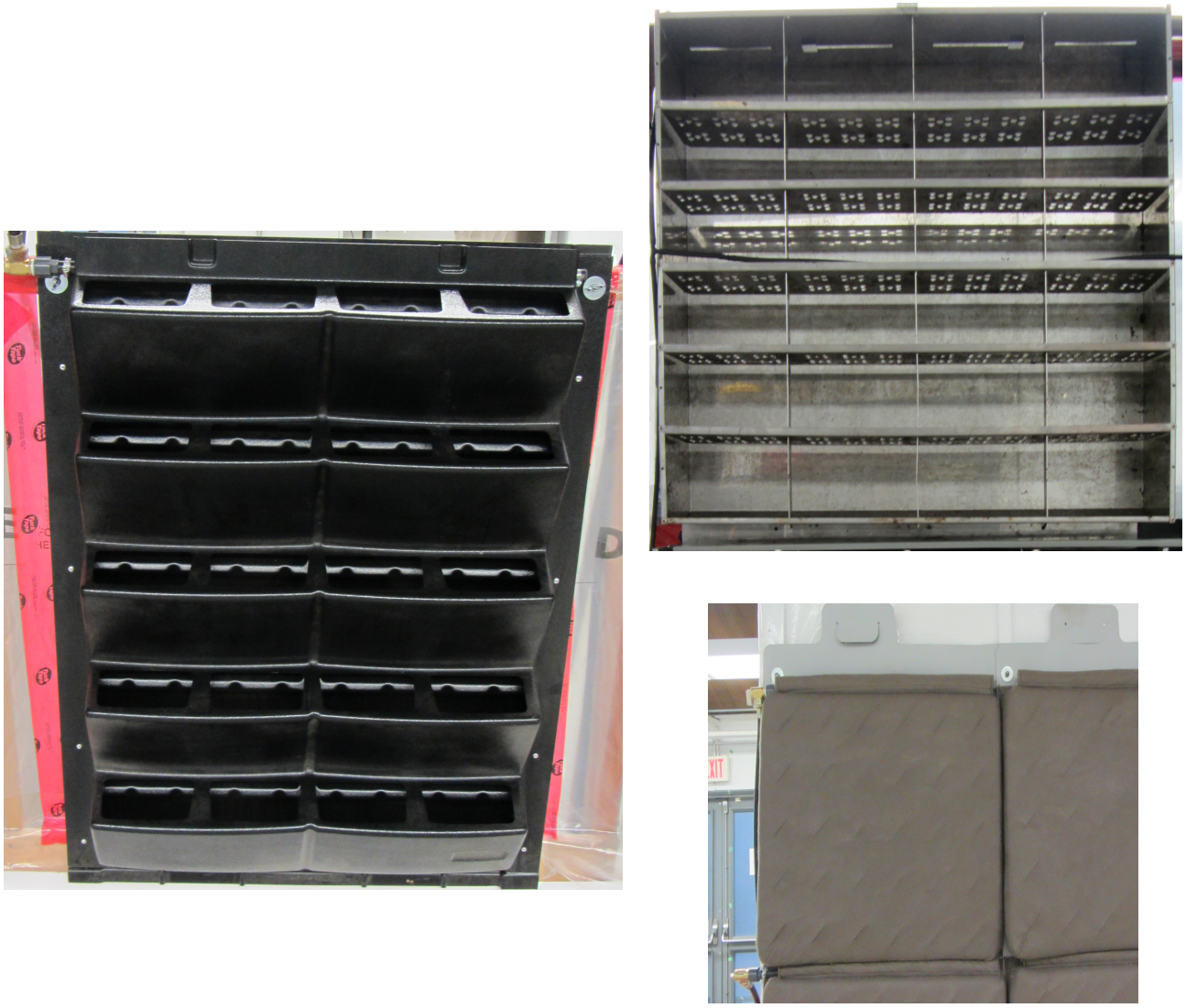

Figure 2.1: The empty living wall carriers. Left: Modulogreen™ (106.2 cm x 91.0 cm x 16.6 cm). Top right: G-O_2™ (61 cm x 61 cm x 15.6 cm). Bottom right: Evergreen® Flexipanels (60 cm x 51 cm x 6 cm).

All-Purpose Potting Mix)[1] and red scoria in a ratio of 7:3, in accordance with the living wall designers' instructions[2]. The growing substrate was added to the cells of G-O_2 and Modulogreen living walls as they were planted. Plants in four-inch pots were transplanted into G-O_2 and Modulogreen living walls to replace dead plants due to pest infestations.

In contrast, Evergreen Flexipanels do not require additional growing substrate. Since the Evergreen Flexipanel has a non-soil based substrate, it requires the addition of fertilizer to supplement the nutrients that are otherwise provided to the plant by a soil-based growing substrate. The fertilizer (AAT ProHort 15-15-18 soil-free feed, Direct Solutions Inc.) was added into the water used to irrigate at approximately 1.30 mg/L (6 g of fertilizer in every 4608 L of water), according to its designer's instructions. At the time of planting the walls, some of the propagation soil in which the plants had been growing was incorporated into the living wall assemblies to reduce damage to the roots during transplantation from the two-inch pots.

Six species of houseplants were used in this study. They were: spider plant (*Chlorophytum comosum*), creeping fig (*Ficus pumila*), common ivy (*Hedera helix*), golden pothos (*Epipremnum aureum*), Pilea (*Pilea depressa*), and a type of fern (*Pteris cretica*). All of these were acquired from Burnaby Lake Greenhouses Ltd (Surrey, BC, Canada). These were chosen due to their shade-tolerance, their availability in two-inch pots, and their use as ornamental plants as recommended by a living wall installer.

A total of 40 positions for the plants were prepared on each of the three living wall assemblies. Seven spider plant, seven creeping fig, seven common ivy, seven golden

1 Some of the potting soil was acquired at least one year before planting, and was used only in the planting of Modulogreen (since those were planted first). The potting soil was also a general potting soil mix. The rest of the potting soil was acquired afterwards.

2 The same recommendations from Modulogreen was applied to both Modulogreen and G-O_2.

pothos, six Pilea, and six fern were planted in each living wall. For the purposes of this study, an extra 10 slits were made evenly across the Evergreen living wall to fit a total of 40 plants. Each slit fitted one plant on the Evergreen living wall carrier. Of the 24 cells in G-O_2 living wall carrier, 16 cells were planted with 2 plants and the remaining 8 were planted with one plant. The cells with one plant and those with two plants were evenly separated in such a way that the living wall resembled those installed by professionals. Two plants were placed in each of the 20 cells in Modulogreen living wall carrier. Three panels of each living wall design were planted, leading to a total of nine living walls.

The living walls were grown indoors in the main laboratory/classroom at the Centre for Architectural Ecology (CAE) at the British Columbia Institute of Technology (Figure 2.2). The wall assemblies were planted and supported on frames facing the south clerestory windows, which provided some sunlight to the growing area of the walls. Grow lights, which consist of an array of LED lights of various non-green visible light wavelengths facilitated the growth of the living walls. The living walls received about 40 μmol/m^2/s of photosynthetic photon flux density (PPFD) for 12 hours of the day, which is the average maximum amount of light that can be used by the shade-tolerant plant species at ambient CO_2 concentrations (Giorgioni & Neretti, 2009). A light meter (LI-250, LI-COR Enviromental, Lincoln, NB) was used to ensure that the surface of the living wall received adequate PPFD in Giorgioni & Neretti (2009). The living walls were irrigated according to the installers' guidelines using an automated irrigation system.

2.2 Test Chamber

A small room within the CAE was modified for the purpose of this study to be the test chamber (Figure 2.3). The chamber dimensions were: length of 2.14 m (north and

south wall), width of 1.90 m (east and west wall), and height of 2.25 m. A glass sliding door (the entrance of the test chamber) is located on the east wall. A window and a passive vent opening of 20.2 cm by 20.8 cm were located on the south side (Figure 2.4). A wall-mounted disc-bladed ventilation fan, located on the north wall, drew air into the test chamber when turned on by a switch. Gaps around the test chamber's envelope were sealed with duct tape. A space was renovated on the north wall to allow one living wall panel to be hung, slightly above the ventilation fan, when required for the duration of the study. (Figure 2.5). The living walls were moved between the growing area and test chamber for sampling.

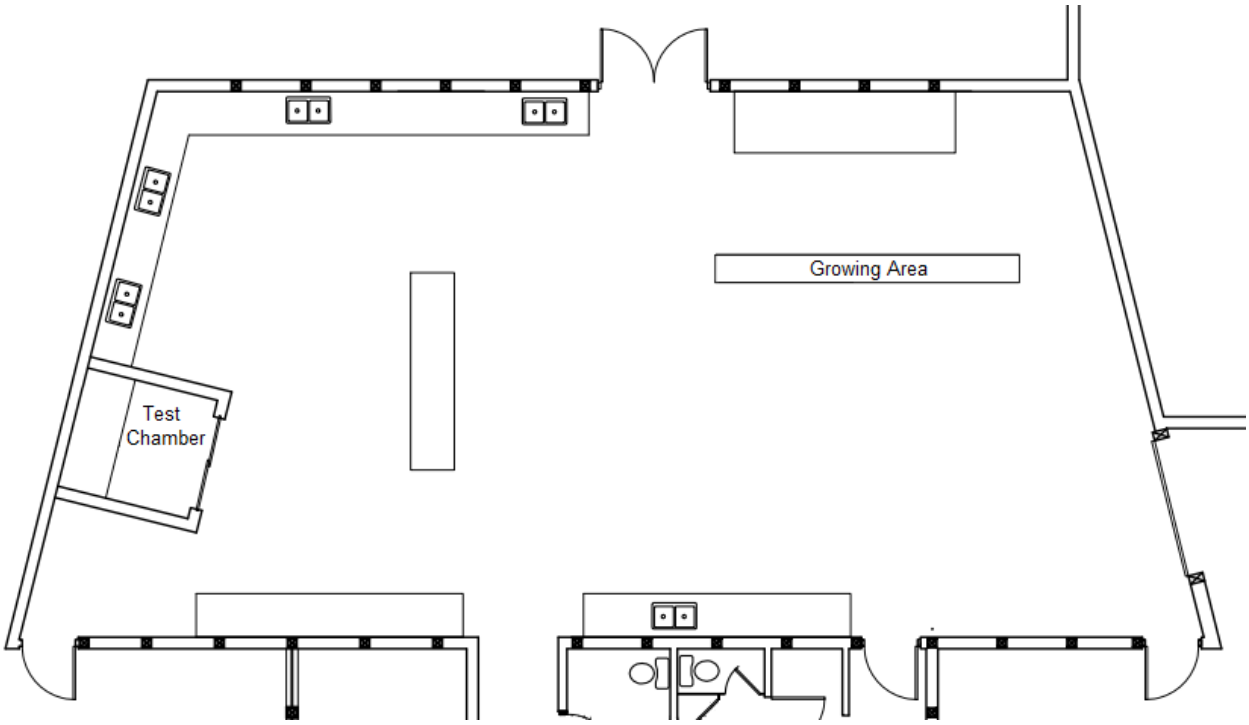

Figure 2.2: Floor plan of the northern part of the Centre for Architectural Ecology. The living walls were planted and grown at the growing area on the east side of the building. The test chamber is on the west side of the building.

Figure 2.3: View of the test chamber from the outside. A 36” measuring stick is placed on the left side of the sliding door for scale.

Figure 2.4: Partial view of the south wall inside the test chamber, showing two of the three LED lights, the room fan, and the exhaust vent above the base of the light stand.

Figure 2.5: Partial view of the north wall inside the test chamber, along with an empty Modulogreen panel. The ventilation fan is located near the base of the wall.

Since most living walls are placed in public spaces such as lobbies or reception areas, the test chamber was configured to simulate the building environment of a hotel lobby. This involved controlling the irradiance level, temperature, ventilation rate, and simulating environmental conditions that reflect human occupancy summarized in Table 2.1.

A series of hoses and tubes were installed through the chamber walls to allow automatic irrigation and sampling without entering the test chamber. The flow rate from the irrigation system was measured to determine the amount of water released during each watering event (results found in Section 3.3).

Irradiance

Artificial lighting sources were installed to mimic the lighting level of a lobby. This provided the necessary PPFD to the living wall so that it was between the light compensation point and the light saturation point for various shade-tolerant plant species, which is roughly 40-70 μmol/m^2/s (Aruas et al., 1986; Bauer & Bauer, 1980; Broschat, 2002; Giorgioni & Neretti, 2009; Oh et al., 2011). In the test chamber, two LED

Table 2.1: Summary of environmental conditions in the test chamber

Irradiance level	40-70 μmol/m^2/s across the area where the living wall was placed
Temperature	20-25 °C
Ventilation Rate	7.66 L/s, pedestal fan to facilitate air mixing in the chamber
Simulated Human Occupancy	Added the following compounds in the test chamber until the steady state concentration reached a mean of • 250 ppm of CO_2 above ambient concentration • 492 μg/m^3 of α-pinene (VOC) • 31 μg/m^3 of toluene (VOC) • 55 μg/m^3 of 2-butanone (VOC)

lamps (19.5 W, 120 V, 5000 K, 1300 lumens) were placed by the south wall to provide lighting across the height of the living walls. Another LED lamp (8 W, 120 V, 4000 K) was placed on the ceiling above where the living walls were hung. The PPFD was kept within the acceptable irradiance range as measured by a light meter (LI-250, LI-COR Environmental, Lincoln, NB). The light was kept on when any samples were taken.

Temperature

The temperature of the test chamber was kept around 20-25 °C, which is within the thermal comfort range proposed by ASHRAE (2010b) given that the RH of the test chamber is kept between 30-60%.

Ventilation

The ventilation of the test chamber was modified to simulate a lobby environment. First, the air flow within the test chamber was adjusted such that the air circulated to simulate mixing in the space. The vents on the south wall were sealed and the windows were kept ajar with a width of 9.8 cm to ensure air circulated to the top half of the chamber before exiting the space through the window opening. A pedestal fan was placed by the south side of the chamber and faced a single direction to facilitate air moving in the bottom half of the chamber (Figure 2.3). The air movement within the chamber was examined by following the movement of the smoke generated by a smoke tube (Wizard Stick, Zero Toys, Concord, MA) when released at several points of the chamber to visualize mixing.

Second, the ventilation rate of the test chamber was modified to reflect the conditions inside buildings built to energy sustainability standards where the living wall

would likely be placed, while achieving an adequate level of ventilation based on the ASHRAE guideline (2010a).

Pilot fungal samples taken in the CAE were overgrown with fungi. For that reason, four electrostatic filters were placed upstream of the ventilation fan that drew air into the test chamber to remove a majority of the particulate matter entering the test chamber. This resulted in a flow rate of 7.66 L/s across the fan, as measured by a thermoanemometer (VelociCalc 9565, TSI, Shoreview, MN). Since the estimation of CO_2 and VOC removal was based on the dilution ventilation model (ACGIH, 2013), a mixing factor for the flow rate with respect to the change in concentration of contaminants in a room was determined. The ventilation rate was measured with the tracer gas decay method using sulfur hexafluoride outlined by Bearg (1993). A portable infrared spectrophotometer (Foxboro Wilks MIRAN 1A CVF, Thermo Fisher Scientific Inc., Waltham, MA) was set to detect the presence of sulfur hexafluoride by the increase in absorbance of infrared radiation with a wavelength of 10.7 μm at a pathlength of 0.75 m. The mixing factor (K_m) was calculated as shown below.

$$Km = Q / Q'$$

where Q is the volumetric flow rate as determined by the thermoanemometer, and

Q' is the volumetric flow rate as determined by the tracer gas decay method.

Simulating Human Occupancy

Since human activity releases CO_2 and a variety of VOCs, these were added into the test chamber. Pure CO_2 gas (Praxair Canada Inc., Mississauga, ON) was released into the test chamber at 60 mL/min, such that the steady-state concentration can show appreciable reduction, but below what is recommended by ASHRAE (2010a). It was

found to be sufficient to evaluate the reduction of CO_2 by the living walls at 250 ppm of CO_2 above ambient concentration in the CAE. A Mason jar with 2:1:1 mixture (based on liquid volume) of α-pinene (Aldrich Chemical Co., cat. 147524, St. Louis, MI), toluene (Fisher Scientific, cat. T324, Pittsburgh, PA), and 2-butanone (Aldrich Chemical Co., cat. 360473, St. Louis, MI) was placed in the chamber. The VOCs were chosen due to their different chemical structure, prevalence in indoor environments, and relatively similar vapour pressures (Bluyssen et al., 1996; Kostiainen, 1995; Wolkoff & Nielsen, 2001). The ratio of the mixture was based on creating relatively equal vapour volume as estimated using Raoult's law such that the mixture released roughly a total of 300 $\mu g/m^3$ when all three VOCs are added together. A candle wick was inserted through a cut opening on the lid of the Mason jar to slowly release α-pinene, toluene, and 2-butanone to reach a mean equilibrium concentration of 492, 31, and 55 $\mu g/m^3$ respectively. The VOC jar and CO_2 was released throughout the duration of the study.

2.3 Sample Collection Method

Volatile Organic Compounds Sampling

Air samples were collected using activated charcoal tubes (100/50 mg) (SKC, Inc. Eighty Four, PA) drawn by GilAir Plus air sampling pumps (Sensidyne, St. Petersburg, FL) at 0.2 L/min for 8 hours based on NIOSH method 1501, 1552, and 2500 (The National Institute for Occupational Safety and Health, 1994). A mass flow meter, TSI flow meter 4146, (TSI, Shoreview, MN) was connected upstream of the activated charcoal tube to determine the flow rate before and after each sample. The samples were void if

the measured flow rates differed by more than 10%. The mean of the measured flow rates of a sample was taken as the flow rate of that sample.

The samples were kept at -18 °C until analysis. Given the low sensitivity of the analytical method used in NIOSH method 1501, 1552, and 2500, a mass spectrometer was used instead of a flame ionization detector. The samples were extracted with carbon disulfide (Fisher Scientific, cat. C573, Pittsburgh, PA), then analyzed by gas chromatography-mass spectrometry for α-pinene, toluene, and 2-butanone. The extracted samples were carried through a wax column (J&W DB-Wax Ultra Inert GC Column 122-7032, Agilent Technologies, Santa Clara, CA) at 250 °C and 9.1 psi by helium with the flow split at 50:1 before the sample reaches the end of the capillary column. The analysis was calibrated for each compound with seven standards between the range of 1 to 140 ng/μL of extracted sample.

Carbon Dioxide and Relative Humidity Sampling

The CO_2 and the RH of the test chamber were sampled with a datalogging device (Q-Trak, TSI, Shoreview, MN) at 5-minute intervals. Each 24-hour period was considered a single sample. One Q-trak was placed in the test chamber, while another was placed outside the test chamber near the ventilation fan, for comparison purposes.

Due to the reliability of the two Q-traks and greater drift from the Q-trak in the test chamber, CO_2 concentrations measured by the Q-traks were adjusted based on the bump test results done after each calibration (details in Appendix A). The Q-traks were calibrated for CO_2 with zero-gas (total hydrocarbon <0.1 ppm) and 1000 ppm CO_2 (Praxair Canada Inc., Mississauga, ON) after three samples.

Since RH is affected by the temperature of the environment, and the temperature in the test chamber and the CAE were different, RH measurements were converted to absolute humidity (AH) and were utilized in data analysis. The conversion to AH is done applying the ideal gas law and the Antoine equation of water vapour (de Nevers, 2012):

$$AH = \frac{\frac{RH}{100\%} \times P \times MM}{R \times (T + 273.15)}$$

$$P = 10^{8.07131 - \frac{1730.63}{233.426 + T}}$$

where P is the saturated vapour pressure of liquid water in mmHg,

T is the air temperature in Celsius measured by the Q-trak,

MM is the molar mass of water,

R is the ideal gas constant, and

AH is the absolute humidity in g/m^3.

As the environmental conditions in the CAE affected those of the chamber and the changes over a day can vary drastically, the samples taken inside the test chamber were adjusted by the difference between the two devices as follows:

$$\Delta C = C_{in} - C_{out}$$

$$\Delta AH = AH_{in} - AH_{out}$$

where ΔC and ΔAH are the difference in CO_2 and AH in the test chamber compared with the laboratory,

C_{in} and AH_{in} are the CO_2 and AH measured in the test chamber, and

C_{out} and AH_{out} are the CO_2 and AH measured in the laboratory outside the test chamber.

To ensure that human activity in the CAE did not affect the results, the mean CO_2 and AH data for both Q-traks were analyzed for the periods between 12:00 am and 6:00 am within the 24-hour period, and each 24-hour period's mean ΔCO_2 and ΔAH measurements during that period of time represented the average CO_2 measurement for that sample. The maximum and minimum ΔAH of the entire period of a sample was used for analysis.

Viable Bioaerosols Sampling

Airborne fungi and airborne bacteria samples were collected onto agar plates by impaction using Andersen 6-stage and Andersen N-6 sampling heads (Grasby-Andersen, Atlanta, GA) respectively, drawing air into the sampler using Gilian Air-Con 2 pumps (Sensidyne, St. Petersburg, FL) at 28.3 L/min for 10 minutes. The Andersen 6-stage sampling head was employed to separate the large concentration of fungal spores found previously into six different plates based on decreasing particle sizes. A specific calibration head for the Andersen 6-stage and Andersen N-6 sampling heads was connected to a rotameter for calibration. The flow rates were calibrated before and after each sample. The samples would be considered void if the measured flow rates differed by more than 10%. The mean of the measured flow rates of a sample was taken as the flow rate of that sample.

For fungal samples, the plates contained malt extract agar (MEA) (Difco, Sparks MD) with 0.304 g/L of chloramphenicol (Aldrich Chemical Co. cat. 857440, Milwaukee, WI). For bacterial samples, tryptic soy agar (TSA) (Difco, Sparks MD) was used. Approximately 45 mL of the appropriate agar was poured into 100 mm petri plates (Phoenix Biomedical, Bolton ON). Air samples were taken 1) inside the test chamber, 2)

outside the test chamber by the ventilation fan (to be referred as CAE regarding the locations of fungi and bacteria samples), and 3) outside the CAE (outdoor). A reference building (RB) next to the CAE was also sampled to control for the higher fungal concentrations found in CAE, which did not represent typical indoor concentrations.

Fungal samples were incubated at room temperature for seven days. Fungal colonies on the plates were counted and characterized by genus according to the reference books by Flannigan, Samson and Miller (2011), Gravesen, Frisund, and Samson (1994), Malloch (1981), and St-Germain and Summerbell (1996). If the colony did not form conidiophore within the seven day incubation period, it was classified as sterile mycelia. All single-celled fungi were classified as yeasts.

Bacterial samples were incubated at 37 °C for 48 hours. Bacterial colonies on the plates were counted and characterized by staining characteristics and morphology. Gram stain was applied to all bacterial colonies after being heat-fixed upon a microscope slide. Crystal violet was first applied, then iodide, then washed with acetone/alcohol, and finally counter-stained with safranin. The samples were quickly rinsed with water one minute after the application of each of the Gram stain components. Actinomycetes, a Gram positive, rod-shaped bacteria, was also examined as a classification group for bacteria due to the frequency found among samples. The classification group were thus: Gram positive cocci; actinomycetes; Gram positive rod, not including actinomycetes; Gram negative cocci; and Gram negative rod.

Endotoxin Sampling

Air samples were collected for endotoxin in particulate matter using a 7-hole sampling head (SKC, Inc. Eighty Four, PA) to capture the inhalable fraction of dust.

Glass fibre filters (Type A/E 37 mm, Pall Corporation, Ann Arbor, MI) were depyrogenated by baking the filters at 180 ºC for two hours prior to use. GilAir Plus air sampling pumps (Sensidyne, St. Petersburg, FL) drew air for 9 hours at 2 L/min. A special calibration head connected to a mass flow meter, TSI flow meter 4146 (TSI, Shoreview, MN) was held tightly at the opening of the 7-hole sampling head to determine the flow rate before and after each sample. The samples would be void if the measured flow rates differed by more than 10%. The mean of the measured flow rates of a sample was taken as the flow rate of that sample.

Endotoxin was extracted from the filter using the extraction method found to have the greatest sensitivity (Spaan, Heederik, Thorne, & Wouters, 2008). The glass fibre filters were stored at 4 °C until extraction and the analytical test was run. The filters were extracted with 0.05% v/v of Tween-20 (Fisher Chemical cat. BP33, Fisher Scientific, Hampton, NH) in depyrogenated water (LAL Reagent Water, Lonza, Walkersville, MD). The filters were then vortexed briefly to keep the whole filter in water. Afterwards, the filters were placed on a shaker for 60 minutes, then in a sonicator bath for 60 minutes, and finally in a centrifuge at 1000 g for 15 minutes at room temperature to complete the extraction.

The concentration of endotoxin was determined using a kinetic Limulus amebocyte lysate assay, Kinetic-QCL (Lonza Group Ltd., Walkersville, MD). A standard 4-parameter fit curve was generated using E. coli O55:B5 endotoxin (Lonza, Walkersville, MD) over the range of 50 EU/mL to 0.049 EU/mL based on the following equation:

$$V_{max} = \frac{A - D}{1 + \left(\frac{X}{C}\right)^{B}} + D$$

where V_{max} is the maximal velocity of the reaction involving endotoxin,

x is the concentration of endotoxin in EU/mL extracted, and

A, B, C, D are the constants of the 4-parameter fit curve.

Samples and standards and their duplicates were dispensed in 96 well microtitre plates, and incubated at 37 °C for 75 minutes. The absorbance of light at a wavelength of 405 nm was read at 30 second intervals using a spectrophotometer, Molecular Devices SpectraMAX 190 microplate reader (Thermo-Fisher Scientific, Waltham MA). Samples of a kinetic assay were accepted for further analysis if the standard curve generated had a coefficient of determination (r^2) greater than 0.98. Samples were rejected if the coefficient of variation was greater than 25% between the sample and its duplicate.

Background Samples

The background samples taken are shown in Figure 2.6. Three samples of airborne fungi, airborne bacteria, endotoxin, and VOCs were each taken when the chamber had 1) no living wall carrier, 2) an empty Modulogreen carrier, 3) an empty Evergreen carrier, and 4) an empty G-O_2 carrier. An additional three samples of airborne fungi, airborne bacteria, and endotoxin were taken in the chamber without a living wall after all the treatments were sampled.

Similarly, three samples (24-hour periods) of CO_2 and RH were taken when the chamber had no living wall carrier with 50-60 mL/min of 100% CO_2 released into the chamber. Three samples were taken without the VOC mixture placed in the test

chamber to ensure that the CAE does not have a source of the three VOC, and all were found below the limit of detection of 20 μg/m^3. Seven samples were taken one day after the VOC mixture was placed in the test chamber without a wall assembly.

2.4 Experimental Treatments

The nine living walls were grown in May 2013 until full coverage in CAE (when the foliage of the plants visually covers 90% of the living walls) before experiments between February 2015 to December 2016 (Figure 2.7). The same methods and parameters as for the background samples were taken for the six IAQ indicators (Figure 2.5). For all 9 living walls, 6 samples of airborne fungi and airborne bacteria and three samples of endotoxin were taken at least 12 hours after the living walls were moved in the test chamber so that it could adapt to the new environment. Twice the samples were taken for fungi and bacteria to account for the wide variability with fungi and bacteria samples. Three samples of CO_2 and RH were taken with one planted Modulogreen, one planted Evergreen, or one planted G-O_2 in the test chamber.

Due to instrument errors that led to a change in the sampling and analytical method, the number of VOC samples in total was reduced to accommodate the timeline of the study. Seven samples of VOCs were taken with one Modulogreen living wall after the living wall was placed in the test chamber for seven days. This was repeated with another Modulogreen living wall, leading to a total of 14 samples. Five samples were taken when the concentration of VOCs in the test chamber did not reach steady-state equilibrium, and were removed from analysis.

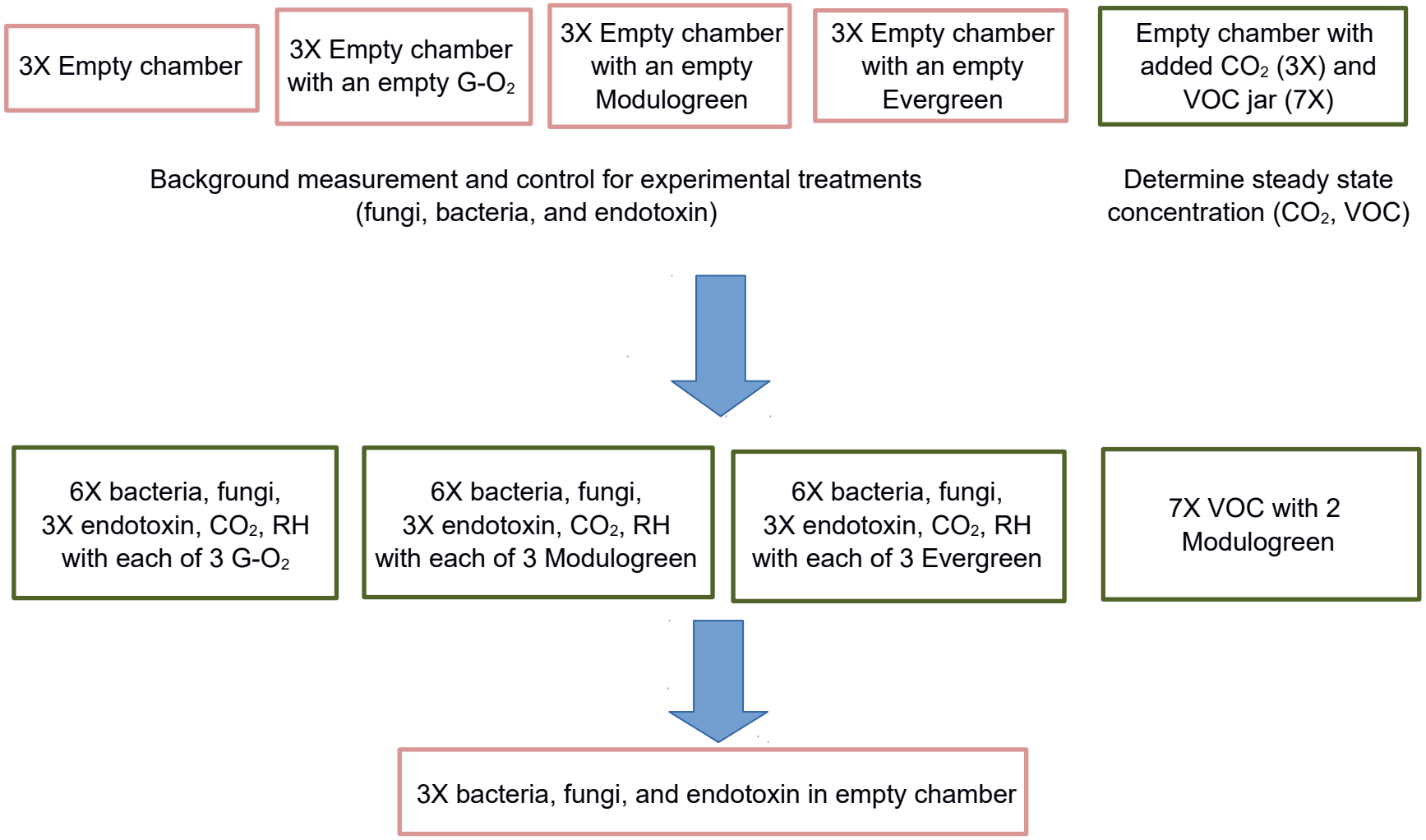

Figure 2.6: The background, control, and experimental treatments that was completed. The order completed is from top to bottom. There is no order to the items listed horizontally.

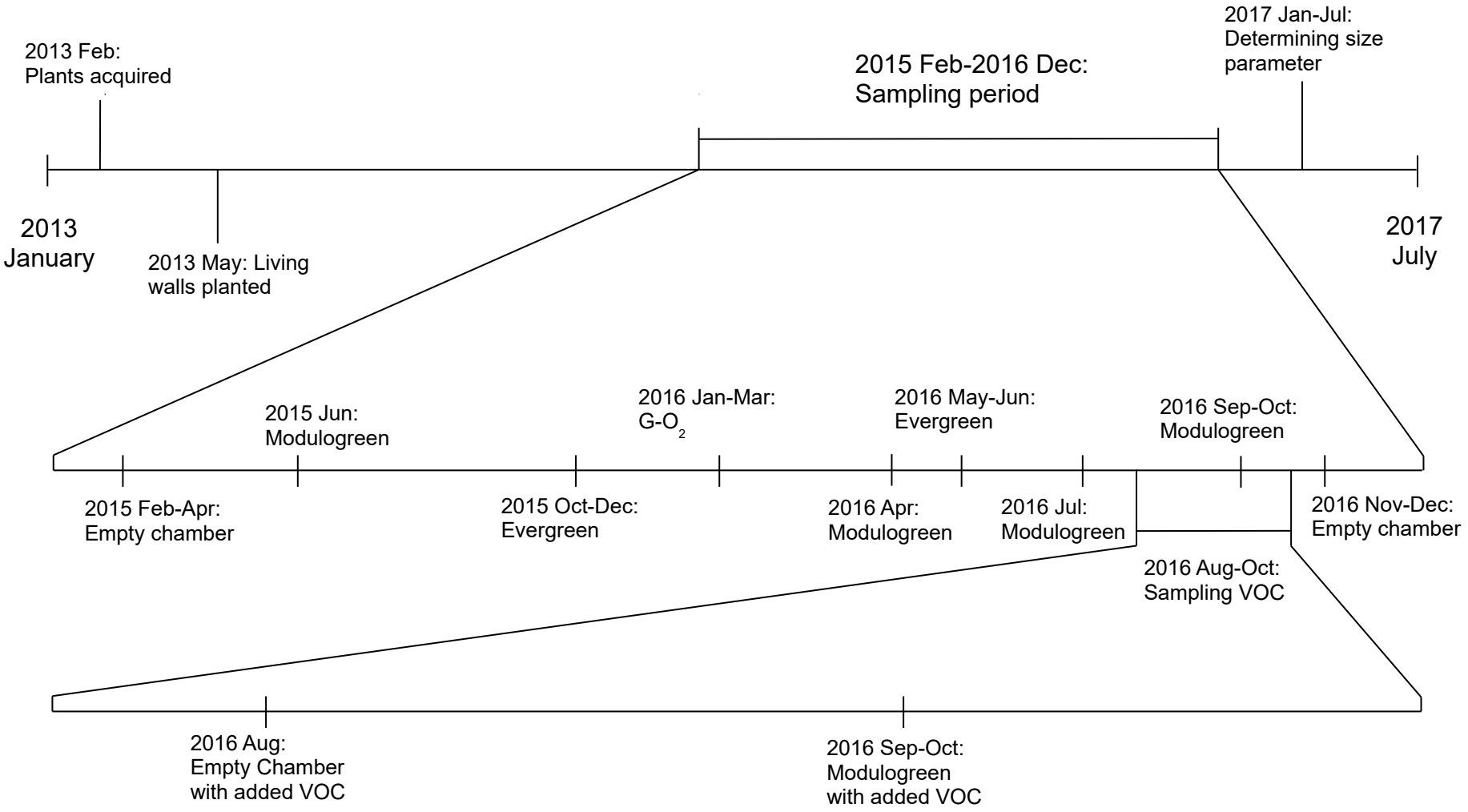

Figure 2.7: Timeline of the events that occurred during the study.

Determining the Size Characteristics of the Living Wall

Some of the size characteristics of the living wall are based on those found in Orwell et al. (2004). These include the dry weight of the shoots, the roots, the growing substrate, and the area of the leaves.

The dry weight and the organic mass of the shoots (which include all the parts of the plant above ground), the roots, and the growing substrate of each living wall were taken after the living walls were sampled in the test chamber. The plants and the growing substrate of each living wall was taken out of the living wall, then the plants were separated by their shoots and roots with a single-edge razor blade. All the roots were shaken inside a sieve with 3/8” holes to remove excess dirt. The pieces of roots large enough to be caught on the sieve were retrieved using a pair of forceps. To estimate the remaining root pieces that were not caught on the sieve, samples of the growing substrate (of 473 mL) were sifted in a series of sieve with holes ranging from 9.5- to 1-mm. All the pieces of roots from the sample were separated from the growing substrate on a piece of aluminum foil, and the remaining growing substrate was placed back in a glass container. Three samples of the growing substrate were taken from each Modulogreen carrier, five from each G-O_2 carrier, and one from each Evergreen carrier. More samples of the substrate were taken from the carriers with more substrate to ensure consistency among samples. One Modulogreen, one G-O_2, and two planted Evergreen walls were compromised near the end of the study, and the dry weight of the plant and the growing substrate were not examined for these living walls. The dry weight of the plant parts and the growing substrate were determined after dry-heating the components at 105 °C for 24 hours. The dry weight of the roots and growing substrate

for each sample were used to determine the ratio of remaining roots left in the growing substrate, and the weight was added to the total dry weight of the roots separated at first.

The organic mass of the shoots, roots, and growing substrate were determined by ashing the dried substance at 550 °C for two hours, then weighing the inorganic ash. The difference between the initial mass of the sample and the mass of the remaining ash is the mass of the organic matter of the sample. Seven samples of the shoots and roots from each living wall and three samples of the soil from each living wall were taken.

While the area of the leaves of the living wall has been reported in many of the studies evaluating the VOC reduction by potted plants and plant leaves, it was not possible to measure the leaf area after sampling due to several incidents leading to the death of the plants in some of the living walls after the living walls were sampled.

Some studies have reported an approximate volume that the potted plants take up relative to the volume of the room (Kim et al., 2009; Song, Kim, Sohn, 2007; Song, Kim, Sohn, 2011; Tudiwer & Korjenic, 2017). However, they have not mentioned how they have determined this number. As such, the volume of the living wall is estimated as the volume of the carrier when filled with substrate. While this underestimates the true value, there is at least some way of comparing with other works that report the relative volume of the potted plants.

Finally, living wall installers measure their growth by the area of living wall installed. It is a simple size characteristic of the living wall that can be easily determined and easily related to the impacts upon IAQ to living wall installers.

2.5 Statistical Analysis

Volatile Organic Compounds and Carbon Dioxide

Values below the limit of detection were substituted with a value at half the limit of detection if more than half of the samples were below the limit of detection, and at the limit of detection divided by the square root of two if less than half of the samples were below the limit of detection. Goodness-of-fit to normality and lognormality were determined for concentration of 2-butanone, α-pinene, and toluene with Shapiro-Wilks test upon the untransformed data and the log-transformed data. Based on the Goodness-of-fit tests, Student's t-tests were conducted between samples of α-pinene and toluene taken without a planted living wall and those with the planted Modulogreen using the untransformed data, and Student's t-test was conducted between samples of 2-butanone taken without a planted living wall and those with the planted Modulogreen using the log-transformed data.

Goodness-of-fit to normality were determined for ΔCO_2 concentration with Shapiro-Wilks test upon the original data and the log-transformed data. ANOVA was then conducted with ΔCO_2 concentration to assess difference depending on the living wall design. Post-hoc t-tests were conducted between the samples taken without a planted living wall and those with each living wall.

Relative Humidity (and Absolute Humidity)

Since AH reflects the true change in the concentration of water vapour in the air, it is used in the data analysis. ANOVA was conducted on the difference in AH among samples taken in the test chamber with each of the three living wall designs and without

a living wall. Student's t-tests were conducted between the average peak ΔAH after each watering event for each planted living wall design and the average background ΔAH. The change in RH at 20 °C and 25 °C from the change in AH were determined as follows:

$$\Delta RH = \frac{\Delta AH \times R \times (T + 273.15)}{P \times MM} \times 100\%$$

$$P = 10^{8.07131 - \frac{1730.63}{233.426 + T}}$$

where P is the saturation vapour pressure of water as determined by the Antoine equation,

R is the ideal gas constant,

MM is the molar mass of water, and

T is the air temperature in Celsius.

Microbiological Community

If there were no colony of a classification group detected in a sample in any location, the sample value was substituted. If more than or equal to half of the samples did not detect a colony of a classification group, the sample is substituted with a value half of the limit of detection. If less than half of the samples did not detect a colony of a classification group, the sample is substituted with a value that is the limit of detection divided by the square root of two. If both sample location for the determination of the above ratios found no colony of a classification group, the ratio is removed from analysis. Otherwise, the ratio would be 1, which would skew the analysis.

To determine whether there is a difference in the size of the fungal and bacterial community due to the presence of a living wall, the ratio of the concentration of total

airborne fungi, and total airborne bacteria were computed between the test chamber and in CAE (TC/CAE). Kruskal-Wallis tests were conducted among TC/CAE ratios based on the living wall design in the test chamber (planted Modulogreen, planted Evergreen, planted G-O_2, or no planted living wall) for total airborne fungi, and total airborne bacteria. Post-hoc Dunn's tests was conducted between TC/CAE without a planted living wall and those in the chamber with each living wall design.

To determine whether there was a difference in the similarity of the fungal and bacterial community between the CAE, the reference building (RB), and outdoors, Spearman's rho was determined between the ranked total fungal and bacterial concentration sampled 1) in CAE and outdoor, 2) in RB and outdoor, and 3) in CAE and RB based on the genus for fungi and staining characteristics and morphology for bacteria. A greater Spearman's rho approaching 1.0 means there are greater similarities in the fungal community between the two environments. Mann-Whitney's U tests were also conducted on the concentration of *Pencillium* spp., *Cladosporium* spp., *Aspergillus* spp., *Paecilomyces* spp., *Botrytis* spp., *Cunninghamella* spp., *Aureobasidium* spp., actinomycetes, cleistothecium, total airborne fungi, and total airborne bacteria sampled in CAE and in RB relative to the outdoor concentration measured at that time. Since the number of samples that can detect *Botrytis* spp., *Cunninghamella* spp., *Aureobasidium* spp., and cleistothecium are less than half, and at least one living wall design would not have any sample detecting these fungal classification groups, the differences among living wall designs could not be evaluated for classification groups. Differences in the concentration of a particular classification group shows which type of fungi is responsible for the differences between the fungal community of the two environments.

To determine whether there is a difference in the similarity of the fungal and bacterial community due to a living wall, Spearman's rho was determined between the ranked concentration sampled in TC and CAE for *Pencillium* spp., *Cladosporium* spp., *Aspergillus* spp., *Paecilomyces* spp., *Botrytis* spp., *Cunninghamella* spp., *Aureobasidium* spp., actinomycetes, and cleistothecium. Kruskal-Wallis test was conducted on Spearman's rho correlation results based on the living wall design in the test chamber for each classification group. Post-hoc Dunn's test was conducted with between the Spearman's rho results for samples taken in the test chamber without a planted living wall and those in the chamber with each living wall design. If the results of the Kruskal-Wallis were not significant, Mann-Whitney U test was also conducted on the Spearman's rho correlation result between samples taken with and without a living wall to observe a possible trend that may be obscured by the wide variability in fungal concentration.

Since some classification groups had many samples below the limit of detection, observations were made upon the percent of samples above the limit of detection by sample locations (TC to CAE) and the presence of a living wall for each classification group.

Endotoxin

Samples with a coefficient of variation greater than 25% were removed from further analysis. Sample average that yielded a V_{max} below the bottom 10% of the calibration curve were considered to be below the limit of quantitation, and were assigned a value corresponding to half of 0.098 EU/mL of lab analyte (which is 0.454 EU/m^3). Goodness-of-fit to lognormality were determined for endotoxin concentration

with Shapiro-Wilks test upon the log-transformed data. One-way ANOVA was then conducted upon the log-transformed samples of endotoxin concentration to compare the different living wall designs. Post-hoc t-tests were conducted on the log-transformed data between the samples taken without a planted living wall and those with each of the three living wall design.

Determining the Impact of the Living Wall on IAQ

The impact of a living wall panel was calculated from the average relative to the size characteristic of the living wall. A living wall's removal rate of CO_2, butanone, toluene, and α-pinene was calculated based on the following equation, which was derived by the dilution ventilation equation (details in Appendix B):

$$X = \frac{Q' \times MM \times (C - C_X)}{C_X \times MV}$$

where X is the plant's removal rate of a gaseous compound,

Q' is the effective ventilation rate of the test chamber,

C is the mean concentration of a gaseous compound in the test chamber without a living wall,

C_X is the mean concentration of a gaseous compound in the test chamber with a living wall,

MM is the molar mass of the gaseous compound, and

MV is the molar volume of the gaseous compound, which is 24.45 L/mol at 1 atm and 25 °C.

The IAQ indicators were examined for trends associated with the plant quantifiers. The correlation and the coefficient of determination (r^2) were calculated to

determine the strength and direction of the relationship among the plant quantifiers and the IAQ indicators.

3 Results

3.1 Size Characteristics of the Living Wall

Table 3.1 lists some of the size characteristics of the living wall. It is clear that the three living wall designs vary in the different ways that of the size characteristics are correlated with one another among the living wall designs. Evergreen and G-O_2 takes up a similar area on a wall, the Modulogreen takes up more than twice that area. Despite the larger area of the Modulogreen, G-O_2 takes up more than twice the volume. For that reason, the G-O_2 also holds less substrate than the Modulogreen. Since Evergreen is a non-soil based living wall, the amount of substrate is much less than soil-based living walls.

Table 3.1: The size characteristics of the three living wall designs

	G-O_2	Modulogreen	Evergreen
Number of plants	40	40	40
Area of living wall (m^2)	0.372	0.799	0.306
Relative Volume of living wall (%)	0.63	0.25	0.17
Dry weight – shoots (kg)	0.321	0.236	0.242
Dry weight – roots (kg)	0.641	1.278	0.169
Dry weight – substrate (kg)	17.52	12.54	0.170
Organic weight – shoots (kg)	0.276	0.207	0.222
Organic weight – roots (kg)	0.578	0.990	0.151
Organic weight – substrate (kg)	2.53	2.00	0.110

3.2 Volatile Organic Compounds

The average concentration of toluene did not differ significantly ($p = 0.068$) between samples taken with and without a living wall. This was likely influenced by a large standard deviation among samples taken in the presence of a living wall (Figure 3.1).

The concentration of 2-butanone in the test chamber significantly decreased ($p < 0.05$) from a geometric mean of 54.1 µg/m^3 without the living wall to a geometric mean of 18.0 µg/m^3 (Figure 3.2). Seven of nine samples were below the limit of detection, which would likely lead to an overestimation of the true geometric mean. There was an

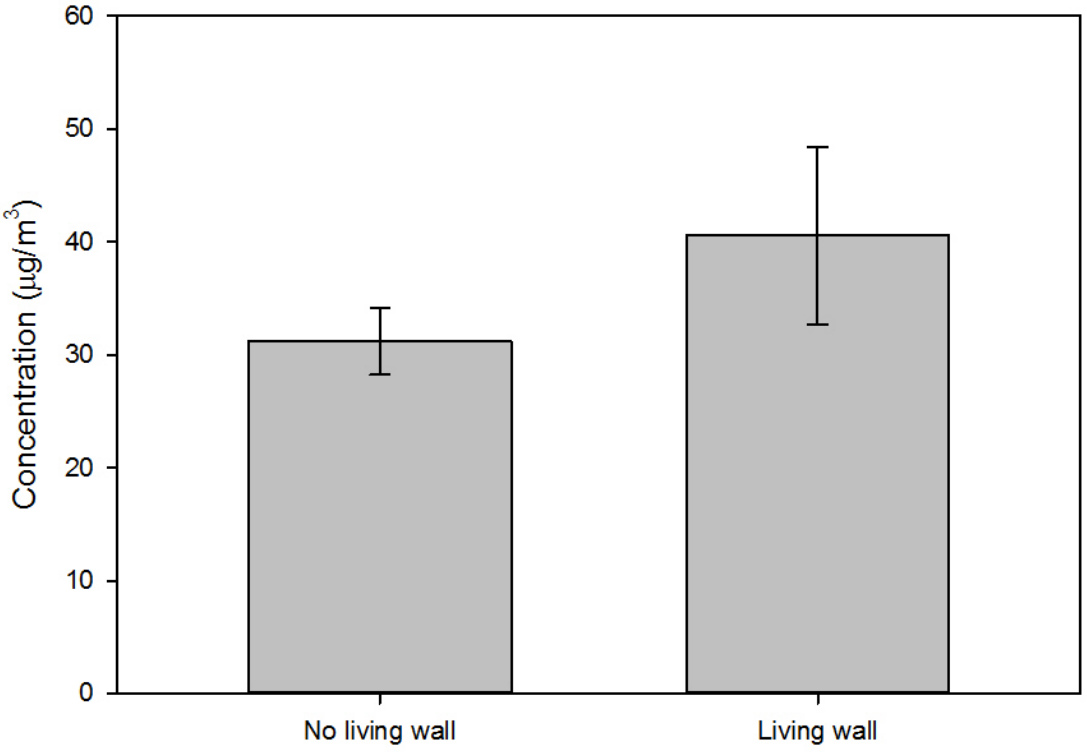

Figure 3.1: No change in toluene concentration in the presence of the Modulogreen living wall. The bars represent the mean concentration, and the error bars denote the 95% confidence interval around that mean. n = 7 for No living wall; n = 9 for Living wall.

estimated 65 µg/h of 2-butanone removed from the test chamber by one living wall, or 36.6 µg/h/kg dry weight of plants in the living wall.

In contrast, the mean concentration of α-pinene significantly increased from 492 µg/m^3 to 843 µg/m^3 in the presence of the living wall ($t = 3.29$, $p < 0.05$) (Figure 3.3). Based on the equation for removal rate, there was an estimated 5.1 mg/h of α-pinene added into the test chamber from the living wall, or 2.9 mg/h/kg dry weight of plants in the living wall.

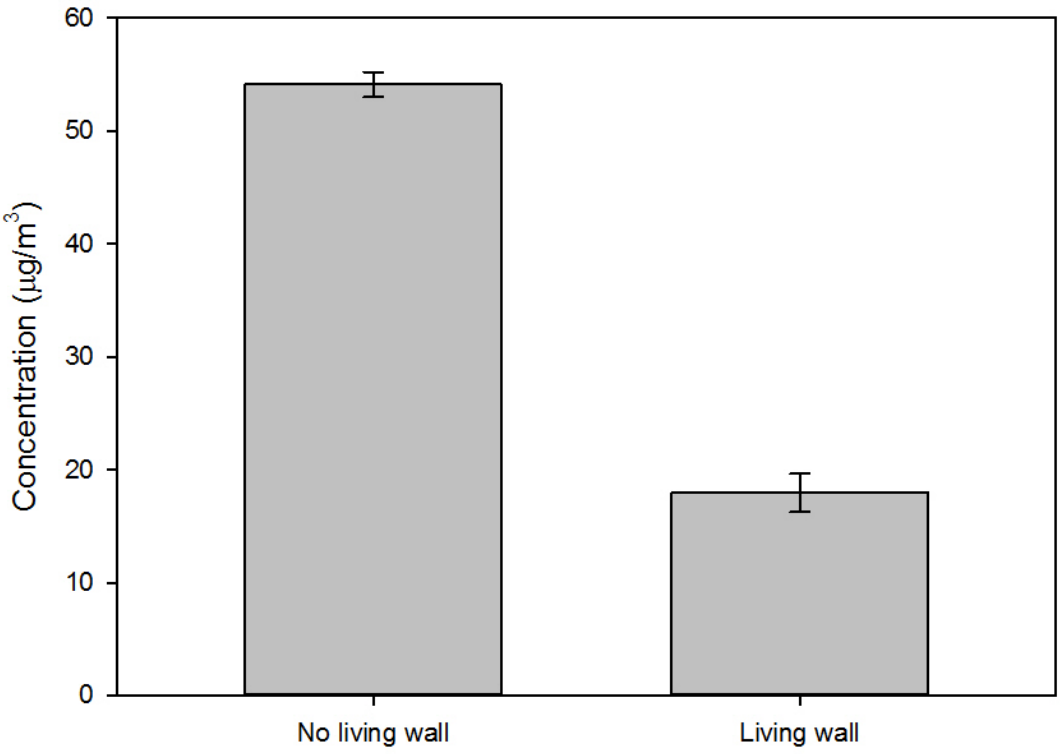

Figure 3.2: Change in 2-butanone concentration in the presence of the living wall. The bars represent the geometric mean concentration, and the error bars denote the 95% confidence interval around that mean. Values below the limit of detection is substituted with a value at half the limit of detection (10.8 µg/m^3). n = 7 for No living wall; n = 9 for Living wall.

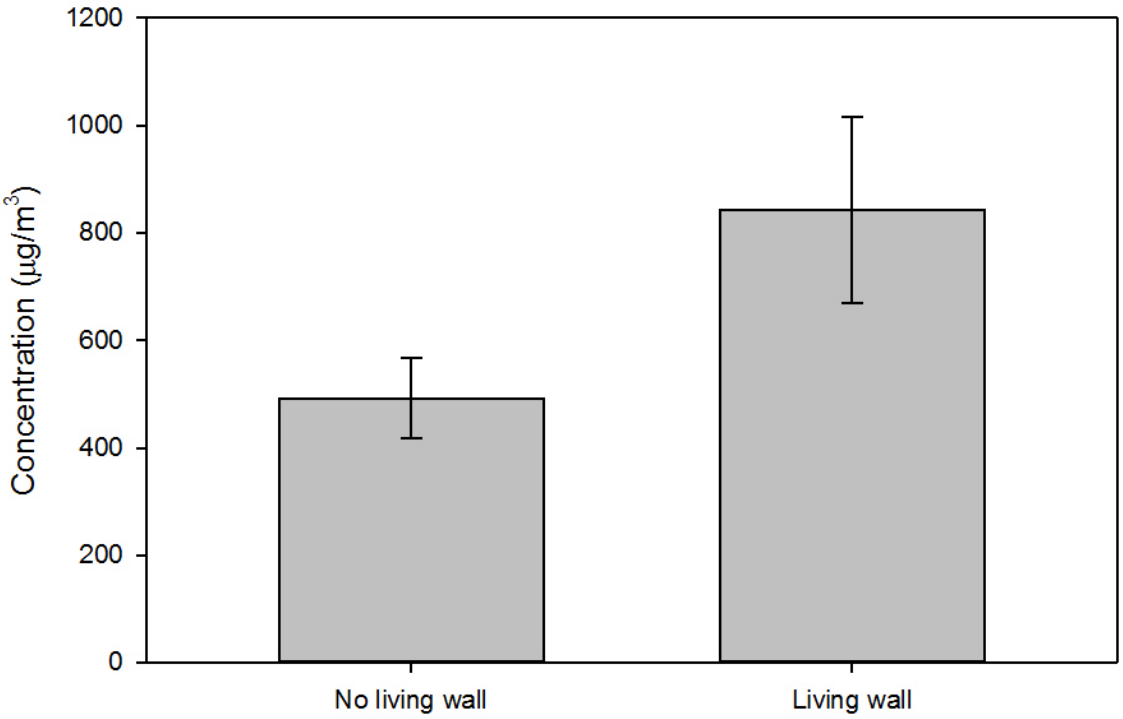

Figure 3.3: Change in α-pinene concentration in the presence of the living wall. The bars represent the mean concentration, and the error bars denote the 95% confidence interval around that mean. n = 7 for No living wall; n = 9 for Living wall.

Table 3.2: Summary of t-tests for the removal of three volatile organic compounds by a living wall

	Mean concentration without living wall ($\mu g/m^3$)	Mean concentration with living wall ($\mu g/m^3$)	t-test (p-value)	Removal rate per living wall (µg/h/wall)	Removal rate per dry weight of plants (µg/h/kg)	Removal rate per area of living wall ($\mu g/h/m^2$)
Toluene	31.2	40.6	1.98 (0.068)	---	---	---
2-butanone*	54.1	18	15.3 (3.1×10^{-9})	65	43	81
α-pinene	492	842	3.29 (5.3×10^{-3})	- (5.1×10^3)	- (2.9×10^3)	- (6.4×10^3)

*t-test was done based on the log-transformed data, and the average reported is the geometric mean.
---Not reported for toluene because the change was not significant.

3.3 Carbon Dioxide

There was a significant difference among samples taken in the test chamber without a living wall and with each of the planted living wall designs ($F = 9.93$, $p < 0.05$). Post-hoc t-test showed significant reduction of CO_2 with any of the three living wall designs, and found no difference in CO_2 concentration among the three living wall designs (Table 3.3). The presence of one G-O_2, Modulogreen, or Evergreen living wall significantly reduced a mean concentration of 253 ppm above ambient concentration of the CAE (as measured by the Q-trak outside the test chamber by the ventilation fan of the test chamber) to 170, 149, and 165 ppm above ambient concentration respectively (Figure 3.4). This corresponded to an estimated removal rate of 4.6, 5.8, and 4.9 mg/h at steady-state equilibrium respectively.

The average CO_2 concentration in the CAE was roughly 550 ppm, and it ranged from 500 to 600 ppm. On average, the CO_2 concentration in the test chamber was 800 ppm without a living wall, a value which under normal circumstances would require actions to reduce the concentration.

Table 3.3: Summary of post-hoc t-test for the removal of carbon dioxide by three living wall designs

	Concentration without living wall (ppm)	Concentration with a living wall (ppm)	Post-hoc t-test (p-value)	Removal rate per living wall (mg/h/wall)	Removal rate per dry weight of plants (mg/h/kg)	Removal rate per area of living wall (mg/h/m^2)
G-O_2	253	170	4.12 (0.001)	4.6	4.8	12.4
Modulogreen		149	5.42 (<0.001)	5.8	3.9	7.3
Evergreen		165	4.66 (<0.001)	4.9	12.0	16.1

*Non-significant post-hoc t-tests between G-O_2 and Modulogreen ($p = 0.78$), G-O_2 and Evergreen ($p > 0.9$), and Modulogreen and Evergreen ($p > 0.9$).

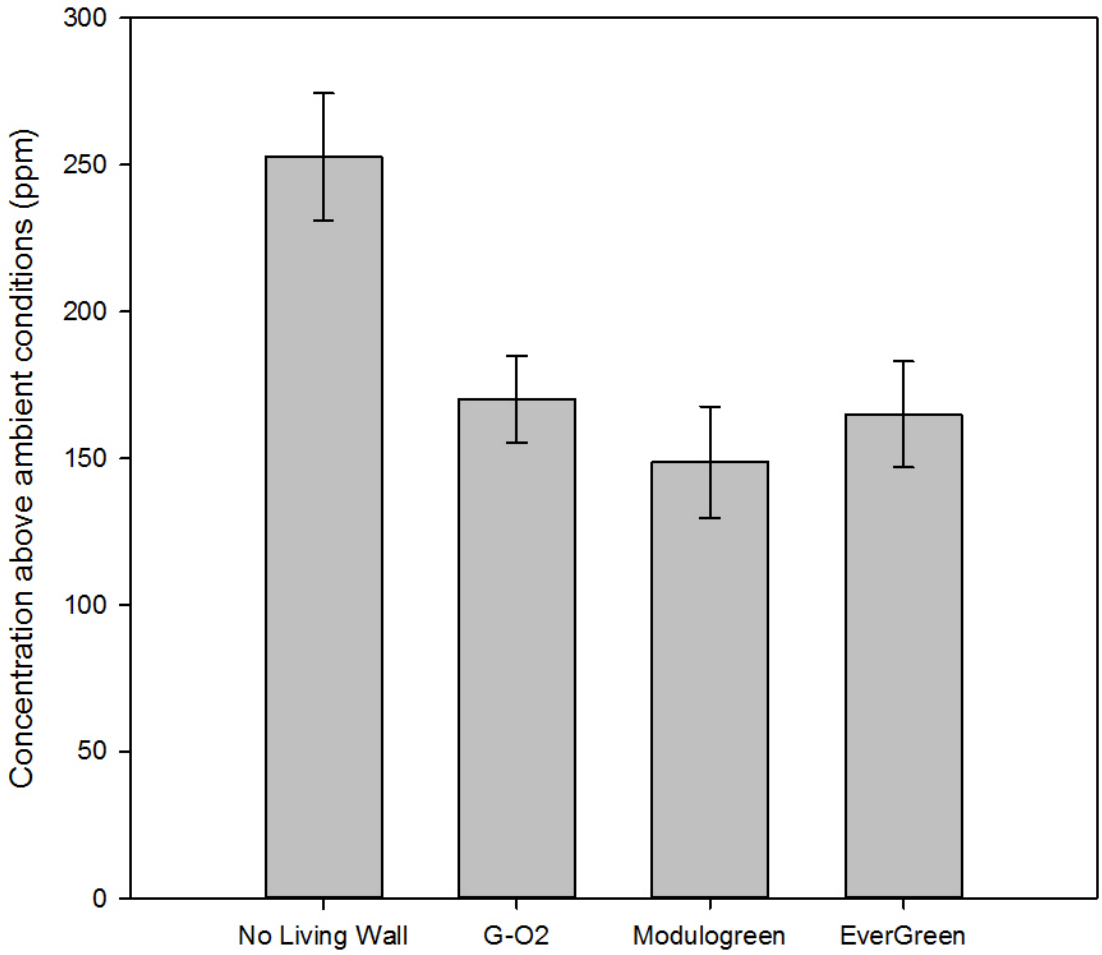

Figure 3.4: Change in CO_2 concentration due to the presence of a living wall. The bars represent the mean concentration, and the error bars denote the 95% confidence interval around that mean. There is no significant difference in the reduction of CO_2 between the three living wall designs. n = 4 for No living wall; n = 10 for G-O_2; n = 14 for Modulogreen; n = 17 for Evergreen. Average CO_2 concentration in the CAE during the study is roughly 550 ppm.

Correlation of various size characteristics to the rate of CO_2 removal by the living wall did not show a significant correlation (i.e. $r \neq 0$) based on t-test (Figure 3.5), despite the differences in various size parameters.

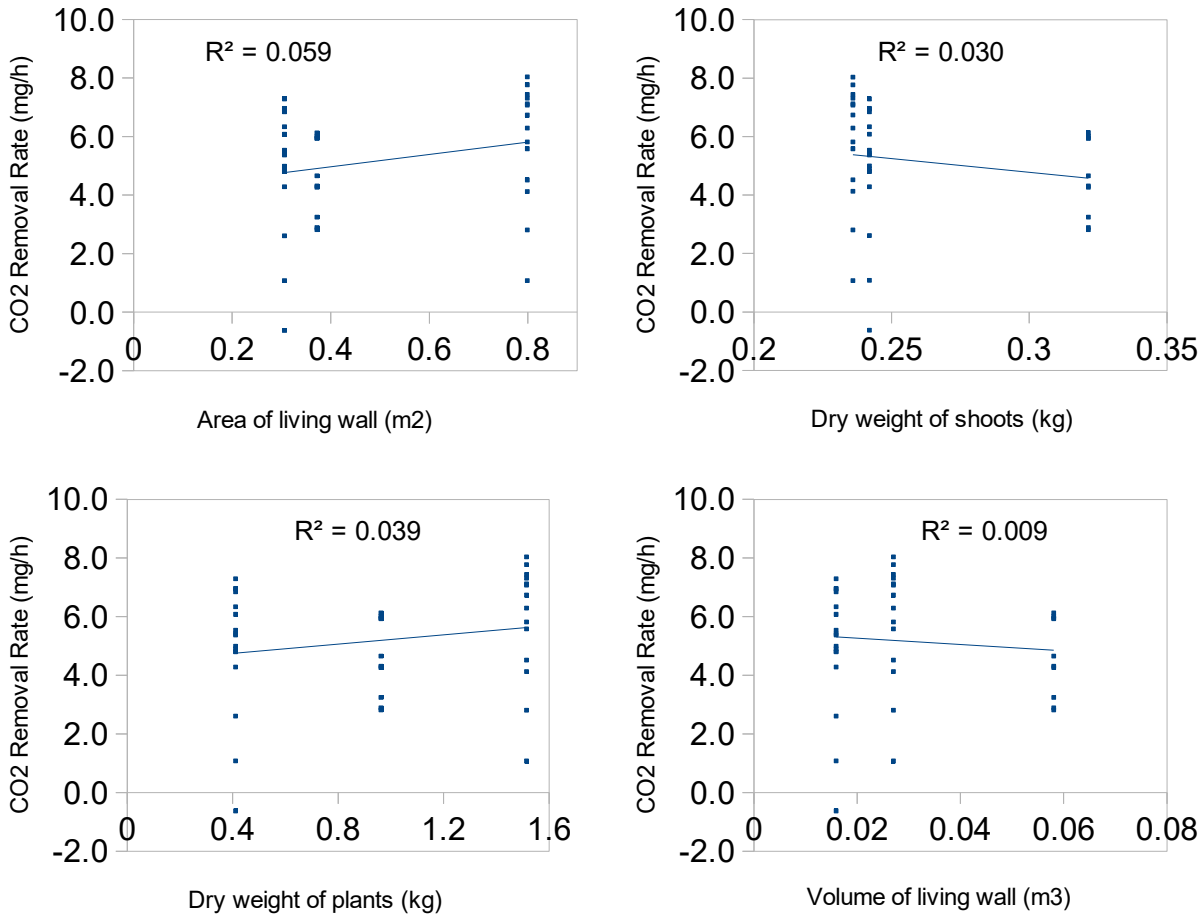

Figure 3.5: Scatterplots of the rate of carbon dioxide removal to various size characteristics of the living wall. The coefficient of determination (R^2) of each plot is shown. There was no significant difference for all size characteristics (Area of living wall: t = 1.56, df = 39, p = 0.13; dry weight of shoots: t = 1.09, df = 39, p = 0.28; dry weight of plants: t = 1.25, df = 39, p = 0.22; volume of living wall: t = 0.58, df = 39, p = 0.57).

3.4 Relative Humidity (and Absolute Humidity)

There was an increase in the average AH of the test chamber relative to the CAE in the presence of any of the three living wall designs (Figure 3.5). The change in AH was associated to the amount of irrigation (Table 3.4). A panel of Evergreen living wall requires more water to maintain (17.2 L/day) than a panel of G-O_2 (7.69 L/day) and Modulogreen living walls (6.31 L/day), which corresponds well to the average AH of Evergreen (2.61 g/m^3), G-O_2 (1.38 g/m^3), and Modulogreen (1.51 g/m^3) living wall. There

is a strong and significant correlation (r = 0.81) between irrigation and the average AH (Figure 3.7).

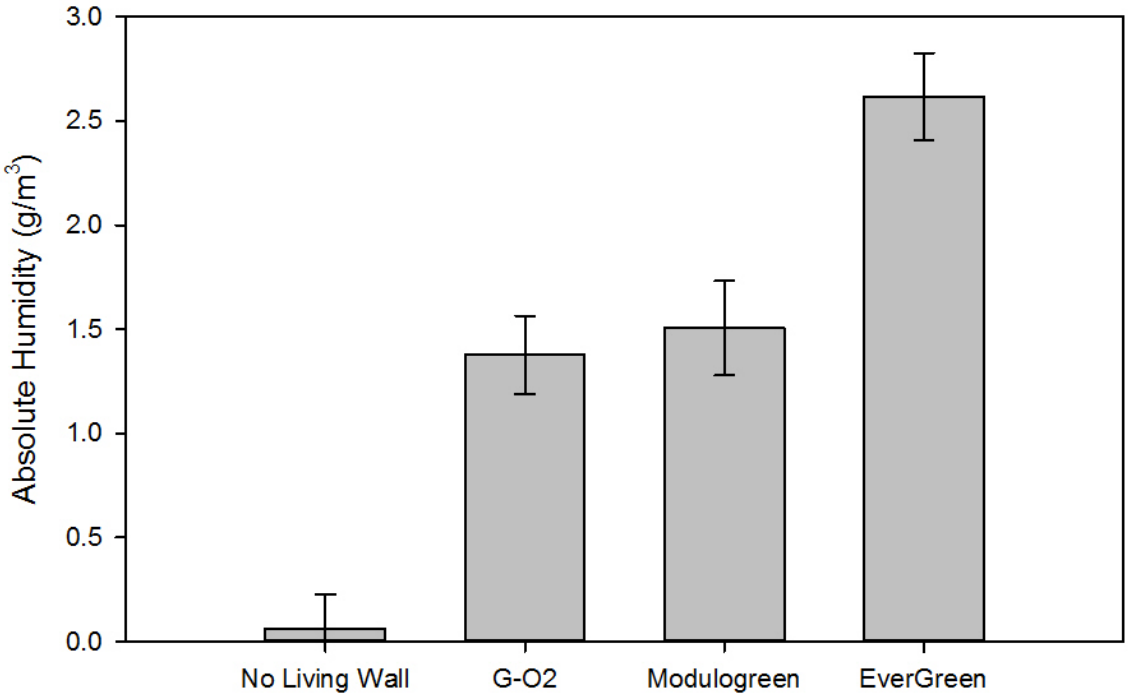

Figure 3.6: Change in average absolute humidity due to the presence of a living wall. The bars represent the mean concentration, and the error bars denote the 95% confidence interval around that mean. There is significant difference in AH among all groups except between G-O_2 and Modulogreen in the test chamber. n = 4 for No living wall; n = 10 for G-O_2; n = 14 for Modulogreen; n = 17 for Evergreen.

Table 3.4: Summary of post-hoc t-test for absolute humidity change by three living wall designs

	Mean ΔAH (g/m^3)	Minimum ΔAH (g/m^3)	Post-hoc t-test (p-value)	Irrigation (L/day)
G-O_2	1.38	0.47	5.6 (<0.001)	7.69
Modulogreen	1.51	0.32	6.4 (<0.001)	6.31
Evergreen	2.61	1.5	11.6 (<0.001)	17.2
G-O_2 to Modulogreen			0.79 (>0.9)	
G-O_2 to Evergreen			7.9 (<0.001)	
Modulogreen to Evergreen			7.8 (<0.001)	

AH did not change significantly when there was no living wall in the test chamber (t = 0.19, p = 0.86).

The effect of irrigation was clearly shown when examining the change in AH over a day (Figure 3.8). The peaks in AH for the Evergreen living wall occurred every four hours, which was the frequency of irrigation for this living wall. The single point of drastic increase in AH for the G-O_2 living wall was from irrigation that occurred every 48 hours, along with the other living walls in the growing area of the CAE. The peak in AH taken in the test chamber without a living wall was due to the irrigation of the living walls in the CAE. Among all RH data taken, the minimum AH increase due to the presence of a G-O_2 and Modulogreen living wall in the test chamber was 0.32 g/m^3. The minimum AH increase due to the presence of an Evergreen living wall in the test chamber was 1.5 g/m^3.

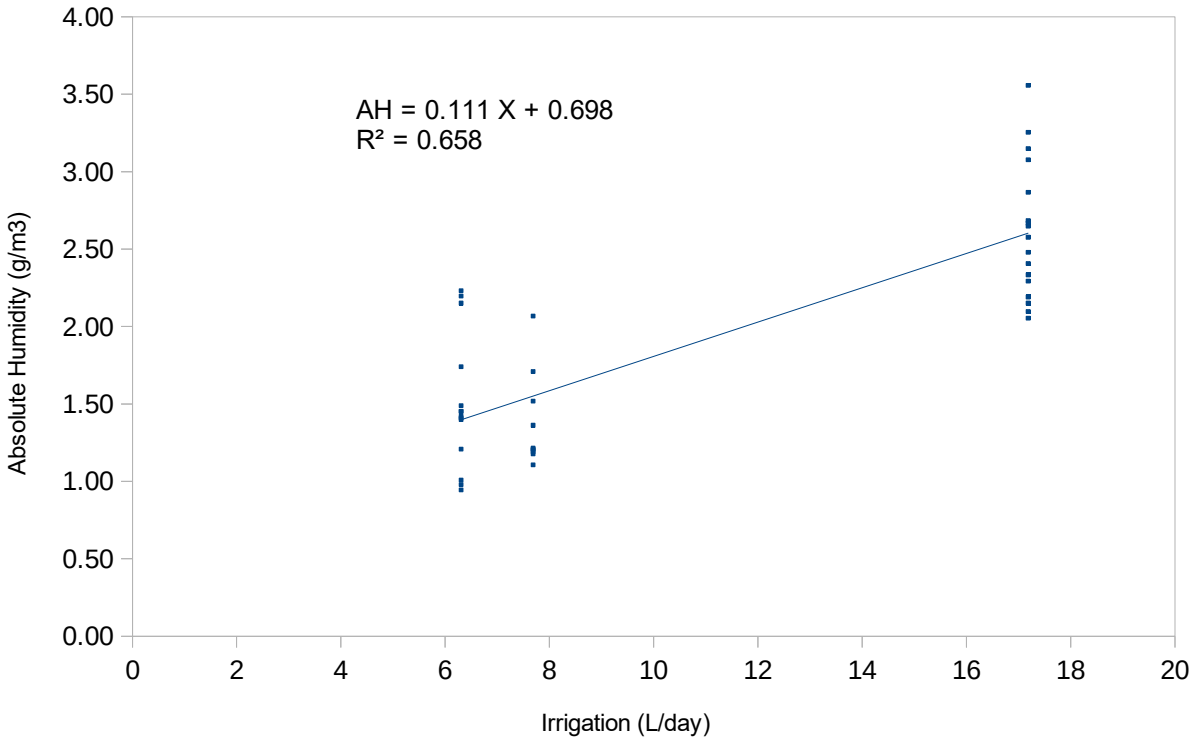

Figure 3.7: Scatterplot of absolute humidity increase in the test chamber and the amount of irrigation. The linear regression equation (where X is Irrigation) and the coefficient of determination (R^2) is shown on the plot.

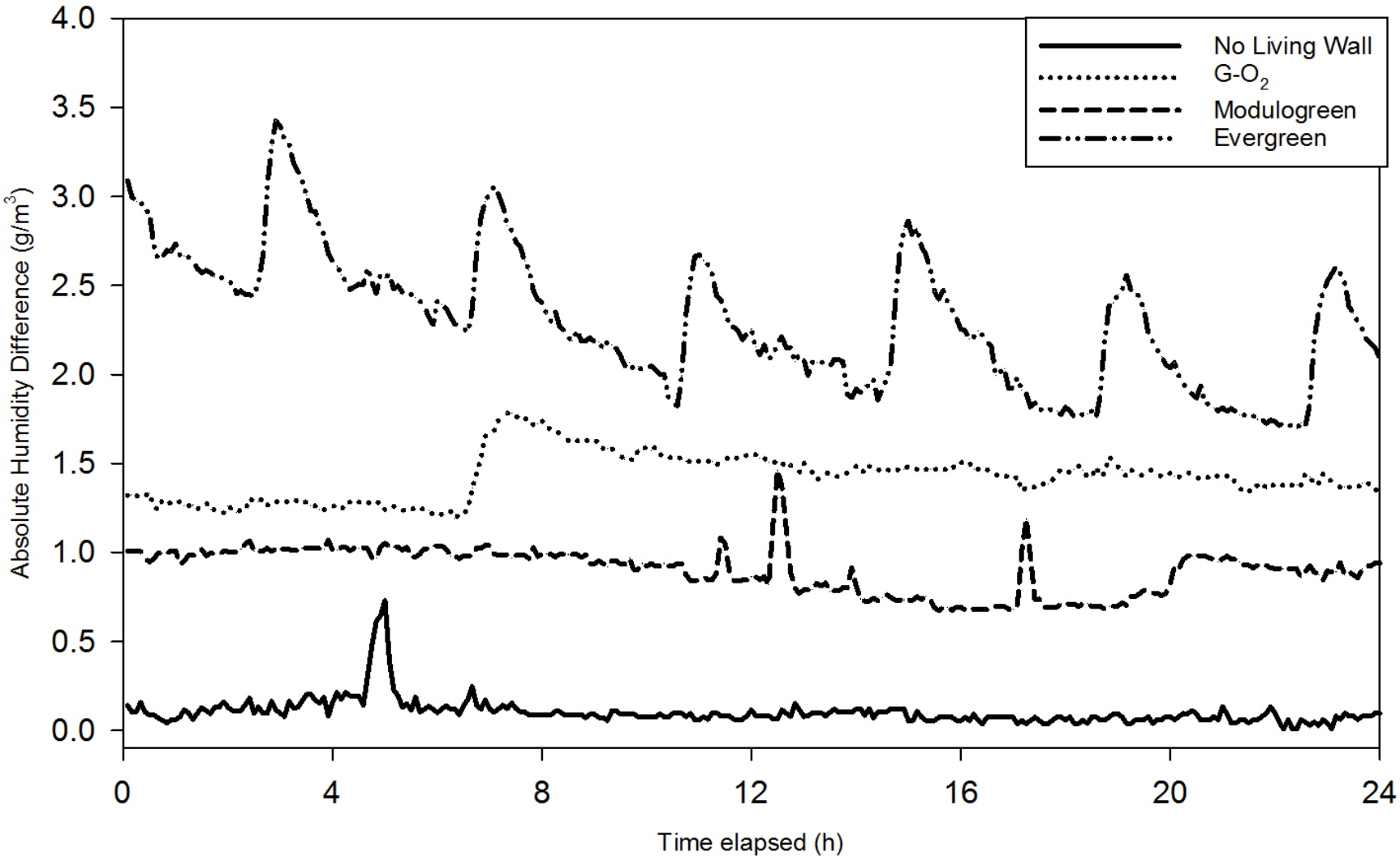

Figure 3.8: Time series plot showing the increase in absolute humidity over a 24-hour period due to the presence of a living wall.

3.5 Bacteria

Concentration of total bacteria were significantly higher in CAE (median = 3.93 times relative to the outdoor) than the RB (median = 0.89 times relative to the outdoor) ($p < 0.05$). It suggests suggests that either there were source(s) of bacteria within CAE, or more adequate ventilation in the RB. The median ratio of concentration of total bacteria in the test chamber and CAE is 0.25, which is likely due to the absence of other organisms shedding bacteria in the test chamber. Despite this difference, there was no significant difference in the TC/CAE for total bacteria for samples taken in the test chamber with and without a living wall ($H = 1.39$, $df = 3$, $p > 0.05$).

There were wide variations in the similarity of bacterial community of samples taken at the same time between 1) the test chamber and the CAE, 2) the CAE and outdoor, and 3) RB and outdoor (Table 3.6). No Gram-negative cocci were detected in any of the samples. Few samples detected Gram-negative rods (26 out of 272), and fewer samples taken in the test chamber (5 out of 68) and the CAE (7 out of 68) detected Gram-negative rods.

Table 3.5: Relative concentration of bacteria and fungi in the test chamber, the Centre for Architectural Ecology, and the reference building

	Median Ratio of Bacteria	Mann-Whitney's U* (p-value)	Median Ratio of Fungi	Mann-Whitney's U* (p-value)
CAE/outdoor	3.94	1012 (7.7×10^{-9})	0.89	431 (3.5×10^{-16})
RB/outdoor	0.89		0.29	
TC/CAE	0.25	1077.5 (6.4×10^{-8})	0.42	1724 (0.01)

TC = Test chamber, CAE = Centre for Architectural Ecology, RB = Reference Building.
*Comparison between the two ratios that the value is adjacent to the left. The lower U (which is used to determine significance) is reported.

Based on the median and the interquartile range of the Spearman's rho (ρ) of the samples, there did not appear to be any strong influence from any of the sampling location upon the similarity of bacterial community in the area. The wide interquartile range is likely influenced by the low number of groups to rank, and very similar concentration of some classification groups, which leads to the ranks between these groups to not be consistent across all sample comparisons. However, there was difference in the degree of similarity of the bacterial community between samples taken in the TC and CAE with and without a living wall ($H = 11.0$, $df = 3$, $p < 0.05$). Post-hoc Dunn tests revealed significantly dissimilar bacterial community from samples taken when the G-O2 was in the test chamber, and a weaker degree of similarity ($p = 0.068$) when the Evergreen was in the test chamber compared with samples taken without a living wall. This difference may be due to the population of actinomycetes growing in some of the living walls. The ratio of concentration of actinomycetes was significantly higher between CAE and outdoor (median = 3.0) than between the test chamber and The CAE (median = 0.707) and between RB and outdoor (median = 0.707). Despite the

Table 3.6: Diversity of bacterial community sampled in the test chamber and several nearby locations.

Comparison of bacterial community	n	Median Spearman's ρ	25% quartile ρ	75% quartile ρ	Range of ρ
TC and CAE, all samples	68	0.700	0.400	0.837	-0.650 to 1.0
CAE and outdoor, all samples	68	0.750	0.362	0.937	-0.350 to 1.0
RB and outdoor, all samples	68	0.750	0.450	0.937	-0.200 to 1.0
TC and CAE, no living wall	16	0.800^{A}	0.575	0.950	0.150 to 1.0
TC and CAE, Evergreen	16	0.500^{B}	0.100	0.763	-0.650 to 0.95
TC and CAE, G-O_2	18	0.525^{B}	-0.050	0.800	-0.550 to 0.95
TC and CAE, Modulogreen	18	0.800^{AB}	0.400	0.950	-0.150 to 1.0

TC = Test chamber, CAE = Center for Architectural Ecology, RB = Reference Building.
Groups with the same letters are not significantly different from one another ($p < 0.05$)

similar average ratios of actinomycetes found in RB and the test chamber relative to the space around that location, nearly half (30 of 66) of the samples for RB and outdoor did not contain actinomycetes colonies.

The concentration of actinomycetes were higher on average in the CAE than other sample locations (Figure 3.9). There was a difference in TC/CAE of actinomycetes in the presence of a living wall, after removing the outliers due to work on soil during

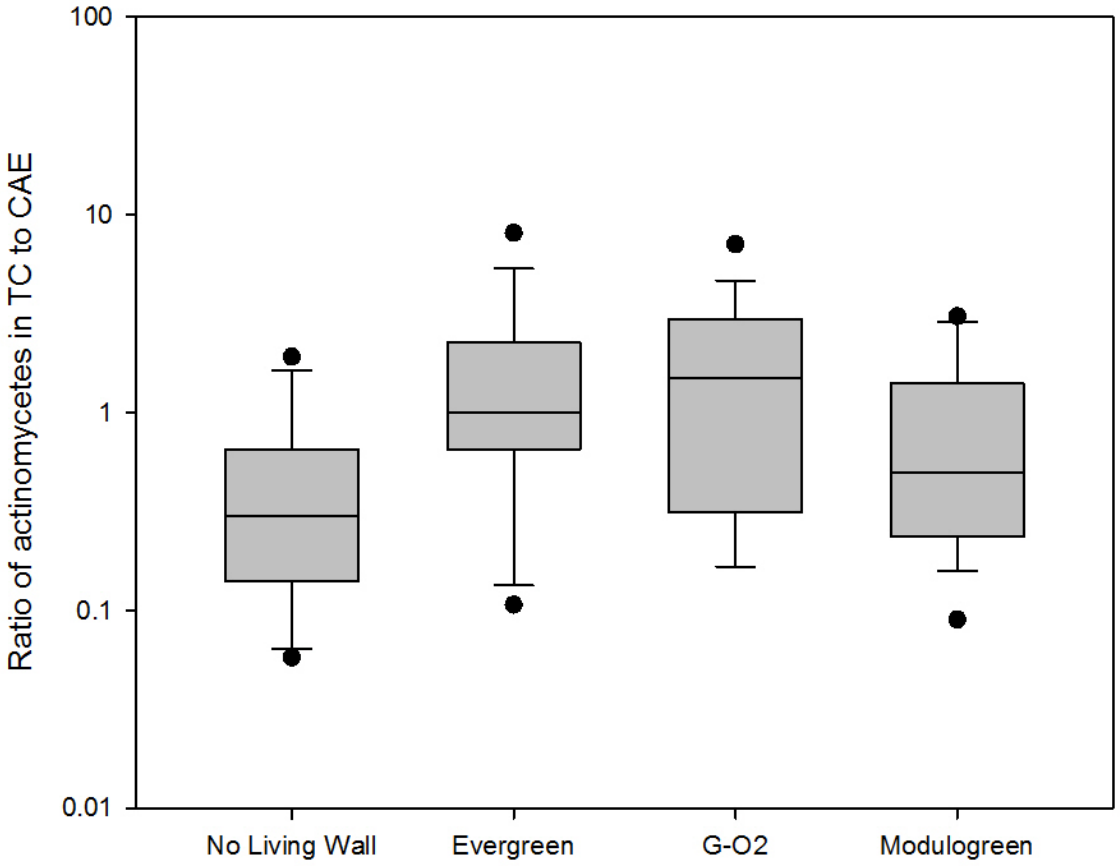

Figure 3.9: Box plot of the TC/CAE of actinomycetes without and with each of the three living wall designs. The line in the middle of the box represents the median. The top and bottom line of the box represent the 75th and 25th percentile. The lines at the end of the whiskers represent the 10th and 90th percentile. The dots in the graph represent outliers. n = 12 for No living wall; n = 16 for Evergreen; n = 18 for G-O_2; n = 17 for Modulogreen.

those days in CAE ($H = 10.26$, $df = 3$, $p < 0.05$). Post-hoc tests found that there was significantly higher TC/CAE of actinomycetes between samples taken in the test chamber with a G-O_2 living wall and samples taken in the test chamber without a living wall. While the difference between samples taken in the test chamber with an Evergreen living wall and those in the test chamber without a living wall did not reach significance ($p = 0.068$), there was a substantial increase in TC/CAE of actinomycetes.

3.6 Fungi

Concentration of total fungi were significantly higher in CAE (median = 0.89 relative to the outdoor) than the RB (median = 0.29 times relative to the outdoor), which suggests there were source(s) of fungi within CAE (Table 3.5). The median ratio of concentration of total bacteria in the test chamber and CAE is 0.42, which suggested that the electrostatic filters are working to reduce the impact of the fungi growing in the CAE, but the concentration of fungi is not as low as it would be in surrounding buildings. This difference was obvious when comparing the concentration of certain fungal classification groups, almost all of which were higher in the CAE than nearby buildings (Figure 3.10).

There was a significant difference in the ratio of all fungi between the TC and CAE ($H = 12.93$, $df = 3$, $p < 0.05$). Post-hoc Dunn's test found the presence of G-O_2, Modulogreen, or Evergreen significantly increased TC/CAE from a median of 0.14 to 0.45-0.48, but the three living wall designs were not significantly different from one another (Figure 3.11). Most of this difference in concentration was due to *Pencillium* spp. (Figure 3.12), which showed the same trend with greater differences. The presence of Evergreen and Modulogreen living wall significantly increased the *Pencillium* spp.

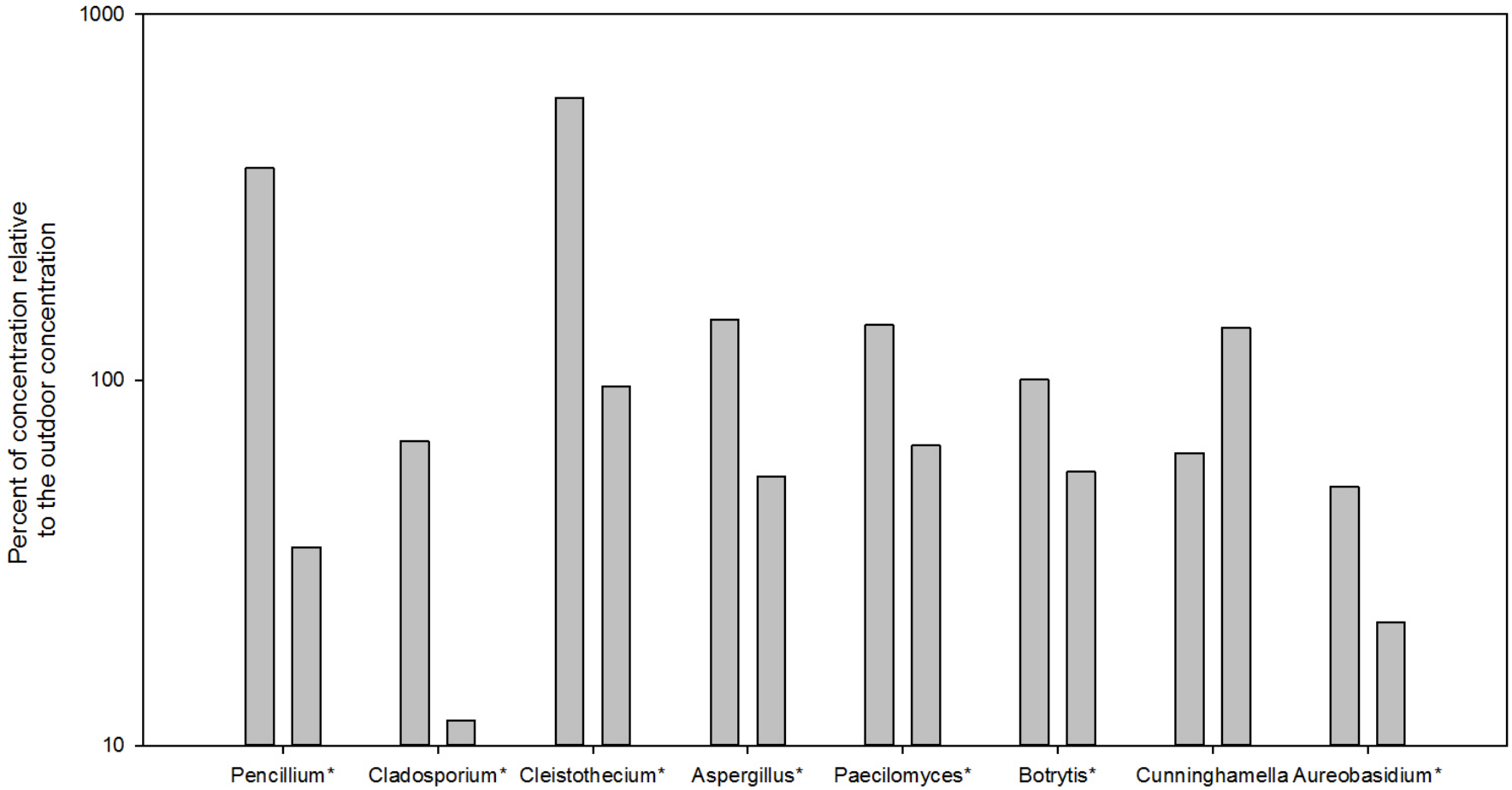

Figure 3.10: Median concentration of various fungal classification groups found in the Centre for Architectural Ecology (left bar) and in the reference building (right bar) relative to the outdoor concentration. All except *Cunninghamella* spp. have significantly different ($p < 0.05$) concentration between the two sample locations based on Mann-Whitney's U test.

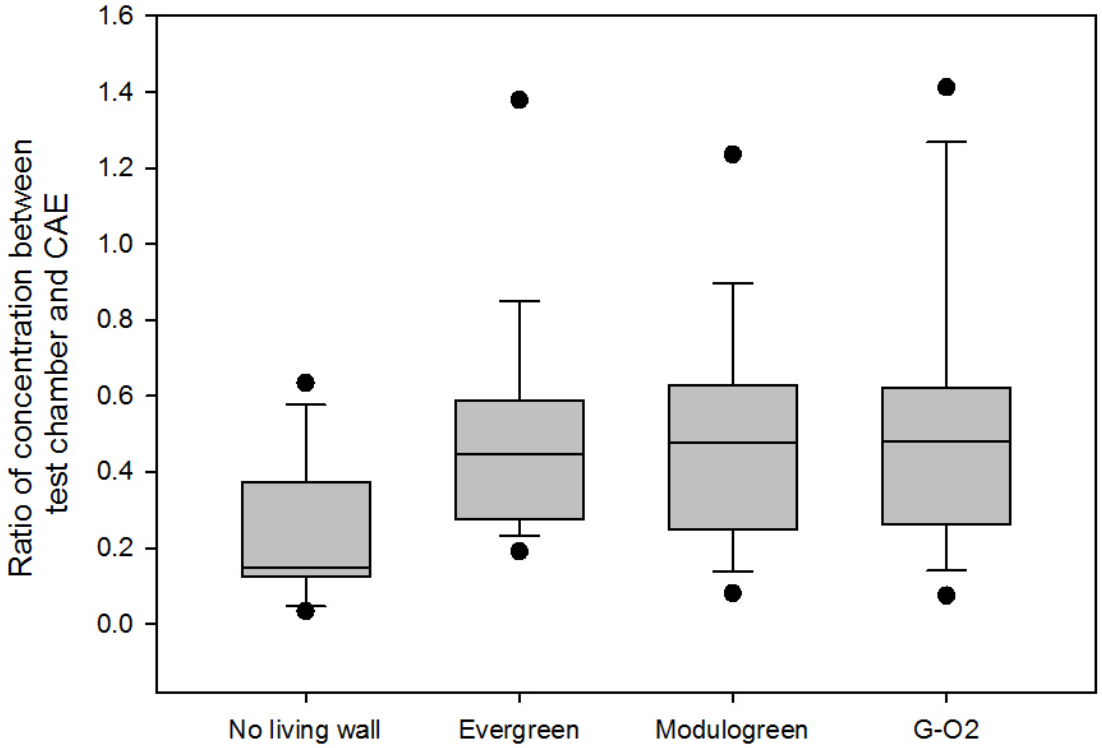

Figure 3.11: Box plots of the TC/CAE of total fungi without and with each of the three living wall designs. The line in the middle of the box represents the median. The top and bottom line of the box represent the 75th and 25th percentile. The lines at the end of the whiskers represent the 10th and 90th percentile. The dots in the graph represent outliers. The living walls contribute significantly more fungi, but the amount does not significantly differ among living wall designs. n = 15 for No living wall; n = 16 for Evergreen; n = 18 for Modulogreen; n = 18 for G-O_2.

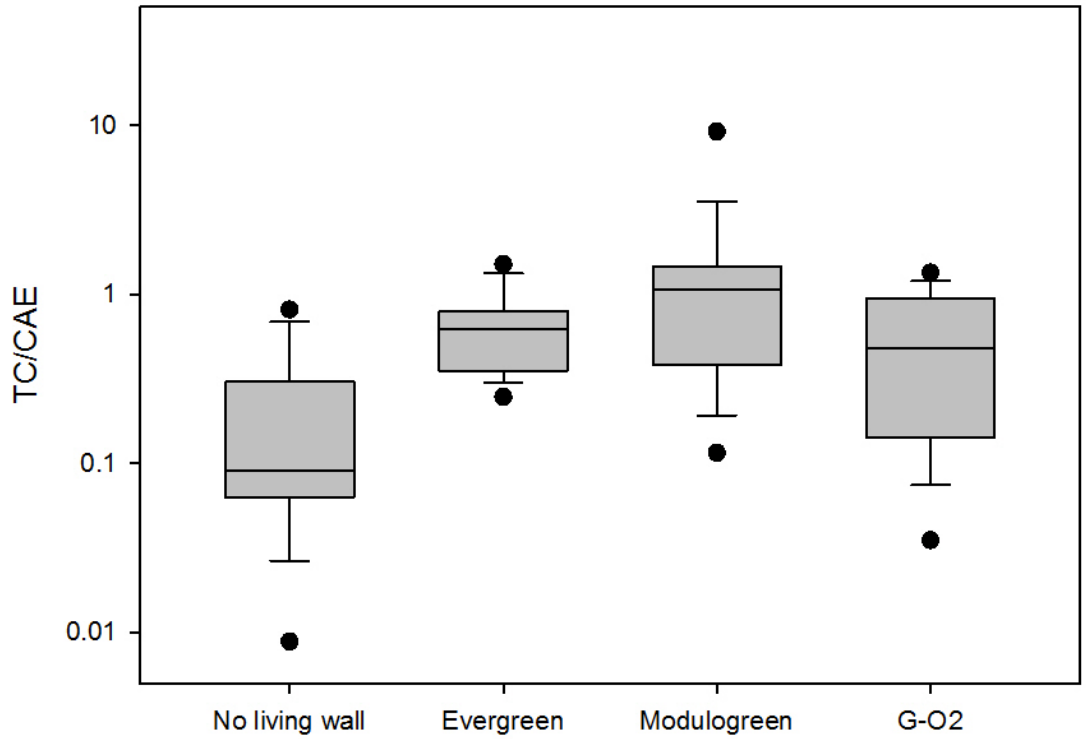

Figure 3.12: Box plots of the TC/CAE of *Pencillium* spp. without and with each of the three living wall designs. The line in the middle of the box represents the median. The top and bottom line of the box represent the 75th and 25th percentile. The lines at the end of the whiskers represent the 10th and 90th percentile. The dots in the graph represent outliers. The living walls contribute significantly more *Penicillium* spp., but the ratios of concentration do not significantly differ among living wall designs. n = 15 for No living wall; n = 16 for Evergreen; n = 18 for Modulogreen; n = 18 for G-O_2.

concentration in the test chamber based on post-hoc Dunn's test, while there was a non-significant increase in the presence of G-O_2 ($p = 0.055$). This difference in concentration was not apparent with the other fungal genera (Figure 3.13).

There were wide variations in the degree of similarity of fungal community from samples taken at the same time between 1) the test chamber and the CAE, 2) the CAE and outdoor, and 3) RB and outdoor (Table 3.7). Based on the median and the interquartile range of the Spearman's rho of the samples, there appeared to be at least one fungal classification group that is predominately found in all of these locations. There is a slight, non-significant difference in the degree of similarity of the diversity of fungal community between the test chamber and the CAE with and without any of the living walls in the test chamber ($p = 0.285$) (Table 3.6). The presence of the any living wall may contribute a different proportion of fungi into the test chamber, which decreases the degree that the CAE shares the same ranked proportion of fungi as the test chamber. Some of the differences may be attributed to the population of *Pencillium* spp. As mentioned earlier, the presence of any of the three living walls increased the population of *Pencillium* spp. in the test chamber.

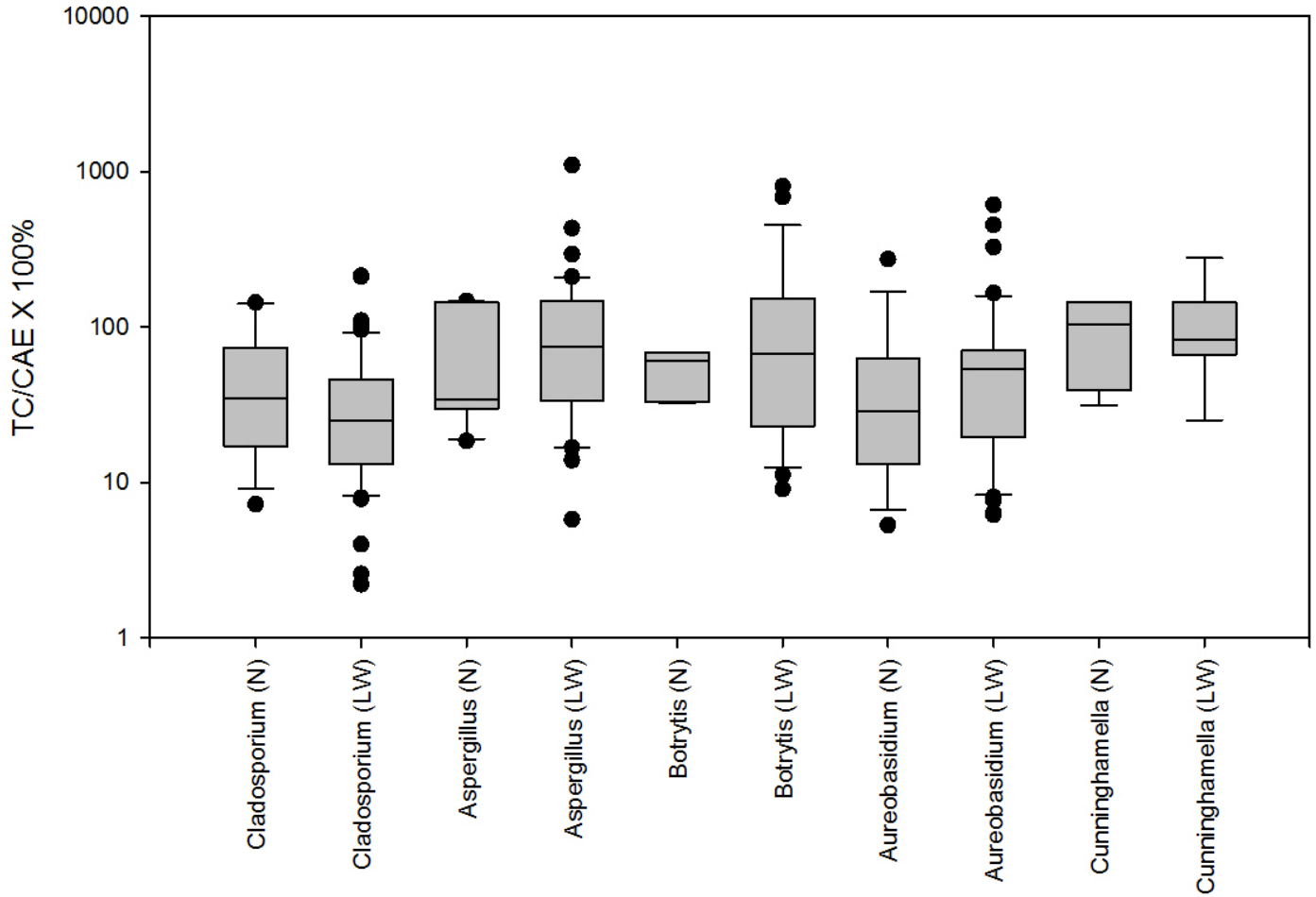

Figure 3.13: Box plots of the TC/CAE of fungal genera without (N) and with (LW) a living wall. The line in the middle of the box represents the median. The top and bottom line of the box represent the 75th and 25th percentile. The lines at the end of the whiskers represent the 10th and 90th percentile. The dots in the graph represent outliers. Refer to Table 3.8 for sample size. There were no significant differences between samples taken with and without a living wall for all groups above based on Mann-Whitney's U test.

Table 3.7: Similarity of fungal community sampled in the test chamber and several nearby locations.

Comparison of fungal community	n	Median Spearman's ρ	25% quartile ρ	75% quartile ρ	Range of ρ
TC and CAE, all samples	67	0.841	0.721	0.903	0.110 to 0.973
CAE and outdoor, all samples	67	0.773	0.657	0.858	0.233 to 0.979
RB and outdoor, all samples	67	0.828	0.686	0.883	0.488 to 0.982
TC and CAE, no living wall*	15	0.869	0.827	0.910	0.454 to 0.926
TC and CAE, Evergreen*	16	0.834	0.708	0.910	0.502 to 0.949
TC and CAE, G-O_2*	18	0.816	0.670	0.872	0.110 to 0.913
TC and CAE, Modulogreen*	18	0.804	0.744	0.890	0.650 to 0.973

TC = Test chamber, CAE = Center for Architectural Ecology, RB = Reference Building.
*not significantly different from one another (H = 3.79, p = 0.285)

This difference may also be attributed to the population of *Paecilomyces* spp. in the living walls. When examining the number of samples detecting at least one colony of certain fungal classification groups, there are generally a greater proportion of samples detecting a specific fungal classification group in the CAE than the test chamber (Table 3.8). Within either the CAE or the test chamber, the proportion of samples detecting at least one colony of a fungal classification group taken without a living wall is similar to those taken with a living wall. This trend holds for all the classification groups with or without a living wall in the test chamber. The exception was *Paecilomyces* spp., which were twice as likely to be detected in the test chamber than the CAE with a living wall, and nearly no difference between the test chamber and the CAE without a living wall in the test chamber.

Table 3.8: Number of samples (and percent of total samples with or without a living wall) detecting colonies of specific fungal classification groups in the test chamber and the Centre for Architectural Ecology (CAE) with and without the living wall in the test chamber.

	Test chamber (n = 67)	CAE (n = 67)
Pencillium spp.		
No living wall (n = 15)	14 (93%)	15 (100%)
With living wall (n = 52)	52 (100%)	52 (100%)
Cladosporium spp.		
No living wall (n = 15)	10 (67%)	13 (86%)
With living wall (n = 52)	44 (85%)	48 (92%)
Aspergillus spp.		
No living wall (n = 15)	6 (40%)	8 (53%)
With living wall (n = 52)	29 (55%)	36 (69%)
Botrytis spp.		
No living wall (n = 15)	0 (0%)	5 (33%)
With living wall (n = 52)	14 (27%)	25 (48%)
Paecilomyces spp.		
No living wall (n = 15)	1 (7%)	0 (0%)
With living wall (n = 52)	28 (53%)	12 (23%)
Cunninghamella spp.		
No living wall (n = 15)	2 (13%)	2 (13%)
With living wall (n = 52)	5 (10%)	5 (10%)
Aureobasidium spp.		
No living wall (n = 15)	5 (33%)	13 (87%)
With living wall (n = 52)	22 (42%)	40 (77%)
Cleistothecium		
No living wall (n = 15)	0 (0%)	0 (0%)
With living wall (n = 52)	27 (51%)	30 (58%)

3.7 Endotoxin

A total of 33 samples had a coefficient of variation below 25%, 11 of which were taken in the test chamber without a living wall (Table 3.9). There was no significant difference in endotoxin concentration in the test chamber with or without any of the three living wall designs (Figure 3.14). The low concentration of endotoxin corresponded to

the absence of Gram-negative bacteria in most of the samples taken in the test chamber and the CAE.

Table 3.9: Endotoxin concentration in the test chamber

	n	Geometric Mean (EU/m^3)	Geometric Standard Deviation
No living wall	11	4.75	5.44
G-O_2	7	1.69	4.3
Modulogreen	8	1.64	3.09
Evergreen	7	2.02	2.09

*No group is significantly different from one another (F = 1.71, p = 0.19)

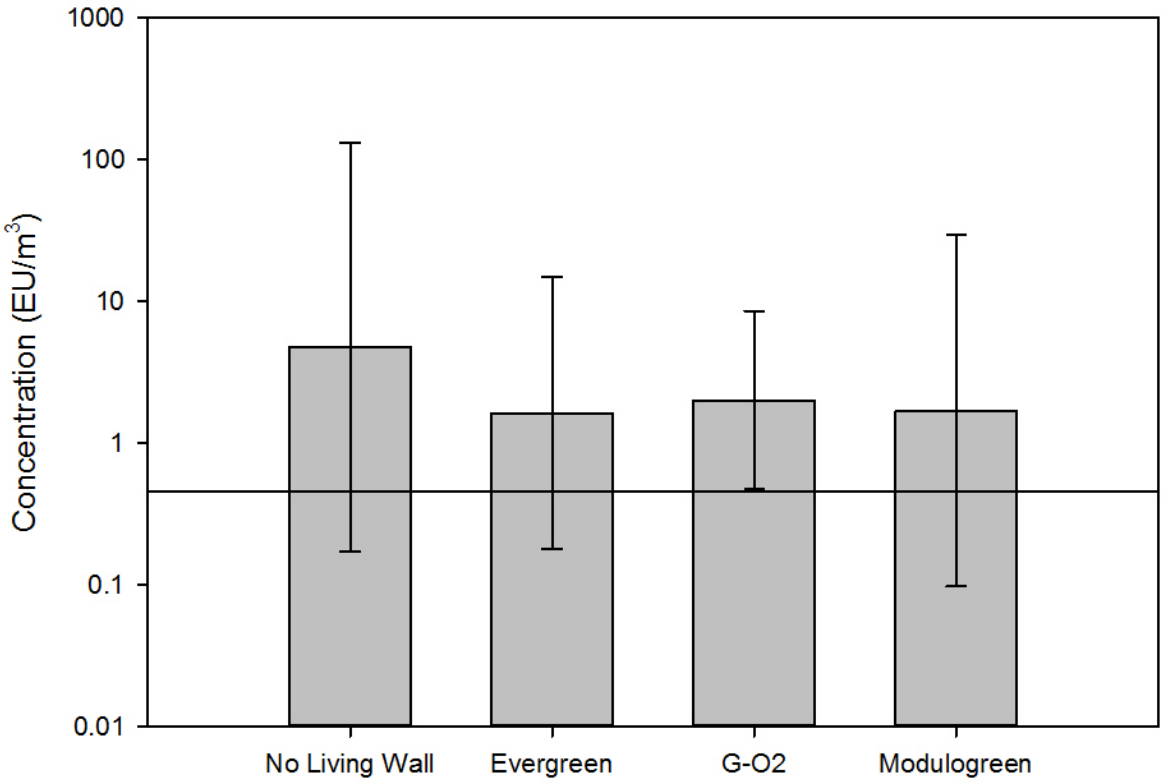

Figure 3.14: Change in endotoxin concentration with or without a living wall. The bars represent the geometric mean concentration, and the error bars denote the 95% confidence interval around that mean. The line across the bar graph represent the limit of quantitation (0.454 EU/m^3). n = 11 for No living wall; n = 7 for Evergreen; n = 7 for G-O_2; n = 8 for Modulogreen.

3.8 Summary of Results

From the results of the study, the following conclusions can be drawn.

It is possible for interior living walls to contribute either to the removal or the release of VOCs, or to both at the same time. Based on measurements taken during steady-state equilibrium of VOCs, Modulogreen removed a significant portion of 2-butanone, had no effect on toluene, and added a significant amount of α-pinene.

Interior living walls can remove 90 ppm of the 250 ppm CO_2 added in the test chamber with adequate lighting for shade-tolerant plants (40-70 μmol/m^2/s of PPFD). Despite the differences in the size of the living walls, there were no trends found among the size parameters and CO_2 reduction capacity among the living wall designs.

Interior living walls can humidify a space. This is related to the amount of irrigation. G-O_2 and Modulogreen significantly humidify into the test chamber by 1.4 g/m^3, and Evergreen significantly humidify the test chamber more than the other two living wall design by 2.5 g/m^3.

Interior living walls have the potential to increase bioaerosols. The living walls significantly increased the median concentration of fungi in the test chamber by 30-34% relative to the concentration of fungi in the CAE. This is mostly driven by *Pencillium* spp., and partly by actinomycetes, and *Paecilomyces* spp. The change in proportion of microorganisms by classification groups indicates that there is a source of microorganisms when the living wall is present.

In the present study, the interior living walls do not appear to increase endotoxin concentration.

4 Discussion

Since the removal of VOCs relates to many of the other IAQ factors, it will be discussed last among IAQ factors.

4.1 Living Walls as Carbon Dioxide Control

In the present study, interior living walls were able to remove one-third of the CO_2 added into the test chamber. The results are similar to Irga et al. (2013). They examined the removal of CO_2 by plants (grown in potting mix or in hydroculture) in a sealed test chamber with 1000 ppm of CO_2 at dim lighting (10 μmol/m^2/s of PPFD) and strong indoor lighting (350 μmol/m^2/s of PPFD) for 40 minutes. In dim lighting, there was an average of 27% reduction in CO_2 by plants grown in hydroculture, and no significant change by plants grown in potting mix. In strong indoor lighting, there was a more pronounced reduction in CO_2, with an average of 61% and 37% respectively. Unlike Irga et al. (2013), Evergreen, the non-soil based living wall did not generate greater CO_2 reduction than the G-O_2 and Modulogreen, which contain at least 12 kg more soil (or more than 50 times the amount of soil).

A recent study by Tudiwer and Korjenic (2017) examined the rate of CO_2 removal by living walls by comparing the change in CO_2 concentration from an initial concentration of 1900 ppm in two classrooms (in Austria) that were not well sealed, one of which had a living wall. Out of four tests, the rate of CO_2 drop in the classroom with the living wall was an average of 3.4% faster than the classroom without living walls, though the change was not significant. The study attributed the non-significant effect to the small amount of living wall in the room (1% of the room volume), which the present study has shown can have a significant reduction on CO_2 with similar relative volume to

the room (0.2-0.6%). It is also possible that the rate of CO_2 removal by living walls do not have as much impact when there are air infiltration through gaps in the building envelope and ventilation.

The correlation of various size parameters of the living walls and removal rate of CO_2 did not show a significant relationship, and did not show a trend that is affected by the size of the living wall. The lack of correlation may be due to the narrow range of size parameters examined. It is possible that a wider range (e.g.: area of living wall between 0.4 m^2 to 4 m^2) may show a stronger relationship.

Despite the significant reduction in the present study, the removal capability of the living wall does not compare to that of a HVAC system in removing pollutants within the building. A ventilation system ideally brings in outside air at ambient outdoor CO_2 concentration, which would eventually dilute the concentration of pollutants to that of ambient outdoor concentration. All three living wall designs in this study had reduced the added CO_2 by 30-40%, which may be removed by a ventilation system. It is not certain how many living walls (or area of living wall installed) is required to remove all of the CO_2.

4.2 Living Walls as Source of Humidity

The results of the study showed increased AH in the test chamber with a living wall in the test chamber. The increased humidity is related to the amount of irrigation. While the change is more pronounced during irrigation, the AH was higher throughout the whole period with the living wall in the test chamber.

ASHRAE recommends a range of temperature between 20-25 °C for comfort. An AH of 1 g/m^3 of water vapour is around 5.8% at 20 °C and 4.4% at 25 °C (and 1 atm).

Based on the results of the study, the G-O_2 and Modulogreen living wall would add a minimum of 1-2% of RH, and an average of 5-7.5% of RH, while the Evergreen living wall would add a minimum of 6.6-8.7% of RH, and an average of 11.4-15% of RH. Tudiwer and Korjenic (2017) found that the classroom with a living wall increased the range of RH throughout the year (25-66%) when compared with a similar classroom without a living wall (21-57%). The range of RH increase (4-9%) is slightly higher than what the G-O_2 and Modulogreen produces. This likely reflects the size of the living wall in that classroom (5.488 m^2), which is much larger than a single panel of G-O_2 (0.372 m^2) or Modulogreen (0.799 m^2).

On the other hand, studies using potted plants do not exhibit much of the humidifying effect. Torpy et al. (2013) examined the RH contributed by two plant species (*S. wallisii* and *D. deremensis* 'Janet Craig') in one or three large self-watering pot plants (20 cm diameter for *S. wallisii* or 30 cm diameter for *D. deremensis* 'Janet Craig') in several office rooms, for which the water was replenished weekly (the amount of which was not mentioned). They found a non-significant change in RH with pot plants in the office compared with the offices with no pot plants. In contrast, Lohr and Pearson-Mims (1996) measured the temperature and RH difference between an office space with and without plants that were watered by capillary action. Out of a total of 22 measurements, they found a significant increase of 0.8% RH with plants (without a significant change in temperature). While significant, 0.8% RH is not likely to make a substantial change to the environment. It is possible that the low water requirements for potted plants contributed little to the humidity of the office rooms.

Since RH is affected by irrigation event and the transpiration of the plants, increasing the size of the living wall would increase the RH added into a room. In a high RH environment, the increased RH due to a living wall may promote the growth of fungi within the building. In a low RH environment, the increased RH due to a living wall may create a more comfortable environment. Greater control of ventilation is required to remove the excess humidity in the room containing a living wall.

4.3 Living Walls as Source of Bioaerosols

In this study, the living wall contributed to increased concentrations of actinomycetes, *Pencillium* spp., and *Paecilomyces* spp. in the test chamber and acted as a reservoir for microorganisms. This likely occurred because these microorganisms were introduced by the growing environment. Actinomycetes were first detected from samples taken when the bags of potting soil were placed in the living walls (and during other planting activities in the CAE). *Pencillium* spp. were detected in very high concentrations within the CAE from samples taken before the study period. The presence of cleistothecium and *Paecilomyces* spp. were detected frequently in samples half way into the study. It is likely that the environmental conditions in the living wall favoured the growth of these microorganisms.

Tudiwer & Korjenic (2017) took one fungal sample in each of the four locations: the classroom with a living wall, the classroom without a living wall, the outdoor courtyard of the school, and the street. The samples showed that both classrooms had a concentration of fungi roughly 15% of the concentration outdoors. The predominate fungal genus were *Pencillium* spp. and *Cladosporium* spp., which are typical outdoor fungi.

Although the increase in the concentration of certain fungi and bacteria was significant, the amount added on average did not exceed the concentrations found in the air entering the test chamber (i.e. none of the living walls had a median TC/CAE of total fungi greater than 1). None of the species found in excess in the test chamber were associated with opportunistic infections (Flannigan, Samson & Miller, 2011; Gravesen, Frisund, & Samson, 1994; Malloch, 1981; St-Germain & Summerbell, 1996). The three groups of microorganisms found to be significantly increased by the presence of the living wall are primarily microbes that thrive in soil. This suggests that the microbial community in the living wall can be controlled and maintained such that it would not promote the growth of harmful microorganisms.

Torpy et al. (2013) placed one to three potted plants with two plant species (*Dracaena deremensis* 'Janet Craig' and *Spathiphyllum wallisii* 'Petite') in 44 offices from two buildings with no dampness or mould issues to determine the change in airborne fungal concentration caused by a potted plant when compared with 11 offices with no plants. There were minor non-significant increases in airborne fungi among all offices, from 61.48 CFU/m^3 in offices without plants to 72.94-126.11 CFU/m^3 in offices with plants during fall (April to June), and from 52.26 CFU/m^3 in offices without plants to 47.76 to 96.41 CFU/m^3 in offices with plants during spring (September to October). Similar to the present study, there were higher average concentration of *Penicillium* spp., *Cladosporium* spp., *Scopulariopsis* spp., *Scytalidium* spp., *Acremonium* spp. (only in offices with *Dracaena deremensis*), *Aspergillus* spp., and *Epicoccum* spp. in offices with plants during fall, and *Scytalidium* spp., *Scopulariopsis* spp., *Acremonium* spp., *Cladophialophora* spp. and sterile mycelia during spring (uncertain of statistical

significance). There was also lower average concentration of Malbranchea spp. in offices with plants during spring (uncertain of statistical significance). While the concentration of most of the classification groups are also found outdoors, it is possible that the potted plants are able to maintain the fungal community, since these offices have a HVAC system.

While these microorganisms are not harmful per se, some individuals may be allergic to them and to the metabolites they produce. As a precaution, the living wall should be placed in locations where the airflow moves away from the occupants and towards the air exhaust of the room.

4.4 Living Wall as Volatile Organic Compounds Control

In this study, several VOCs were added into a test chamber to evaluate the removal effectiveness of a living wall. The results were mixed.

Toluene

In this study, toluene was not removed by the living wall, despite many studies having positive results (De Kempeneeer, 2004; Husti et al., 2016; Orwell et al., 2006; Yoo et al., 2006). This may be due to several reasons. One, the concentration was very low, which did not lead the plants and microorganisms to begin metabolism which remove toluene. Wood et al., (2006) examined the effect of indoor potted plants on TVOCs in three office buildings (and not in a laboratory chamber) using a photoionization detector. Significant reduction of TVOC concentration was found in offices with plants when TVOC concentration were above 200 ppb in the offices without plants for that week. On weeks when the TVOC concentration was below 100 ppb in

offices without plants, there was no visible change to TVOC concentration. There was non-significant TVOC concentration reduction between 100 and 200 ppb. The same may be occurring with the living walls in the test chamber, where the concentration ranged from 23.4 to 35.5 $\mu g/m^3$.

The previous studies examining the removal of toluene were working with higher concentrations. De Kempeneer (2004), Orwell et al (2006), and Yoo et al. (2006) were using doses ranging from 0.2 ppm to 90 ppm (0.75-340 mg/m^3). Husti et al. (2016) examined the toluene reduction before and three months after placing a plant in an office for two months, which dropped from 14.97 to 11.5 $\mu g/m^3$, both start and endpoint are below the limit of detection of the living wall samples in the CAE.

The absence of toluene change is not unique to the present study. Kim et al. (2011) and Lim et al. (2009) found that toluene either did not change or increased during their study period in newly constructed homes with plants. They attributed the additional toluene to 1) the sources of toluene in the building materials, 2) lack of ventilation (with closed windows during winter), and 3) lack of plant maintenance near the end of the study period, when interest in the study waned. All of these points are likely important in the maintenance of a living wall, and can aid the living wall in improving IAQ.

Two, the microorganisms that remove toluene are not normally favoured by the plants that were grown on the living walls in this study. Wolverton and Wolverton (1993) have noted that the presence of Gram-negative rods, such as *Pseudomonas* spp., are effective in the removal of organic pollutants. Similarly, de Kempeneer et al. (2004) introduced *P. putida* to boost the removal of toluene. In this study, there was very few samples with Gram-negative rods, and very low concentrations of endotoxins detected.

It is likely that the living wall in this study did not remove toluene due to the absence of Gram-negative rods. In order to remove certain VOCs, it may be necessary to introduce the necessary microorganisms, and foster their growth in the substrate or upon the plants with the appropriate plant species.

2-butanone

In the present study, the estimated removal rate of 2-butanone was 81 μg/h/m^2 of living wall when constantly exposed to 31 μg/m^3 of 2-butanone. Tani and Hewitt (2009) found that the removal rate was 50-65 μg/h/m^2 of leaf of the original concentration when constantly exposed to 117.8-1178 μg/m^3 of 2-butanone in a sealed chamber. Despite a lower concentration exposed to the living wall, the living wall was able to remove 2-butanone at a similar rate. Unlike toluene, it appears that the living wall in the present study can readily take up 2-butanone.

Many of the VOCs studies were carried out with plant leaves or shoots to evaluate their removal effectiveness of aldehydes and ketones (those with a carbon bonded to an oxygen molecule with two pairs of electron) (Kim et al., 2008; Orwell et al., 2004; Schmitz, Hilger, & Weidner, 2000; Tani & Hewitt, 2009; Yoo et al., 2006). For those studies that changed the lighting scheme upon the plants, there was little uptake of aldehydes and ketones by the leaves or shoots. When the leaves or shoots are in the dark, there is almost no stomatal conductance, which measures the amount of activity by the leaves to open stomata (Kim et al., 2008; Tani & Hewitt, 2009). The uptake of formaldehyde during dark periods are negligible (see Table 1.1 for results from Kim et al., 2008) Similar results can be found from time-series plots by Tani & Hewitt (2009), which show negligible difference in concentration of an aldehyde between the inlet that

supplied the aldehyde and the outlet that draws air out of the test chamber. A similar trend can be seen with ketones. This suggest that lighting plays a role in the uptake of aldehydes and ketones that may be metabolized by the plant.

α-pinene

In this study, the concentration of α-pinene increased in the test chamber containing a living wall. It is likely that the plants in the living wall produced α-pinene in response to some environmental stimuli, leading to a higher concentration and a larger standard deviation among the samples taken in the test chamber with a living wall than samples taken in the test chamber without a living wall.

It may be inferred that VOCs produced by the plants and the microorganisms in the living wall would be added to the indoor environment. The effects of VOCs from plants and microorganisms remain uncertain. Given the wide range of VOCs that may be produced by microorganisms, some of these VOCs may be undesirable, especially the musty odour associated with fungi. On the other hand, α-pinene belongs to a group of VOCs called terpenes, which have been touted to have possible health benefits (Cho et al., 2017; Li et al., 2007; Mao et al., 2012). These compounds can be found in air collected in a forest, and the presence of interior living walls may potentially provide the same benefit.

The mixed results in this study were similar to the TVOC measurements in Wood et al. (2006). The TVOC in the test chamber without a living wall was on average 578 $\mu g/m^3$ (Table 2.1), but increased to an average of 900 $\mu g/m^3$ with Modulogreen in the test chamber. It is likely that the lack of VOC reduction at low concentration is due to the potted plants and the living walls' metabolism.

4.5 Living Wall on the Improvement of IAQ

Overall, the living wall may be able to improve certain aspects of indoor air quality, but there are many considerations in order to reap this benefit.

Ventilation.

In order for the living wall to remove pollutants, ventilation may be needed to mix the air within a room such that the living wall may affect all the air. In this study, the test chamber had fair mixing (an average mixing factor of 3.5), which was achieved by the standing fan blowing air in a circular pattern around the test chamber. In many rooms, there are spots where the air remains stagnant due to the location of the air inlet and air exhaust within that room. Placing a living wall in these spaces would reduce the air available to the living wall, thus reducing its effectiveness.

Despite the pollutant removal capacity demonstrated in this study, the living wall can produce excess humidity, and possibly promote the growth of microorganisms to which some occupants may be allergic. As a precaution, the living wall should be placed downstream of the air movement within the room to mitigate these issues (ACGIH, 2013). When placed downstream of the occupants, the pollutants would not be carried to the occupants. In order to make use of the living wall's pollutant removal capacity, the air in that room would need to be recirculated through a HVAC system, which would filter out most bioaerosols and adjust the relative humidity as required.

Plant Selection

Since not all plants have the same pollutant removal capacity or promote the growth of microorganisms that can remove pollutants effectively (Wolverton &

Wolverton, 1993), choosing the appropriate plants that can improve IAQ and survive in the indoor environment may limit the choices available. While there are many plant species under research (see Table 1.1 for all the plant species), many of these plants are more suited to large pots, and living walls are not ideal places to grow them. Two of the plant species used in this study have been well recommended for living walls: *Epipremnum aureum*, and *Chlorophytum comosum*. One additional consideration when choosing plants for interior living walls is to find plants that do not produce flowers, and thus pollen, which can be unpleasant for those who are allergic.

Lighting

Prior research have shown that the stomata plays an important role in the reduction of CO_2 and some VOCs (Irga et al, 2013; Kim et al., 2008). Since plants can uptake gaseous pollutants through the stomata, and light induces the opening of stomata, lighting is important not only to the survival of the living wall, but also to its pollutant removal capacity.

If the living wall is placed in an area without daylighting, artificial lighting would have to be provided. The lighting is especially important for CO_2 removal when occupancy increases near the living wall and CO_2 concentration subsequently rise. This lighting may not be appropriate in all settings, where the glare from the light can become distracting to activities.

Maintenance

Proper maintenance of the living wall can foster the appropriate microbial community in the living wall. This ensures that beneficial microorganisms (e.g.: those

that remove VOCs) thrive, and detrimental microorganisms (e.g.: those that produce bioaerosols that may lead to allergic symptoms) do not thrive. In order for the living wall to remove certain VOCs, one would need to acquire the appropriate microorganism and keep them alive on the living wall. This may involve regular innoculation with those microorganisms, or introduction of small doses of the VOCs to promote their growth, potentially making the management of the wall onerous.

4.6 Strengths and Limitations

The present study was able to show, with some confidence, that there are both beneficial and possibly detrimental impacts from an interior living wall. However, with appropriate control and maintenance, the detrimental impacts can be mitigated and the beneficial impacts enhanced. The preliminary evaluation of these IAQ factors will aid in the future building designs that incorporate interior living walls.

While the present study was able to evaluate many of the IAQ factors that may be affected by a living wall, there are a few gaps that this study could not address.

Size

Given the volume of the test chamber, it was not possible to install more than one living wall with appropriate lighting and irrigation. Despite the many size characteristics examined, this study did not find any of the size characteristics to be associated with the impacts upon IAQ factors between the living wall designs, except for RH, which was mostly due to the amount of irrigation. As mentioned earlier, it is likely that the range of size parameters examined was too narrow, and did not show a difference. Future studies can plant living walls of various sizes (for each of those parameter) with a

greater range of difference to see whether and which size parameter(s) are most correlated to the impact of various IAQ factors.

VOC

The present study looked at three VOCs with different chemical groups to examine how the living wall interacted with them. These are only a few of the many VOCs that are used by humans. An evaluation of the effectiveness of various plant species to remove other VOCs that are commonly found within residential and commercial buildings may provide a clearer view on whether plants can effectively remove the VOCs that are of concern to human health and comfort.

The study was not able to examine whether non-soil based living walls would remove less VOCs than soil based living walls. Future study can show whether the presence of soil (or a substrate that fosters the growth of microbial community in the living wall) increases the impact of the living wall on the removal of VOCs.

Carbon Dioxide

Since the living wall designs did not differ significantly in the reduction of CO_2, it is unknown whether increasing the size of living walls in a room would increase this effect, and at what size of the living wall would lead to a plateau in the reduction of CO_2.

Endotoxin

This study did not find a significant difference in the concentration of inhalable endotoxin due to the presence of the living wall. It is likely that there were few Gram-negative rod in the living walls. If the living wall supported a greater proportion of Gram-negative rod, it is not certain whether there would be more endotoxin in the test

chamber. This is especially important for living walls placed in buildings whose occupants may be more susceptible to the effects of endotoxin.

4.7 Conclusion

Despite the few studies that examined the full range of factors affecting IAQ, interior living walls have the potential to both enhance and reduce IAQ when placed in building environments. Interior living walls can reduce some VOCs and add other VOCs, remove some CO_2, add RH, and add specific microbial populations favoured by the living walls in the space which they occupy, which all interact in some way with environmental factors and the plant species in the living walls. All of these factors can be controlled through dilution ventilation and careful consideration of the environmental factors that allow the living wall to thrive. Given the findings in the present study, future research can focus on aspects that can inform the public on the overall IAQ impact of living walls.

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Appendices

Appendix A: Adjusting CO_2 Concentration Data from Q-trak

The bump test results with zero-gas (0 ppm) and 1000 ppm after calibration would show what the Q-trak records at those concentrations. The adjustment can be done by determining the equation based on the difference between bump test results and the Q-trak measurement as follows:

Let X be the CO_2 concentration displayed by the Q-trak.

Let Y be the corrected CO_2 concentration based on the bump tests data point.

Let m be the slope of the line, and b be the y-intercept of the linear equation in slope-intercept form.

Table A.1: Variables in the equation for adjusting CO_2 concentration data from Q-trak.

Bump test gas concentration	0 ppm	1000 ppm
Q-trak measurement	X_1	X_2
"Actual" measurement	Y_1 = 0 ppm	Y_2 = 1000 ppm

First, solve for the slope m.

$$m = (Y_2 – Y_1) / (X_2 – X_1)$$

$$m = 1000 \text{ ppm} / (X_2 – X_1)$$

Then solve for the y-intercept b in the linear equation using the slope m.

$$Y = mX + b$$

$$Y_1 = m \cdot X_1 + b \text{ OR } Y_2 = m \cdot X_2 + b$$

$$b = Y_1 – m \cdot X_1 \text{ OR } Y_2 – m \cdot X_2$$

Then substitute m and b into the linear equation and it can be used to adjust for the concentration.

Here's an example:

If X_1 = 20 ppm, X_2 = 980 ppm, then

$$m = (1000 – 0) \text{ ppm} / (980 – 20 \text{ ppm})$$

$$m = 1.042$$

$$b = 1000 \text{ ppm} – 1.042 \cdot 980 \text{ ppm}$$

$$b = -20.83 \text{ ppm}$$

$$Y = 1.042 X – 20.83 \text{ ppm}$$

If X = 750 ppm, then

$$Y = 1.042 (750 \text{ ppm}) – 20.83 \text{ ppm}$$

$$Y = 802.1 \text{ ppm}$$

Appendix B: Dilution Ventilation Accounting for Living Wall as a Source/Sink of Volatile Organic Compound and Carbon Dioxide

Estimates of the removal rate of each VOC and CO_2 within a test chamber by each living wall were calculated using the dilution ventilation model.

Let X be the removal rate of a gaseous compound.

Let C_X be the mean concentration of a gaseous compound in the room with a living wall.

Recall Equation [1] from Section 1.3 referring to carbon dioxide:

$$V\,dC = G\,dt - Q' \cdot C\,dt$$

Due to the removal capacity of the living wall, we introduce a term to represent the removal rate by the bolded component in the following equation:

$$V\,dC_X = G\,dt - Q' \cdot C_X\,dt \mathbf{- X \cdot C_X\,dt}$$

$$G\,dt = Q' \cdot C_X\,dt + X \cdot C_X\,dt, \qquad \text{if } dC_x = 0$$

$$\int G\,dt = \int (Q' \cdot C_X + X \cdot C_X)\,dt$$

$$G\,(t_2 - t_1) = C_X\,(Q' + X)\,(t_2 - t_1)$$

$$G = C_X\,(Q' + X) \qquad [4]$$

$$G = C_X \cdot Q' \qquad [3]$$

Substituting Equation [3] and Equation [4] based on G would yield

$$C \cdot Q' = C_X\,(Q' + X)$$

$$X = (C - C_X)\,(Q' / C_X)$$

The molar mass and molar volume of the compound is used to convert the volume of the gas, since the removal rate of a gaseous compound were reported in

mass per time in the literature. A living wall's removal rate of CO_2, butanone, toluene, and α-pinene was calculated based on the following equation:

$$X = \frac{Q' \times MM \times (C - C_X)}{C_X \times MV}$$

where C is the mean concentration of a gaseous compound in the room without a living wall,

MM is the molar mass of the gaseous compound, and

MV is the molar volume of the gaseous compound.