UNIVERSITY OF BIRMINGHAM

IMPACT OF PLANTS ON INDOOR AIR QUALITY

by

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Abstract

An indoor environment that does not detrimentally affect our health and is comfortable to spend time in should be considered a fundamental human right. Previous studies have shown that various species of indoor plants are able to remove pollutants, including those shown to be both prevalent and harmful in indoor environments and contributors to poor Indoor Air Quality (IAQ).

However, a lack of consensus exists on whether plants are able to improve IAQ in real dynamic environments with complex and everchanging ventilation, indoor sources and occupant numbers. Additionally, the processes underlying pollutant removal in plants are evidently equally complex, with numerous environmental parameters and likely physiological traits influencing species' removal ability. In terms of Indoor Environmental Quality (IEQ), relative humidity regulation and control are vital for both occupant comfort and reductions in disease transmission – the latter ever more relevant with the current pandemic. However, humidity control indoors through mechanical systems is energy intensive, thus, exploration of plants as a passive technique is worthwhile.

The focus of this study was to investigate a representative range of houseplants – with differing metabolisms, leaf types, and sizes – for their potential to improve indoor environments through pollutant removal (namely, CO_2 and NO_2) and relative humidity regulation under differing environmental conditions and experimental scales. Alongside this, the study looked to address some of the inherent issues with plant-pollutant removal experiments in literature, namely, pollutants tested at much higher concentrations than what is typically measured in indoor environments.

For CO_2 removal, both studied experimental scales (leaf and chamber scale) drew the conclusions that for measurable removal supplementary lighting is required (at ~ 22 200 lux), and to elicit room scale concentration changes the number and density of plants offered by a green wall is necessary. At typical indoor light levels (0 – 500 lux), little potential is offered for CO_2 removal, however, respiration rates were equally found to be negligible in terms of increasing CO_2 concentrations at the room scale. The type of growing media (GM) was found to have a significant influence, with peat GM contributing to a greater reduction of CO_2. Additionally, substrate moisture content (SMC) was deemed to have a negligible effect, especially when removal rates were extrapolated to the room scale.

All studied plant types were able to reduce NO_2 concentrations representative of a polluted urban environment to varying degrees at typical indoor light levels (0 – 500 lux). Few statistical differences were measured between differing environmental factors at the single plant scale namely, GM type, light level, and substrate moisture content. This research suggests that approximately five plants in a small, unventilated office could provide broadly similar health benefits in terms of life years saved, as are estimated to result from clean air policies in urban areas.

As a method for measuring low VOC concentrations to improve the current plant-pollutant experimental methodology, little potential was offered by solid phase micro extraction (SPME) as a technique over what has been previously utilised, the gas-tight syringe.

Moreover, we found that plant species which assimilated the most CO_2 also contributed most to increasing relative humidity (RH) namely, *Hedera helix* and *Spathiphyllum wallisii* 'Verdi'.

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CONTENTS

List of Figures

List of Tables

List of Definitions/abbreviations

ABA	Abscisic acid
AER	Air exchange rate
ALM	Agglomeration of Lausanne-Morges
ANOVA	Analysis of variance
ASHRAE	The American society of heating, refrigeration and air-conditioning engineers
BTEX	Benzene, toluene, ethylbenzene and xylene
CADR	Clean air delivery rate
CO_2	Carbon dioxide
DEFRA	Department for Environment, Food and Rural Affairs
DLI	Daily light integral
ET	Evapo-transpiration
ETLA	Evapo-transpiration per unit leaf area
GC	Gas chromatography
GCMS	Gas chromatography mass spectrometry
GM	Growing media/medium

HPLC	High performance liquid chromatography
HVAC	Heating, ventilation and air conditioning system
I/O Ratio	Indoor-outdoor ratio
IAQ	Indoor air quality
IEQ	Indoor environmental quality
IRGA	Infra-red gas analyser
K_{AW}	Air-water partition coefficient
K_{OA}	Octanol-air partition coefficient
K_{OW}	Octanol-water partition coefficient
LA	Leaf area
LCP	Light compensation point
LOD	Limit of detection
LOQ	Limit of quantification
LSD	Least significant differences
m/z	Mass to charge ratio
MS	Mass spectrometer
NO_2	Nitrogen dioxide

NO_3	Nitrate radical
NO_X	Nitrogen oxides
O_3	Ozone
OEHHA	California Office of Environmental Health Hazard Assessment
OH	Ozone hydroxyl radical
PAHs	Polycyclic aromatic hydrocarbons
PDMS	Poly(dimethylsiloxane)
PM	Particulate Matter
$PM_{0.1}$	$PM_{0.1}$ is the mass concentration of airborne particulate matter with aerodynamic diameter less than 0.1 µm
PM_{10}	PM_{10} is the mass concentration of airborne particulate matter with aerodynamic diameter less than 10 µm
$PM_{2.5}$	$PM_{2.5}$ is the mass concentration of airborne particulate matter with aerodynamic diameter less than 2.5 µm
ppb	Parts per billion
PPM	Uptake or emission of CO_2 by potted plant microcosm
ppm	Parts per million

PTR-MS	Proton-transfer-reaction mass spectrometry
RH	Relative humidity
%RSD	Relative standard deviation
RHS	Royal Horticultural Society
SBS	Sick building syndrome
SEM	Standard error of the mean
SIM	Selected ion monitoring
SMC	Substrate moisture content
SOA	Secondary organic aerosol
SPME	Solid phase micro extraction
SVOCs	Semi-volatile organic compounds
t_R	Retention time
UN	United Nations
USEPA	United States Environmental Protection Agency
VOCs	Volatile organic compounds
VVOCs	Very volatile organic compounds
WHO	World Health Organisation

List of Conversions

NO_2	1 ppb = 1.9125 µg m^{-3} at 20°C, 1013mb (European commission)
Light level	1 µmol m^{-2} s^{-1} = 74 lux (Cool white fluorescent) (Thimijan and Heins, 1983)

List of Publications

1. Gubb, C., Blanusa, T., Griffiths, A. and Pfrang, C. (2018) 'Can houseplants improve indoor air quality by removing CO_2 and increasing relative humidity?', *Air Quality, Atmosphere and Health,* 11(10), pp. 1191-1201. doi: https://doi.org /10.1007/s11869-018-0618-9 — see Appendix D
2. Gubb, C., Blanusa, T., Griffiths, A. and Pfrang, C. (2019) 'Interaction between plant species and substrate type in the removal of CO_2 indoors', *Air Quality, Atmosphere, & Health,* 12(10), pp. 1197-1206. doi: https://doi.org/10.1007/s11869-019-00736-2 — see Appendix E
3. Gubb, C., Blanusa, T., Griffiths, A. and Pfrang, C. (2020) 'Can plants be considered a building service?', *Building Services Engineering Research and Technology*, 41(3), pp. 374–384. doi: https://doi.org/10.1177/0143624419899519 — see Appendix F
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The authors contribution:

1. Curtis Gubb collected and analysed the data. Curtis Gubb wrote the paper. Dr Tijana Blanusa, Prof. Alistair Griffiths and Dr Christian Pfrang reviewed the manuscript. Curtis Gubb edited the paper.
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1.0 Introduction

1.1 What is 'air quality'?

Air quality, outdoor and indoor, is a term used to describe how polluted or clean the air we breathe is, with clean air regarded by the United Nations (UN) as a fundamental human right — not just a policy objective (Boyd, 2019). More than 90% of the global population lives in regions where air pollution exceeds World Health Organisation (WHO) standards, causing a vast impact on public health and in turn the economy, yet, the problem is entirely preventable and created predominately by anthropogenic sources (Boyd, 2019; WHO, 2010; Holgate, 2017).

1.2 Why should we be concerned about air quality?

Globally every minute, a child dies of an illness caused by air pollution whilst ten adults die, prematurely, because of exposure to poor air quality — leading to at least five million deaths annually. Thus, poor air quality causes more deaths annually than the annual number of deaths caused by war, murder, car accidents, plane crashes, malaria, tuberculosis, HIV/AIDS, and Ebola, combined (Boyd, 2019).

In the UK, the impact of air pollution is associated with an excess of 40,000 deaths per year (Holgate, 2017). With the adverse effects ranging across the life course, from impairing fetal growth and lung development in childhood to respiratory or cardiovascular disease, impaired cognition, type 2 diabetes, and cancers in older life. With this, comes an immense economic burden estimated at £20 billion a year annually in the UK just from ambient (outdoor) pollution — not even considering indoor impacts associated with a minimum of 99,000 deaths a year in Europe (Holgate, 2017).

1.3 What causes 'poor' air quality?

Poor air quality is caused by a wide array of airborne pollutants exceeding standards or guidelines set by bodies such as the WHO, Department for Environment, Food and Rural Affairs (DEFRA) and the EU commission. A complex mixture of these pollutants − including particulate matter (PM) and both, organic and inorganic gases − are present in both outdoor and indoor environments (Molhave, 2003).

Organic pollutants are divided by their vapour pressures or boiling points into further sub-groups of very volatile organic compounds (VVOCs) e.g. formaldehyde and acetaldehyde, volatile organic compounds (VOCs) e.g. alpha-pinene and benzene and semi-volatile organic compounds (SVOCs) e.g. naphthalene and polycyclic aromatic hydrocarbons (Molhave, 2003; Weschler and Nazaroff, 2008; Salthammer, 2016). Inorganic pollutants include carbon dioxide (CO_2), nitrogen dioxide (NO_2) and ozone (O_3). PM is classified by size, the most common include $PM_{0.1}$, $PM_{2.5}$ and PM_{10} – defined as PM with a diameter less than or equal to 0.1/2.5/10 µm respectively (Seinfeld and Pandis, 2016).

1.4 What is the relationship between ambient and indoor air quality?

Numerous studies have found that the indoor air quality is affected by ambient air quality (Baek, Kim and Perry, 1997; Leung, 2015; Meadow et al., 2014) and less commonly vice-versa (Leung, 2015). The concentration and fate/lifetime of pollutants differs between both environments. Indoors there is an absence of sunlight, rain, and extreme temperature fluctuations but the deposition onto surfaces, filtration, infiltration and other internal sources heavily influence a pollutants 'Chemistry' (Weschler and Carslaw, 2018). Consequently, knowledge of the indoor-outdoor concentration gradient (I/O ratio) is of

paramount importance to assess exposure and possible health impacts in indoor environments (Leung, 2015).

1.5 Is indoor air quality something we need to be concerned about?

On average, Americans spend 87% of their time indoors (Klepeis et al., 2001) and in western Europe people may be exposed for more than 20 hours per day to indoor air (Molhave et al., 1997). As we spend so much of our time inside breathing indoor air, quantifying the concentration of indoor pollutants with relevant safe exposure guidelines or standards is imperative.

Pollutants all possess varying toxicity and prevalence indoors. Prolonged exposure to a pollutant (for longer than the set averaging period), at a concentration greater than the health guideline, can cause symptoms from mild sensory irritation (i.e. alpha-pinene) to significant respiratory problems (i.e. NO_2) to cancer (i.e. benzene) (WHO, 2010; Wolkoff and Nielsen, 2017).

Indoor pollutants cause an array of acute and long-term (chronic) health problems, and are the probable cause of sick building syndrome (SBS), a phenomenon describing health issues experienced by the occupants of a building, caused by spending time within the building but, where no specific cause can be found (HSE, 2000; WHO, 2010). Indoor pollutants also react with indoor ozone and produce radicals and secondary organic aerosol (SOA) – all, considered harmful to health (Weschler and Shields, 1997; Weschler and Shields, 1999; Wolkoff et al., 2006).

1.6 What influences pollutant concentrations indoors?

The main sources of indoor pollutants are humans, their activities indoors, construction materials and the infiltration of outdoor produced particles and pollutants (Ekberg, 1994; Myers and Maynard, 2005; WHO, 2010). Typical activities such as cooking, cleaning and painting produce numerous indoor pollutants (Afshari, Matson and Ekberg, 2005; WHO, 2010). Moreover, with the built environments push towards net zero carbon, increased air tightness – in an attempt to reduce energy consumption – can decrease infiltration and lead to an accumulation of internally generated indoor pollutants (Myers and Maynard, 2005; Satish et al., 2012).

Once a pollutant is indoors its lifetime is primarily dependent on the partition between the vapour and particulate phase, this is subject to a pollutant's molecular weight, water solubility and associated partition coefficients (octanol-water K_{OW}, octanol-air K_{OA} and air-water K_{AW}) (Weschler and Nazaroff, 2008; Krol, Zabiegala and Namiesnik, 2011). SVOCs often have the longest lifetime and can persist indoors for years post introduction, because of a stronger tendency to partition into sorbed states than other pollutants (Weschler and Nazaroff, 2008).

One of the most important factors influencing pollutant concentrations indoors is the amount of ventilation, be it provided naturally or mechanically to the space; this in turn is linked to the air exchange rate (AER) — the volume of air removed from a space divided by the volume of the space itself, measured in number of air changes per hour (h^{-1}). Increasing the amount of ventilation will increase the AER and generally decrease the concentration of indoor pollutants by introducing more fresh air into the space and increasing the dilution

rate (Emenius, Egmar and Wickman, 1998; Weschler and Shields, 2000; Hult *et al.*, 2015). However, several studies found that both the chemical and physical properties of the pollutant dictated the effectiveness of increased AER on concentration reductions (Hodgson et al., 2003; Zuraimi, Tham and Sekhar, 2003).

Additionally, the AER can be considered of importance for indoor air quality (IAQ) because of airborne secondary reactions. Nazaroff and Weschler (2004) — in a seminal study on the topic — measured the reaction time of numerous pollutants with the common indoor radical species: ozone, hydroxyl (OH) and nitrate (NO_3). The study determined that the reaction rate of common indoor pollutants with OH was generally too long and would only have a significant impact on IAQ at a very low AER (i.e. with poor ventilation). However, reactions with NO_3 and ozone were found to have a larger pseudo first-order rate constant and thus, are likely to be of greater significance for IAQ and pollutant Chemistry indoors.

Moreover, the absorption of pollutants onto common indoor surfaces is an important factor for IAQ. The time phase considerations previously described above in the Nazaroff and Weschler (2004) study do not relate to surface reactions, this coupled with high surface to volume ratios indoors (typically 2-4 m^2/m^3) and the enhanced ozone-reactivity of certain terpenoids contributes to numerous reactions of great importance for indoor environments (Weschler, 2011; Weschler and Carslaw, 2018).

Measuring the rate of infiltration i.e. the I/O ratio can confirm if indoor or outdoor sources are predominantly affecting the IAQ. A review of 40 studies measuring VOC concentrations in several indoor environments (including schools, offices and homes) concluded that the I/O was generally greater than one. This suggests that indoor VOC concentrations are primarily

influenced by indoor factors (i.e. sources and poor ventilation) and not pollutants infiltrating from outdoors (Paciencia et al., 2016).

Humans themselves are one of the main sources of indoor pollutants (WHO, 2010). Thus, an increased occupancy leads to higher concentrations of pollutants such as CO_2 from respiration, increased relative humidity and skin oil reactions with ozone and nitrate radicals (Langer et al., 2016; Cheng et al., 2016; Weschler and Carslaw, 2018). In domestic environments, increased occupancy also creates secondary implications which may be detrimental to IAQ, namely, a higher number of household activities being undertaken such as cooking, cleaning and crafts (Guo et al., 2009; Cheng et al., 2016).

On average American adults spend between 20 and 30 minutes a day cleaning (Nazaroff and Weschler, 2004), most of which will involve some kind of cleaning product. The most abundant products in terms of VOC emissions are air fresheners, general purpose cleaners and floor polish/waxes (Nazaroff and Weschler, 2004). The emission profiles — and thus, lifetime indoors — of the various pollutants will vary with each compound's volatility. VVOCs will evaporate very quickly after or during use, causing a short-term spike in emission concentrations. Less volatile VOCs and SVOCs produce a delayed emission profile over time, increasing the probability of exposure to the compounds (Wolkoff et al., 1998). Another major health concern is the formation of secondary pollutants. Ozone, a common indoor radical for these reactions reacts fastest with un-saturated compounds, such as terpenes — commonly seen in household cleaning products (Cheng et al., 2016). Such reactions have been shown to produce: aldehydes, including formaldehyde; organic acids; ultra-fine

particles; and free radicals many of which can be dangerous to health, if produced at high enough concentrations (Weschler, 2000; Kim et al., 2015).

Cooking and especially frying has been shown to produce concentrations of $PM_{2.5}$ in the thousands of $\mu g/m^3$ — several orders of magnitude greater than recommended annual/24 hour health guidelines (He et al., 2004; Abdullahi, Delgado-Saborit and Harrison, 2013; Sofuoglu et al., 2015). Similar results have been found with a group of SVOCs known as polycyclic aromatic hydrocarbons (PAHs), where concentrations in kitchen air have been measured in $\mu g/m^3$ and maximum annual exposure guidelines in ng/m^3 (Zhu and Wang, 2003; Li et al., 2003; He et al., 2004; See, Karthikeyana and Balasubramanian, 2006). Alongside this, cooking equipment such as stoves are considered one of the main sources of NO_2 indoors (WHO, 2010). Additionally, SOA can be produced via the thermal desorption of SVOCs from surfaces to which they have sorbed, such as cooking utensils and stovetops (Weschler and Carslaw, 2018).

In the US approximately 80 % of homes are located in urban areas (Weschler and Carslaw, 2018), with this in mind, numerous studies have found concentrations of PM or certain VOCs indoors to be higher in urban than rural environments (Gallego et al., 2008; Yoon, Lee and Park, 2011; Rufo et al., 2016). In rural areas, a connection between the house and the garage was found to have a major influence on pollutant concentrations (Weisel, Alimokhtari and Sanders, 2008; Gallego et al., 2008). Whereas in urban settings, the proximity to traffic had the biggest effect on increasing PM and NO_2 concentrations indoors (Gallego et al., 2008).

1.7 How do we know which pollutants are dangerous indoors?

Several review articles have previously compiled data on indoor pollutants (i.e. concentration and identity) measured in several different indoor environments namely, homes, offices and schools (Logue et al., 2011; Cometto-Muniz and Abraham, 2015; Mandin et al., 2016).

Cometto-Muniz and Abraham (2015) contrasted pollutant concentrations between differing indoor environments and between outdoor and indoor environments. The study found that between home/school and commercial environments the same pollutants could measure a concentration of up to 68 times greater in commercial environments (e.g. tetrachloroethylene) and 25-fold in home vs schools (e.g. 1-butanol). Additionally, between indoor and outdoor, concentrations were measured up to 152 times greater indoors for the same pollutant e.g. methyl isobutyl ketone.

The EU-commissioned OFFICAIR project investigated 37 recently built or refurbished office buildings across Europe over two seasons for indoor pollutants. The study found that concentrations of pollutants varied significantly across seasons, with significantly higher concentrations measured in summer for formaldehyde and ozone, and in winter for benzene, α-pinene, D-limonene, and NO_2. $PM_{2.5}$ was the only pollutant to be measured above relevant acute (24-h) and annual WHO ambient air quality guidelines during the study period (Mandin et al., 2016).

Logue et al. (2011) compared indoor pollutant concentrations to relevant health guidelines produced by the United States Environmental Protection Agency (USEPA) and California Office of Environmental Health Hazard Assessment (OEHHA) for 67 home environments

between 1998 and 2010. The author identified nine 'priority' indoor pollutants which were considered to be harmful, chosen on the basis of the measured concentration data exceeding health guidelines and the fraction of homes impacted (Table 1-1).

Table 1-1: The identity of the 'priority' pollutants identified by (Logue et al., 2011), and the range of concentrations which they have been measured at in home environments in literature is taken from (Gubb *et al.*, 2020) and displayed in Appendix A; * Only one appropriate measurement.

Priority Indoor Pollutant	The range of mean indoor concentrations ($\mu g\ m^{-3}$)
Acetaldehyde	5.0 - 22.0
Acrolein	0.8 - 2.3
Benzene	1.0 - 43.7
Butadiene -1,3	*0.5
Dichlorobenzene -1,4	0.2 - 120.0
Formaldehyde	12.4 - 69.0
Naphthalene	0.0 - 2.6
Nitrogen dioxide	13.1 - 489.7
$PM_{2.5}$	12 - 47

An assessment of these 'priority' pollutants and their mean concentrations in indoor environments has not been carried out since 2010. The author of this thesis has compiled data from home environments, post-2011 to determine if concentrations have changed over this time period and compared concentrations to up to date chronic health guidelines produced by the WHO and USEPA (Figure 1-1).

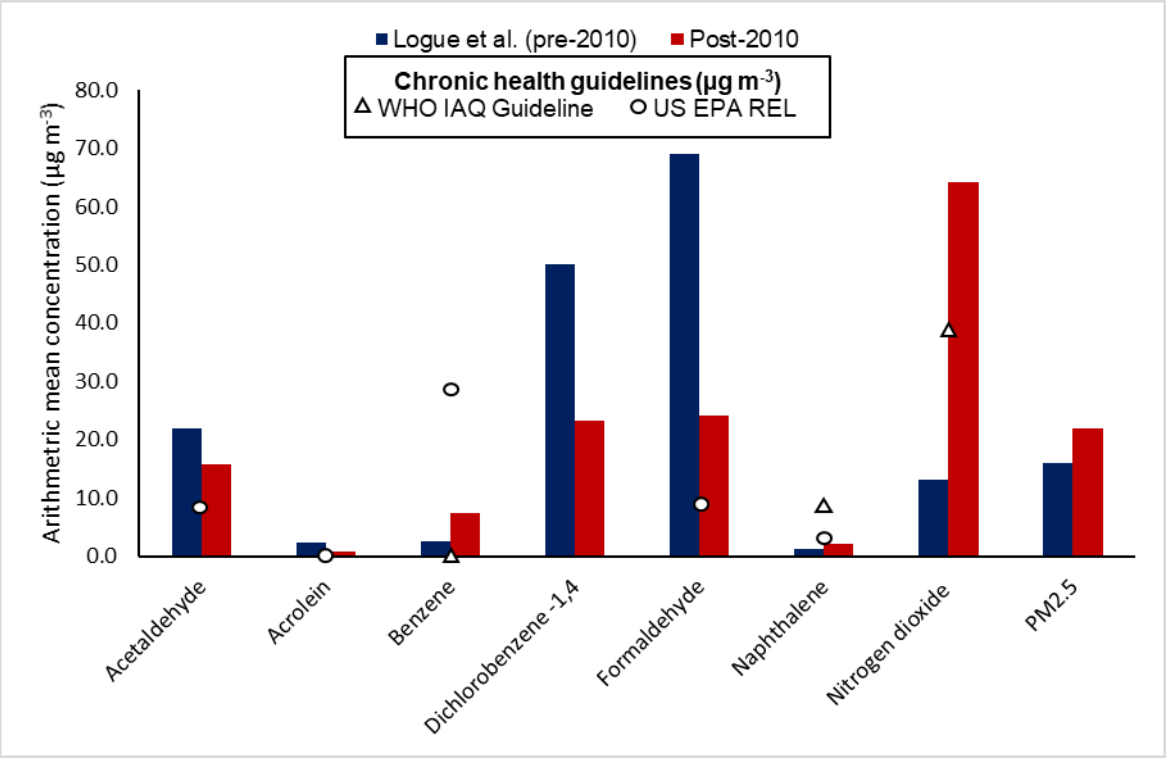


Figure 1-1: Arithmetic mean concentrations of the 'priority' pollutants designated by (Logue et al., 2011) both, pre and post 2010 (i.e. date of Logue study). Butadiene - 1,3 was omitted from the figure, as no data was found post-2010 in home environments. Chronic health guidelines are taken from (WHO, 2010; USEPA, 2014). Full health guideline tables can be found in Appendix B — none were available from the above sources for dichlorobenzene - 1,4.

The data collected in Figure 1-1 suggests that the mean concentrations of three indoor pollutants have increased post-2010 namely, benzene, naphthalene and NO_2. Reductions in concentrations of acetaldehyde, acrolein, dichlorobenzene – 1,4, and formaldehyde were measured; perhaps, due to a large body of research focusing on lowering pollutant emissions from building materials (Barry and Corneau, 2006; Seo et al., 2009; Gunschera et al., 2013; Giosue et al., 2017; da Silva et al., 2017). However, comparing with health guidelines the research suggests that acetaldehyde, benzene, formaldehyde, and NO_2 are the indoor pollutants commonly present at concentrations to cause long term health issues.

Contrary to the compiled data in Figure 1-1, the WHO found insufficient evidence for acetaldehyde to be included in their IAQ guidelines report, it was however categorised with 12 other indoor pollutants in a secondary 'warning' group (WHO, 2010). Acetaldehyde is present in numerous consumer products such as deodorants, foods and alcoholic drinks and is likely produced by wood burners indoors (Lovreglio *et al.*, 2009; Gustafson *et al.*, 2007; WHO, 2010). Acetaldehyde is an irritant to the eyes and airways (WHO, 1995; Lovreglio et al., 2009) and is categorised as a carcinogen (Soffritti et al., 2002).

Sources of benzene indoors include building materials/furniture (Ezeonu et al., 1994; Yu and Crump, 2003), heating and cooking (Heavner, Morgan and Ogden, 1995; Ilgen et al., 2001; Kim, Harrad and Harrison, 2001; Lee, Li and Ao, 2002), attached garages (Graham et al., 2004; Batterman, Jia and Hatzivasilis, 2007; Dodson et al., 2008), and other various human activities (i.e. cleaning, painting and the use of consumer products) (Wallace et al., 1987; Kim, Harrad and Harrison, 2001; Brown, 2002) — benzene also infiltrates indoors from outdoor air (WHO, 2010).

Benzene causes numerous chronic health issues, both carcinogenic and non-carcinogenic (haematological and immunological), most are caused by inhalation (the exposure route 95 – 99% of the time indoors) (MacLeod and Mackay, 1999; WHO, 2010). Research suggests that the two main critical health outcomes from long term benzene exposure are blood dyscrasias and leukaemia (WHO, 2010). Additionally, exposure at typical indoor concentrations positively correlated with mortality due to lung cancer, all haematological cancers and multiple myeloma. WHO therefore suggests that exposure to any concentration of benzene is considered unsafe (WHO, 2010).

Indoors formaldehyde is produced by numerous sources including building materials, furniture, consumer products and combustion processes (i.e. heating, cooking and smoking) (Kelly, Smith and Satola, 1999; Salthammer, Mentese and Marutzky, 2010; WHO, 2010). It should be noted that secondary formaldehyde is formed indoors through reactions between ozone and terpenes (alkenes) (Nazaroff and Weschler, 2004; Uhde and Salthammer, 2007) and by the oxidation of VOCs (WHO, 2010).

Formaldehyde is a classified carcinogen (WHO, 2010) with inhalation causing an array of chronic health problems. Research suggests that exposure at typical indoor levels can play a key role in the development of airway cancer (McGregor *et al.*, 2006; WHO, 2010) and cause myeloid leukaemia (Baan et al., 2009; Hauptmann et al., 2009; Zhang et al., 2010).

Outdoors NO_2 is produced primarily by road traffic and infiltrates indoor environments — the main indoor sources are combustion processes (i.e. heating appliances, fireplaces and stoves) (WHO, 2010). As expected the main factor influencing indoor concentration is the proximity of the building to roads (Nakai, Nitta and Maeda, 1995; Janssen et al., 2001; Kodama et al., 2002), or the presence of an attached garage (WHO, 2010). The WHO chronic (annual) health guideline of 40 μg m^{-3} was set to prevent respiratory illnesses, the main symptom of long-term exposure — especially in children (Hasselblad, Eddy and Kotchmar, 1992). Other health problems caused by NO_2 exposure include airway inflammation and decreases in immune defence (WHO, 2010).

1.8 What about carbon dioxide (CO_2) indoors?

Research suggests that CO_2 is likely to only cause severe health outcomes at high concentrations, uncommonly measured in indoor spaces (6500 to 18500 ppm) (Persily and

de Jonge, 2017). Nevertheless, a number of studies have found that more realistic concentrations (< 5000 ppm) may increase absenteeism and impair cognitive function and productivity (Erdmann and Apte, 2004; Satish et al., 2012; Gaihre et al., 2014; Du et al., 2020). Additionally, it is still considered an important pollutant in the built environment as it is relatively easy to measure and monitor whilst acting as an indicator for both, IAQ conditions and ventilation rates within a space (Persily and de Jonge, 2017). Thus, for this thesis, the author considered CO_2 an important pollutant to study alongside other previously identified harmful indoor pollutants.

1.9 What mitigation strategies do we have to minimise exposure to harmful pollutants indoors?

There are many mitigation strategies which can be implemented indoors to reduce pollutant concentrations. These include filtration of the outdoor air through mechanical ventilation systems (Ginestet et al., 2013) and filtration on fan coil units which re-circulate indoor air (Quang et al., 1994). Also, ensuring that sufficient fresh air is provided to a space through mechanical or natural ventilation (Quang et al., 1994; HM-Government, 2015; Chartered Institution of Building Services, 2005). However, most mitigation techniques require energy input and as we try to reduce our impact on the climate and move towards net zero carbon, passive, and more sustainable options should be further explored. Indoor plants would mostly fall into this category with the simple potted plant only requiring watering, albeit active green walls require energy for fans and complex irrigation systems. Nevertheless, in comparison to many of the engineered solutions mentioned above the energy use would be negligible.

1.10 Can plants remove indoor pollutants, particularly ones which are identified as particularly harmful?

More than 200 indoor plants (i.e. the plant and substrate system) have been tested for their ability to remove pollutants. The most common organic pollutants tested include benzene, toluene, ethylbenzene and xylene (BTEX), formaldehyde and trichloroethylene (Orwell et al., 2004; Kim et al., 2008; Sriprapat and Thiravetyan, 2013; Irga, Pettit and Torpy, 2018). A full list of studies is presented in Appendix C.

A number of indoor plant species have also been investigated for their sequestration ability of the 'priority' pollutants identified in Figure 1-1 namely, benzene (Wolverton, Johnson and Bounds, 1989; Porter, 1994; Cornejo et al., 1999; Oyabu et al., 2001; Wood et al., 2002; Orwell et al., 2004; Yoo et al., 2006; Liu et al., 2007; James et al., 2008; Baosheng et al., 2009; Yang et al., 2009; Chun et al., 2010; Treesubsuntorn and Thiravetyan, 2012; Irga, Torpy and Burchett, 2013; Sriprapat and Thiravetyan, 2013) and formaldehyde (Wolverton and Mcdonald, 1982; Godish and Guindon, 1989; Wolverton, Johnson and Bounds, 1989; Wolverton and Wolverton, 1993; Kondo et al., 1995; Oyabu et al., 2001; Hasegawa et al., 2003; Oyabu et al., 2003a; Hasegawa et al., 2004; Oyabu et al., 2005; Sawada et al., 2007; Kim and Kim, 2008; Kim and Lee, 2008; Kim et al., 2008; Sawada and Oyabu, 2008; Baosheng et al., 2009; Kim et al., 2009; Lim et al., 2009; Kim et al., 2010; Aydogan and Montoya, 2011; Xu, Wang and Hou, 2011; Zhou et al., 2011; Jin et al., 2013; Su and Liang, 2015; Li, Pemberton and Zheng, 2015; Lin, Chen and Chuah, 2017).

One study investigated the ability of seven indoor plant species (*Dracaena marginata* and *Dracaena* 'Janet Craig', *Epipremnum aureum*, *Howea forsteriana*, *Schefflera* 'Amate' and

Spathiphyllum cvs 'Petite' and 'Sensation') to remove the 'priority' pollutant benzene. Plants were enclosed inside a static Perspex chamber (0.216 m^3) and tested in duplicate at a light level of ~ 120 µmol m^{-2} s^{-1}. The study looked to determine benzene removal rates after (1) several doses of 80 µg m^{-3} (25 ppm) over 24 hr periods (2) a 24-hr dark regime i.e. no light (3) a double dose of benzene (160 µg m^{-3}) with no light (4) plant removal i.e. bare substrate with no light. The authors found varying removal rates from 12 (*Howea forsteriana*) to 27 (*Dracaena* 'Janet Craig') ppm d^{-1} (40 to 88 mg m^{-3} d^{-1}) from stage (1). These rates were maintained in the dark, rose linearly with concentration increase but, could mostly be attributed to the substrate and not the plant (Orwell et al., 2004).

Another study investigating benzene removal by *Syngonium podophyllum*, compared plants grown in traditional substrate and hydroculture (i.e. no substrate). The study utilised eight glass chambers (15 L) and plants were placed inside at a light level of 20 µmol m^{-2} s^{-1} (1480 lux). A dose of 80 µmol m^{-2} s^{-1} was added, with the concentration measured and quantified by gas chromatography mass spectrometry (GC-MS) at 24 hr intervals. The authors concluded that indoor plants potted in traditional substrate possessed a higher removal rate than hydroculture potted plants but, both treatments removed significant amounts of Benzene (Irga, Torpy and Burchett, 2013).

The ability of *Fatsia japonica* and *Ficus benjamina* (aboveground parts and root zone) to remove formaldehyde — in the day and night — were determined in one study. Species were exposed to 2 µL L^{-1} in airtight 1 m^3 chambers at a light level of 20 + 2 µmol m^{-2} s^{-1} (1480 lux), with the time taken for the initial concentration to reduce by 50% determined. The authors found that with the whole plant-substrate system, *Fatsia japonica* removed

formaldehyde faster than *Ficus benjamina* (50% decay in 96 and 123 min respectively). Both species removed formaldehyde in a 1:1 ratio (aboveground parts: root zone) in the day, and 1:11 in the night. The root zone was found to remove formaldehyde primarily through the microorganisms and roots (90%) and a small amount through GM absorption (10%) (Kim et al., 2008).

Another similar study investigated formaldehyde removal by *Chlorphytum comosum*, *Aloe vera* and *Epipremnum aureum* with potted soils. The study utilised a plexiglass chamber (height 60 cm, inner diameter 40 cm) and a flow system generating 1 mg m^{-3} initially and adding a further 1 mg m^{-3} every three days until phytotoxicity of the studied species. Formaldehyde concentrations were measured at the inlet and outlet, every three days, with a specific analyser. Species were subjected to 12 hr of light at 240 µmol m^{-2} s^{-1} (17 760 lux) per day. The authors found that all plant-substrate systems removed formaldehyde, with *Chlorphytum comosum* removing the most. The authors also determined that microorganisms in the substrate accounted for ca. 50% of the formaldehyde removal in all the plant-substrate systems (Xu, Wang and Hou, 2011).

To my knowledge no studies have investigated the potential of indoor plants to sequester acetaldehyde. Concerning NO_2 a couple of studies have investigated indoor plants and their removal abilities, these are covered in detail in Chapter 5 (Morikawa et al., 1998; Pettit et al., 2019). Additionally, the removal of NO_2 by outdoor plants has been thoroughly studied (Okano, Machida and Totsuka, 1989; Ammann et al., 1999; Takahashi et al., 2005; Nowak, Crane and Stevens, 2006). A full list of studies investigating potted plants and pollutant removal (VOCs) is presented in Appendix C.

1.11 Can plants also help with indoor environmental quality (IEQ), specifically relative humidity (RH)?

A fundamental aspect of indoor environmental quality (IEQ) and both human comfort and health indoors is relative humidity (RH); keeping RH within the recommended range of 40 - 60 % will help avoid the majority of associated adverse health effects and increased likelihood of pathogen transmission — ever more important with the current pandemic (Arundel *et al.*, 1986; Zhang and Yoshino, 2010; Butcher *et al.*, 2015; Gubb *et al.*, 2018). However, controlling humidity via (de)humidification in mechanical systems is incredibly energy intensive thus, a great importance should be placed on finding passive solutions, namely, indoor plants. Plants, through the natural process of transpiration, release water vapour into the air which in turn, contributes to raising the humidity (Gubb *et al.*, 2018). A number of studies have investigated the effect of indoor plants on RH indoors, results however, have not confirmed the assumed hypothesis that plants would raise RH within a space (Lohr and PearsonMims, 1996; Lim *et al.*, 2009; Jeong *et al.*, 2008; Pegas *et al.*, 2012).

One study looked at RH changes with various foliage plants (including *Aglaonema sp.*, *Chamaedorea seifrizii, Dracaena marginata, Epipremnum aureum*, and *Spathiphyllum sp*.) in a 32 m^3 office where taxa covered 5 % of the room. A small but, statistically significant increase in RH was measured, over a period of several months(Lohr and PearsonMims, 1996).

Another study evaluated 27 different indoor plants for their effects on RH in 1.7 m^3 growth chambers, where taxa occupied 20 % of the total volume of the chamber. On average, RH increased by 15.5 %RH (referring to absolute increase) from the initially controlled 40 – 50 %

RH provided by the chamber. The most effective taxon *Cyrtomium caryotideum*, gave rise to a 30.3 %RH increase in RH with all 27 species showing some level of RH increase. The study's authors suggest that the RH increase was not related to total leaf area but more each taxon's inherent transpiration rate (Jeong et al., 2008).

One study observed that the presence of six houseplants selected from the following species (the study did not specify which were used and how many of each) *Dracaena deremensis* (cvs 'Striped Dracaena' or 'Janet Craig'), *Dracaena marginata* (cvs 'Rededge Dracaena', 'Madagascar Dragon Tree', or 'Marginata') and *Spathiphyllum* (cvs 'Mauna Loa' or 'Peace lily') in a 52.5 m^2 classroom and measured in two separate measuring periods a reduction of 60.8 to 45.4 %RH in RH over a 90 day testing period and no statistically significant change over a nine week measuring period. Although, this was not associated with the plants specifically or speculated on by the authors of the study. It is likely that other environmental factors within the classroom, that were not tightly controlled were behind the reduction — highlighting the difficulty in undertaking this type of experiment in 'real-life' situations (Pegas et al., 2012).

1.12 How do plants remove indoor pollutants?

Indoor plants remove pollutants via three distinct pathways — aboveground parts (i.e. stomata and cuticle); GM and the roots (Cruz et al., 2014). Research suggests the dominant pathway is governed by the physical properties of the pollutant but, more than one pathway can remove pollutants simultaneously (Irga, Torpy and Burchett, 2013). Research suggests that certain pollutants may have an affinity for a particular pathway (Cruz et al., 2014). Non-polar pollutants are likely removed by the aboveground plant parts due to low mobility in

the substrate or transport system and a higher permeability for plant surfaces (Sabljic et al., 1990). Whereas hydrophilic VOCs (such as formaldehyde) will not diffuse as easily through the cuticle as a lipophilic VOCs (such as benzene) (Cruz et al., 2014).

No relationship has been measured between an indoor plant species or characteristic (i.e. amount of hair, stomata characteristics/density or wax layer) and its pollutant removal ability (Cruz et al., 2014). Research suggests that indoor plants may be utilised in more of an indirect role, helping maintain and support a GM microorganisms rather than removing indoor pollutants directly (Wood *et al.*, 2002; Orwell *et al.*, 2004; Kim *et al.*, 2008; Irga, Torpy and Burchett, 2013). Research into hydroponic planting — substrate-less systems — have also found less effective pollutant removal (Wood et al., 2002; Irga, Torpy and Burchett, 2013). Different indoor plants can support different microorganisms in their GM (Zhang et al., 2013) and different GM has been shown to have varying pollutant removal abilities (Oyabu et al., 2003b); thus, the GM — namely, the microorganisms within, may be the primary factor in pollutant removal. This poses a significant problem for researchers, due to the variability within soil microorganism communities, setting up a well-characterised experiment is challenging.

1.13 What has been shown to influence a plants ability to remove indoor pollutants?

Numerous studies have found that different indoor plant species (Wolverton and Wolverton, 1993; Aydogan and Montoya, 2011; Treesubsuntorn and Thiravetyan, 2012); and cultivars (Orwell et al., 2004; Zhou et al., 2011) have a widely differing potential for pollutant removal. One study measuring formaldehyde sequestration observed differences between five cultivars of *Aglaonema commutatum* (Golden Jewellery, White Rajah, Red Narrow, Silver

Queen and Treubii), two cultivars of *Dieffenbachia amoena* (Camilla and Green Magic) and two cultivars of *Sansevieria trifasciata* (Hahnii and Laurentii) under the same experimental conditions (Zhou et al., 2011). Moreover, differences in benzene sequestration ability were measured to the cultivar level in *Spathiphyllum floribundum* ('Petite' and 'Sensation'); both, measured different average 24hr removal rates in both light and dark conditions — *Spathiphyllum floribundum* 'Petite' was more effective in both conditions (Orwell et al., 2004).

As the concentration of the pollutant in question is increased, the rate of pollutant removal increases linearly (Kondo *et al.*, 1995; Orwell *et al.*, 2004; Orwell *et al.*, 2006; Xu, Wang and Hou, 2011) but, the removal efficiency (defined as the percentage of removal per unit time per leaf area) can vary (Oyabu et al., 2003b; Orwell et al., 2006). One study tested the efficiency and rate of toluene removal by *Dracaena deremensis* 'Janet Craig' and *Spathiphyllum* 'Sweet Chico'. The study observed that as the concentration was increased between 764– 439,844 μg m^{-3} the rate of removal increased, and the efficiency decreased. The study's authors suggested that an efficiency decrease is due to the houseplant reaching its pollutant sequestration capacity (Orwell et al., 2006). However, another study found a slight increase in removal efficiency in *Epipremnum aureum* with increasing formaldehyde concentration (Oyabu et al., 2003b).

Several studies have reported a clear link between decreasing removal efficiency and increasing molecular size of the indoor pollutant (Oyabu et al., 2003b; Baosheng et al., 2009; Oyabu et al., 2001). Additionally, it has been found that as water solubility increases, both

aldehyde and ketone sequestration increased — with aldehydes often selectively sequestered over ketones in one study (Tani and Hewitt, 2009).

In 'real life' indoor environments, pollutant mixtures — rather than single pollutants — are present in indoor air for plants to remove. Several studies have investigated both binary and more complex mixtures of indoor pollutants (Cornejo et al., 1999; Orwell et al., 2006; Yoo et al., 2006; Yang et al., 2009).

For binary mixtures *Kalanchoë blossfeldiana* was found to selectively remove benzene over toluene. Although, the study used different concentrations of each compound in a tested mixture (51,805 μg m^{-3} benzene, 7,609 μg m^{-3} toluene) likely influencing the results (Cornejo et al., 1999). Another study conversely found that toluene was selectively removed over benzene by *Hedera helix*, *Spathiphyllum wallisii*, *Syngonium podophyllum* and *Cissus rhombifolia* — suggesting it may be species specific, moreover, concentrations were comparatively more similar (1,602 μg m^{-3} benzene, 1,890 μg m^{-3} toluene) than in the above described study. The same authors observed higher removal rates for each taxon exposed to a single pollutant (toluene or benzene) than the mixture. However, the single pollutant concentrations used in the study were approximately double that of the mixture (3,204–3,779 μg m^{-3}), perhaps, explaining the removal rate disparity (Yoo et al., 2006).

A more robust investigation measured the pollutant removal ability of a *Dracaena deremensis* 'Janet Craig' when exposed singly, and to a mixture of toluene and m-xylene. A number of concentrations were tested (0.2, 1, 10 and 100 ppm) — the same for both single compounds and mixtures — with a synergistic relationship identified whereby, toluene accelerated the removal of m-xylene at lower studied concentrations (Orwell et al., 2006).

Further, more complex pollutant mixtures have been investigated in one study on 28 indoor plants. Plants were exposed to ~ 10 ppm of each pollutant i.e. benzene, trichloroethylene, toluene, octane, and a-pinene with the concentration decline measured over a 6-hr period. Unfortunately, however, the species were not tested with individual pollutants thus, how the mixture may have altered the removal ability is unknown (Yang et al., 2009).

1.14 Can the environmental conditions also effect a plant's removal ability?

Plants utilise UV and visible light in fundamental processes such as photosynthesis and depend on it for survival. Light is not a constant source, it changes spatially and temporally therefore, plants must constantly adapt and move to absorb what is required. It is generally acknowledged that UV light induces stomatal movement and the effect (i.e. aperture) influenced by the wavelength of the radiation (Farquhar and Sharkey, 1982; Zeiger, 1983; Shimazaki et al., 2007; Lawson, 2009).

Increasing light intensity induces stomatal pore opening, which in turn, would increase gas exchange (i.e. CO_2 assimilation) and the stomatal assimilation of other pollutants. Varying the light intensity, however, may have little effect if the pollutants are removed by the cuticle or microorganisms (Godish and Guindon, 1989; Wood et al., 2002; Orwell et al., 2004). Formaldehyde and NO_2 have been found to degrade photochemically under light without indoor plants, and this should be carefully controlled in any experiments to ensure the light itself is not contributing to the removal (Cruz et al., 2014).

Soil water stress is the main environmental factor which effects photosynthesis and respiration, it also causes stomatal closure and reduced mesophyll and stomatal conductance (Lawlor and Cornic, 2002; Flexas et al., 2006). Thus, effecting the pollutant

assimilation ability of the plants — especially concerning any removal occurring via the stomatal pathway (Torpy, Zavattaro and Irga, 2017; Gubb *et al.*, 2018; Gubb *et al.* 2019).

A plant species response to water deficiency is dependent on its genetic makeup (Chaves, 1991). In a 'plant-substrate' system as water deficiency in the substrate increases, stomatal pores begin to close preventing more water loss by transpiration and evaporation from the leaves (Webb and Mansfield, 1992; Hsiao, 1973). A plant achieves this by detecting stress and utilising the hormone abscisic acid (ABA) to rapidly close the stomata (Hsiao, 1973; Jones and Mansfield, 1970; Wilkinson and Davies, 2002). Stomatal closure is an initial response by a plant to the effect of water stress and this in turn limits carbon assimilation. With stomata closure comes a reduction in both carbon assimilation and transpiration (Willis and Balasubramaniam, 1968; Hsiao, 1973; Chaves, 1991).

If a plant is water deficient the stomata will shut, preventing any gas exchange and any possible pollutant sequestration. For sequestration experiments, knowledge of where the substrate moisture content (SMC, $m^3\ m^{-3}$) of each species is low enough to cause a stomatal shutdown is significant — because experiments will not measure the plant working to its full ability.

Increasing the ambient temperature —whilst retaining plant functionality — will generally increase pollutant sequestration. One study determined that pollutant removal increased as the temperature was increased from 17 – 35 °C. The authors observed that an increase in temperature increased the permeability of the cuticle (Baur and Schonherr, 1995). Increasing the temperature has also been shown in certain cases to increase microbiological growth — possibly aiding microorganism pollutant removal (Cruz et al., 2014).

1.15 How is plants' ability to remove pollutants tested?

Experimental setups often utilise small (< 1 m^3) static (i.e. sealed, no air circulation) (Irga, Torpy and Burchett, 2013; De Kempeneer et al., 2004; Orwell et al., 2004; Orwell et al., 2006; Sriprapat and Thiravetyan, 2013; Siswanto, Chhon and Thiravetyan, 2016) or dynamic (i.e. with air circulation) (Tani and Hewitt, 2009; Yang et al., 2009; Kim et al., 2008; Cornejo et al., 1999) chambers to enclose the plants and broadly follow a similar methodology to measure removal ability. This comprises of the injection of a desired pollutant (i.e. fumigation), followed by an equilibration time for concentration homogeneity, the sampling of chamber air over a period of time and finally quantification and analysis through a headspace chromatography instrument (e.g. gas chromatography mass spectrometry, GCMS).

Dynamic setups utilise a continuous air stream through the chamber, and in contrast to static, are easier to control the inevitable increases in RH and temperature brought by enclosing a plant. However, care must be taken to introduce a clean air stream and not introduce impurities into the chamber. Static chambers are, therefore, advantageous for low concentrations of pollutants as impurities are not introduced, but consequently, measures to control RH and temperature are often required (Tholl et al., 2006).

Small chamber experiments – both dynamic and static – are often criticised for an inability to accurately extrapolate results to building scale *in situ* conditions (Irga, Pettit and Torpy, 2018; Torpy et al., 2018). Buildings – unlike small chambers — possess complex heating, ventilation, and air conditioning (HVAC) systems with significant air exchange and numerous sources and mixtures of pollutants (Irga, Pettit and Torpy, 2018). However, other than static

room-scale chambers – requiring substantial resources – small chambers are often considered the best alternative.

1.16 Other common issues with these experiments?

Another issue with a number of studies is that they report on pollutants which have little practical relevance (e.g. are infrequently present indoors or in concentrations too low to cause damage to human health) (De Kempeneer et al., 2004; Tani et al., 2007; Kim et al., 2011; Siswanto, Chhon and Thiravetyan, 2016). Additionality, pollutant concentrations in some experiments are often 1000-fold greater than what is typically measured *in situ* — altering a plants removal efficiency (Aydogan and Montoya, 2011; Torpy et al., 2018; Cruz et al., 2014). A number of studies have investigated the indoor pollutant formaldehyde, typically measured in the range of 12.4 – 69 µg m^{-3} (Table 1-1) in home environments (Ohura et al., 2006; Logue et al., 2011). Example formaldehyde concentrations used in indoor plant sequestration experiments include: 2000000 µg m^{-3} (Aydogan and Montoya, 2011); 2500 µg m^{-3} (Jin et al., 2013); and 2400 µg m^{-3} (Lim et al., 2009), — an inherent problem across nearly all experiments of this type. A number of studies have shown that increasing the pollutant concentration will linearly increase the removal rate (Kondo et al., 1995; Orwell et al., 2006; Xu, Wang and Hou, 2011), but not necessarily the removal efficiency (Orwell et al., 2006; Cruz et al., 2014). However, often no attempt is made to extrapolate the removal rate results back to typical indoor concentrations.

Furthermore, where fans are used to circulate air within chambers, any mass transfer resistance from the bulk air to the boundary layer is substantially reduced. In ‘real-life’ indoor settings at the room scale, boundary layers may be thick and thus further inhibit the

removal of pollutants (Soreanu and Dumont, 2020). Moreover, to extrapolate from the chamber to the room scale, differences in terms of pollutant diffusion namely, the distance and thus time taken for pollutants to move from the source to the site of removal can affect results. This can be additionally exacerbated by large differences in air distribution, indoor air stability and the complex air patterns created by ventilation. Thus, the transport of pollutants (sink to source) is not easily replicated by chamber experiments (Gong et al., 2010; Soreanu and Dumont, 2020; Deng and Gong, 2021).

1.17 How will this thesis look to address these issues and add to the literature already available?

Initially, this thesis looked to address the issues surrounding high pollutant concentrations being used in plant-pollutant sequestration experiments. In an attempt, to facilitate experiments for the testing of various VOCs at typical indoor concentrations. The author found a clear pattern in current literature of high concentrations being used far exceeding what would be found in indoor environments. Firstly, this adds uncertainty to any measured removal ability, in terms of will the plants be able to replicate the same removal ability in 'real-life' environments. Additionally, it had also been shown to affect the removal efficiency of plants in several studies, therefore, testing at *in situ* concentrations will add increased certainty to any measured removal rates.

The author hypothesised that both the sampling of chamber air after the fumigation period and the transfer method into a suitable headspace chromatography instrument were likely responsible for the use of elevated concentrations due to losses in sensitivity. To address this, the author proposed validating a technique known as solid phase micro extraction

(SPME), whereby, pollutants are sampled via adsorption onto a specially coated fibre and transferred directly to the analytical instrument. These measurements were to be contrasted with the traditionally used gas-tight syringe method (Chapter 6).

Houseplants have previously been investigated for their CO_2 removal ability in a number of studies. The initial study looked to determine the inherent ability of seven UK-common indoor plant species to remove CO_2 via leaf stomatal assimilation with an infra-red gas analyser (IRGA) — an instrument commonly used in horticultural experiments (Chapter 3). It was hypothesised that choosing species representing a variety of metabolisms, leaf types and sizes would link a particular characteristic to superior removal ability — something that has not been previously identified. Plants were tested under both wet and dry substrate conditions and at a variety of light levels.

Further, I looked to investigate if the differing species would transpire and lose water at differing rates and thus, contribute to the passive increase of relative humidity indoors (Chapter 2). Studies confirm that humidity between 40 – 60 % is best for human health, and in the UK — especially in the winter — humidity inside is often measured at around 30%. Humidification indoors for mechanically ventilated buildings is energy consuming and thus, it was hypothesised that plants could offer a passive low-energy alternative to increasing humidity.

A custom-built chamber was then utilised to enclose the whole plant — including the substrate — and test if the studied species could reduce a concentration of CO_2 commonly measured indoors of 1000 ppm (Chapter 4). The best and worst performing plant species were chosen from the previous leaf-level gas exchange (IRGA) experiments, to additionally

investigate if the leaf-level results on gas exchange could be effectively scaled up to indoor environments. The plant species were tested at several typical indoor light levels and with two differing substrates. The latter was hypothesised to provide differing removal rates— if a pathway of removal was also via the substrate — because of numerous other studies confirming that the substrate was responsible for most of the pollutant removal.

NO_2 is an incredibly dangerous pollutant in terms of its human health impacts and is commonly measured above recommended health guidelines for longer than relevant averaging periods indoors. However, there has only been a very limited number of studies investigating houseplants ability to remove NO_2, especially in real-time and at in situ concentrations. To address this, the author designed an experiment to test the removal ability of three different taxa — potted in two different substrates under typical indoor and night-time conditions — to remove the WHO acute (1-hour) guideline concentration of 200 $\mu g\ m^{-3}$ (Chapter 5). It was hypothesised that the studied species would be able to remove at least some NO_2, because of previous studies measuring the ability of outdoor plants and trees to do the same.

Specific research aims are therefore:

- To test the inherent ability of common houseplants with differing structural/functional characteristics for CO_2 removal, and determine if a link between a particular trait and superior removal ability could be identified (Chapter 3).
- Determine if a number of common houseplants would all contribute differing amounts to raising the relative humidity indoors and identify which of the studied species were superior (Chapter 3).

- Measure the ability of houseplants to remove a commonly experienced — and recommended maximum health guideline (over an 8-hour averaging period) — 1000 ppm concentration of CO_2 in a static chamber setup (Chapter 4).
- Address the lack of research focusing on houseplants and their ability to remove NO_2 with a focus on both real-time and *in situ* concentration experiments (Chapter 5).
- Develop a new method for VOC removal experiments, allowing measurements to be carried out at typically measured indoor concentrations (Chapter 6).

2.0 General Materials and Methods

2.1 Environmental conditions for experiments

Experiments were carried out at both the Glasshouse Complex of the School of Agriculture, Policy and Development, at the University of Reading (UK) and in the Bioscience building of the School of Geography, Earth and Environmental Sciences at the University of Birmingham. An experimental overview is presented below in Table 2-1 detailing both where each experiment was undertaken and in what order these were carried out.

At both experimental sites, plants were maintained in indoor office environments prior to the experiments starting. Once the experiments in those environments begun, air temperature and RH were measured with a Tinytag RH/temperature logger (Gemini data loggers, Chichester, West Sussex, UK). All species were acclimatised to the environment for a minimum of 90 days with plants maintained in 3 L containers, with a slow release fertiliser feed provided (Osmocote, Marysville, OH, USA). Taxa were prepared for experiments with GM moisture at the container capacity (SMC, > 30%) and plants were thus considered optimally watered on the commencement of each experiment (Vaz Monteiro *et al.*, 2016). To ascertain GM moisture, the SMC was measured prior to experimentation for each plant, in two locations per container using a SM300 capacitance-type probe connected to a HH2 Moisture Meter (Delta-T Devices, Cambridge, Cambridgeshire, UK; 0–100% range and an accuracy of ± 2.5%).

2.2 Plant Material

The following species and cultivars of houseplants: *Dracaena fragrans* (cvs 'Lemon Lime' and *'*Golden Coast'), *Guzmania* 'Indian Night', *Hedera helix*, *Spathiphyllum wallisi* (cvs 'Bellini'and

'Verdi') and *Zamioculas zamiifolia* were chosen for the experiments (Figure 2-1). The genera / species / cultivars were chosen to represent a range of leaf types (succulent and herbaceous), metabolisms (C3 and CAM) and plant sizes. Furthermore, houseplants common to the UK market were prioritised during the selection process, with most of the studied in the top 100 bestselling plants from the Royal Horticultural Society (RHS) plant centre.

Guzmania 'Indian Night' is an evergreen perennial from the *Bromeliaceae* family and is native to Florida and Tropical America (WCSP, 2017). *Guzmania* sp. are epiphytic, using other plants (often trees) as growing sites (Nieder, Prosperi and Michaloud, 2001). The plants cannot take the sufficient moisture and nutrients required from the substrate and employ aboveground parts for these processes (Nieder, Prosperi and Michaloud, 2001); thus, the aboveground part of the species was hypothesised to be more active in terms of gas exchange and possibly pollutant removal.

Dracaena fragrans is a tropical shrub of the *Asparagaceae* plant family and is native to tropical Africa (WCSP, 2017); in this study two cultivars of this family are used, differing in plant habit and size. Due to its waxy leaves and anecdotally low water use requirements it was hypothesised to be less physiologically active than other studied species. In this study the cultivars 'Golden Coast' and 'Lemon Lime' were selected, it was hypothesised that an inherent difference in removal capacity could be measured between cultivars of the same species. In horticultural practice a range of cultivated varieties 'cultivars' are usually available within a single species. They are usually bred for different ornamental qualities (e.g. flower colour or size, leaf colour). Commercially, a range of these will be available to homeowners and there is a merit in understanding if they perform differently in terms of

pollutant removal but additionally, as they are more similar in comparison than two regular plant species, it was hypothesised this may help determine if a particular plant trait is linked to greater removal.

Hedera helix is a shrub and a climber and a member of the *Araliaceae* family. *Hedera helix* is native to Europe, the Mediterranean and Iran (WCSP, 2017). It was chosen on the basis of it traditionally being an outdoor plant that can be kept indoors, and therefore, possessing a more active gas exchange and possibly increased pollutant removal rate.

Spathiphyllum wallisi is an evergreen herbaceous perennial of the *Araceae* family and is native to the south American region (WCSP, 2017). In this study two *Spathiphyllum wallisi* cultivars were used ('Bellini' and 'Verdi'), alongside being a very common plant in the UK, we wanted to test the cultivar hypothesis as with *Dracaena fragrans* for another species.

Zamioculas zamiifolia is an evergreen perennial and a member of the *Araceae* family. It inhabits dry and humid forests, open bushland and savannas in southeast Africa; the species possesses a weak CAM that is activated under water stress and was selected specifically because of this (Holtum *et al.*, 2007). A CAM metabolism enables plants to successfully live in a range of environments as they become highly water use efficient, possessing lower transpiration rates because decarboxylation and CO_2 refixation are accompanied by reduced stomatal aperture (Winter and Holtum, 2005; Holtum *et al.*, 2007). Under water stress this plant would therefore limit its daytime CO_2 uptake which may have implications on its capacity for the removal of pollutants.

Dracaena fragrans ‘Golden Coast’

Dracaena fragrans ‘Lemon Lime’

Spathiphyllum wallisi ’Bellini’

Spathiphyllum wallisi ‘Verdi’

Guzmania ‘Indian Night’

Hedera helix

Zamioculas zamiifolia

Figure 2-1: Images of the seven houseplant taxa selected for study

2.3 Growing Media

Three different GM were used in total across all plant-based experiments with the peat-free Sylvamix GM (6:2:2 sylvafibre: growbark pine: coir; Melcourt, Tetbury, Gloucestershire, UK) used in all. This GM was initially selected on the basis of it being both peat free — peat is a limited resource with the UK government pushing for a voluntary phasing out by 2030 (Defra, 2018) — and possessing the ability to support a wide range of houseplants effectively in prior experiments (horticultural advice provided by the University of Reading glasshouse team).

The two other GMs selected were both peat-based, for CO_2 removal experiments described in Chapter 4, Clover professional pot bedding substrate (100% Irish Moss Peat; Clover, Dungannon, Co. Tyrone, UK) was chosen. The author wanted to contrast plants potted in very differing substrates, hence, with and without peat. Peat-based substrates are still

commonly used across the UK because of their uniformity, providing easier water management (Schmilewski, 2008; Alexander, Williams and Nevison, 2013). Peat has a very high-water retention capacity, unlike certain peat-alternatives such as coir, sand and wood fibres (Schmilewski, 2008). As several studies have linked soil moisture to microbial respiration an investigation into moisture content is of significance to pollutant removal (Cook, Orchard and Corderoy, 1985; Manzoni, 2012).

For NO_2 removal experiments presented in Chapter 5, Wyevale Multipurpose Compost (58 % peat; 42% green compost/coir, exact ratios not disclosed, Wyevale, Brentford, Middlesex, UK) was selected to contrast against the peat-free Sylvamix. This GM was recommended by the RHS horticultural houseplant advisor after growth and survival issues for certain species in the previous peat GM (Clover professional pot bedding substrate; see Chapter 4). Further, this GM was awarded a Which? best buy for multi-purpose compost and was therefore deemed to be commonly used in the UK market and looking to provide further impact and relevance to the experiments.

Table 2-1: Provides a summary of and the order in which experiments were undertaken. Different equipment specific to each chapter is described in the individual materials and methods section; Key: UoR – University of Reading; UoB – University of Birmingham; V - *Spathiphyllum wallisii* 'Verdi'; GC - *Dracaena fragrans* 'Golden Coast'; HH – *Hedera helix*; ZZ - *Zamioculcas zamiifolia*; SY - Sylvamix growing medium; CL - Clover professional pot bedding substrate; WY - Wyevale Multipurpose Compost; GC-MS – Gas chromatography mass spectrometry; SPME – Solid phase micro extraction; IRGA – Infrared gas analyser.

Experiment	Location	Plant Material	Growing Media	Main Equipment
Is solid-phase micro-extraction (SPME) a suitable technique for determining a houseplant's removal ability of volatile organic compounds (VOCs)? (Chapter 6)	UoR	N. A	N. A	GCMS, SPME
Can houseplants improve indoor environmental quality by removing CO_2 and increasing relative humidity? (Chapter 3)	UoR	All (Section 2.2)	SY	IRGA
Interaction between plant species and substrate type in the removal of CO_2 indoors (Chapter 4)	UoR/UoB	V, GC, HH	SY, CL	Chamber, CO_2 Sensor
Houseplants can remove the pollutant nitrogen dioxide indoors (Chapter 5)	UoB	V, GC, ZZ	SY, WY	Chamber, NO_2 Sensor

2.4 Statistical Analysis

For both CO_2 experiments (Chapters 3 &4), experimental data (i.e. CO_2 concentrations) were analysed using GENSTAT (17th Edition, VSN International, Hemel Hempstead, Hertfordshire, UK). NO_2 concentrations (Chapter 5) were analysed using SPSS (26th Edition). For all, an analysis of variance (ANOVA) was performed to compare means for each measured parameter between different species and/or over time. Variance levels were checked for homogeneity and values were presented as means with either associated least significant differences (lsd) at a 5% significance level, standard error of the mean (SEM) or as Tukey's

95% confidence intervals for multiple comparisons. Where a lsd or Tukey's confidence interval has been used the associated p-value is presented.

For VOC experiments presented in Chapter 6, the linearity was assessed by calculating a R^2 value for the linearity plot, with an $R^2 > 0.99$ considered an excellent fit for validation purposes (Rood, 2007). Additionally, the reproducibility was also assessed at each measured concentration (n = 3), with a relative standard deviation (%RSD) of < 20 % considered acceptable (UNODC, 2009). %RSD was calculated as shown in Equation 2-1.

$$\%RSD = 100 * \text{Standard deviation} / \text{Mean} \quad (2\text{-}1)$$

3.0 Can houseplants improve indoor environmental quality by removing CO_2 and increasing relative humidity?

This chapter was based on the published paper presented in Appendix D. Substantial additions to this chapter are as follows:

- Section 3.3.3 (Further analysis included)
- Figure 3-4

3.1 Introduction

Indoor CO_2 concentrations are primarily dependent on the occupancy level and outdoor air supply rate (Zhang, Wargocki and Lian, 2017). Humans produce and exhale CO_2; therefore, a greater occupancy coupled with lower ventilation rates – intended to reduce energy consumption – gives rise to higher and often harmful CO_2 concentrations indoors (Satish *et al.*, 2012). Additionally, even when ventilation by ambient air is employed, the problems may be exacerbated in the future: ambient CO_2 concentrations increased by 40% over the last century, to 400 ppm – with a rise to 670 ppm expected by 2100 (Hersoug, Sjodin and Astrup, 2012).

The American Society of Heating, Refrigerating and Air-conditioning Engineers (ASHRAE) recommends a maximum indoor CO_2 concentration of 1000 ppm (Torpy, Zavattaro and Irga, 2017). Concentrations indoors (e.g. in fully occupied offices or meeting rooms) often reach 2000 to 2500 ppm but can rise as high as 5000 ppm (Zhang, Wargocki and Lian, 2017). Although discrepancies in the maximum safe exposure concentration are commonplace in literature, prior research suggests indoor CO_2 concentrations may present unwanted health issues (Zhang, Wargocki and Lian, 2017). These include mucus membrane symptoms (i.e.

sore/dry throat, dry eyes and sneezing) and respiratory problems (i.e. tight chest, wheezing/coughing and shortness of breath) (Seppanen, Fisk and Mendell, 1999; Erdmann and Apte, 2004). Elevated CO_2 can also reduce the cognitive performance of students in schools, while long-term, regular exposure has been linked to increased absenteeism, weight gain and obesity (Hersoug, Sjodin and Astrup, 2012; Satish *et al.*, 2012; Gaihre *et al.*, 2014; Nieuwenhuis *et al.*, 2014; Vehvilainen *et al.*, 2016; Zhang, Wargocki and Lian, 2017).

An additional challenge in indoor environments is low relative humidity (RH). An RH below 30% has been shown to cause eye irritation and skin dryness, with an RH below 10% causing dryness of the nasal mucus membrane. Low RH can also increase the likelihood of influenza transmission, enhance indoor ozone concentration and produce static electricity (Arundel *et al.*, 1986; Berglund, 1998; Sunwoo *et al.*, 2006; Lowen *et al.*, 2007; Zhang and Yoshino, 2010; Abusharha and Pearce, 2013). However, high RH (> 60%) can also cause issues by encouraging fungal/mould growth and contributing to the deterioration of building materials (Berglund, 1998; Bin, 2002; Zhang and Yoshino, 2010; Frankel *et al.*, 2012). The majority of adverse health effects concerning RH can be avoided by maintaining indoor levels between 40 and 60% (Arundel *et al.*, 1986).

Various techniques are used in the built environment to control and regulate CO_2 levels. They include highly engineered approaches to ventilation (Hesaraki, Myhren and Holmberg, 2015; Mateus and da Graca, 2017) as well as low-tech approaches which can include the use of plants (Raji, Tenpierik and van den Dobbelsteen, 2015; Charoenkit and Yiemwattana, 2016). A number of studies investigate a houseplants' potential to sequester CO_2 from indoor environments (Oh *et al.*, 2011; Pennisi and van Iersel, 2012; Torpy, Irga and Burchett,

2014; Gubb *et al.*, 2018). Studies vary in scale and focus – from those focusing on individual plants in experimental chambers, to room scale studies *in situ*.

A range of studies investigated houseplants' ability to sequester CO_2 in home, school, and office environments. Various combinations of houseplants were found to generally reduce room CO_2 concentrations and increase RH; however, studies rarely specify exact plant numbers and plant types. Plant species commonly used include *Dracaena deremensis, Dracaena marginata, Ficus benjamina*, *Hedera helix*, and *Spathiphyllum clevelandii* (Raza *et al.*, 1991; Lohr and PearsonMims, 1996; Jeong *et al.*, 2008; Lim *et al.*, 2009; Oh *et al.*, 2011; Pegas *et al.*, 2012).

Light levels and substrate moisture are the key factors influencing gas exchange between the plant and the environment, with 'low' light and 'dry' substrate both reducing houseplants' ability to sequester CO_2 and contribute to RH increases indoors *via* transpiration (Lawlor and Cornic, 2002; Flexas *et al.*, 2006; Gubb *et al.*, 2018). In indoor environments light levels are typically at least 100-fold lower compared to outdoors (on a clear summer day for example) and are maintained in the range of approximately 0 – 50 µmol m^{-2} s^{-1} (3700 lux) (Thimijan and Heins, 1983; Boyce and Raynham, 2009; Lai *et al.*, 2009; Hawkins, 2011). Research suggests however, that having higher indoor light levels (approximately 30 – 50 µmol m^{-2} s^{-1}; 2200 – 3700 lux) would greatly increase occupant comfort (Lai *et al.*, 2009; Huang *et al.*, 2012). As previously proposed, indoor light and particularly low light levels (0 – 500 lux) are the most limiting factor for CO_2 assimilation (Pennisi and van Iersel, 2012).

The positive contribution of plants to the reduction of CO_2 levels and RH increases indoors are based on the premise that plants function optimally and are sequestering CO_2 or

releasing water vapour at their maximum capacity. However, the main challenges for maintaining plant function in the indoor environment are 'low' indoor light levels and issues arising from plants' (mis) management, most frequently plants' being under or over watered without the correct nutrients (RHS, 2017). A few studies addressed these questions in part by investigating a wide range of light levels and their effect on CO_2 assimilation (Pennisi and van Iersel, 2012; Torpy, Irga and Burchett, 2014; Torpy, Zavattaro and Irga, 2017). However, no study to the knowledge of the author has investigated the effect of differing substrate moisture content (SMC) – namely investigating the effect of 'wet' (> 0.30 m^3 m^{-3}) and 'dry' (< 0.20 m^3 m^{-3}) SMC conditions. Additionally, previous studies have not specifically focused on plants' cultivar-level differences; this may be of interest as for many houseplant species there are a range of cultivars available, which may potentially offer augmented service compared to straight species if they are larger in size or more physiologically active.

Pennisi and van Iersel (2012) investigated the CO_2 assimilation of 17 houseplant species in both a simulated controlled environment utilising light levels of 10, 20 and 30 µmol m^{-2} s^{-1} (740, 1480, 2220 lux) and a public office building in Atlanta (USA). In the public office, the amount of CO_2 assimilated by plants varied depending on plant size. In the controlled environment, most species exhibited positive carbon assimilation over a 10-week period. The study found that in both environments larger, woody plants (such as *Ficus benjamina*) assimilated more CO_2 than herbaceous species.

Torpy et al. (2014) investigated the CO_2 assimilation of eight common indoor plant species by producing light response curves and light compensation points (LCPs) using an infra-red gas analyser. The results indicated that at least some CO_2 sequestration could be expected

from the studied species under current indoor lighting systems and plants could be effectively utilised in the built environment to sequester CO_2 given a moderate increase in the targeted lighting levels.

This research aims to improve the understanding of which taxa (i.e. plant species and cultivars) as well as which light and substrate moisture conditions are best placed to regulate indoor CO_2 and RH. Specifically, the aims of the study were to determine:

- The impact of drying substrate on CO_2 removal capacity by different taxa
- The impact of light levels on net CO_2 assimilation of taxa (i.e. to test the potential to improve the performance by supplementing indoor light levels)
- The evapo-transpiration (ET) rates of each taxon and their potential contribution to increasing indoor RH.

3.2 Materials and Methods

3.2.1 Plant material

Five common houseplant species, including two cultivars, were selected for the study to represent a range of leaf types (succulent and herbaceous), plant sizes and plant metabolisms often found in indoor environments (Table 3-1). Selected plants were 2-years old at the time of purchase in July 2016 from the RHS plant centre (Wisley, Surrey, UK), ranging between 10cm - 60cm in height, depending on the taxon. Within the species, plant height and stature were uniform (data not shown). Plants were maintained in Sylvamix GM (6:2:2 sylvafibre: growbark pine: coir; Melcourt, Tetbury, Gloucestershire, UK) in 3 L containers, with a slow release fertiliser feed (Osmocote, Marysville, OH, USA). For three months prior to experimentation plants were kept at ambient temperatures (17 – 22 °C) and

typical light levels (< 10 µmol m^{-2} s^{-1}, 500 lux) in an indoor office environment within the Crops Laboratory in the Glasshouse Complex of the School of Agriculture, Policy and Development, at the University of Reading (UK).

Table 3-1: Characteristics of the houseplants (i.e. plant species and cultivars) chosen for experiments. Leaf area (n = 2) and plant height (n = 5) are means ± SEM. Species' Latin name is given in italic and cultivar, where applicable, follows.

Species/cultivars	Family	Metabolism	Leaf area (cm^2)	Plant height (cm)
Dracaena fragrans 'Lemon Lime'	*Asparagaceae*	C3	1742 ± 91	51 ± 1
Dracaena fragrans 'Golden Coast'	*Asparagaceae*	C3	1438 ± 10	60 ± 1
Guzmania ' Indian Night'	*Bromeliaceae*	C3/CAM	1230 ± 6	32 ± 1
Hedera helix	*Araliaceae*	C3	1509 ± 243	9 ± 0
Spathiphyllum wallisii 'Bellini'	*Araceae*	C3	1766 ± 189	35 ± 1
Spathiphyllum wallisii 'Verdi'	*Araceae*	C3	5451 ± 1104	36 ± 1
Zamioculcas zamiifolia	*Araceae*	CAM	1388 ± 88	57 ± 1

3.2.2 Net leaf-level CO_2 assimilation at 'low' and 'high' indoor light levels under 'dry' and 'wet' conditions

Experiments were conducted on five plants per taxon. Measurements of the net CO_2 assimilation rate (µmol m^{-2} s^{-1}) were made using a LCPro infrared gas analyser (ADC Bioscientific, Hoddesdon, Hertfordshire, UK) on three young, fully expanded leaves per plant (with consistent leaf selection i.e. third fully expanded leaf from the plant tip) under office conditions (16.6 – 21.8 °C, RH > 35%) at 'low' and 'high' indoor light levels (Hawkins 2011; Huang et al. 2012). 'Low' ~10 µmol m^{-2} s^{-1} (~ 740 lux) lighting was achieved in the usual lighting conditions of the room (eight fluorescent lights, Osram, Munich, Germany lighting a floor area of 20 m^2). To achieve 'high' 50 µmol m^{-2} s^{-1} (3700 lux) during measurements, the photosynthetic photon flux density (i.e. light level, µmol m^{-2} s^{-1}) was supplemented at the

leaf by an external halogen source (50 W, 12 V). Each light level was administered for seven minutes and the net CO_2 assimilation rate recorded at the end of the seven-minute period.

SMC based on volume of water per volume of substrate was measured daily for each plant, in two locations per container using a SM300 capacitance-type probe connected to a HH2 Moisture Meter (Delta-T Devices, Cambridge, Cambridgeshire, UK; 0–100% range and an accuracy of ± 2.5%). At the start of the experiment, substrate moisture was at the container capacity (SMC > 30%, 0.3 m^3 m^{-3}) and plants were thus considered optimally watered (Vaz Monteiro *et al.*, 2016). Measurements were also made on 'dry' plants (SMC < 20%, 0.2 m^3 m^{-3}). Measurements were made over approximately one month.

3.2.3 Calculation of the respiration of the potted-plant microcosm

To ensure that CO_2 removal by the aboveground parts of the plant (i.e. leaves and stem) was not cancelled out by respiration of the potted-plant microcosm (PPM) (i.e. substrate and non-photosynthetic plant parts) the PPM was investigated for CO_2 contributions at both 'high' and 'low' light and under 'wet' and 'dry' SMC conditions (n = 3). The PPM respiration values were then subtracted from all the leaf CO_2 assimilation values made, to obtain the overall contribution of the plant and substrate.

Measurements of the PPM respiration were made utilising a 150 L (45 x 45 x 75 cm, 0.15 m^3) Perspex chamber (The plastic people, Leeds, West Yorkshire, UK) sealed with Swagelok fittings (Swagelok, Bristol, South Gloucestershire, UK). Enclosed inside the Perspex chamber was a HOBO MX1102 CO_2 logger (Onset Computer Corporation, Bourne, MA, U.S.A), a 12 V DC brushless fan (RS Components, Corby, Northants, UK), and a calibrated (20 – 90 % RH, 0 – 40 °C) Tinytag RH/temperature logger (Gemini data loggers, Chichester, West Sussex, UK).

This was achieved by enclosing representative substrate samples inside the chamber with an identical methodology as described in Section 4.2.2. Whereby, experiments were made on one substrate enclosed inside the Perspex chamber at a CO_2 concentration of 1000 ppm (± 10%). Experiments were for a duration of 1 hr with the CO_2 concentration logged every second. Substrate results were an average from experiments with bare substrate (and no roots) and substrate where the aboveground parts were removed (leaving roots). The author acknowledges that this is not an ideal replication but felt this 'average', was the best representative sample that could be achieved with the experimental setup.

The external RH/temperature surrounding the chamber was also monitored with another, identical Tinytag logger. Inside the chamber 'low' light levels were achieved as described in Section 3.2.2; 'high' levels were generated by two LED lights (V-TAC Europe Ltd, Sofia, Bulgaria) and measured with a calibrated light sensor (Skye instruments, Llandrindod Wells, Wales, UK). Bare substrate was prepared for the experiment as explained in Section 3.2.2. Experiments were undertaken for 2 hr, with the chamber analysed for leakage prior, during and after experimentation; leakage was found to be < 2% of the starting concentration over a 2-hr test period. Measurements were made over approximately one week.

Data obtained in Section 3.2.2 was scaled by leaf area by multiplying CO_2 assimilation (mg m^{-2} hr^{-1}) with leaf area (m^2), providing CO_2 assimilation in mg hr^{-1} $plant^{-1}$ for each taxon. Data were also corrected for PPM respiration and leakage by calculation of an average conversion value (mg hr^{-1}) for both 'wet' and 'dry' SMC conditions.

3.2.4 Generating light response curves

To generate light response curves, measurements of the net photosynthetic rate (µmol m^{-2} s^{-1}) were made as explained in Section 3.2.2 on four plants per taxon. Environmental conditions within the leaf cuvette were temperature controlled at 25 °C, ambient CO_2 concentration (~400-450 ppm) and an ambient RH of 35-45 %. Plants were prepared for the experiment as explained in Section 3.2.2, achieving a SMC > 0.30 m^3 m^{-3} and were considered optimally watered on the commencement of each experiment (Vaz Monteiro *et al.*, 2016). SMC was maintained at this level for the duration of the experiment.

To generate the light response curve light was supplemented in the following set levels : 0, 50, 300, 1200 µmol m^{-2} s^{-1} (0, 3700, 22 200 and 88 800 lux) as described in Section 3.2.2. 0 µmol m^{-2} s^{-1} was chosen to investigate each species CO_2 assimilation in the dark; 50 µmol m^{-2} s^{-1} the highest indoor light level; 300 µmol m^{-2} s^{-1} was chosen to represent the highest feasible light level which could be engineered (with supplementary artificial lighting) in an indoor environment; 1200 µmol m^{-2} s^{-1} (a sunny day in a UK climate) was chosen to present information on a plant's maximal capacity for net CO_2 assimilation. Measurements were made over approximately one week.

The light response curves were based on an equation proposed by Prioul and Chartier (1977) and were produced using the model by Lobo et al. (2013). Light compensation points – (LCPs) (which represent the light level where the CO_2 assimilation is equal to zero) (Torpy, Irga and Burchett, 2014) were calculated with the same model (Lobo et al. 2013) for all taxa apart from *Guzmania* 'Indian night', which was omitted due to very low assimilation rates and therefore, unreproducible results.

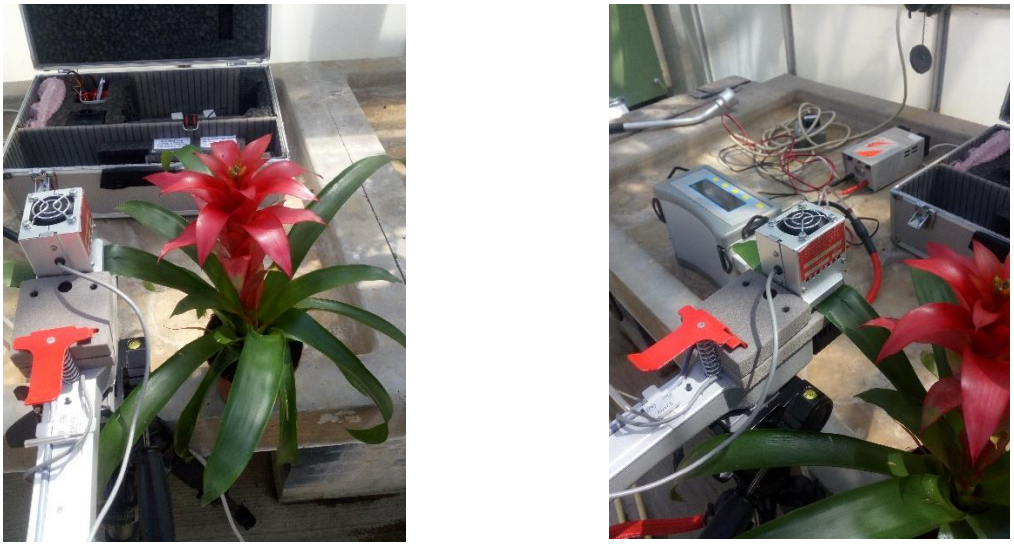


Figure 3-1: Images of the experimental setup for leaf CO_2 assimilation measurements, equipment pictured includes infra-red gas analyser, leaf cuvette and external halogen light source.

3.2.5 Plants' water use/evapo-transpiration (ET) experiments

Water use/ET of the plants were inferred by consecutive plant/pot weight measurements using a precision balance (CBK 32, Adam Equipment, Milton Keynes, Buckinghamshire, UK) under indoor office conditions (RH > 35% and at typical light levels, < 10 µmol m^{-2} s^{-1} (500 lux). Plants were prepared for the experiment as explained in Section 2.2, starting the experiment with SMC at full water-holding capacity and were not watered for the duration of the experiment. Measurements were made at 0 h and then every 24 hr over a three-week period on a whole 'plant – substrate system' (i.e. potted plant, with uncovered substrate) enabling the calculation of the water loss at each time-point. The work was interested in total potential RH contribution of the plant along with substrate, mimicking a real-life scenario of an indoor plant. Each plant was removed from the experiment when its SMC dropped < 20% (0.2 m^3 m^{-3}). Destructive measurements of LA were made using a LA meter (Delta-T Devices, Cambridge, Cambridgeshire, UK) on two plants per taxon, at the end of the

experiment. While it is appreciated that measuring the leaf area at the end of the experiment may lead to under/over-estimating assimilation measured earlier in the experiment, I was limited by the number of experimental plants that could be destructively harvested. Given that this approach was applied to all species, that the leaf areas were assessed within two months of the assimilation experiments, and that plants did not increase in size significantly over this period (as evidenced by height measurements made at the start and the end of the experiment), I believe that the risk of the error is small and evenly spread. SMC was measured daily as explained in Section 3.2.2. Water use/ET per unit leaf area (ETLA, expressed in g cm^{-2}) was calculated by dividing the ET (i.e. water loss) from a plant in a 24-hr period by the mean leaf area.

3.3 Results

3.3.1 Net leaf-level CO_2 assimilation at 'low' and 'high' indoor light levels under 'dry' and 'wet' conditions

At 'low' indoor light 'dry' *Spathiphyllum wallisii* 'Verdi' was statistically significantly respiring the most (-88 mg hr^{-1}, $p < 0.001$), and was therefore the only taxon to measure significant differences between 'dry' and 'wet' substrate. In 'dry' substrate statistically significant differences in CO_2 assimilation were measured between the cultivars of *Spathiphyllum wallisii* 'Bellini' and 'Verdi' (-20 and -61 mg hr^{-1}, respectively; $p < 0.001$). In 'wet' substrate, there were no significant differences in CO_2 between any studied taxa (Table 3-2).

At 'high' indoor light only *Spathiphyllum wallisii* 'Verdi' measured statistically significant differences between 'dry' and 'wet' substrate (-61 and 60 mg hr^{-1}, respectively; $p < 0.001$; Table 3-2). No statistically significant differences in CO_2 assimilation were measured

between cultivars under the same SMC conditions; significant differences were measured with *Spathiphyllum wallisii cvs '*Bellini' and 'Verdi' between 'dry' (-20 and -61 mg hr^{-1}, respectively) and 'wet' (12 and 60 mg hr^{-1}, respectively) SMC conditions ($p < 0.001$, Table 3-2).

Table 3-2: Net leaf-level CO_2 assimilation of each species at 'low' and 'high' indoor light (< 10 and 50 µmol m^{-2} s^{-1}) in 'wet' (> 0.30 m^3 m^{-3}) and 'dry' (< 0.20 m^3 m^{-3}) conditions. Data are a mean of five plants of each species, three young, fully expanded leaves per plant ± SEM (n=15). Data are adjusted to account for PPM respiration and chamber leakage and is normalised by leaf area (Table 3-1). (–) values signify respiration (i.e. the release of CO_2).

'Low' Light (< 10 µmol m^{-2} s^{-1})	Net CO_2 assimilation per plant (mg hr^{-1})	
Taxa	'Wet' (> 0.30 m^3 m^{-3})	'Dry' (< 0.20 m^3 m^{-3})
Dracaena fragrans 'Lemon Lime'	-17 ± 2	-36 ± 5
Dracaena fragrans 'Golden Coast'	-28 ± 3	-25 ± 2
Guzmania ' Indian Night'	-14 ± 1	-24 ± 1
Hedera helix	-10 ± 2	-27 ± 1
Spathiphyllum wallisii 'Bellini'	-15 ± 5	-23 ± 3
Spathiphyllum wallisii 'Verdi'	4 ± 5	-88 ± 33
Zamioculcas zamiifolia	-18 ± 2	-24 ± 2
'High' Light (50 µmol m^{-2} s^{-1})	Net CO_2 assimilation per plant (mg hr^{-1})	
Taxa	'Wet' (> 0.30 m^3 m^{-3})	'Dry' (< 0.20 m^3 m^{-3})
Dracaena fragrans 'Lemon Lime'	-6 ± 6	-42 ± 11
Dracaena fragrans 'Golden Coast'	-22 ± 5	-24 ± 5
Guzmania ' Indian Night'	-12 ± 7	-20 ± 1
Hedera helix	-7 ± 8	9 ± 5
Spathiphyllum wallisii 'Bellini'	12 ± 9	-20 ± 4
Spathiphyllum wallisii 'Verdi'	60 ± 31	-61 ± 25
Zamioculcas zamiifolia	-12 ± 3	-21 ± 1

3.3.2 Generating light response curves and light compensation points

Light compensation points (LCPs), which represent the light level where the net CO_2 assimilation is equal to zero, were calculated for each species (Table 3-3). Of the studied species, *Spathiphyllum wallisii* 'Verdi' and *Hedera helix* had the lowest LCPs of 20 and 31 µmol m^{-2} s^{-1} respectively. The highest LCP was recorded for *Dracaena fragrans* 'Golden Coast' (96 µmol m^{-2} s^{-1}), with both *Dracaena fragrans* 'Lemon Lime' and *Zamioculcas zamiifolia* also

having LCP values outside of the light level typically experienced in indoor environments (93 and 65 µmol m^{-2} s^{-1} respectively, Table 3-3).

Table 3-3: Light compensation points (LCPs) are means of 8 leaves per species ± SEM for each of the studied species. Comparative lux values are provided in brackets.

Taxa	LCP µmol m^{-2} s^{-1} (lux)
Dracaena fragrans 'Lemon Lime'	93 ± 7 (6882)
Dracaena fragrans 'Golden Coast'	96 ± 13 (7104)
Guzmania 'Indian Night'	N. A
Hedera helix	31 ± 4 (2294)
Spathiphyllum wallisii 'Bellini'	32 ± 12 (2368)
Spathiphyllum wallisii 'Verdi'	20 ± 10 (1480)
Zamioculcas zamiifolia	65 ± 16 (4810)

At 0 µmol m^{-2} s^{-1}, *Hedera helix* was statistically significantly respiring the most (- 1 µmol m^{-2} s^{-1}, $p < 0.001$; Figure 3-2), no significant differences were measured in net assimilation between other studied species.

At 300 µmol m^{-2} s^{-1}, all taxa were assimilating CO_2. Net assimilation was highest in *Hedera helix* (8 µmol m^{-2} s^{-1}) and was statistically significantly different to all other species($p < 0.001$). *Spathiphyllum wallisii* 'Bellini' and *S. wallisii* 'Verdi' (2 and 2 µmol m^{-2} s^{-1} respectively) measured a net assimilation that was statistically significantly higher than three other studied species (*Dracaena fragrans* 'Lemon Lime', *Dracaena fragrans* 'Golden Coast' and *Guzmania* 'Indian Night', $p < 0.001$; Figure 3-2). At this highest indoor photosynthetic photon flux density, there were no cultivar level differences within the same species in net assimilation.

At 1200 µmol m^{-2} s^{-1}, all plants were assimilating CO_2. Net assimilation was highest in *Hedera helix* (11 µmol m^{-2} s^{-1}) and was statistically significantly higher than all other species ($p < 0.001$). *Spathiphyllum wallisii* 'Bellini' (3 µmol m^{-2} s^{-1}) measured a net assimilation that was statistically significantly higher than three other studied taxa (*Dracaena fragrans* 'Lemon Lime', *Dracaena fragrans* 'Golden Coast' and *Guzmania* 'Indian Night', $p < 0.001$; Figure 3-2). Again, no net assimilation was statistically significantly different between cultivars of the same species.

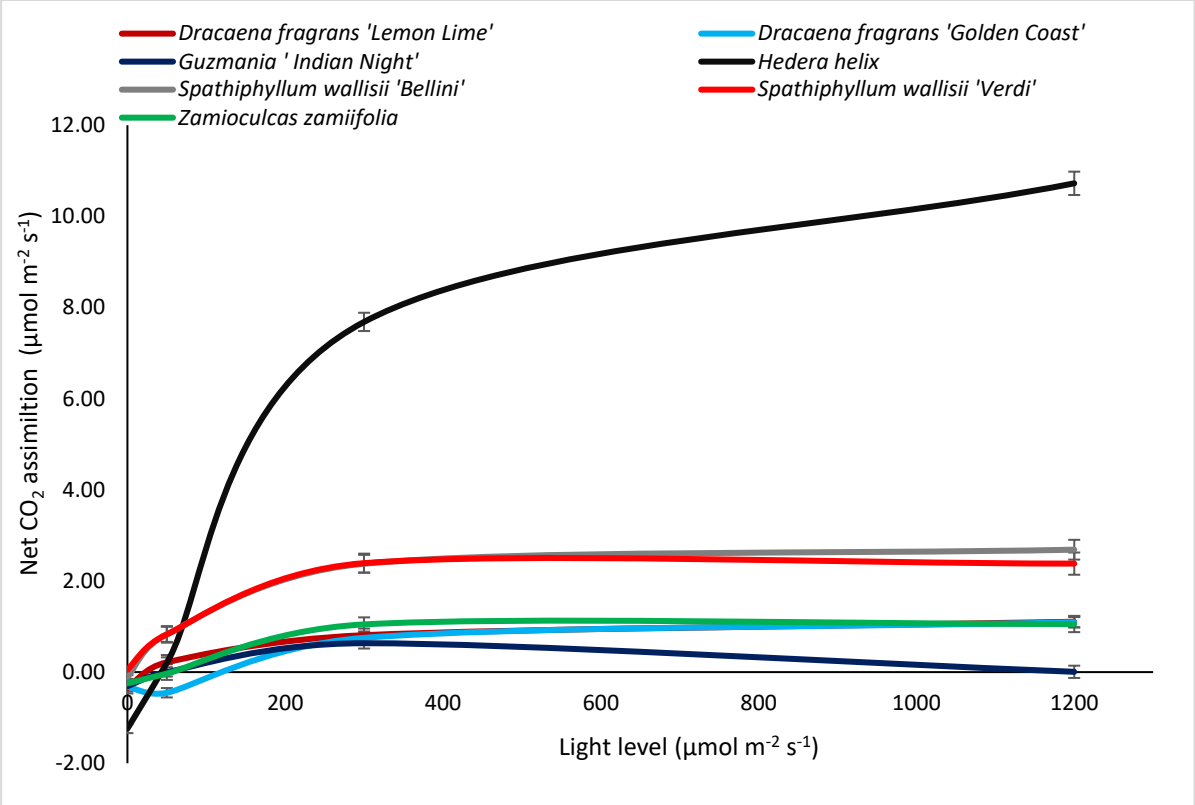


Figure 3-2: Net CO_2 assimilation across three light levels (0, 50, 300, 1200 µmol m^{-2} s^{-1}); data are a mean of four containers of each species and two young fully expanded leaves per plant (n=8). Tukey's 95% confidence intervals are used for species comparison in text – error bars represent SEM.

3.3.3 Plants' water use/evapo-transpiration experiments

In terms of ET per plant per day, when well-watered, the ET was statistically significantly higher for *Hedera helix* (70.5 g) and *Spathiphyllum wallisii* 'Verdi' (71.0 g) compared to all the other taxa ($p < 0.001$). ET per plant was also statistically significantly different between *Guzmania* 'Indian Night' (28.0 g) and *Dracaena fragrans* 'Lemon Lime' (44.3 g, $p < 0.001$); ET per plant at 24 hr was statistically significantly different between *Spathiphyllum wallisii* cultivars ($p < 0.001$; Figure 3-3).

In terms of ET per leaf area per day, when well-watered the ET was statistically significantly higher for *Hedera helix* (0.047 gcm^{-2}) in comparison to other taxa ($p < 0.001$). ET per leaf area was statistically significantly lower for *Spathiphyllum wallisii* 'Verdi' (0.013 g cm^{-2}), in comparison to the other taxa tested ($p < 0.001$) - no ET per leaf area was statistically significantly different between any other species. The ET per leaf area was statistically significantly different between one pair of cultivars: *Spathiphyllum wallisii* 'Bellini' and *Spathiphyllum wallisii* 'Verdi' (0.02 g cm^{-2} and 0.013 g cm^{-2}, respectively; $p < 0.001$; Figure 3-3).

At the time when SMC decreased to 20%, ET reduction ranged between 7% (*Spathiphyllum wallisii* 'Verdi') and 63% (*Guzmania* 'Indian Night') (data not shown). The time taken for the SMC to decrease to < 20% ranged between 10 days (*Dracaena fragrans* 'Golden Coast' and *Spathiphyllum*) and 23 days (*Zamioculcas zamiifolia*) across studied taxa (Figure 3-4).

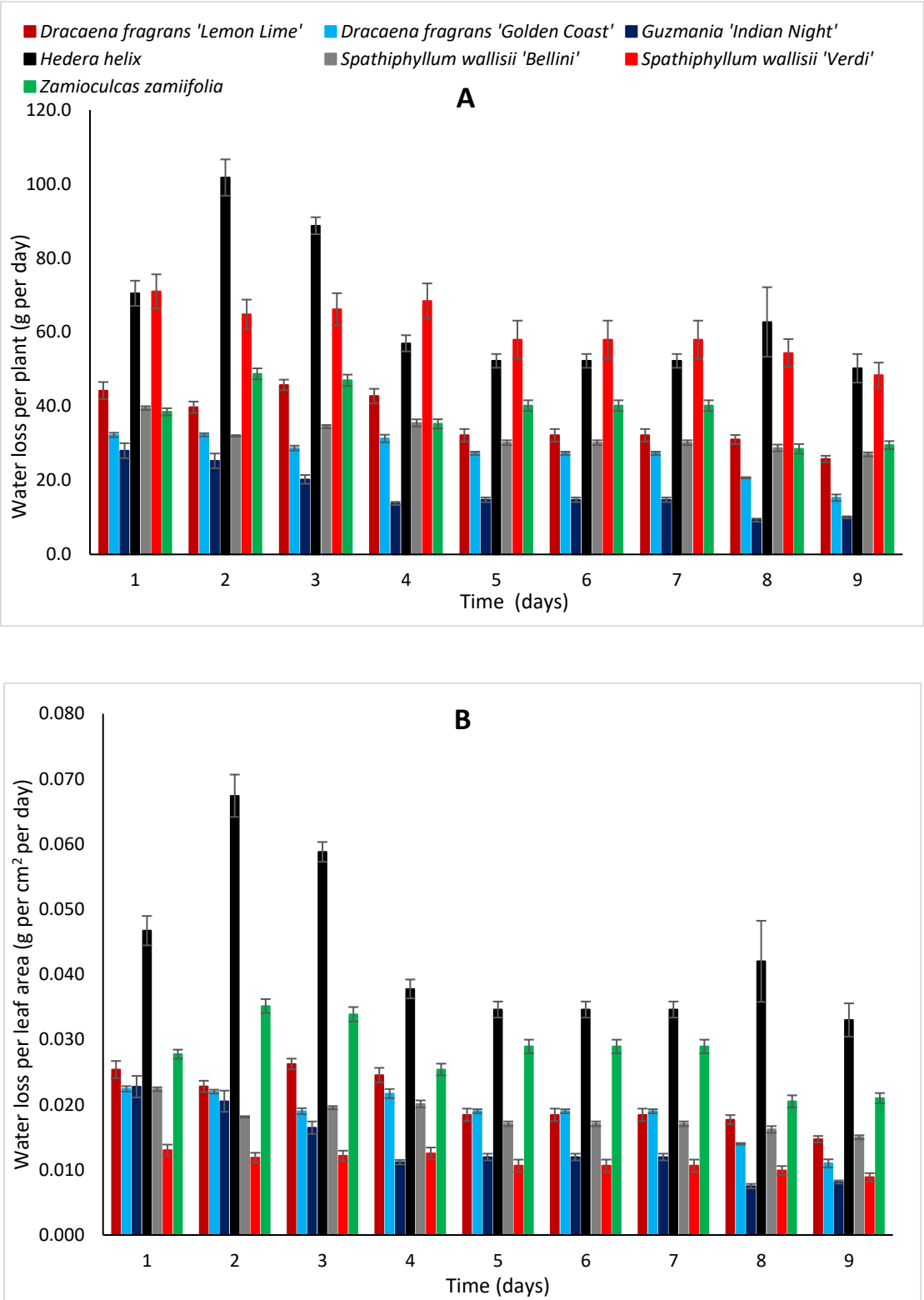


Figure 3-3: Water use per plant (A) and per leaf area (B) over a nine-day period with loss not carried over to the next day (the time taken for one species to drop below 0.2 $m^3 m^{-3}$, Figure 3-4) - data are a mean of four containers of each species ± SEM (n=4).

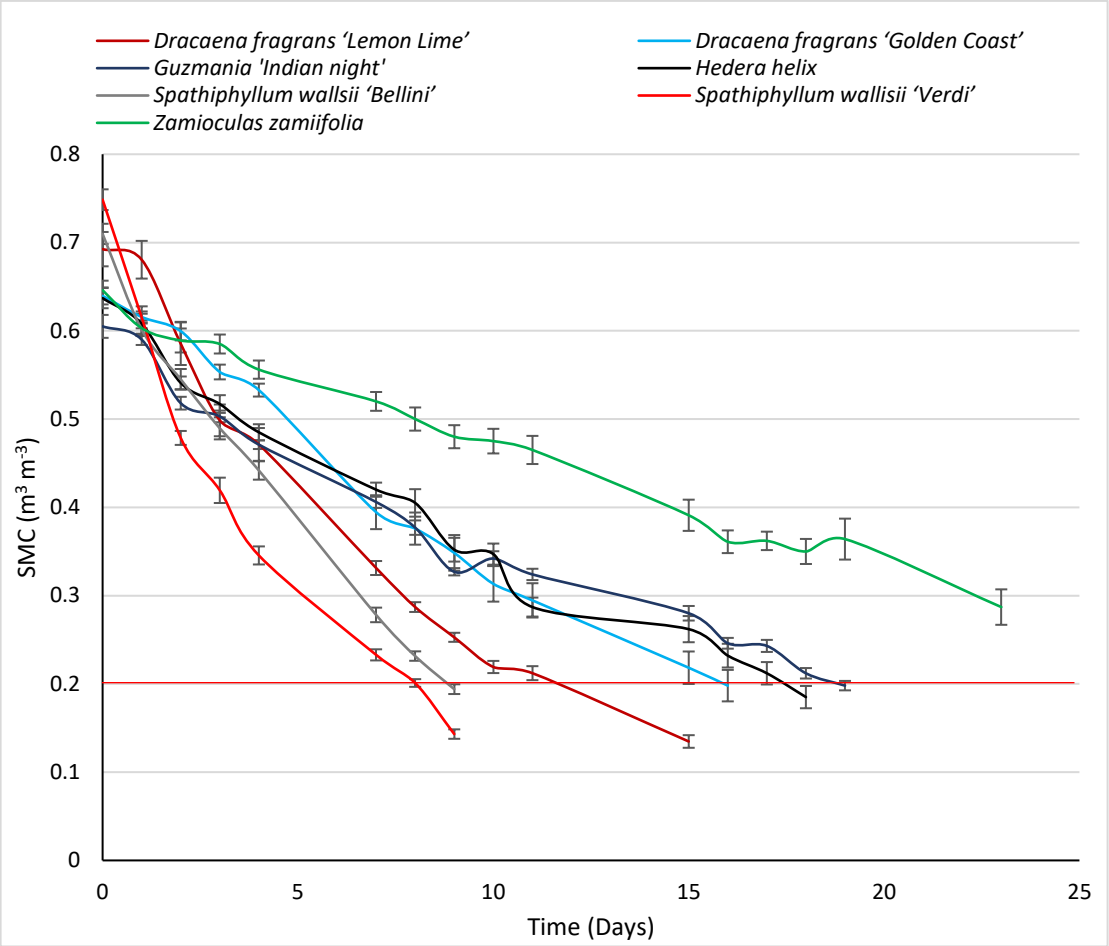


Figure 3-4: The time taken (Days) for the SMC to decrease to < 20% (< 0.2 m^3 m^{-3}) for each of the studied species ± SEM (n = 10).

3.4 Discussion

This current work presents the first insight into leaf-level CO_2 assimilation — from plants in both 'dry' and 'wet' substrate – and potential RH increases for a range of common houseplant taxa (i.e. species and cultivars), differing in structure and physiological function.

In this study it was demonstrated that little potential is offered by the studied houseplants alone to reduce CO_2 concentrations in 'low' light indoor environments – with only three taxa's light compensation points falling within the typical indoor light level range (0 – 50 µmol m^{-2} s^{-1}; 0 – 3700 lux Table 3-3). However, findings demonstrate that although respiration was generally occurring in houseplants grown in 'dry' substrate, the net CO_2 exchange recorded was extremely low and thus likely to have little or no negative impact on the CO_2 levels at a room scale. Results suggest that increasing light levels to a technically feasible 300 µmol m^{-2} s^{-1} (22 200 lux e.g. through use of supplementary lighting) would provide a significant increase in CO_2 assimilation in most of the studied taxa. The study also indicates that the best performing taxa for CO_2 assimilation will also contribute the most to raising RH indoors.

From the results of this study I estimated the mass (in grams) of CO_2 removed per hour, per plant and per m^2 of each taxon. In home and office environments, each person contributes 30g (CO_2)/hour and 36g (CO_2)/hour, respectively (Persily and de Jonge, 2017) and these different values are consequences of the level of individual's activity in various environments. Using both these values, I calculated the number of plants required to remove 10% of a single person's CO_2 contribution at the 'very high' (300 µmol m^{-2} s^{-1}) indoor light level (Table 3-4). The plant numbers range from 15 (for more active plants like *Hedera*

and *Spathiphyllum*) to >100 for physiologically less active plants, highlighting how correct plant choice can result in a different air quality outcome. Of the taxa investigated *Guzmania, Dracaena* and *Zamioculcas* would be better placed to provide services other than CO_2 reduction (e.g. pollutant sequestration (Yang et al. 2009; Kim et al. 2010)). *Hedera* and *Spathyphyllum* would have more effect on room-level CO_2 exchange, and in numbers which can be realistically installed in small living walls. Estimates of the number of plants required to remove the CO_2 generated by human contributions were also made by Pennisi and van Iersel (2012) and Torpy et al. (2014) albeit, at different experimental scales making direct comparisons difficult.

In typical indoor environments with 'low' light levels, only one taxon, in 'wet' substrate conditions was assimilating CO_2 (*Spathiphyllum wallisii* 'Verdi') and would contribute to CO_2 concentration reduction (3.9 mg hr^{-1}, respectively; Table 3-2). Additionally, only three taxa were found to possess light compensation points that fall within the range of typical indoor light levels (i.e. *Hedera helix and Spathiphyllum wallisii* 'Verdi' and 'Bellini'). Both *Hedera helix and Spathiphyllum wallisii* would require an unrealistic number of plants to see any significant CO_2 concentration reduction (data not shown); at typical 'low' indoor light levels, the study indicates that a plants' potential benefits psychologically or in productivity terms (Thomsen, Sonderstrup-Andersen and Muller, 2011; Raanaas *et al.*, 2011; Nieuwenhuis *et al.*, 2014) would be more important than their contribution to indoor CO_2 removal.

In typical 'low' light indoor environments, when grown in 'dry' substrate, all studied taxa were respiring. The results also indicated that in the range of typically observed indoor light levels, six of the studied species (*Dracaena fragrans* cvs 'Lemon Lime' and 'Golden Coast',

Guzmania 'Indian Night', *Hedera helix, Spathiphyllum wallisii* 'Bellini' and *Zamioculcas zamiifolia*) were respiring in both 'dry' and 'wet' SMC conditions (Table 3-2). The (mis) management and under watering of houseplants is anecdotally a common problem; therefore, determining if a 'dry' houseplant is releasing significant amounts of CO_2 into an indoor environment and detrimentally impacting health is important; results however, suggest this is not the case. In 'dry' SMC conditions, in typical office light, *Spathiphyllum wallisii* 'Verdi' was releasing the most CO_2 into the indoor environment out of all studied taxa at 0.0876 g hr^{-1}. In comparison, a single person, in an office environment would release 36 g/hour into the indoor environment (Persily and de Jonge 2017). This confirms that in typical office light conditions – even for plants growing in drying substrate – the contribution of plants to room-level CO_2 is negligible.

At a 'high' indoor light level (50 µmol m^{-2} s^{-1}), a greater net CO_2 assimilation was generally measured for all taxa, but no statistically significant differences were found between cultivars of the same species in 'dry' or 'wet' conditions. Although measurements were only made under 'wet' SMC conditions, this trend for the lack of cultivar differences continued at higher light levels of 300 and 1200 µmol m^{-2} s^{-1} suggesting that cultivar level differences were not pronounced in this study.

Results suggest that for most studied taxa, light saturation occurs at around 300 µmol m^{-2} s^{-1} and further increases beyond this show little difference in assimilation terms (Figure 3-2). As discussed in Torpy et al. (2014) targeted indoor lighting could be used to maximise a houseplants CO_2 assimilation potential. Extensive research has been undertaken into various light systems for plant cultivation and development on indoor living walls but not specifically

with potted houseplants or concerning CO_2 assimilation (Yeh and Chung, 2009; Egea *et al.*, 2014). The findings support the notion that increased light levels maximise plant gas exchange and suggest future research should investigate the suitability of testing targeted lighting installations in indoor environments. Light compensation points calculated in this study are generally higher, but comparable with other indoor species previously tested (Burton, Pennisi and van Iersel, 2007; Pennisi and van Iersel, 2012; Torpy, Irga and Burchett, 2014; Torpy, Zavattaro and Irga, 2017; Tan *et al.*, 2017).

Earlier attempts at estimating the CO_2 removal of houseplants did not take into account ambient CO_2 concentrations or consider the effects of substrate moisture on CO_2 assimilation (Pennisi and van Iersel, 2012). A more robust study by Torpy et al. (2014) investigated several factors which could influence assimilation including different acclimatisation treatments, the respiration of the 'potted-plant microcosm', but again did not consider impact of substrate moisture conditions. Other studies did not specify the exact number or type of houseplant (Lim *et al.*, 2009; Pegas *et al.*, 2012) which contributed to any CO_2 concentration reduction or, only considered a single light level (Oh *et al.*, 2011).

The results from the ET experiment indicate that the best performing species in CO_2 assimilation terms (*Hedera helix* and *Spathiphyllum wallisii* 'Verdi') both have the highest ET rates per plant. However, the comparative water use per area results show *Spathiphyllum wallisii* 'Verdi' having the lowest ET *per leaf area*; this species is therefore, inherently more water use efficient and only uses more water *per plant* due to its large size. This study found a difference between the *Spathiphyllum wallisii* cultivar pair in terms of water use per plant and per area – with no difference per plant or per area measured for the *Dracaena fragrans*

pair. This confirms the hypothesis that inherent physiological differences can be measured in water use terms down to a cultivar level. The results also suggest that certain species (i.e. *Spathiphyllum wallisii* 'Verdi') do not restrict their water loss under water stress conditions (SMC < 20%). *Spathiphyllum wallisii* 'Verdi' would therefore, in a drying substrate, continue to contribute the most to RH increases. To achieve the optimal function for the studied taxa, which would then support biggest improvements in IAQ – based on results from Section 3.3.3 and experience – I suggest a watering regime of 200 ml per week for all studied species other than *Spathiphyllum wallisii* 'Verdi' and *Hedera helix*, where 250 ml is recommended twice a week. Future studies should also evaluate the CO_2 assimilation ability of other more physiologically active, vigorous species (i.e. *Osmunda japonica*, *Selaginella tamariscina* and *Hemigraphis alternata*), which also performed well in other pollutant sequestration experiments (Yang *et al.*, 2009; Kim *et al.*, 2010) under much higher indoor light levels (~300 µmol m^{-2} s^{-1}).

From the results of the ET experiment the contribution of studied taxa to raising RH indoors was estimated. Calculations of the amount of water vapour in the air were made through the equation: RH (%) = 100 * actual vapour density (g m^{-3}) / saturation vapour density (g m^{-3}) (using a saturation vapour density of 19.1 g m^{-3} at 22 °C) (Galindo *et al.*, 2005). A RH of 40 – 60% is considered optimal in terms of human health (Arundel *et al.*, 1986), therefore the number of plants were calculated – per taxon - required to raise RH from 40 to 60 % in a static 100 m^3 office (Table 3-5). Calculations assume that 100% of the water vapour 'lost' by species (Figure 3-3 (A)) was released into the surrounding environment. The results do not take into account the impact of ventilation, occupancy or the feedback effect (i.e. as RH increases plants release less water vapour into the indoor environment). These calculations

are intended to act as a guide on how the studied taxa could influence RH indoors. With results indicating that five *Spathiphyllum wallisii* 'Verdi' or *Hedera helix* plants growing in an unmulched (i.e. uncovered) GM - over a 24-hr hour period - could raise the RH from 40 to 60% (Table 3-5). It also suggests that less physiologically active plants (such as *Guzmania, Dracaena* and *Zamioculcas*) could be used in larger numbers (10+) as part of installations such as indoor living walls within even smaller offices, without a risk of office RH raising above 60%. Conversely, *Hedera* and large *Spathiphyllum* cultivars would be suitable in smaller numbers (5 or below) or in larger rooms with greater overall volume where their RH-influencing effect would be diluted.

Table 3-4: Net CO_2 assimilation (mg hr^{-1}) of each species and number of species required to remove 10 % of the CO_2 generated per person at 'very high' indoor light (300 µmol m^{-2} s^{-1}, 22200 lux) in 'wet' (> 0.30 m^3 m^{-3}) conditions. Data is taken from Figure 3-2 and adjusted to account for PPM respiration and chamber leakage and is normalised by leaf area (Table 3-1). Plant numbers for each taxon were calculated by dividing the 30 g (CO_2)/hour or 36 g (CO_2)/hour exhaled per person in home and office environments respectively (Persily and de Jonge 2017) by the net CO_2 assimilation of each taxon (mg hr^{-1}).

'Very high' Light (300 µmol m^{-2} s^{-1})	mg hr^{-1}	Number of plants	
Taxa	'Wet' (> 0.30 m^3 m^{-3})	Home	Office
Dracaena fragrans 'Lemon Lime'	11 ± 8	273	327
Dracaena fragrans 'Golden Coast'	6 ± 6	500	600
Guzmania ' Indian Night'	1 ± 5	3000	3600
Hedera helix	172 ± 10	17	21
Spathiphyllum wallisii 'Bellini'	55 ± 11	55	65
Spathiphyllum wallisii 'Verdi'	195 ± 36	15	18
Zamioculcas zamiifolia	12 ± 7	250	300

Table 3-5: Number of plants required to raise the RH from 40 to 60% in a static 100 m^3 office. Numbers of plants were generated from data in Figure 3-3 at a temperature of 22 °C, where ventilation, occupancy and the feedback effect were not considered. Calculations of the amount of water vapour in the air were made through the equation: RH (%) = 100 * actual vapour density (g m^{-3}) / saturation vapour density (g m^{-3}) (using a saturation vapour density of 19.1 g m^{-3} at 22 °C) (Galindo et al. 2005).

Species/cultivar	Number of Plants
Dracaena fragrans 'Lemon Lime'	9
Dracaena fragrans 'Golden Coast'	12
Guzmania ' Indian Night'	14
Hedera helix	5
Spathiphyllum wallisii 'Bellini'	10
Spathiphyllum wallisii 'Verdi'	5
Zamioculcas zamiifolia	10

3.5 Conclusion

The results indicate that net CO_2 assimilation of all studied plants was generally 'low', with *Spathiphyllum* cultivars and *Hedera helix* removing most CO_2.

While CO_2 assimilation of plants in 'wet' substrate was higher than in 'dry' conditions, in practical terms however (*i.e*. when considering the plant's potential to influence indoor CO_2 levels), net CO_2 assimilation differences between 'dry' and 'wet' plants at 'high' and 'low' indoor light levels were negligible for the taxa studied. Light compensation points were in the typical indoor light range for both *Spathiphyllum wallsii* 'Verdi' and *Hedera helix,* suggesting that these plants would be best suited to provide most CO_2 removal in a typical indoor setting. Additionally, both, per plant, had the highest transpiration rates, suggesting the highest potential for influencing the RH. Finally, this study indicates that increasing

indoor light levels to 300 μmol m^{-2} s^{-1} would, in most taxa, have a significant impact on the potential for houseplants to assimilate CO_2 and increase RH in indoor environments.

4.0 Interaction between plant species and substrate type in the removal of CO_2 indoors

This chapter was based on the published paper presented in Appendix E

4.1 Introduction

Elevated indoor concentrations of CO_2 (> 600 ppm) are harmful to human health, increase absenteeism and reduce cognitive performance (Seppanen, Fisk and Mendell, 1999; Erdmann and Apte, 2004; Shendell *et al.*, 2004; Shaughnessy *et al.*, 2006; Gaihre *et al.*, 2014; Zhang, Wargocki and Lian, 2017). Traditional ventilation systems are designed to keep CO_2 concentrations near-ambient with outdoor air infiltration albeit, increasing building energy consumption (Perez-Lombard, Ortiz and Pout, 2008). Indoor plants can act as a simple low-cost form of ventilation, reducing indoor requirements (by ~ 6%) with CO_2 removal, but only under certain environmental conditions i.e. a very high light level (~ 22200 lux) – as confirmed by several previous studies (Torpy, Irga and Burchett, 2014; Torpy, Zavattaro and Irga, 2017; Gubb *et al.*, 2018).

Numerous health guidelines exist for maximum safe CO_2 concentrations, the lowest of these being 1000 ppm (over an 8-hour period) produced by the American society of heating, refrigeration and air-conditioning engineers (ASHRAE) (Torpy, Irga and Burchett, 2014; Torpy, Zavattaro and Irga, 2017). This concentration is often exceeded in indoor environments, especially classrooms (Shendell *et al.*, 2004; Gaihre *et al.*, 2014). Concentrations indoors are typically less than 2000 – 2500 ppm, but can rise as high as 5000 ppm, with the main source of CO_2 indoors being humans themselves (Zhang, Wargocki and Lian, 2017).

Elevated CO_2 concentrations (> 600 ppm) can cause an array of health issues including eye irritation, mucus membrane symptoms (i.e. sore/dry throat, dry eyes and sneezing) and respiratory problems (i.e. tight chest, wheezing/coughing and shortness of breath) (Seppanen, Fisk and Mendell, 1999; Erdmann and Apte, 2004; Tsai, Lin and Chan, 2012). Additionally, elevated concentrations have been associated with declines in cognitive function (at ~ 950 ppm); absenteeism, with increases of 100 ppm associated with a reduced annual attendance of half a day per annum and reductions in cognitive performance, with concentrations of 600 – 1000 ppm found to significantly reduce decision making ability (Shaughnessy *et al.*, 2006; Satish *et al.*, 2012; Gaihre *et al.*, 2014; Vehvilainen *et al.*, 2016; Allen *et al.*, 2016).

Numerous studies have shown that light levels significantly influence a plants ability to remove CO_2 via their impact on stomata as a main pathway for CO_2 uptake (Pennisi and van Iersel, 2012; Torpy, Irga and Burchett, 2014; Torpy, Zavattaro and Irga, 2017; Gubb *et al.,* 2018). Indoors, the light level is typically between 0 – 500 lux but, can be as high as 3000 lux in certain workplace environments (Boyce and Raynham, 2009; Lai *et al.*, 2009; Hawkins, 2011; Huang *et al.*, 2012). Often, supplementary lighting is required to support specific plant installations such as a green-wall, where higher light levels are utilised above the installation and not throughout the entire room – this supplementary light can be engineered at least as high as 22200 lux (Gubb *et al.*, 2018). Plants' under- or over-watering also affects a plant's ability to remove CO_2 (Sailsbury and Ross, 1991) but previous work showed that indoors light levels was the primary driver of CO_2 uptake and the soil drying had a smaller impact (Gubb *et al.*, 2018).

Plants remove airborne pollutants via four different pathways, the aboveground plant part, the roots, and two of which directly involve the substrate namely, the substrate itself, along with the microbial activity within the substrate (Cruz *et al.*, 2014). It can therefore be expected that both the type and condition (wet/dry) of the substrate will affect a plants CO_2 removal ability. Experiments investigating the ability of plants to remove volatile organic compounds (VOCs) have found that the removal of VOCs is predominately associated with the microflora in the substrate, plants themselves are only utilised in-directly to maintain and support substrate microorganisms (Wood *et al.*, 2002; Orwell *et al.*, 2004; Kim *et al.*, 2008; Cruz *et al.*, 2014; Irga, Pettit and Torpy, 2018; Kim *et al.*, 2018); these microorganisms – especially those associated with the root system – have been shown to metabolise an array of different pollutants (Weyens *et al.*, 2015).

Various substrates are available in the UK for growing indoor plants, each can be classified between two extremes, peat and peat-free. Peat – an organic material – is a limited resource hence, attempts by the UK government for voluntary phasing out of peat by 2030 (Defra, 2018). However, peat-based substrates are still commonly used across the UK because of their uniformity, providing easier water management (Schmilewski, 2008; Alexander, Williams and Nevison, 2013). Peat has a very high-water capacity, unlike certain peat-alternatives such as coir, sand and wood fibres (Schmilewski, 2008). As several studies have linked soil moisture to microbial respiration an investigation into moisture content is of significance to CO_2 removal (Cook, Orchard and Corderoy, 1985; Manzoni, 2012). Furthermore, with different substrate types able to support different microorganisms (Zhang *et al.*, 2013) it was hypothesised that differences in removal would be measured between peat and a peat free substrate.

The type of substrate used in prior CO_2 removal experiments has often been peat-free the author wanted to test plants potted in differing substrates, both with and without peat. Therefore, two different substrates – from now on referred to as peat and peat-free – were chosen for this experiment to determine if they affected plants' ability to remove CO_2 and the concentration of CO_2 in the external environment. It was hypothesised that growing the same taxa in differing substrates might provide differing CO_2 removal abilities.

If houseplants are to reduce elevated CO_2 concentrations, they must be functioning optimally i.e. experience appropriate light levels, feeding and watering (i.e. substrate moisture content - SMC). A few studies have investigated these issues in part, testing various plants potted in different peat-free substrates (Torpy, Irga and Burchett, 2014; Torpy, Zavattaro and Irga, 2017; Gubb *et al.*, 2018) However, none to the knowledge of the author have investigated how different substrate types affect CO_2 removal ability.

Torpy, Irga and Burchett (2014) determined the light response curves of eight common plants potted in a peat-free substrate consisting of composted hardwood, sawdust, composted bark fines, and coarse river sand (2:2:1). The authors suggested that in typical 'low' indoor light some CO_2 removal could be expected but, moderately increasing light levels would mean the studied plants could be effectively utilised in a built environment setting.

Torpy, Zavattaro and Irga (2017) also investigated the ability of two taxa (*Chlorophytum comosum* and *Epipremnum aureum*) potted in a peat-free substrate comprising of coconut fibre – as part of an active green-wall – to remove 1000 ppmv of CO_2 at light levels of 50 and 250 µmol m^{-2} s^{-1}. The study found removal was much more effective at 250 µmol m^{-2} s^{-1} and

found that removal from a 5 m^2 wall of *C. comosum* could balance the respiratory emissions of a full-time occupant.

This study aims to present which houseplants together with the substrate they are grown in (from now on referred to as houseplants or taxa) can reduce a CO_2 concentration of 1000 ppm at 'very high' (~ 22200 lux), typical 'low' light (~ 500 lux) and 'no' indoor light (0 lux) in 'wet' (SMC > 30 %, 0.3 m^3 m^{-3}) and 'dry' (SMC < 20 %, 0.2 m^3 m^{-3}) SMC conditions and also, for the first time – to the knowledge of the author – with two different substrates. 0 lux was chosen to investigate CO_2 assimilation/respiration in the dark; ~ 500 lux was chosen to represent typical office conditions; 22200 lux was chosen to represent the highest technically feasible light level which could be engineered indoors (with supplementary artificial lighting) (Torpy, Zavattaro and Irga, 2017).

This experiment was undertaken on a whole plant/substrate scale as opposed to leaf-level experiments investigated in prior work (Gubb *et al.*, 2018). It was hypothesised that experiments on this larger scale would provide more accurate estimations for how plants can influence 'room-scale' concentrations of CO_2. Additionally, this study looks to highlight if substrate type can make a difference to the CO_2 removal ability of taxa and justify the need for further research with a more extensive range of appropriate substrates in subsequent studies.

4.2 Materials and Methods

4.2.1 Plant material

Three common houseplant taxa (*Dracaena fragrans* 'Golden Coast', *Hedera helix* and *Spathiphyllum wallisii* 'Verdi') which were shown in the previous study to have a range of

CO_2 removal capacities were selected for this study (Chapter 3). They represented a range of leaf types (succulent and herbaceous) and plant sizes (Table 4-1). Plants were maintained in peat-free substrate i.e. Sylvamix GM (6:2:2 sylvafibre: growbark pine: coir; Melcourt, Tetbury, Gloucestershire, UK) or in peat substrate i.e. Clover professional pot bedding substrate (100% Irish Moss Peat; Clover, Dungannon, Co. Tyrone, UK) in 3 L containers, with a slow release fertiliser feed (for 6 months, Osmocote, Marysville, OH, USA). Selected houseplants were one-year old at the time of purchase. Within the taxon, plant height and stature were uniform (data not shown). Prior to experimentation (for > 90 days) plants were kept at room temperatures (17 – 22 °C) and 'low' light levels (~ 500 lux) in an indoor office environment within the Crops Laboratory in the Glasshouse Complex of the School of Agriculture, Policy and Development, at the University of Reading (UK) and for a shorter period of time under similar conditions at the School of Geography, Earth and Environmental Sciences at the University of Birmingham (UK). *Hedera helix* could not be successfully grown in the peat substrate and was omitted from the study in this substrate after several failed attempts.

Table 4-1: Characteristics of the houseplants chosen for experiments in both peat and peat-free substrate. Leaf area (n = 3) and plant height (n = 5) are means ± SEM. Species' botanical Latin name is given in italic and cultivar, where applicable, follows.

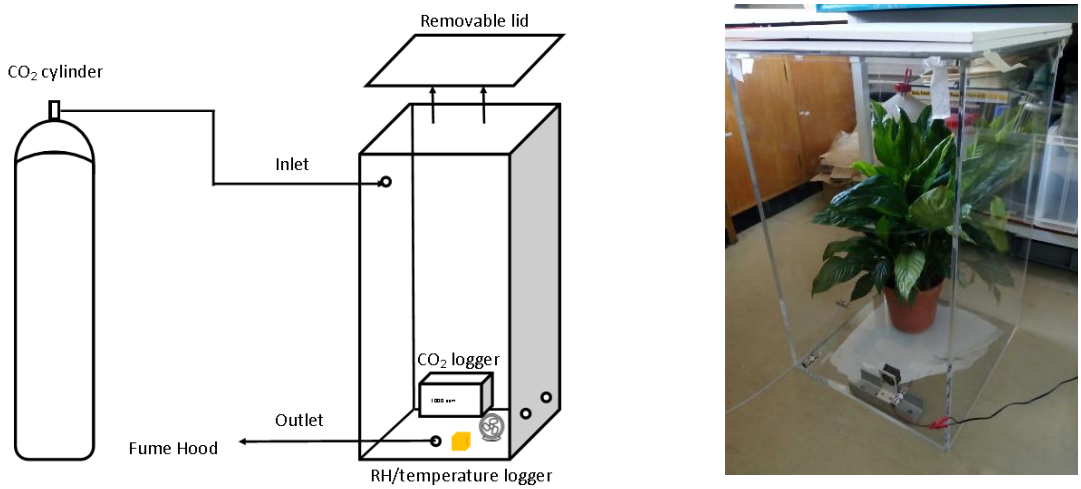
Taxa – Peat-free	Family	Metabolism	Leaf area (cm^2)	Plant height (cm)
Dracaena fragrans 'Golden Coast'	*Asparagaceae*	C3	4057 ± 337	83 ± 1
Hedera helix	*Araliaceae*	C3	1542 ± 122	8 ± 1
Spathiphyllum wallisii 'Verdi'	*Araceae*	C3	6033 ± 128	38 ± 1

Taxa – Peat	Family	Metabolism	Leaf area (cm^2)	Plant height (cm)
Dracaena fragrans 'Golden Coast'	*Asparagaceae*	C3	1417 ± 112	48 ± 1
Spathiphyllum wallisii 'Verdi'	*Araceae*	C3	2591 ± 442	42 ± 2

4.2.2 CO_2 Chamber experiments

Experiments were carried out in an experimental laboratory with a non-bypass fume hood at the University of Reading (UK). The experimental setup (Figure 4-1) consisted of a ~150 L (45 x 45 x 75 cm, 0.15 m^3) Perspex chamber (The plastic people, Leeds, West Yorkshire, UK) connected to a CO_2 cylinder (CO_2 > 99% purity, Air Liquide, Coleshill, West Midlands, U.K) with a combination of Teflon tubing (¼ inch diameter) and Swagelok fittings (Swagelok, Bristol, South Gloucestershire, UK). Enclosed inside the Perspex chamber was a HOBO MX1102 CO_2 logger (Onset Computer Corporation, Bourne, MA, U.S.A), a 12 V DC brushless fan (RS Components, Corby, Northants, UK), 500 g of silica gel (Sigma – Aldrich Company Ltd, Gillingham, Dorset, U.K) and a calibrated (20 – 90 % RH, 0 – 40 °C) Tinytag RH/temperature logger (Gemini data loggers, Chichester, West Sussex, UK). The external RH/temperature surrounding the chamber was also monitored with another, identical Tinytag logger. Inside the chamber 'no' (0 lux) light was achieved by undertaking at experiments at night; 'low' (~

500 lux) light levels were achieved in the usual lighting conditions of the room (four fluorescent ceiling lights, Osram, Munich, Germany lighting a floor area of 11 m^2); 'very high' levels were achieved with two LED lights (V-TAC Europe Ltd, Sofia, Bulgaria) which were positioned on stands externally, one at an ~ 30 cm height above the chamber and another ~ 30 cm from the side of the chamber. Colour temperature of those lights was 6000k and both lights combined produced a 'very high' (~ 22200 lux) light level inside the chamber — all three levels were measured with a calibrated light sensor (SKP 200, Skye instruments, Llandrindod Wells, Wales, UK). This 'very high' light level approximately corresponds to the light saturation for the studied species on a light response curve (Gubb *et al.*, 2018) and was chosen to represent the highest feasible light level which could be engineered (with supplementary artificial lighting) in an indoor environment.



A B

Figure 4-1: Schematic diagram (A) and image (B) of the CO_2 chamber experimental setup

Measurements of the ability of studied taxa to reduce CO_2 concentrations of 1000 ppm (ASHRAE recommended maximum 8 hr exposure guideline taken from (Torpy, Irga and Burchett, 2014; Torpy, Zavattaro and Irga, 2017) were undertaken on either three ('no' and 'low' light) or five ('very high' light) plants per taxon. Taxa were prepared for experiments with substrate moisture at the container capacity (SMC > 30%) and plants were thus considered optimally watered on the commencement of each experiment (Vaz Monteiro *et al.*, 2016). Measurements were also made on each houseplant's 'dry' substrate (SMC < 20%) after a period of drying – the length of which was dependent on the type of plant and its inherent evapo-transpiration rate (Gubb *et al.*, 2018). To ascertain when each taxon was 'dry' SMC was measured prior to experimentation for each plant, in two locations per container using a SM300 capacitance-type probe connected to a HH2 Moisture Meter (Delta-T Devices, Cambridge, Cambridgeshire, UK; 0–100% range and an accuracy of ± 2.5%). Experiments were made on one whole 'plant – substrate system' (i.e. potted plant, with uncovered substrate) enclosed inside the Perspex chamber at a CO_2 concentration of 1000 ppm (± 10%). Experiments were for a duration of 1 hr with the CO_2 concentration logged every second. Appropriate 'control' measurements were run at all three light levels on both the empty chamber and pot with substrate, but no plant (in both 'wet' and 'dry' SMC). The number of runs with only substrate and pot were either three for 'no' and 'low' light or five for 'very high' light.

Experimental parameters for each lighting treatment were as follows: 'no' light, ambient (CO_2 < 500 ppm; Temperature 17 – 26 °C; RH 23 – 64 %) and inside chamber (Temperature 17 – 26 °C; RH 31 – 90 %); 'low' light, ambient (CO_2 < 500 ppm; Temperature 13 – 23 °C; RH 24 – 61 %) and inside chamber (Temperature 13 – 24 °C; RH 36 – 90 %); and high light,

ambient (CO_2 < 500 ppm; Temperature 15 – 22 °C; RH 21 – 60 %) and inside chamber (Temperature 15 – 24 °C; RH 32 – 90 %). The chamber was also analysed for leakage prior, during and after experimentation; leakage was found to be < 5% of the starting concentration over the test period. All results were corrected for leakage. This was achieved – for 'no' and 'low' light - by adding the average CO_2 concentration lost through leakage (ppm) to the amount of CO_2 respired by each taxon (ppm) – correcting for the fact that each taxon would have measured a greater concentration of CO_2 if the chamber was airtight. The opposite was done for 'very high' light, correcting for the fact that each taxon would have removed more CO_2 if the chamber was airtight.

Based on the findings of the previous leaf-level work with the same taxa (Chapter 3)(Gubb *et al*., 2018) it was hypothesised that at 'no' and 'low' indoor light levels taxa would increase CO_2 concentrations within the enclosure. The CO_2 concentration (ppm hr^{-1}) removed by each taxon were calculated with the data measured directly every second by the appropriate logger and divided by the leaf area in m^2 presented in Table 4-1 to give a unit of ppm m^{-2} h^{-1}.

4.3 Results

4.3.1 CO_2 chamber experiments – 'no' light

At 'no' indoor light no species reduced CO_2 from the initial 1000 ppm concentration, and the CO_2 concentration inside the chamber increased with all treatments; no statistically significant differences in concentration were measured within species between 'dry' or 'wet' conditions (Table 4-2). Additionally, statistical differences were measured between the peat and peat free substrates for *Dracaena fragrans* 'Golden Coast' in both 'dry' (331 and 138

ppm m^{-2} hr^{-1}, respectively; Table 4-2) and 'wet' conditions (332 and 151 ppm m^{-2} hr^{-1}, respectively; Table 4-2).

4.3.2 CO_2 chamber experiments – 'low' light

At 'low' indoor light *Spathiphyllum wallisii* 'Verdi' potted in the peat substrate reduced the concentration of CO_2 from the initial 1000 ppm concentration ('dry' and 'wet', 43 and 1 ppm m^{-2} hr^{-1}, respectively; Table 4-3). All other plant/substrate combinations increased the CO_2 concentration. Statistically significant differences were measured within taxon between 'dry' and 'wet' conditions for *Hedera helix* in the peat-free substrate (379 and 518 ppm m^{-2} hr^{-1}, respectively; Table 4-3). Additionally, statistical differences in removal were measured between the peat and peat-free substrates for *Spathiphyllum wallisii* 'Verdi' in 'wet' conditions (227 and -1 ppm m^{-2} hr^{-1}, respectively; $p = 0.03$; Table 4-3) but not 'dry' (192 and -43 ppm m^{-2} hr^{-1}, respectively, $p = 0.126$,; Table 4-3) and for *Dracaena fragrans* 'Golden Coast' in 'dry' conditions (147 and 7 ppm m^{-2} hr^{-1}, respectively, Table 4-3).

4.3.3 CO_2 chamber experiments – 'very high' light

At 'very high' indoor light all treatments reduced the concentration of CO_2 from the initial 1000 ppm. Significant differences were measured in CO_2 reduction between all taxa, under both 'dry' and 'wet' conditions between the peat and peat-free substrates. The range of removal rates was the smallest at 15 mins and the largest at 60 mins in both 'wet' and 'dry' conditions. After 15 minutes, no statistically significant differences in CO_2 reduction were measured within the same taxon in either substrate between 'dry' and 'wet' conditions. After 60 minutes, statistically significant differences were measured in both *Spathiphyllum*

and *Dracaena* potted in the peat substrate between 'dry' and 'wet' conditions, but not in the peat-free substrate (Figure 4-2).

In 'wet' conditions after 15 minutes, no statistically significant differences were measured between any studied taxa in either peat or peat-free substrate (Figure 4-2, $p = 0.550$). After 60 minutes, *Dracaena fragrans* 'Golden Coast' in the peat substrate reduced statistically the largest amount of CO_2 from the initial 1000 ppm concentration (1420 ppm m^{-2} hr^{-1}; $p < 0.001$). No statistically significant differences in CO_2 removal were measured between *Spathiphyllum wallisii* 'Verdi' (623 ppm m^{-2} hr^{-1}) in the peat substrate or any of the taxa potted in the peat-free - *Hedera helix*, *Spathiphyllum wallisii* 'Verdi' and *Dracaena fragrans* 'Golden Coast' (541, 436 and 463 ppm m^{-2} hr^{-1}, respectively; $p < 0.001$; Figure 4-2).

In 'dry' conditions after 15 minutes, no statistically significant differences were measured between any studied taxa in the peat or peat-free substrate (Figure 4-2, $p = 0.221$). After 60 minutes, *Dracaena fragrans* 'Golden Coast' in the peat reduced statistically the largest amount of CO_2 from the initial 1000 ppm concentration (1703 ppm m^{-2} hr^{-1} $p < 0.001$). A statistically significant difference was measured between *Spathiphyllum wallisii* 'Verdi' (820 ppm m^{-2} hr^{-1}) in the peat substrate and *Hedera helix* in the peat-free (401 ppm m^{-2} hr^{-1}; $p < 0.001$). No statistically significant differences were measured between other studied species i.e. *Spathiphyllum wallisii* 'Verdi' and *Dracaena fragrans* 'Golden Coast' (524 and 470 ppm m^{-2} hr^{-1}, respectively; $p < 0.001$; Figure 4-2).

Table 4-2: Mean CO_2 increase in the chamber per m^2 of leaf area for each taxon potted in the peat and peat-free substrates at 'no' (0 lux) indoor light in 'wet' (SMC > 30 %, 0.3 m^3 m^{-3}) and 'dry' (SMC < 20 %, 0.20 m^3 m^{-3}) conditions. Data are a mean of three plants per taxon ± SEM.

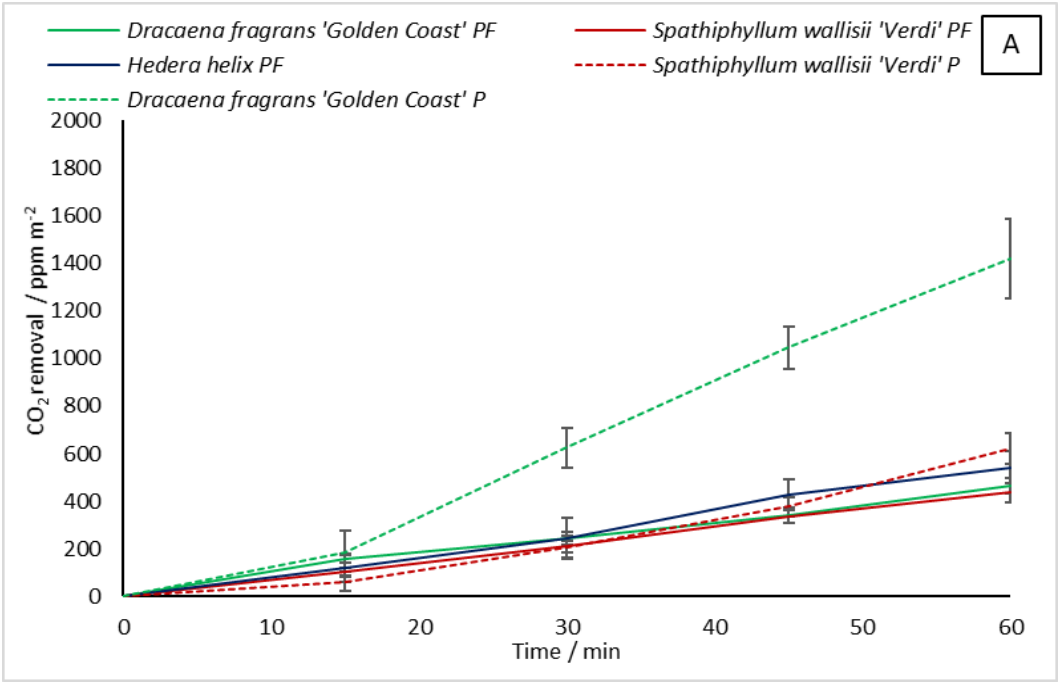
Taxa – Peat-free	Mean CO_2 increase at 'no' light ppm m^{-2} hr^{-1}	
	'Wet' (> 30 % SMC)	'Dry' (< 20 % SMC)
Dracaena fragrans 'Golden Coast'	332 ± 24	331 ± 18
Hedera helix	745 ± 189	408 ± 148
Spathiphyllum wallisii 'Verdi'	177 ± 30	155 ± 15

Taxa – Peat	Mean CO_2 increase at 'no' light ppm m^{-2} hr^{-1}	
	'Wet' (> 30 % SMC)	'Dry' (< 20 % SMC)
Dracaena fragrans 'Golden Coast'	151 ± 78	138 ± 67
Spathiphyllum wallisii 'Verdi'	228 ± 42	185 ± 18

Table 4-3: Mean CO_2 increase in the chamber per m^2 of leaf area for each taxon potted in peat and peat-free substrates at 'low' (~ 500 lux) indoor light in 'wet' (SMC > 30 %, 0.3 m^3 m^{-3}) and 'dry' (SMC < 20 %, 0.20 m^3 m^{-3}) conditions. Data are a mean of three plants per taxon ± SEM, (-) values signify CO_2 assimilation (i.e. CO_2 uptake by the plant thus its removal from the chamber).

Taxa – Peat-free	Mean CO_2 increase at 'low' light ppm m^{-2} hr^{-1}	
	'Wet' (> 30 % SMC)	'Dry' (< 20 % SMC)
Dracaena fragrans 'Golden Coast'	142 ± 8	147 ± 13
Hedera helix	518 ± 42	379 ± 54
Spathiphyllum wallisii 'Verdi'	227 ± 57	192 ± 104

Taxa – Peat	Mean CO_2 increase at 'low' light ppm m^{-2} hr^{-1}	
	'Wet' (> 30 % SMC)	'Dry' (< 20 % SMC)
Dracaena fragrans 'Golden Coast'	66 ± 68	7 ± 52
Spathiphyllum wallisii 'Verdi'	-1 ± 38	-43 ± 64



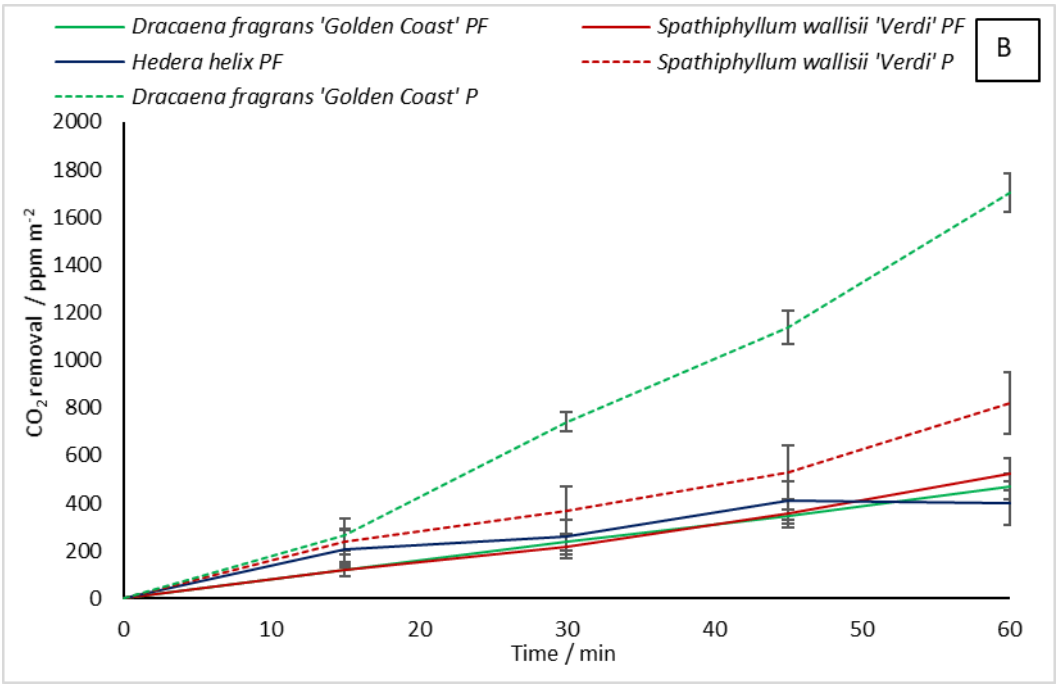


Figure 4-2: Mean CO_2 removal by each taxon in peat (P) and peat free (PF) substrates at 'very high' indoor light (~ 22200 lux) per m^2 of leaf area in **'**wet' (SMC > 30 %, 0.3 m^3 m^{-3}) (A), and 'dry' (SMC < 20 %, 0.20 m^3 m^{-3}) (B) conditions over a 60 min period. Data are a mean of five plants per species – error bars represent SEM.

4.4 Discussion

This work is the first to investigate how potting common houseplants in two differing substrates influenced their ability to reduce a harmful CO_2 concentration of 1000 ppm at a whole plant/substrate scale.

This study demonstrated that at 'low' light in 'dry' substrate conditions assimilation occurred with *Spathiphyllum wallisii* 'Verdi' potted in peat substrate (- 43 ppm m^{-2} hr^{-1}) but not in peat-free (192 ppm m^{-2} hr^{-1}), contrary to the initial hypothesis where an increase in CO_2 concentration was expected from all studied taxa (Gubb *et al.*, 2018). Similarly, the study found that *Dracaena fragrans* 'Golden Coast' was the most effective taxon at reducing high concentrations of CO_2 at 'very high' indoor light levels when potted in the peat substrate, but this was not the case with the peat-free where removal was measured at a significantly lower rate (1703 ppm m^{-2} hr^{-1} and 470 ppm m^{-2} hr^{-1}, respectively).

These measurements suggest that differing substrate types (i.e. peat and peat-free) are able to influence CO_2 assimilation. A taxon may grow more effectively and be more physiologically active in a particular substrate, facilitating a superior CO_2 removal ability. Peat is often described as a perfect substrate for plants, providing a large air-space, high water capacity and due to its formation, a relatively pest and pathogen free environment (Schmilewski, 2008). Moreover, peat contains a carbon concentration in the range of 30 -70 kg/m^3 (18 -60%) whereas, for other mineral soils this concentration is typically < 20% (Agus, Hairiah and Mulyani, 2011), this additional carbon is hypothesised by the authors as a possible reason for superior CO_2 sequestration. Alternatively, the substrate and plant combined may support differing microorganisms, which in turn could provide a superior

removal ability (Zhang *et al.*, 2013). This, however, would need to be explored further by evaluation of the differing microorganisms in both substrates and additional inoculation experiments with the microorganism in question (De Kempeneer *et al.*, 2004). Clearly, the substrate type is of critical importance in terms of CO_2 removal, and this should be further investigated in subsequent studies.

At 'no' and 'low' light levels typically experienced in indoor environments (Hawkins, 2011), most of the studied taxa would increase the concentration of CO_2 in indoor environments as measured in earlier leaf-level work (Chapter 3) (Gubb *et al.*, 2018).

This study clearly suggests that increasing the lighting levels indoors – made possible with targeted lighting installations – would allow taxa to significantly reduce harmful concentrations of CO_2. Agreeing with other similar studies, which show that light is the limiting factor for CO_2 reduction indoors (Pennisi and van Iersel, 2012; Gubb *et al.*, 2018) and that houseplants can be expected to aid ventilation systems – by providing additional CO_2 removal - but not replace them completely (Torpy, Irga and Burchett, 2014).

The results of the current study allow us to estimate the number of houseplants required to reduce CO_2 concentrations to a safe acceptable indoor level – literature suggests that concentrations of 600 ppm and below cause fewer health issues then elevated CO_2 concentrations (Section 4.1). Therefore, for a small office of 15 m^3 (11 m^3 is the minimum space required per person (HSE, 1992), I calculated the time required for a 'dry' *Dracaena fragrans* 'Golden Coast' potted in the peat substrate (the best performing combination) to remove 400ppm of CO_2 (i.e. reduce CO_2 concentration from 1000 to 600 ppm), at a 'very

high' light level assuming a sealed environment with no other sources of and without estimate of transport or diffusion differences between the scales (Equation 4-1).

Time per m^2 of LA (hr) =

Concentration of CO_2 to remove (ppm) / Rate of CO_2 removal (ppm m^{-2} hr^{-1}) x 1/100 (4-1)

Taking into account volumetric loading differences (Girman, 1992) between the test chamber (0.15 m^3) and the small office (15 m^3), the rate of CO_2 removal is reduced by a factor of 100. Consequently, from the results in Figure 4-2 it was estimated 2 m^2 of *Dracaena fragrans* 'Golden Coast' (equating to 14 plants) in 'dry' conditions (the best performing combination) would require 12 hr to remove 400 ppm of CO_2 in the office as per the above stipulated conditions.

Differences in removal between 'dry' and 'wet' conditions across taxa at all light levels and substrates was deemed negligible in agreement with (Chapter 3) (Gubb *et al.*, 2018). Indicating that if plants are left to dry out – anecdotally a common occurrence – the impact on a room scale is small, although on a leaf level there are differences in CO_2 assimilation. Additionally, at 'no' and 'low' light levels most taxa (i.e. the overall system) were respiring. This study suggests that although at typical 'no' indoor light all studied taxa added CO_2 to the indoor environment, the highest increase was approximately half the CO_2 concentration removed at 'very high' light levels. This current work therefore confirms that placing a number of the studied houseplants in a typical home/office environment would not significantly damage health by increasing CO_2 concentrations indoors under either 'wet' or 'dry' substrate conditions.

Even at ‘very high’ light levels, both *Spathiphyllum wallisii* 'Verdi' and *Hedera helix* would require an unrealistic number of plants in both substrates to reduce CO_2 concentrations from 1000 ppm to a near-ambient level. This is in contrast with plants’ pronounced benefits in health and productivity terms (Park and Mattson, 2008; Park and Mattson, 2009; Shibata and Suzuki, 2002; Shibata and Suzuki, 2004).

The findings support the notion that the light level significantly impacts CO_2 removal, as suggested in previous studies (Pennisi and van Iersel, 2012; Torpy, Irga and Burchett, 2014; Torpy, Zavattaro and Irga, 2017; Gubb *et al.*, 2018). Other previous work had also determined that unrealistic numbers of plants (> 200) are required to remove a significant amount of CO_2 in indoor environments (Pennisi and van Iersel, 2012; Torpy, Irga and Burchett, 2014). These studies, however, did not take into account substrate moisture differences, or ambient CO_2 concentrations (Pennisi and van Iersel, 2012). Other studies did not specify which, or how many taxa provided any CO_2 removal (Lim *et al.*, 2009; Pegas *et al.*, 2012), or only considered one light level (Oh *et al.*, 2011).

Torpy, Zavattaro and Irga (2017) estimated that a 2 m^2 active green wall of *Chlorophytum comosum* (where substrate is actively ventilated by pushing air through it) in peat-free substrate would be capable of removing 11 g of CO_2 per hour in a 16 m^3 room. Previous work presented in Chapter 3 estimated that 2 m^2 (of leaf area) of *Spathiphyllum wallisii* 'Verdi' in unventilated peat-free substrate removed 0.75 g of CO_2 per hour at a comparable light level (Gubb *et al.*, 2018). This current work estimated that 2 m^2 (of leaf area) of *Dracaena fragrans* 'Golden Coast' at a light level comparable to both of the previous

removes 3 g per m^3 of CO_2 per hour in a 15 m^3 room, clearly highlighting the benefits of 'active' walls (i.e. substrate ventilation) opposed to traditional 'passive' houseplants.

I support the notion that any future work should focus on green walls (Pettit *et al.*, 2017; Torpy, Zavattaro and Irga, 2017) (especially 'active' walls) which yield more effective removal due to an increased LA of taxa and increased substrate airflow. Additionally, plants which have performed well in removing other indoor pollutants at high indoor light levels i.e. *Osmunda japonica* (Kim *et al.*, 2010) should be further examined. Furthermore, more substrate types should also be investigated. This study has shown that the ability of plants to remove CO_2 at typical indoor light levels may be maximised with certain substrate types and moisture conditions, therefore lower – more realistic – numbers of plants may be required to reduce harmful concentrations of CO_2. Additionally, as 'active' walls – which are clearly superior removers – place extra emphasis on the substrate, removal differences between substrate types will likely be further highlighted.

4.4.1 Limitations of the study

Both *Dracaena fragrans* 'Golden Coast' and *Spathiphyllum wallisii* 'Verdi' potted in the peat substrate were smaller in size (LA) and with *D. fragrans* younger – due to sourcing issues in the UK. The author acknowledges that this may have influenced physiology in part. Moreover, the economic viability of running the lights at a 'very high' level of 22200 lux has not been investigated. Additionally, the author recognises the use of peat is contentious and do not necessarily endorse its use. However, this study is designed to highlight how differing substrates can provide various removal abilities and look to conduct more detailed work in the future.

4.5 Conclusion

The study confirmed that growing the same taxa in differing substrates significantly influenced removal ability in most of the studied species – highlighting the key role substrate types play**.** The results from the current work indicates that 2 m^2 of *Dracaena fragrans* 'Golden Coast' would require 12 hr at a 'very high' light level (~ 22200 lux) in 'dry' conditions to reduce 1000 ppm of CO_2 – the ASHRAE recommended maximum 8 hr exposure guideline – to a 600 ppm concentration in a 15m^3 closed environment (i.e. small office) with no other sources of CO_2. Other studied taxa (*Spathiphyllum wallisii* 'Verdi' and *Hedera helix*) were found to require an unrealistic number of plants at the same 'very high' light level.

At typical 'no' and 'low' indoor light levels most studied houseplants increased CO_2 concentrations albeit, for the highest respiring plants at approximately half the concentration removed at 'very high' light. Therefore, none of the studied houseplants would significantly elevate CO_2 concentrations indoors and thus, cause detrimental health effects. Differences between 'dry' and 'wet' substrates in their capacity for CO_2 removal at either 'no', 'low' or 'very high' light can be considered negligible. These findings support the notion that raising the light level indoors is paramount for studied taxa to remove CO_2.

5.0 Houseplants can remove the pollutant nitrogen dioxide indoors

5.1 Introduction

Nitrogen oxides (NO_x) have been shown to produce ground level ozone, increase susceptibility to ill health, particularly respiratory infections and also affect soil chemistry (Defra, 2019). Within the UK, 34 % of the NO_x is produced by road transport (Defra, 2019). The most noxious component of NO_X is the pollutant nitrogen dioxide (NO_2) (WHO, 2010). The UK government has set aside £255 million in the form of 'the NO_2 plan', specifically implementing mitigation measures to reduce roadside emissions such as bus retrofits, clean air zones, traffic signal improvements and the phase out of diesel cars by 2040 (DEFRA, 2017; Defra, 2019); as a pollutant NO_2 also infiltrates indoor environments (WHO, 2010).

Indoor concentrations are a function of both indoor and outdoor sources, elevated outdoor concentrations (i.e. in cities with a greater density of traffic) will inevitably produce elevated indoor concentrations (WHO, 2010). The most important indoor sources of NO_2 are combustion processes (i.e. heating appliances, fireplaces and stoves) with a building's proximity to roads or the presence of an attached garage shown to be the largest factor influencing indoor concentrations (Nakai, Nitta and Maeda, 1995; Janssen *et al.*, 2001; Kodama *et al.*, 2002; WHO, 2010). Indoor concentrations often exceed those outdoors because of the presence of these additional indoor sources (Kattan *et al.*, 2007).

The World Health Organisation (WHO), EU Commission and the Department for Environment, Food and Rural Affairs (DEFRA) all set a chronic (annual) NO_2 health guideline of 40 µg m^{-3} (21 ppb)— aiming to prevent respiratory illnesses and decreases in lung

function, the main symptoms of long term exposure (especially in children) (Hasselblad, Eddy and Kotchmar, 1992; Koistinen *et al.*, 2008) — and an acute (1 hour mean) health guideline of 200 µg m^{-3} (105 ppb, with 18 permitted exceedances per year) appropriate for both indoor and outdoor environments. However, it has been suggested that the < 40 µg m^{-3} chronic guideline is unlikely to be achievable everywhere, especially in areas with a high density of traffic (Koistinen *et al.*, 2008). Acute exposures to high concentrations of NO_2 significantly affect vulnerable groups, e.g. asthmatics, causing minor changes in pulmonary function (at 560 µg m^{-3} for two and a half hours) (WHO, 2010) and increased airway reactivity (at 500 µg m^{-3}) (Tunnicliffe, 1994; Strand *et al.*, 1998; Niimi et al., 2003). Additionally, acute exposures have been associated with airway inflammation in both healthy and asthmatic study participants (Ezratty, 2014; Defra, 2019).

The EU-commissioned INDEX report collected data on mean NO_2 concentrations across Europe pre-2004 and found indoor concentrations to be in the range of 13 – 62 µg m^{-3} but, in homes with gas cooking and heating equipment, the short-term peak concentrations were measured between 180 – 2500 µg m^{-3}. The study found that 25% of homes exceeded a 60 µg m^{-3} NO_2 concentration (Koistinen *et al.*, 2008).

Reducing the indoor NO_2 concentration indoors would likely reduce health issues alongside economic savings – one study estimated savings of £60,000 per school through a reduction in asthma flare-ups and associated medical costs (based on parents willingness to pay model (Guerriero, 2016)). Indoors, a variety of techniques can be utilised to reduce NO_2 concentrations; these include filtration, designing ventilation systems to provide sufficient

fresh air and appropriate fans and indoor ventilation for combustion systems. These all require ongoing maintenance and often large initial costs for installation. This study investigates the feasibility of a using a simpler approach — low-cost potted houseplants to remove NO_2 indoors and supplement already existing mitigation techniques.

The ability of vegetation outdoors (i.e. trees/plants) to remove NO_2 has been extensively studied (Morikawa *et al.*, 1998; Teklemariam and Sparks, 2006; Jim and Chen, 2008; Vallano and Sparks, 2008; Nowak *et al.*, 2014). Plants have been shown to remove NO_2 through the stomata, simultaneously with CO_2 or O_2, or through absorption by the water present in the leaves – it can therefore be hypothesised that houseplants would do the same and the water content of plant and GM would play an important role (Nowak *et al.*, 2014; Gourdji, 2018). Moreover, a clear variation between plant types ability to remove NO_2 has been previously measured in a study looking at 217 different plant taxa (including houseplants, albeit from dry leaf analysis post-fumigation not *in situ* (Morikawa *et al.*, 1998)). Additionally, it has been suggested that plants with elevated leaf ascorbate concentrations are able to remove more NO_2 (Teklemariam and Sparks, 2006). It can therefore also be hypothesised that different types of cultivated houseplants will remove NO_2 at different rates.

The uptake of NO_2 by plants has previously shown to be concentration-dependent, thus testing at an appropriate guideline concentration is important (Hu and Sun, 2010). Additionally, as NO_2 is removed via the plant stomatal pathway, it can be assumed – as with CO_2 (Gubb *et al.*, 2018; Gubb *et al.,* 2019) – that the light levels will influence NO_2 removal ability. It has been previously shown that if more UV radiation reaches the plants, a higher

NO_2 removal is measured (Teklemariam and Sparks, 2006; Gourdji, 2018). This study will therefore investigate the impact of two light levels on a plants' ability to remove NO_2.

Morikawa et al. (1998) investigated the ability of 217 plant taxa to assimilate ^{15}N labelled NO_2 via leaf fumigation and dry leaf analysis. This included several houseplants such as *Spathiphyllum* spp. and *Dracaena sanderiana* – both possessing a removal ability at the lower end of their respective families. The study found uptake of NO_2-N content to differ as much as 657-fold between all the studied taxa, 62-fold within a particular family (*Theaceae*) and 26-fold within a species (*Solidago altissima*). Additionally, the authors suggest that the metabolic pathway of NO_2-N differs among different plant types (Morikawa *et al.*, 1998).

Pettit et al. (2019) recently reported the removal of NO_2, NO_X and O_3 via an active green wall in a closed loop flow reactor. The authors tested *Spathiphyllum wallisii* and *Syngonium podophyllum* for their ability to remove NO_2 at ambient 70 ppb (134 µg m^{-3}) and elevated concentrations 6.656 ppm (6656 ppb, 12730 µg m^{-3}) at an average photosynthetic flux density of 9.95 µmol m^{-2} s^{-1} (~ 740 lux). The results suggested that at ambient NO_2 concentrations and high indoor light levels both plant types were able to remove NO_2, with a *Clean Air Delivery Rate (CADR)* of 79.92 and 87.84 m^3 h^{-1} m^{-3} of biofilter substrate, respectively. The authors, however, did not investigate how humidity inside the closed reactor — which would have risen sharply due to the presence of a plant — may have affected the sensors' ability to accurately measure concentrations. Additionally, the light level and 'elevated concentrations' for the indoor measurements was far exceeding what you would normally find indoors, likely elevating the removal ability of the plants above what could be observed in real indoor environments.

This research investigates the ability of three houseplants to remove, in real-time, an *in-situ* concentration of 100 ppb NO_2 (chronic 1-hour WHO guideline) on a whole plant/GM scale with 'dry' (SMC < 20 %, 0.2 m^3 m^{-3}) and 'wet' (SMC > 30 %, 0.3 m^3 m^{-3}) substrate moisture content (SMC) at 'low' from now on known as 'typical' (~ 500 lux) and 'no' (0 lux) indoor light. 0 lux was chosen to investigate plants NO_2 removal ability in the dark (measured at night) and ~ 500 lux was chosen to represent typical office conditions. The effect of the GM was investigated in further detail (Figure 5-3) once a potentially significant contribution was identified in initial tests.

5.2 Materials and Methods

5.2.1 Plant material

Three common houseplant taxa (*Dracaena fragrans* 'Golden Coast', *Spathiphyllum wallisii* 'Verdi' and *Zamioculcas zamiifolia*) were selected for this study. They represented a range of leaf types, physiology (succulent and herbaceous) and plant sizes (Table 5-1). Plants were maintained in peat-free GM i.e. Sylvamix growing medium (6:2:2 sylvafibre: growbark pine: coir; Melcourt, Tetbury, Gloucestershire, UK) in 3 L containers, with a slow release fertiliser feed (Osmocote, Marysville, OH, USA). Selected houseplants were purchased one-year prior to the study. Within the taxon, plant height and stature were uniform (data not shown). Prior to experimentation (for > 160 days) plants were kept at room temperatures (21 - 22 °C) and 'typical' light levels (ca. 500 lux) in an indoor office environment within the School of Geography, Earth & Environmental Sciences, at the University of Birmingham (UK).

Table 5-1: Characteristics of the houseplants chosen for experiments. Leaf area (n = 5) and plant height (n = 5) are means ± SEM. S Latin, botanical name is given in italic followed by cultivar, where applicable.

Taxa	Family	Metabolism	Leaf area (cm^2)	Plant height (cm)
Dracaena fragrans 'Golden Coast'	*Asparagaceae*	C3	3081 ± 72	70 ± 1
Spathiphyllum wallisii 'Verdi'	*Araceae*	C3	5013 ± 220	43 ± 1
Zamioculcas zamiifolia	*Araceae*	CAM	2147 ± 249	77 ± 1

5.2.2 Growing media-only experiments

For the GM only experiments (Section 5.3.4), the GM selected were Melcourt Sylvamix medium (6:2:2 sylvafibre: growbark pine: coir; Melcourt, Tetbury, Gloucestershire, UK) and Wyevale Multipurpose Compost (58 % peat; 42% green compost/coir, exact ratios not disclosed, Wyevale, Brentford, Middlesex, UK). From now on referred to as Sylvamix and Wyevale respectively.

5.2.3 NO_2 Chamber experiments

The experimental setup (Figure 5-1A) consisted of a ~ 150 L (45 x 45 x 75 cm, 0.15 m^3) Perspex chamber (custom-built by The Plastic People, Leeds, West Yorkshire, UK) connected to a 1000 ppm NO_2 in air cylinder (> 99 % purity; Speciality Gases, West Bromwich, West Midlands, U.K) with a combination of PTFE tubing (¼ inch outer diameter; Sigma Aldrich, UK) and Ultratorr fittings (Swagelok London, UK). Enclosed inside the Perspex chamber was an electrochemical NO_2 sensor (Alphasense, Great Notley, Essex, UK; Figure 5-1B) connected via a Raspberry Pi stack with temperature and relative humidity sensor (South Coast Science, Brighton, East Sussex, UK) and a 12 V DC brushless fan (RS Components, Corby, Northants, UK). The external RH/temperature surrounding the chamber was monitored with a

calibrated (20 – 90 % RH ± 3%, 0 – 40 °C ± 0.25 °C) Tinytag RH/temperature logger (Gemini data loggers, Chichester, West Sussex, UK).

Inside the chamber 'no' (0 lux) light was achieved by undertaking experiments at night; 'typical' (~ 500 lux) light levels were achieved in the usual lighting conditions of the room — all levels were measured prior to the experiment commencing with a calibrated light sensor (Professional Light Meter, Brannan, Cumbria, UK).

A B

Figure 5-1: Image of the experimental setup (A) and the electrochemical NO_2 sensor (B)

Measurements of the ability of the different studied plant types to reduce NO_2 concentrations of 100 ppb (WHO acute 1-hour guideline) (WHO, 2010) were undertaken on five plants per taxon. Plants were prepared for experiments with GM moisture at the container capacity (*Substrate Moisture Content, SMC,* > 30%) and plants were thus considered optimally watered on the commencement of each experiment (Vaz Monteiro *et al.*, 2016). To ascertain the GM moisture, the SMC was measured prior to experimentation

for each plant, in two locations per container using a SM300 capacitance-type probe connected to a HH2 Moisture Meter (Delta-T Devices, Cambridge, Cambridgeshire, UK; 0–100% range and an accuracy of ± 2.5%). Experiments were carried out on one whole 'plant – GM system' (i.e. potted plant, with uncovered GM) enclosed inside the Perspex chamber at an initial NO_2 concentration of 100 ppb (± 15 %). Experiments were conducted for a duration of 1 hour.

Appropriate control measurements of the studied NO_2 concentration were run at both light levels on both the empty chamber and pot with GM. The number of runs with only GM and pot mirrored the replication of the number of experiments including plants (n = 5). Further control measurements were undertaken to assess the impact of increasing the humidity within the chamber on both the sensor functionality and the concentration of NO_2 measured. Any humidity increases (within an empty chamber) were found to have a negligible effect on the NO_2 concentration measured by the sensor (data not shown).

Table 5-2: Experimental parameters for each lighting treatment during experimentation.

	'no' light		'typical' light wet		'typical' light dry	
	ambient	inside chamber	ambient	inside chamber	ambient	inside chamber
NO_2 (ppm)	< 0.5	-	< 0.5	-	< 0.5	-
Temperature (°C)	21–26	23–27	18–24	20–26	23–26	25–28
Relative Humidity (%)	29–54	46–86	38–57	48–87	35–57	42–81

The chamber was also analysed for leakage prior, during and after experimentation; NO_2 background loss was found to be on average 4.5 ppb over the one-hour test period. All results were corrected for this loss. The NO_2 concentrations (ppb h^{-1} Table 5-3) removed by each plant taxon were calculated with the data measured/logged directly every six seconds

and divided by the leaf area in m^2 presented in Table 5-1 to enable us to calculate a unit of ppb m^{-2} h^{-1} (Figure 5-2).

5.3 Results

5.3.1 NO_2 chamber experiments – per plant

5.3.1.1 Comparison between different plant types within treatment — per plant

No statistical differences were measured in NO_2 removal between different plant types/GM in 'no' light, 'wet' GM ($p = 0.174$) or 'typical' light 'dry GM ($p = 0.191$; Table 5-3). In 'typical' light under 'wet' GM conditions however, a statistically significant difference in NO_2 removal was measured between *Dracaena fragrans* 'Golden Coast' and bare GM (with Dracaena removing significantly more, 62 vs 44 ppb h^{-1}, respectively; $p = 0.03$; Table 5-3).

5.3.1.2 Comparison between treatments within the same plant type — per plant

Spathiphyllum wallisii removed similar concentrations of NO_2 in all three environments tested. This was also the case for bare GM and *Dracaena fragrans* 'Golden Coast' ($p = 0.802$, 0.109, 0.508, respectively; Table 5-3). However, statistical differences were measured for *Zamioculcas zamiifolia* between the treatments 'no' light 'wet' and 'typical' light 'wet' (where light significantly increased the removal of NO_2 – from 47 to 58 ppb h^{-1}, respectively; $p = 0.03$; Table 5-3).

Table 5-3: Mean NO_2 removal per plant (ppb h^{-1}), from inside the chamber containing 100 ppb at 'no' (0 lux) and 'typical' (~ 500 lux) indoor light in 'wet' (SMC > 30 %, 0.3 m^3 m^{-3}) and 'dry' (SMC < 20 %, 0.2 m^3 m^{-3}) conditions. Data are a mean of five plants per plant type ± SEM.

	NO_2 Removed (ppb h^{-1})		
	'no' wet	'typical' wet	'typical' dry
Dracaena fragrans 'Golden Coast'	57 ± 1	62 ± 6	49 ± 4
Spathiphyllum wallisii 'Verdi'	58 ± 6	60 ± 3	55 ± 6
Zamioculcas zamiifolia	47 ± 2	58 ± 3	49 ± 3
Bare growing media	49 ± 5	44 ± 4	42 ± 3

5.3.2 NO_2 chamber experiments – per m^2 of leaf area

5.3.2.1 'no' light, wet – per m^2 of leaf area

After 60 min, statistical differences in NO_2 removal were measured between *Spathiphyllum wallisii* 'Verdi' (115 ppb m^{-2} h^{-1}) and both, *Dracaena fragrans* 'Golden Coast' and *Zamioculcas zamiifolia* (185 and 218 ppb m^{-2} h^{-1}, respectively; $p < 0.01$; Figure 5-2). However, no statistical differences were measured between *Dracaena fragrans* 'Golden Coast' and *Zamioculcas zamiifolia* ($p = 0.08$; Figure 5-2).

5.3.2.2 'typical' light, dry – per m^2 of leaf area

After 60 mins, statistical differences in NO_2 were measured between *Zamioculcas zamiifolia* and both *Spathiphyllum wallisii* 'Verdi', *Dracaena fragrans* 'Golden Coast' (226, 110 and 158 ppb m^{-2} h^{-1}; $p < 0.01$). However, no statistical differences were measured between *Spathiphyllum wallisii* 'Verdi' and *Dracaena fragrans* 'Golden Coast' ($p = 0.06$; Figure 5-2).

5.3.2.3 'typical' light, wet – per m^2 of leaf area

After 60 min, *Zamioculcas zamiifolia* removed more than *Dracaena fragrans* 'Golden Coast' and *Spathiphyllum wallisii* 'Verdi' (272, 201, 119 ppb m^{-2} h^{-1}, respectively; $p < 0.01$; Figure 5-2). The max. removal rate thus corresponds to ca. 4.5 ppb per m^2 of leaf area per minute.

5.3.3 Comparison between treatments within plant type - per m^2 of leaf area

After 60 min, a statistical difference in NO_2 removal was measured between the treatments of 'typical' light wet and 'typical' light dry for *Zamioculcas zamiifolia* (272 and 226 ppb m^{-2} h^{-1}; $p = 0.04$). No other statistical differences were measured between treatments for either *Spathiphyllum wallisii* 'Verdi' or *Dracaena fragrans* 'Golden Coast' ($p = 0.8$ and $p = 0.1$, respectively; Figure 5-2).

5.3.4 NO_2 chamber experiments – Comparison between two different growing media

No statistical differences in NO_2 removal were measured between any treatments or GM types at any timepoint (15 min, $p = 0.472$; 30 min, $p = 0.909$; 45 min, $p = 0.972$; 60 min, $p = 0.966$; Figure 5-3).

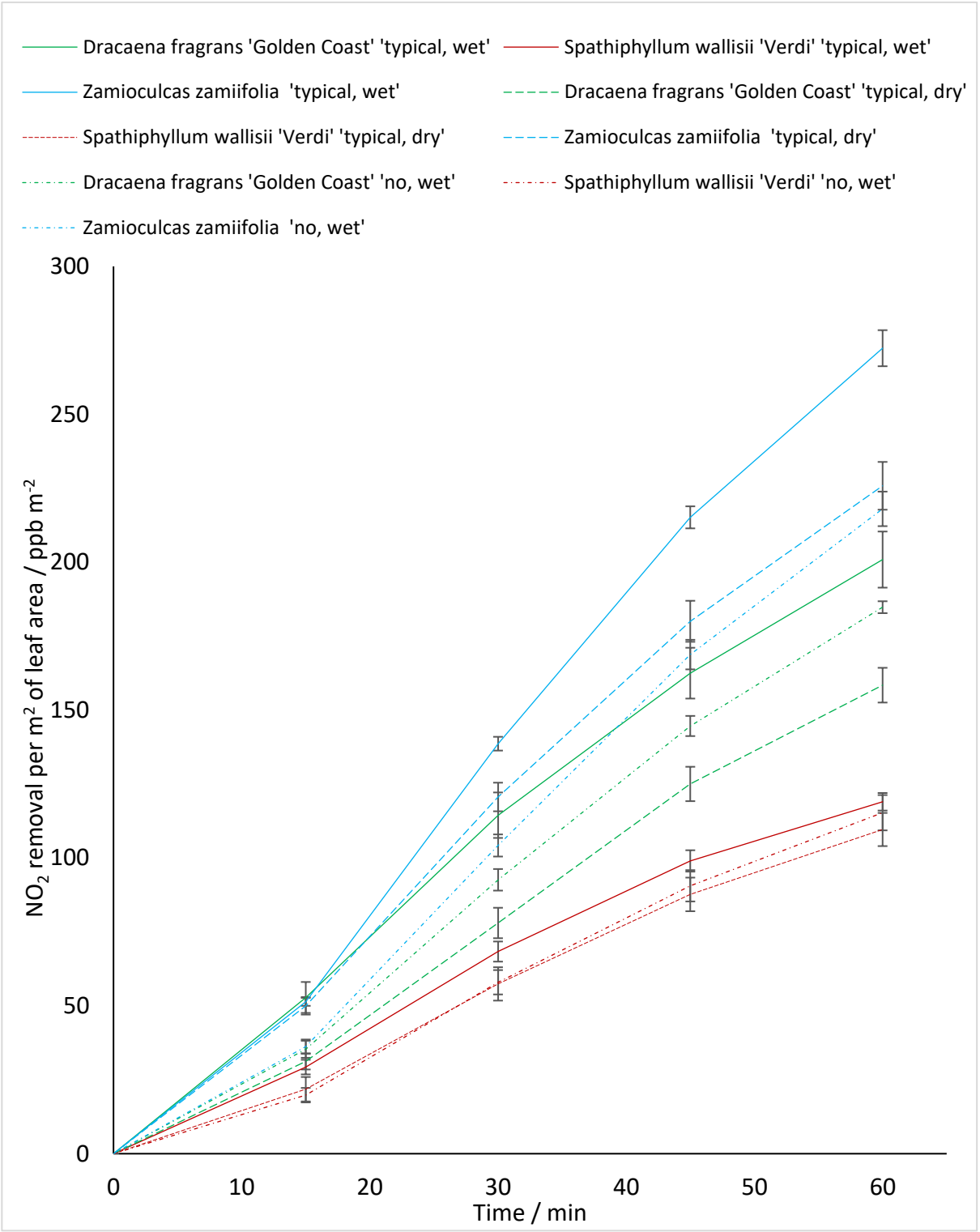


Figure 5-2: Mean NO_2 removal per m^2 of leaf area as a function of time from a concentration of 100 ppb by each plant type under differing environmental conditions per m^2 of leaf area over a 60 min period (see legend). With light level defined as either 'no' (0 lux) or 'typical' (~ 500 lux) and substrate moisture content defined as **'**wet' (SMC > 30 %, 0.3 m^3 m^{-3}) or 'dry'

(SMC < 20 %, 0.2 m^3 m^{-3}). Data are a mean of five plants per plant type – error bars represent SEM.

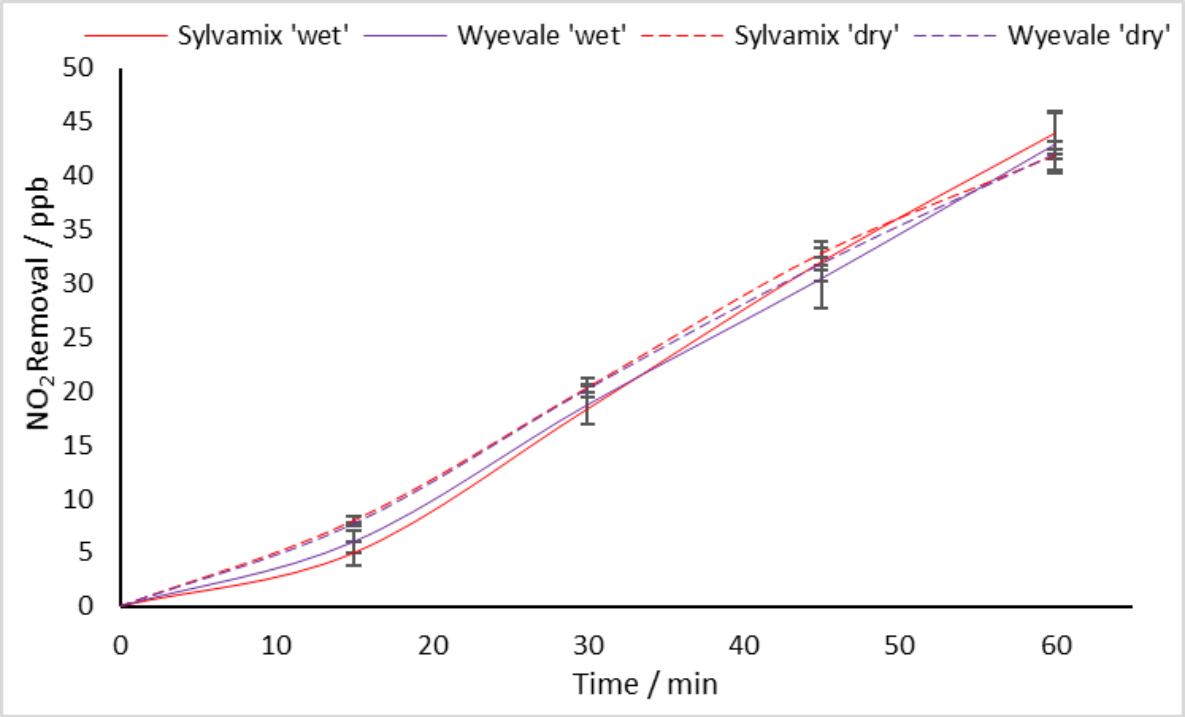


Figure 5-3: Mean NO_2 removal as a function of time from a concentration of 100 ppb over a 60 min period by Sylvamix and Wyevale in 'wet' (SMC > 30 %, 0.3 m^3 m^{-3}) and 'dry' (SMC < 20 %, 0.2 m^3 m^{-3}) substrate moisture conditions at 'typical' (~ 500 lux) light levels. Data are a mean of five growing medias per growing medium – error bars represent SEM.

5.4 Discussion

This work investigates the ability of three common houseplants to remove – in real-time, over a period of 1 h – an *in-situ* concentration of 100 ppb of NO_2 from a 150 L chamber. Demonstrating that the studied houseplants are able to remove significant amounts of NO_2 under common indoor conditions i.e. 0 and 500 lux. As per the hypothesis, different taxa were able to remove NO_2 at differing rates – per m^2 of leaf area (Figure 5-2), suggesting different inherent capacities for NO_2 removal. However, contrary to the initial hypothesis that water content of the GM would influence NO_2 removal, only one plant type –

Zamioculcas zamiifolia – was significantly influenced by this. Additionally, although the GM significantly contributed to NO_2 removal – equal within error for *Spathiphyllum wallisii* 'Verdi' and *Dracaena fragrans* 'Golden Coast' per plant – the type of GM used (peat or peat free), or its water content made no statistically significant difference to the NO_2 removal ability.

In terms of removal per plant (ppb h^{-1}), very few statistical differences were measured within or between treatments across all plant types and bare GM. This suggests that both the light level, and GM moisture had little effect on the NO_2 removal at single plant scale. Moreover, the similarity of removal between bare GM and potted plants across all treatments suggests that most of the removal is achieved via the GM itself. Removal would likely be through breakdown by the microbial activity within the GM or absorption through moisture contained within the soil – as NO_2 is absorbed by water (Dekker, Snoeck and Kramers, 1959). Thus, further investigation and experiments were required (Section 5.3.4).

Investigating another GM for its NO_2 removal ability was hypothesized to clarify if microorganisms were breaking down/metabolising the pollutant. As different GM support different microorganisms (Zhang *et al.*, 2013), it would be expected that variances in removal would be measured. However, no statistical differences in NO_2 removal were measured between 'wet', 'dry' or GM types at any timepoint over the 60-minute experiment (Figure 5-3). This suggests that in the context of this experiment there either were no differences in microbial communities' composition, no differences in microbial activity or that the contribution of this pathway to NO_2 removal is insignificant. Moreover, the fact that

no statistical differences were measured between GM moisture content ('wet' and 'dry', Figure 5-3) suggested that moisture absorption of NO_2 was also not the primary removal pathway. However, further experiments (data not shown) at very low moisture content (SMC < 10%) saw a reduction in removal rate, and it is therefore suggested that even in 'dry' GM conditions (in a biological and practical horticultural sense, 15 – 20 %), enough moisture was still present to remove NO_2.

In terms of removal per m^2 of leaf area, statistical differences were often measured between different plant types for the same treatment — confirming an inherent difference in different plant types removal ability as per the hypothesis and supporting previous work (Morikawa *et al.*, 1998). However, when comparing between treatments within the same plant type statistical differences were only measured for *Zamioculcas zamiifolia* in 'typical' light between 'wet' and 'dry' GM. This suggests that neither light — up to 500 lux — nor water content (down to very low SMC, i.e. below 10%) had much of an effect on the NO_2 removal ability of each species, aligning with the per plant removal results. Furthermore, removal experiments investigating GM only showed a similar pattern of little change in NO_2 removal in response to the change in environmental conditions (Figure 5-3), this lends further weight to the 'per plant result' conclusion that the GM is responsible for a large amount of the removal. While the detailed mechanism could not be resolved in the experiments, a very small amount of moisture appears to be more critical for NO_2 removal efficiency than any of the other parameters investigated here.

From the experiments, taking into account volumetric considerations, estimates of the amount of NO_2 each taxon will remove per plant and per m^2 in a sealed 15 m^3 room (the size of the PI's office) containing 100 ppb – assuming there are no additional sources within the room and the 100 ppb is uniformly distributed throughout. It should be noted that this estimation of the max. impact is not considering natural or mechanical ventilation which is highly variable and may substantially alter the indoor NO_2 exposure in particular if there is a significant indoor–outdoor concentration gradient. In typical buildings the outdoor-to-indoor air exchange provides approximately one air change per hour (~ 1 h^{-1}) (Cummings and Waring, 2019).

For natural ventilation, air change rates are incredibly difficult to predict due to uncertainties around wind speed, pressure co-efficient, air temperature and ventilation area (Chartered Institution of Building Services, 2005; HM-Government, 2015). With mechanical ventilation, various guidance bodies specify a minimum air supply criterion in litres per second per person with UK building regulations (Part F) suggesting a minimum of 10/l/s/per person (HM-Government, 2015)

Taking into account volumetric loading differences (Girman, 1992) between the test chamber (0.15 m^3) and the small office (15 m^3), the rate of NO_2 removal is reduced by a factor of 100. Therefore, using measured removal rates (Figure 5-2) and reducing by a factor of 100 allows us to derive the removal rate in a small office (Table 5-4). This extrapolation does not consider the differences between pollutant transport and diffusion between the experimental scales. It is therefore estimated that 1 m^2 of the highest removing species per m^2 — *Zamioculcas zamiifolia* — in optimal environmental conditions namely, 'typical' light

and ‘wet’ GM would reduce a concentration of 100 ppb at a rate of 3 ppb per hour. Furthermore, the highest removing potted plant (not considering leaf area) namely, *Dracaena fragrans* 'Golden Coast' potted in wet GM under ‘typical’ light conditions was able to reduce a concentration of 100 ppb at 0.62 ppb per hour. Results from all plant types in this condition are presented below in Table 5-4.

Table 5-4: The derived ability of each studied potted plant to reduce a concentration of 100 ppb inside a 15m^3 room in ‘wet’ (SMC > 30 %, 0.3 m^3 m^{-3}) substrate moisture conditions at ‘typical’ (~ 500 lux) light levels

	NO_2 Removed (ppb h^{-1})
	'typical' wet
Dracaena fragrans 'Golden Coast'	0.62
Spathiphyllum wallisii 'Verdi'	0.60
Zamioculcas zamiifolia	0.58

These estimates suggest that five plants in such a small office could remove approximately 3 ppb of NO_2 per hour. At first sight such a removal rate may look relatively low, but it should be noted that the measured rates of NO_2 removal occurred in typical light and even dark conditions while NO_2 exposure peaks tend to appear over short, often rush-hour related periods (Malley *et al.,* 2018; Engström and Forsberg, 2019). It can therefore be expected for any removal to be constant throughout the day or night, even when plants are under mild water deficit — unlike with CO_2 where supplementary lighting was required (Gubb *et al.*, 2018; Gubb *et al.,* 2019)**.** Therefore, plants are able to passively remove NO_2, without additional energy requirements. Furthermore, as plant type seems to have little effect (Table 5-4) on NO_2 removal at the office scale (considering the plant types in this study only), easier-to-maintain plants like *Zamioculcas zamiifolia* and other succulents would likely be just as effective.

Comparing to earlier work on houseplants' ability to remove carbon dioxide (CO_2) indoors (Gubb *et al.*, 2018; Gubb *et al.*, 2019), the fact that removal of NO_2 occurs at typical indoor light levels is a significant advantage. Without the need for supplementary lighting, increasing both energy costs and integration difficulties for designers, passive NO_2 mitigation via potted plants is a much more viable technique — especially with the built environments push towards net-zero carbon buildings.

This work is consistent with recent experiments that have suggested that the GM and the microorganisms within are predominantly involved in the removal of pollutants and plants themselves are only utilized in-directly to maintain and support GM microorganisms (Irga, Pettit and Torpy, 2018; Kim *et al.*, 2018).

As both the concentration of NO_2 and light level — albeit, > 500 lux —affect the removal ability of plants (Teklemariam and Sparks, 2006; Hu and Sun, 2010; Gourdji, 2018), direct comparison to literature is difficult, unless removal with exactly the same conditions were investigated. However, numerous studies have tested outdoor vegetation for NO_2 removal, and this study aligns with those (Morikawa *et al.*, 1998; Nowak *et al.*, 2014; Pettit *et al.*, 2019) in the fact that all studied plant types were able to remove NO_2 to varying degrees at a wide variety of concentrations.

To put the small office estimate of an NO_2 removal rate of ca. 3 ppb per hour from five potted plants into context, it is useful to compare to alternative approaches that have been

taken to reduce NO_2 exposure in urban areas. As an example, the Agglomeration of Lausanne-Morges (ALM) in Switzerland has a long-term record of introduction of successful clean air policies and a recent study compared to the health benefits resulting from these policies over the decade 2005–2015 (Castro, Künzli and Götschi, 2017). Castro et al. (2017) suggested that the NO_2 exposure reduction by 2.8 ppb in the ALM region may have lowered the NO_2-related deaths by 51 and the life-years lost by 550 years. Based on this estimate, five potted plants in each small office may be able to reduce the indoor NO_2 exposure for the occupant to a similar extent. However, studies of mechanical and natural ventilation systems as well as an investigation of the long-term ability of potted plants to retain NO_2 are needed to establish how significant the air quality services of potted plants are both in the long-term and in real-life conditions.

A very similar *Clean Air Delivery Rate (CADR)* per plant from all of the studied taxa was calculated namely, 0.1 m^3/h — unsurprisingly much lower than the results from the active wall experiments by (Pettit *et al.*, 2019), where the CADR was two orders of magnitude higher. The result was comparable to some of the studies summarised by Cummings et al (Cummings and Waring, 2019). This recent review paper calculated the CADR from 12 previously published potted plant pollutant removal studies (but none investigating NO_2 removal). The study found that the distribution of single-plant CADR spanned orders of magnitude, with a median of 0.023 m^3/h (i.e. ca. 4–5 times lower than what this study found). Their median CADR would require the placement of 10–1000 plants/m^2 of floor area for the combined VOC-removing ability by potted plants to achieve the same removal rate that outdoor-to-indoor air exchange already provides in typical buildings (~1 h^{-1}). This review

suggests moving away from passive experiments and onto active technologies (Cummings and Waring, 2019). However, it should be noted that outdoor-to-indoor air exchange for pollutants such as NO_2 may also increase the indoor NO_2 exposure especially since ventilation rates are likely to be higher during outdoor NO_2 peak periods (day-time, particularly during the morning rush-hour period when people tend to arrive at work and thus open doors and potentially windows) than during low NO_2 outdoor concentrations (night-time) unless a smart and active ventilation system is in place. In passively ventilated buildings which still represent the vast majority of buildings in the UK, a continuous NO_2 removal by potted plants can thus provide indoor air quality services.

I do not dispute the notion that future work on green walls (especially 'active' walls) is urgently needed since these yield more effective removal due to an increased leaf area and increased GM airflow (Irga, Pettit and Torpy, 2018). In their study, Pettit et al. found NO_2 removal i.e. the CADR was measured at 79.92 and 87.84 $m^3\ h^{-1}\ m^{-3}$ of biofilter substrate respectively — three orders of magnitude greater than the passive removal studies. Other active wall studies investigating VOCs have found similar CADRs (i.e. 28.3 and 18.9 $m^3\ h^{-1}\ m^{-2}$ green wall area, per different plant type) (Torpy *et al.*, 2018). However, passive low-tech potted plant NO_2 removal should not be overlooked given the wide and immediate availability across the globe and a significant potential to improve indoor air quality, particularly, in small offices in urban environments without smart, active ventilation systems. For occupants of these offices, a realistic number of potted plants may have comparable health benefits to clean air policies that are obviously crucial to reduce NO_2 exposure outdoors.

5.5 Conclusion

This study investigated if a simple set up, with just a potted houseplant could be effective at passively removing the harmful pollutant NO_2. This was carried out by investigating the ability of *Spathiphyllum wallisii* 'Verdi', *Dracaen*a *fragrans* 'Golden Coast' and *Zamioculcas zamiifolia* and two different GM to remove *in situ* concentrations (100 ppb) of NO_2 in real-time at two typical indoor light levels (0 and ~ 500 lux) and in 'wet' and 'dry' (SMC > 30 %, 0.3 m^3 m^{-3} and SMC < 20 %, 0.2 m^3 m^{-3}) GM conditions.

All studied plant types across all treatments were able to reduce the NO_2 concentrations representative of a polluted urban environment, but to varying degrees. It should be noted that in many treatments bare GM alone removed comparable amounts to GM and plant together — suggesting that the GM and/or moisture contained within is the main pathway of removal.

The greatest NO_2 removal measured in a 150 L chamber over 1-h periods was a rate of 4.5 ppb per m^2 of leaf area per minute and 58 to 62 ppb per plant over the 1-h period for each of the three plant types in 'wet' GM at ca. 500 lux. This would correspond to a rate of up to 3 ppb per m^2 of leaf area per hour and 0.62 ppb per plant per hour in a small office when not considering natural or mechanical ventilation which may substantially alter the indoor NO_2 exposure in particular if there is a significant indoor–outdoor concentration gradient. The studied plant types remove clearly measurable amounts of NO_2 passively during the day and night without additional energy requirements (unlike mechanical ventilation or filtration systems), thus adding indoor air quality services and the associated health benefits to the other established building services potted plants provide at minimal cost (Gubb *et al.*, 2020).

Such a passive removal approach is unsurprisingly significantly less effective than the deployment of 'active' green walls, highlighting the need for further research in this area. However, these require a significantly increased amount of energy, irrigation considerations and thorough maintenance in comparison to simple houseplants. This study nevertheless demonstrates that simple houseplants will passively remove NO_2 under normal indoor conditions, but the air quality services of potted plants strongly depend on room size and competing removal routes such as building ventilation as well as on the success of clean air policies aiming to reduce outdoor NO_2 peak events that will in turn impact on indoor NO_2 exposure.

6.0 Is solid-phase micro-extraction (SPME) a suitable technique for determining a houseplant's removal ability of volatile organic compounds (VOCs)?

6.1 Introduction

Volatile organic compounds (VOCs) are a group of organic pollutants defined by the World Health Organisation (WHO) and Environmental Protection Agency (EPA) as having a boiling point of ca. 50 – 260 °C. VOCs can cause an array of health issues, with severity dictated by an individual pollutant's toxicity, measured concentration and lifetime indoors. Prolonged exposure (for longer than the relevant averaging period) at elevated concentrations above the recommended health guidelines can cause symptoms from mild sensory irritation (i.e. alpha-pinene) to cancer (i.e. benzene) depending on the pollutants toxicity (WHO, 2010; Wolkoff and Nielsen, 2017).

Numerous experiments have tested the ability of houseplants in various forms i.e. potted plants, hydroponically and as part of green walls to remove VOCs, ultimately aiming to reduce their concentration and in turn associated detrimental health impacts within indoor environments. More than 200 plant taxa with a variety of GM (i.e. the plant species and/or cultivars) have been tested for their ability to remove pollutants. The most common organic pollutants tested include: benzene, toluene, ethylbenzene and xylene (BTEX), formaldehyde and trichloroethylene (Orwell *et al.*, 2004; Kim *et al.*, 2008; Sriprapat and Thiravetyan, 2013; Irga, Pettit and Torpy, 2018).

Indoor plants can remove pollutants via three distinct pathways — through aboveground parts (i.e. stomata and cuticle); growing GM and roots (Cruz *et al.*, 2014). It is well

established that the removal of organic pollutants is predominately associated with microflora in the substrate, plants themselves are only utilised in-directly to maintain and support substrate microorganisms (Orwell *et al.*, 2004; Wood *et al.*, 2002; Cruz *et al.*, 2014; Kim *et al.*, 2008).

To test a plants removal ability experimental setups often utilise small (< 1 m^3) static (i.e. sealed, no air circulation) (Irga, Torpy and Burchett, 2013; De Kempeneer *et al.*, 2004; Orwell *et al.*, 2004; Orwell *et al.*, 2006; Sriprapat and Thiravetyan, 2013; Siswanto, Chhon and Thiravetyan, 2016) or dynamic (i.e. with air circulation) (Tani and Hewitt, 2009; Yang *et al.*, 2009; Kim *et al.*, 2008; Cornejo *et al.*, 1999) chambers to enclose the plants and often utilise a similar testing methodology. Specifically, this comprises of the injection of a desired pollutant (i.e. fumigation) into a chamber, followed by an equilibration time for concentration homogeneity, and the sampling of chamber air over a period of time with quantification and analysis through a headspace chromatography instrument (e.g. gas chromatography mass spectrometry, GC-MS).

In this type of experiment, indoor plants are often exposed to concentrations up to 1000-fold higher than what is measured *in situ* — altering a plant's removal efficiency (Aydogan and Montoya, 2011; Cruz *et al.*, 2014; Torpy *et al.*, 2018). The sampling of chamber air after fumigation, and the transfer into a suitable headspace chromatography instrument are likely both responsible for the use of elevated concentrations due to losses in sensitivity. Sampling is achieved with either a gas tight syringe (De Kempeneer *et al.*, 2004; Orwell *et al.*, 2004; Orwell *et al.*, 2006; Yoo *et al.*, 2006; Yang *et al.*, 2009; Irga, Torpy and Burchett, 2013; Boraphech *et al.*, 2016; Siswanto, Chhon and Thiravetyan, 2016) or a trap (active or passive)

(Wolverton, Johnson and Bounds, 1989; Kim *et al.*, 2008; Kim *et al.*, 2016) and transfer accomplished via direct injection or desorption into an instrument.

This study is looking to investigate if the sampling of chamber air can be improved to allow for experiments to be carried out effectively at *in situ* VOCs concentrations. It was hypothesised that utilising a sampling technique known as solid phase micro extraction (SPME) would achieve this. Prior studies have shown that SPME can provide a high reproducibility (< 10 %) and linearity (R^2 > 0.99) at low parts per billion (ppb) concentrations of VOCs (Martos and Pawliszyn, 1997; Larroque, Desauziers and Mocho, 2006a; Essah and Sanders, 2010).

SPME works by adsorption and desorption of pollutants from an inert fibre; the fibre is coated with differing compounds at different thicknesses enabling the selective sampling of compounds with varying polarity. Numerous fibre coatings are available, each possessing a specific affinity for certain compounds — enabling a preferred choice for the particular pollutant in question (Essah and Sanders, 2010). Pollutants are sampled by exposing the fibre and are analysed by thermally desorbing the fibre inside an appropriate chromatography instrument (Augusto and Valente, 2002; Tholl *et al.*, 2006).

This study will initially utilise a custom-built gas-handling line ("rig") to contrast the air sampling ability of the commonly used gas-tight syringe and — to the authors knowledge for the first time in experiments of this type – SPME for *in-situ* VOC concentrations.

The study will investigate the VOC alpha-pinene, due to its low toxicity for this validation stage in comparison to other VOCs (Wolkoff and Nielsen, 2017) prevalence in indoor consumer products and lack of previous study in literature (Yang *et al.*, 2009). Two SPME

fibres were selected (Section 6.2.1), chosen on the premise of their coatings being specifically designed for use with VOCs (Spietelun *et al.*, 2010) which are to be assessed via the grab sampling technique — whereby the SPME fibre is left directly exposed to alpha-pinene at one location and at one point in time. Quantification and analysis of samples will be made with a GCMS instrument. Experiments will attempt to investigate an alpha-pinene concentration of 14000 μg m^{-3} (2.5 ppm) – a threshold concentration for sensory irritation (Wolkoff and Nielsen, 2017).

To allow the experiment to be effectively validated, I selected a number of measures to assess each method. These included the limit of detection (LOD), limit of quantification (LOQ), reproducibility and linearity. The LOD can be defined as ca. three times the area of baseline noise provided by the analytical instrument and the LOQ ten times — with any peak areas lower than the LOQ unsuitable for quantification purposes (Rood, 2007; Hubschmann, 2009). Additionally, the reproducibility will be validated by measuring the relative standard deviation (% RSD) between samples — with < 20 % considered acceptable (UNODC, 2009). Furthermore, linearity, which is defined as a methods ability to obtain results, which are directly proportional to the concentration of a desired gas within the sample, will be assessed by calculating a R^2 value for a linearity plot — an $R^2 > 0.99$ is considered an excellent fit for validation purposes (Rood, 2007).

Initially, both the limit of detection (LOD) and limit of quantification (LOQ) of the analytical instrument will be calculated from appropriate blank runs. Furthermore, the reproducibility will be measured at 5 ppm for all methods (described in Section 6.2). Any methods achieving

suitable reproducibility will be taken forward and assessed for linearity— with plots encompassing the desired 2.5 ppm.

6.2 Materials and Methods

6.2.1 Experimental setup

The experimental setup comprised of a rig system (Figure 6-1) coupled with a GC-MS instrument (Thermo Fischer Scientific – ITQ 1100, Waltham, MA, U.S.A). Sampling of gases from the rig were achieved using either a gas tight syringe (Hamilton, Reno, NV, U.S.A) or by solid phase micro-extraction (SPME) (Supelco, Bellefonte, PA, U.S.A)**.** Two fibres were selected for the study, namely, polyacrylate – 85µm, 24 ga, white and; divinylbenzene/carboxen/polydimethylsiloxane – 50/30 µm, 24 ga, grey.

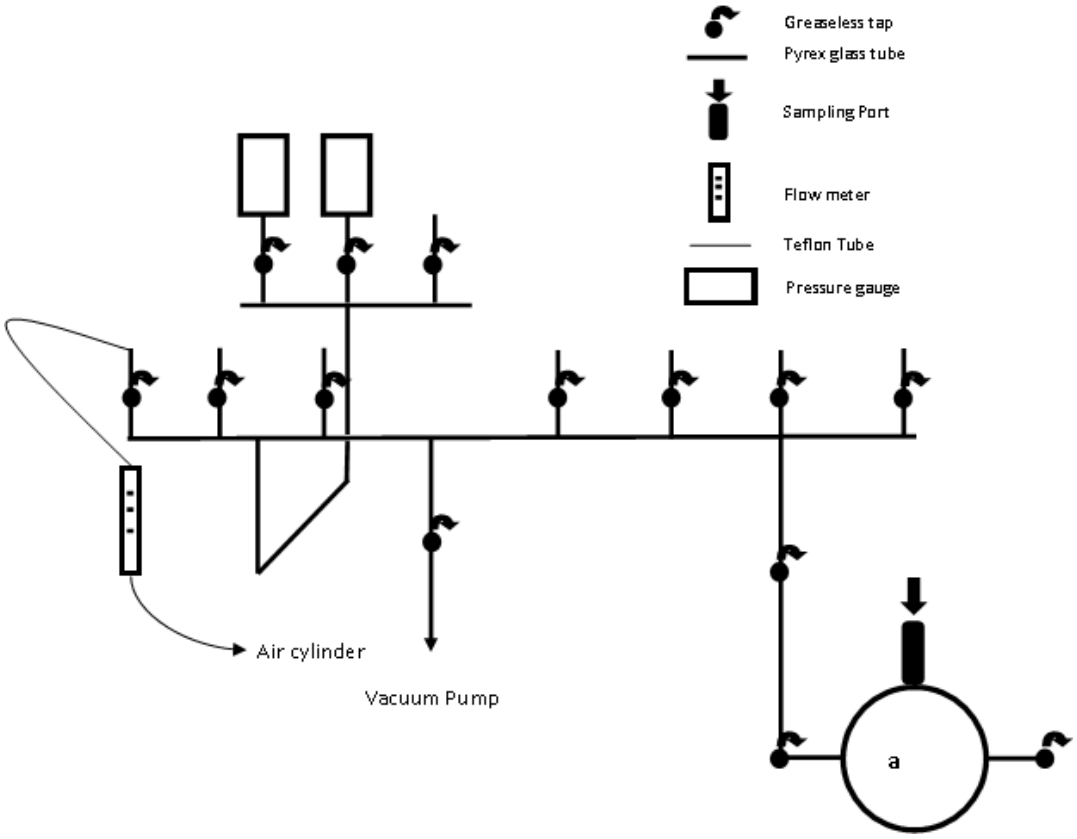


Figure 6-1: Diagram of the rig system – legend is shown on Figure. *a* is the sampling bulb where required concentrations of alpha-pinene are produced and sampled. The cold finger (see Fig. 6-2) can be attached to any empty greaseless tap.

The rig system was made from Pyrex glass (Corning Inc, Corning, NY, USA) and operated with Young's greaseless taps (GPE Scientific Ltd, Leighton Buzzard, Bedfordshire, UK). A vacuum was achieved in the rig by a rotary vacuum pump (Edwards, Burgess Hill, West Sussex, UK) and measured with two separate pressure gauges (Leybold, Cologne, North Rhine-Westphalia, Germany). Experiments were carried out in the Chemistry laboratory of the school of Chemistry, Food and Pharmacy on the Whiteknights campus at the University of Reading (UK).

The desired gaseous sample was introduced into the rig via a cold finger (Figure 6-2) – consisting of Pyrex glass and a Young's greaseless tap. The cold finger can be attached to the rig at any empty port above a greaseless tap. Liquid analytical standard (Section 6.2.6) is then added to the finger, allowing for the headspace gas to be introduced into the rig by opening of the greaseless tap. Prior to rig introduction several 'freeze-pump-thaw' cycles were carried out upon the cold finger, to remove any impurities in the standard. This consisted of the following steps: freezing the standard inside the cold finger with liquid nitrogen; evacuating the headspace inside the cold finger; and a slow warming of the cold finger using a water bath (approx. 25 °C). These steps were continued until no bubbling was observed inside the cold finger.

Figure 6-2: The cold finger used for experiments containing pure alpha-pinene standard.

The Gas Chromatography (GC) part of the GC-MS instrument separated the different analytes in a sample of gas utilising a silica based column (RXi-5HT, 30m, 0.25mmID 0.25µm df; Restek, Bellefonte, PA, USA). Due to the nature of the experiments a standard multi-purpose column was selected for analysis; a single analyte was to be investigated per experiment and therefore, separation was not deemed sufficiently difficult to require specialist columns. The data was displayed in the form of unique retention time (t_R, minute) for each analyte and by peak height/area (µV). The peak height/area reflects the current measured when the analyte passed through the detector and an interaction occurred thus, producing an electrical signal where the size relates to the analyte concentration (Hubschmann, 2009).

The Mass spectrometer (MS) combined with the GC provided the retention time, separation, and the unique ion fragmentation pattern for each analyte within the sample (Tholl *et al.*, 2006). Prior to each day of experiments appropriate calibration experiments were run which

included leak check, k factor calculation and tune. Experiments were carried out in the CAF laboratory at of the school of Agriculture, Policy and Development on the Whiteknights campus at the University of Reading (UK).

Sampling from the glass bulb was carried out using either a gas tight syringe, or by SPME. A number of different sized gas tight syringes (i.e. 250 µl, 5 mL) and two different SPME fibres were trialled throughout the experimental validation period.

6.2.2 General experimental procedure

Concentrations of the desired compound were generated inside the 1 L glass bulb (Figure 6-1) through a series of dilutions using the rig system. A known pressure of compound was emitted into the prior evacuated bulb (P_1), which was isolated, and made to atmospheric pressure (P_{atm1}) using dry synthetic air (Section 6.2.6). The rest of the rig was evacuated to remove unwanted concentrations of the desired compound. The compound inside the bulb was pumped off until the desired pressure was reached (P_2), which was isolated, and made to atmospheric pressure (P_{atm2}) using dry synthetic air (details Section 6.2.6) The finished compound was sampled as per one of the three methods (Section 6.2.3, 6.2.4 and 6.2.5). The dilution steps above are presented more succinctly in Equation 6-1:

$$\text{Concentration (ppmv)} = (P_1/P_{atm1}) \times (P_2/P_{atm2}) \times 10^6 \qquad (6\text{-}1)$$

Where: P_1 = pressure of compound initially isolated in bulb (Torr)

P_{atm1} = pressure to which initial concentration was made up with synthetic air (Torr)

P_2 = pressure of compound subsequently isolated in the bulb (Torr)

P_{atm2} = pressure to which subsequently isolated concentration was made up with synthetic air (Torr)

6.2.3 Gas-tight Syringe — direct injection experiments

Gas samples were taken with a gas-tight syringe via the sampling port on the bulb (Figure 6-1). The sampling port consisted of a swage lock with septum – connected to the bulb via a glass tube. The removed gas was then directly injected into the GC-MS instrument for quantitative analysis. An identical volume of gas (250 µl) was sampled for each experimental reputation with a 250 µl syringe. Two methods were tested in terms of gas generation using the rig: The first (Method 1) generated a new concentration for each repetition as described in Section 6.2.2, synthetic air was generated in the rig and sampled for blank runs in between compounds; the second (Method 2), sampled the same gaseous sample (generated as described in Section 6.2.2) three times with ambient lab air used for blanks.

The injection port temperature was 55 °C and was operated in the splitless mode. Helium was used as the carrier gas at a flow rate of 1.0 mL min^{-1}. The column temperature was held at 50 °C for 1 min and then programmed at 15 °C/ min to 250 °C and held for 5 min. Mass spectroscopy conditions were: ion source 220 °C; MS transfer line 275 °C; and a scan range of 29 to 350 mass units. Method 1 was also run in selected ion monitoring (SIM) mode, with ions 91, 92 and 93 (IFRA, 2016).

6.2.4 SPME — direct injection experiments

Gas samples were collected by leaving the SPME exposed inside the bulb – via the sampling port – for 15 minutes. The time was chosen after a saturation analysis of the fibre was undertaken over an appropriate range (10 – 45 min, data not shown). The time of 15 min

was found to be the shortest required for appropriate saturation to be achieved, with assessment made by analysing peak areas (data not shown). Two methods were trialled for gas generation as described in Section 6.2.3 (i.e. Method 1 and Method 2). The SPME was transferred to the GCMS and thermally desorbed for quantitative analysis.

The injection port temperature was 220 °C and was operated in the splitless mode. Helium was used as the carrier gas at a flow rate of 1.0 mL min^{-1}. The column temperature was held at 40 °C for 1 min and then programmed at 15 °C/ min to 200 °C and held for 5 min. Mass spectroscopy conditions were: ion source 220 °C; MS transfer line 275 °C; and a scan range of 29 to 350 mass units.

6.2.5 SPME — vial experiments

A different method with the SPME was also trialled, adapted from (Essah and Sanders, 2010). Whereby, gas samples were removed from the bulb as described in Section 6.2.3 via a 5 ml Hamilton gas tight syringe and injected into a 25 ml HPLC vial – dilution between the two volumes were calculated so 5 ppm was enclosed inside the vial, as per Equation 6-2. The SPME was then inserted and held inside the vial for 15 mins - a saturation analysis was undertaken as per Section 6.2.4. Two SPME fibres were analysed for the experiment and the instrument setup was as described in Section 6.2.4.

Conc. in vial (mg m^{-3}) = vol. of gas sampled (ml) / vol. of HPLC vial (ml) * conc. In bulb (mg m^{-3}) (6-2)

6.2.6 Analytical standards

All standards – liquid or gaseous – employed for experiments were of analytical grade and sourced from recognised suppliers. Liquid standards used were as follows: Alpha Pinene > 98% purity, CAS: 80-56-8 (Sigma – Aldrich Company Ltd, Gillingham, Dorset, U.K). Gaseous

standards used were as follows: Dry Synthetic Air > 99.9% purity (Air Liquide, Coleshill, West Midlands, UK); Helium > 99.999% purity (Air Liquide, Coleshill, West Midlands, U.K).

6.3 Results

6.3.1 LOD and LOQ

Five random blank runs were selected, spanning across the time period of all experiments in this study to account for any changes to the instrument over time. The background noise was averaged and is presented in Table 6-1. These values were then multiplied by three and ten for LOD and LOQ respectively (see detail in Section 6.1), providing an average LOD and LOQ for the GCMS instrument used for all experiments presented in Section 6.3.

On comparison with the average LOQ and the peak areas of alpha-pinene in prior experiments it was judged that at the concentrations being tested, all peak areas in question exceeded the LOQ and were therefore suitable for quantification.

Table 6-1: Table showing the limit of detection and limit of quantification for the analytical instrument used in all experiments

Background Noise (peak area, µV)	LOD (peak area, µV)	LOQ (peak area, µV)
27404	82212	274040
25713	77139	257130
7015	21045	70150
5714	17142	57140
19241	57723	192410
Average: 17017	**Average: 51052**	**Average: 170170**

6.3.2 Reproducibility experiments

6.3.2.1 Gas-tight Syringe — direct injection reproducibility experiments

Method 2 had the lowest %RSD (9.1%) of the three methods and therefore the greatest reproducibility. Running the MS in SIM mode did not improve the reproducibility in comparison to a full scan in Method 1 (27.4% and 23.8%, respectively) and was therefore not assessed with Method 2. Average peak areas ranged between 7059433 – 8155436 µV (%RSD = 7%; Method 1 and Method 2, respectively) for the 5 ppm alpha-pinene concentration (Figure 6-3).

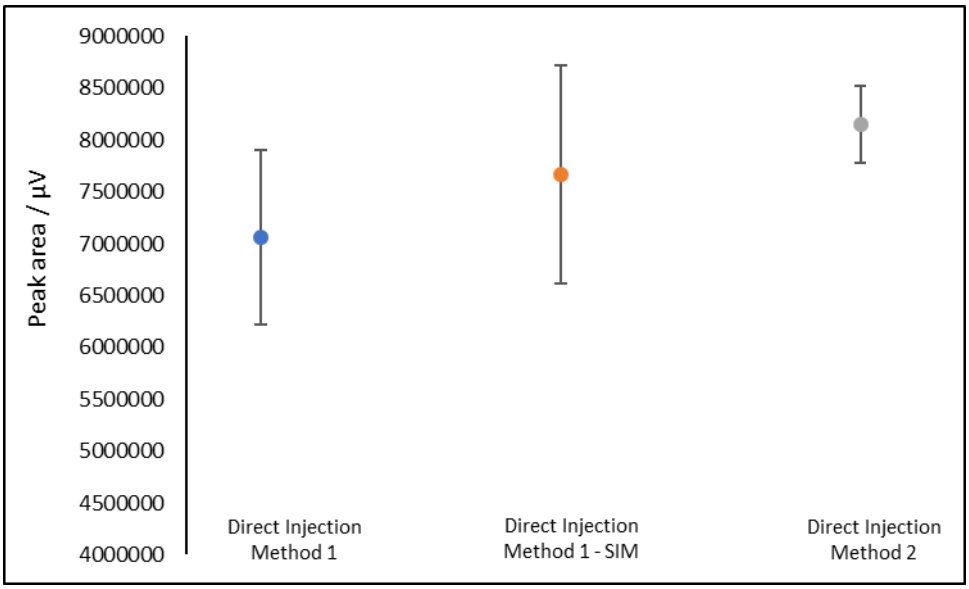


Figure 6-3: Comparison of the reproducibility of three direct injection methods (i.e. Method 1, Method 1 – SIM, and Method 2) at a concentration of 5 ppm alpha-pinene (n = 3) – error bars correspond to %RSD between runs.

6.3.2.2 SPME — direct injection reproducibility experiments

Method 2 had the lowest %RSD (8.8%) compared to Method 1 (10.3%) and therefore had the greatest reproducibility (Figure 6-4). Average peak areas for both Method 1 (3447835764 µV) and Method 2 (2044551376 µv) were much greater (~ 1000-fold higher)

than those measured using a gas tight syringe for direct injection at the same concentration (Section 6.3.2.1).

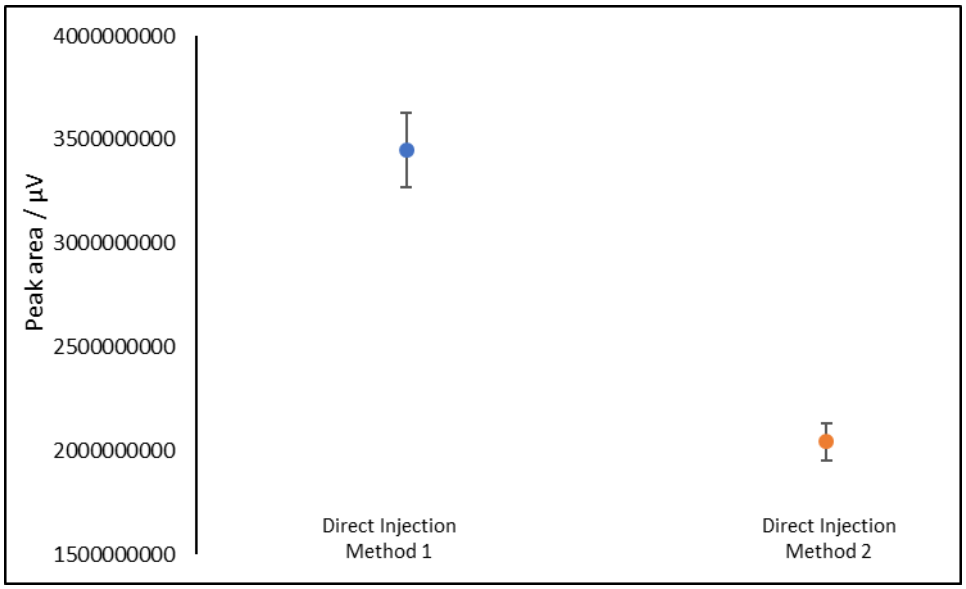


Figure 6-4: Comparison of the reproducibility of two SPME direct injection methods (i.e. Method 1, and Method 2) using a divinylbenzene/carboxen/polydimethylsiloxane, 50/30 µm, 24 ga (grey) fibre at a concentration of 5 ppm alpha-pinene (n = 3) – error bars correspond to %RSD between runs.

6.3.2.3 SPME vial reproducibility experiments

Using the vial method, the white SPME fibre had the lowest %RSD (40.3 %) compared to the grey SPME (85.0 %) fibre and therefore had the best reproducibility (Figure 6-5). Average peak area was 100-fold higher in the grey fibre over the white (127825486 and 4823011 µV, respectively) with both average peak areas significantly lower than the direct injection SPME method described in Section 6.3.2.2.

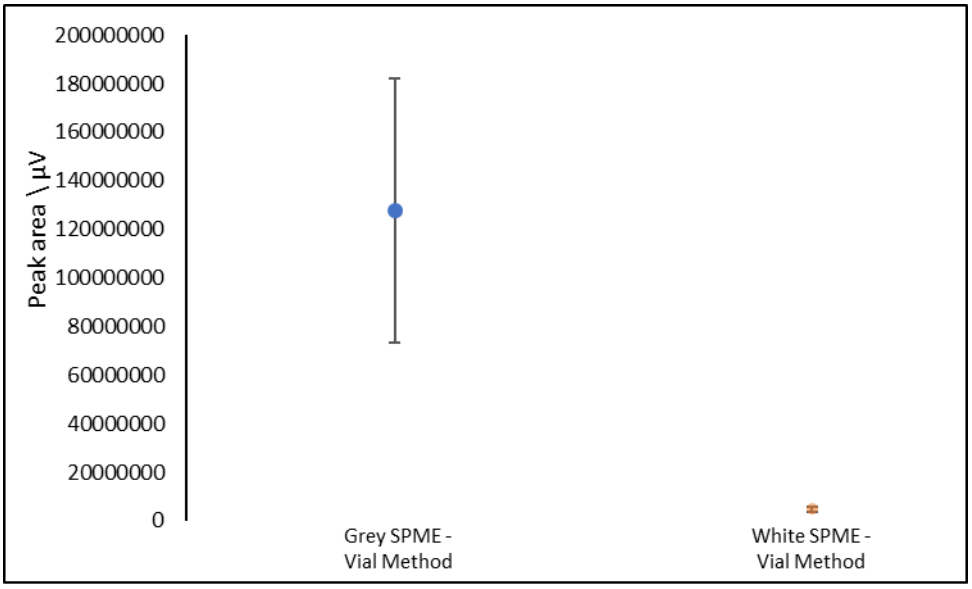


Figure 6-5: Comparison of the reproducibility of two SPME fibres (i.e. polyacrylate – 85µm, 24 ga (white) and divinylbenzene/carboxen/polydimethylsiloxane – 50/30 µm, 24 ga (grey) with the vial method adapted from (Essah and Sanders, 2010) at a concentration of 5 ppm alpha-pinene (n = 3) – error bars correspond to %RSD between runs.

6.3.3 Linearity experiments

6.3.3.1 Gas-tight Syringe — direct injection linearity experiments

Both Method 1 & 2 possessed a low %RSD in the previous experiment (Section 6.3.2.1) and were therefore chosen for a linearity assessment. Three concentrations encompassing 2.5 ppm were chosen to assess both Method 1 (2, 5 and 10 ppm) and Method 2 (3, 4 and 5 ppm). Method 1 produced a linearity curve with an excellent fit (R^2 = 0.9958) according to the prior stated criteria (Section 2.4; Figure 6-6). However, %RSD was high for all tested concentrations i.e. 62.7, 23.8 and 11.5 %, respectively and would be considered unacceptable as per prior stated criteria (Section 2.4).

Experiments undertaken using Method 2 produced a linearity curve with a poor fit for quantitative experimental purposes (R^2 = 0.7621; Figure 6-7). However, %RSD across studied concentrations (22.7, 9.8 and 9.1 %, respectively) were much lower than with Method 1

(Figure 6-7), with concentrations at 5 and 10 ppm considered acceptable as per prior stated criteria (Section 2.4).

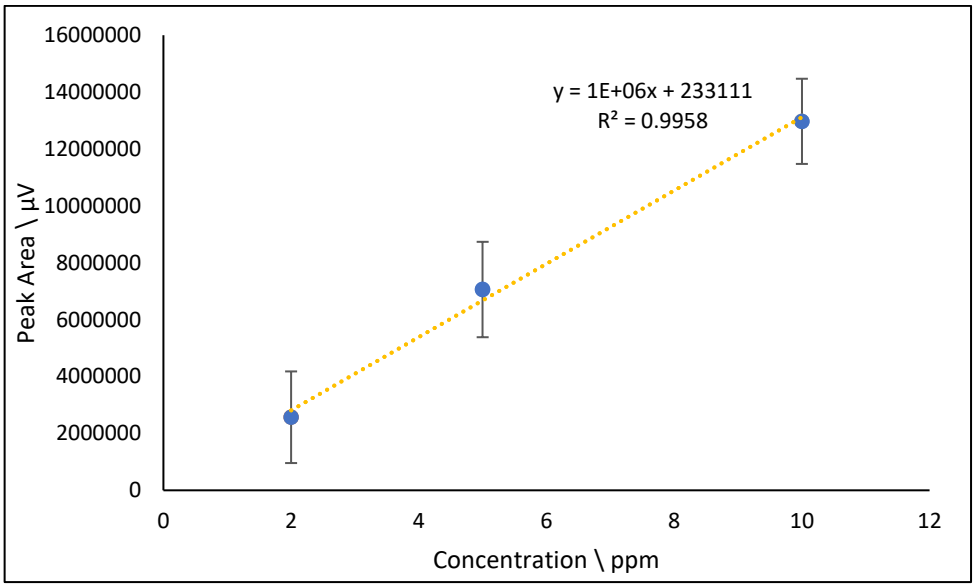


Figure 6-6: Linearity plot of alpha-pinene using Method 1 at concentrations of 2, 5 and 10 ppm (n = 3) – error bars correspond to %RSD between runs.

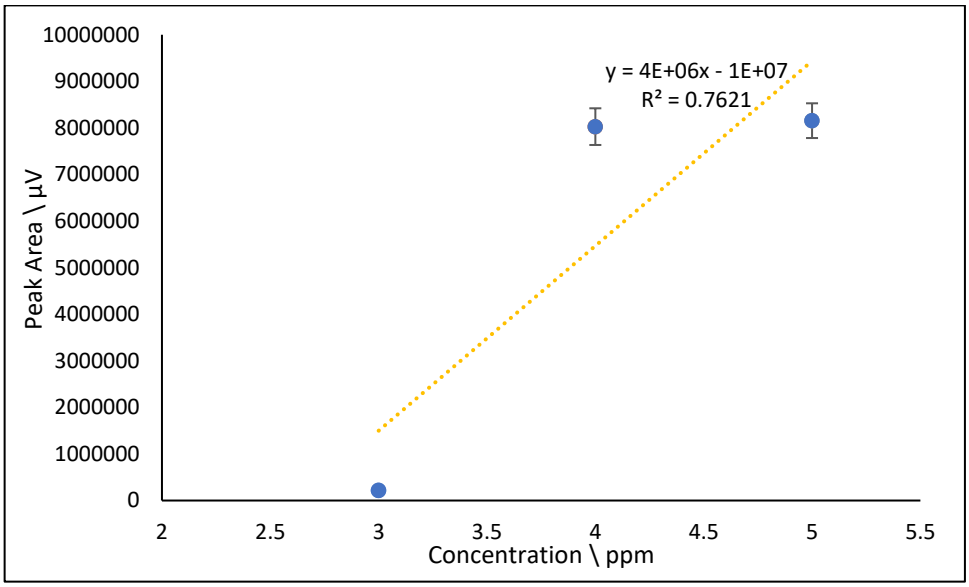


Figure 6-7: Linearity plot of alpha-pinene using Method 2 at concentrations of 3, 4 and 5 ppm (n = 3) – error bars correspond to %RSD between runs (error bar too small at 3 ppm to show).

6.3.3.2 SPME direct injection linearity experiments

Direct injection with the grey SPME fibre (Section 6.3.2.2) produced the lowest %RSD of all the SPME based experiments and was therefore chosen for a linearity assessment. The fit between concentrations (3, 4, 5 and 6 ppm) were too poor to produce a trendline and R^2 value for the experiment (Figure 6-8). %RSD across studied concentrations (0.9, 4.1, 4.4 and 4.0 %, respectively) were low and would be considered acceptable for quantitative analysis as per the prior mentioned criteria (Section 2.4).

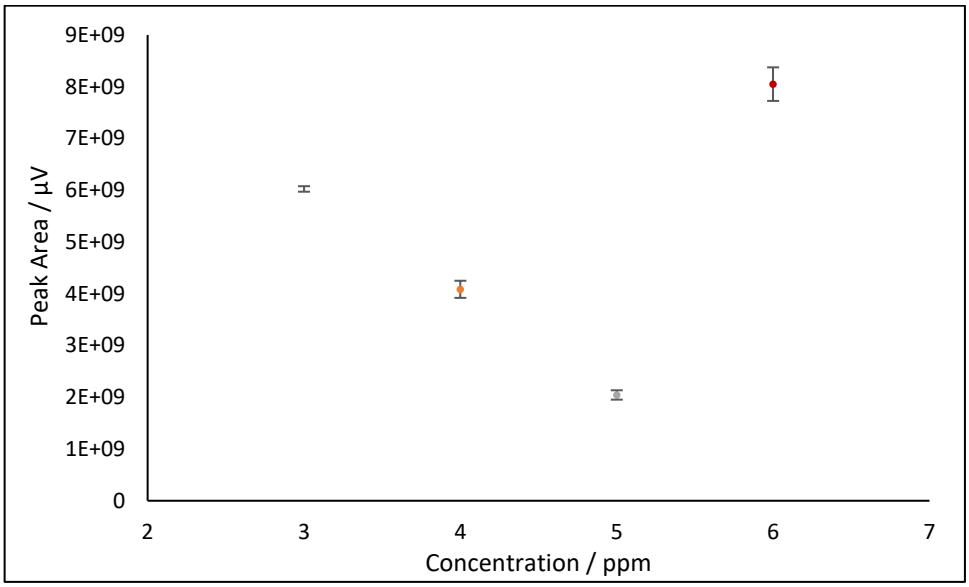


Figure 6-8: Linearity plot of alpha-pinene using Method 2 with an SPME direct injection via a divinylbenzene/carboxen/polydimethylsiloxane – 50/30 µm, 24 ga (grey) fibre at concentrations of 3, 4, 5 and 6 ppm (n = 3) – error bars correspond to %RSD between runs.

6.4 Discussion

The current work is the first to assess the feasibility of SPME as a sampling technique for plant-VOC removal experiments.

In this study it is demonstrated that the SPME has little potential as a sampling technique and should not replace the current – and most commonly used – technique i.e. the gas-tight

syringe. Experiments demonstrated that although the reproducibility with the SPME were significantly improved for the same concentration (in comparison to the gas-tight syringe) linearity was poor, deeming the technique unsuitable for experiments of this type. However, peak areas when using the SPME were significantly larger for the same generated concentration, suggesting that lower concentrations could be more easily measured, without going under the LOQ (Section 6.3.1). Additionally, the author could not replicate the promising results using the SPME with vials as described in (Essah and Sanders, 2010), and the method was not therefore assessed for linearity due to poor reproducibility.

For the reproducibility experiments, the gas-tight syringe direct injection and particularly Method 2 produced the lowest %RSD with both the gas tight syringe and SPME. This was to be expected as it removed the error associated with generating a new concentration for each run. However, this worsened the fit for the linearity plot and exacerbated any possible error associated with the initially generated concentration. The direct injection SPME experiments had better reproducibility in both Method 1 & 2 than the associated gas tight syringe experiments. SIM was trialled (Section 6.3.2.1) as it was hypothesised to give a more accurate peak area and in turn produce a greater reproducibility however, experiments demonstrated this was not the case.

SPME vial experiments were assessed on the premise of a previous study reporting linearity curves with $R^2 > 0.99$ and reproducibility of ± 3 % at low ppb pollutant concentrations (xylene and toluene) (Essah and Sanders, 2010). Results from the study were unable to replicate these results with the two fibres (white and grey) trialled measuring a %RSD of 40.3

and 80.5 % respectively – unsuitable as per the criteria required namely, < 20 %. Therefore, a linearity experiment was not undertaken using this method.

Techniques with the best reproducibility (i.e. direct injection - gas tight syringe Method 1 & 2 and SPME Method 2) were assessed for linearity. Results indicated that although Method 2 produced good reproducibility the linearity of the method was poor and unsuitable. Method 1 with the gas tight syringe produced an excellent fitting linearity curve ($R^2 > 0.99$), but poor reproducibility at concentrations < 10 ppm.

The assessment of SPME as a viable method for gas sampling has been undertaken in numerous studies; excellent linearity fits ($R^2 > 0.99$) and good reproducibility (%RSD < 10 %) with different pollutants, fibres and concentrations have been previously reported (Koziel and Pawliszyn, 2001; Koziel, Noah and Pawliszyn, 2001; Larroque, Desauziers and Mocho, 2006a; Larroque, Desauziers and Mocho, 2006b; Essah and Sanders, 2010). Alpha-pinene has also been investigated with various SPME fibres, often as part of a mixture of pollutants (Martos and Pawliszyn, 1997; Larroque, Desauziers and Mocho, 2006a; Larroque, Desauziers and Mocho, 2006b).

One study validated an SPME experimental method with alpha-pinene at a concentration of 34000 µg m^{-3} (6.1 ppm) for reproducibility experiments and between 0.6 to 1333 µg m^{-3} (0.00011 to 0.24 ppm) for a linearity assessment with a poly(dimethylsiloxane) (PDMS) fibre. Results demonstrated a high reproducibility across two days (%RSD 2.4 and 2.2 %, respectively; n = 10) and a $r^2 = 0.99$ (Martos and Pawliszyn, 1997).

As an acceptable linearity curve was produced in this study with the gas tight syringe using Method 1, it is likely that the problem was not with the rig or other parts of the method

(Section 6.2.1) but the SPME itself. Moreover, as a variety of different fibres were tested it is likely that the problem lies not with the adsorption process, but with desorption and namely, transfer to the analytical instrument. As the rig and experimental instrument were not located in the same lab, unfortunately, movement of the SPME and gas-tight was required post-sampling. It is therefore likely, that the alpha-pinene was preserved better in the gas-tight syringe than on the SPME fibre — explaining the variable peak areas measured during the linearity experiments for the same rig generated concentrations.

This current work suggests that although there was improved reproducibility with the SPME at low concentrations, linearity was poor, and the gas tight syringe can currently still be considered the best option for this type of experiment. Future studies should investigate the feasibility of viable alternatives such as utilising using proton-transfer-reaction mass spectrometry (PTR-MS) for analysis. Prior studies have found that at very low ppb concentrations (75 -750 ppb) of methyl ethyl ketone, an overall error of just 0.8-2.9 % was recorded in comparison with 4.3-14.2 % for the same analysis undertaken with gas chromatography (Tani *et al.*, 2007). Furthermore, alongside ensuring the rig and instrument are located next door to each other, the use of Tenax tubes/traps and gas flow through methods from the rig to the analytical instrument directly should also be further explored (Tholl *et al.*, 2005; Tani and Hewitt, 2009; Kim *et al.*, 2011; Kim *et al.*, 2016; Gong *et al.*, 2019).

6.4.1 Limitations of the study

Due to the PI's move to the University of Birmingham, experiments had a firm end date and additional exploration of differing sampling techniques such as the traps and flow through

systems mentioned above were unable to be explored. Experimental equipment including the rig system had to be left at the University of Reading for other students and thus, other avenues of experimentation namely CO_2 removal experiments were transferred across to the University of Birmingham.

6.5 Conclusion

This study investigated the suitability of two different techniques to accurately and reproducibly determine low concentrations of VOCs, utilising alpha-pinene for the validation stage. It was hypothesised that experiments testing the VOC removal ability of various plant species in literature were unable to test at low, *in situ* concentrations because of the sampling technique used — the common gas-tight syringe. Thus, the author contrasted the gas-tight syringe with a new sampling method (for this type of experiment) namely, solid phase micro extraction (SPME). It was hypothesised that this would allow for measurements at typically measured indoor concentrations, with several other prior studies finding good validity.

However, contrary to the other similar studies, this experiment found that SPME had little potential as a sampling technique and should not replace the current – and most commonly used – technique namely, the gas-tight syringe. This study found that no method tested could provide both adequate reproducibility and linearity together when low concentrations (in the range of 3 – 10 ppm were used), rendering them unfeasible. However, due to prior experiments having success with SPME, further research in this area should be considered.

7.0 Discussion & Concluding Remarks

The field of research focusing on plants and their ability to improve indoor air quality is relatively new but rapidly evolving. The first, now somewhat disputed studies were undertaken by NASA scientist Dr Wolverton in the 1980s looking for ways to clean the air passively, for future dwellings in space (Wolverton and Mcdonald, 1982; Wolverton, McDonald and Watkins, 1984; Wolverton, Johnson and Bounds, 1989; Wolverton and Wolverton, 1993). Issues with these experiments were first noticed by the US Environmental Protection Agency (EPA) (Girman, 1992). This dispute mainly focused on a lack of volumetric considerations when extrapolating to the room scale (i.e. no consideration for how the removal rate would reduce in a larger volume of space), meaning that rates were greatly overestimated often by a factor of 100. However, despite the limitations of the pioneering work, these studies still form the foundation of numerous more recent studies (Baosheng *et al.*, 2009; Chun *et al.*, 2010; Sriprapat and Thiravetyan, 2013; Boraphech and Thiravetyan, 2015; Toabaita, Vangnai and Thiravetyan, 2016; Boraphech *et al.*, 2016), with certain aspects such as the static chamber setup are still considered, in this author's opinion and by some others, the best way to measure pollutant removal (Tholl *et al.*, 2006). Post-2000, the frequency of experiments began to increase with the harmful indoor pollutants benzene and formaldehyde predominately tested in both static and dynamic setups with experiments also adding plants to 'real-life' office environments to measure removal. Furthermore, investigation of the growing media (GM) impact on removal was undertaken with hydroculture based research (Orwell *et al.*, 2004; Wood *et al.*, 2006; Orwell *et al.*, 2006; Kim *et al.*, 2010; Irga, Torpy and Burchett, 2013).

More recently, work has begun to focus on indoor green walls and especially active green walls, where air is pushed through the GM via fans, enhancing removal (Torpy *et al.*, 2018; Pettit, Irga and Torpy, 2019; Pettit *et al.*, 2019). This is especially true of VOCs, where numerous studies have found the GM — and the microorganisms within — predominately associated with any measured removal (Cruz *et al.*, 2014; Kim *et al.*, 2018). This increased rate of removal over traditional potted plants means that active green walls can compete with the removal rates of portable air cleaners — with the added psychological and productivity benefits associated with indoor planting (Shibata and Suzuki, 2004; Thomsen, Sonderstrup-Andersen and Muller, 2011; Nieuwenhuis *et al.*, 2014; EPA, 2018). However, alongside active green walls being anecdotally, more expensive to purchase relative to potted plants, the energy required to run the fans and irrigation systems is moving away from the built environments and UK governments drive towards net zero carbon (The Lancet, 2019), and an ever more climate conscious society (Tompkins *et al.*, 2010).

Traditional potted plants can be considered a relatively passive resource — apart from the watering required — and are currently more realistic for research purposes in terms of budget, and the space and resources required. If the future of pollutant removal is moving towards green walls, new and prior experiments on potted plants still add value through determination of the most effective species to be utilised and studied in green walls (Torpy, Zavattaro and Irga, 2017).

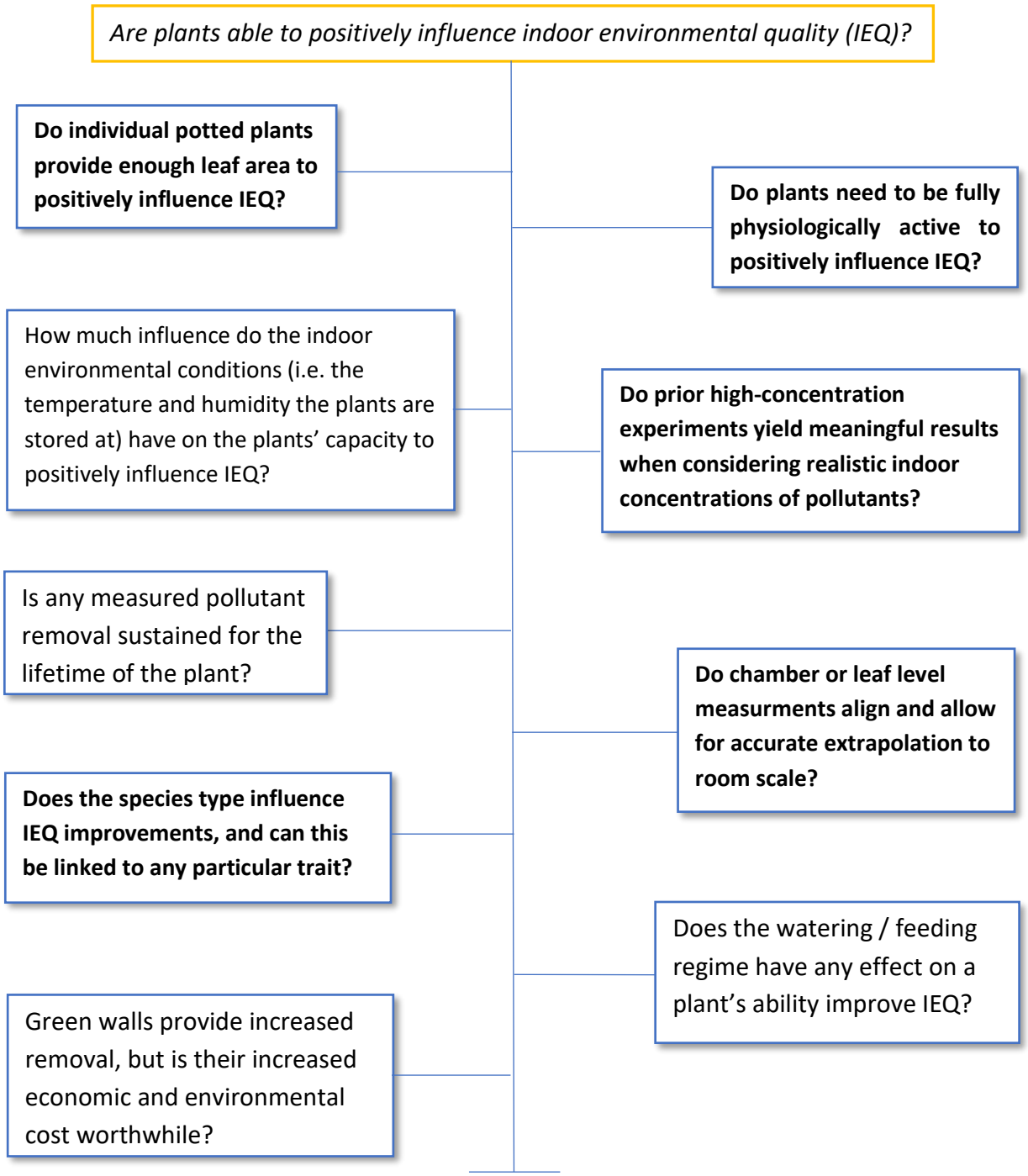


Figure 7-1: A framework of questions which need to be addressed to determine if plants can positively influence indoor environmental quality (which encompasses air quality and humidity) — questions in bold have been addressed at least in part by this study.

Figure 7-1 presents an overarching question of whether plants — in any of their numerous forms (i.e. potted plants, green walls) — provide measurable improvements to indoor environmental quality (which covers both indoor air quality and humidity) at the room scale. To establish this, a number of secondary questions need to be addressed, with those highlighted above — at least in part — considered in this study and discussed below.

7.1 Do individual potted plants provide enough leaf area to positively influence indoor environmental quality?

A recent review paper (Cummings and Waring, 2019) analysed 12 previously published potted plant VOC removal studies and translated all the results into clean air delivery rate (CADR). The review found that the median CADR would require the placement of 10–1000 plants per m^2 of floor area to achieve the same removal rate that outdoor-to-indoor air exchange already provides in typical buildings (~1 h^{-1}) (Cummings and Waring, 2019). However, the studies reviewed in the paper are investigating a small number of VOCs only, with the review not taking into account the differing toxicities across all VOCs and indoor pollutants where a small CADR may still be effective if the pollutant is highly toxic at a relatively low concentration i.e. NO_2 and many SVOCs (WHO, 2010; Krol, Zabiegala and Namiesnik, 2011). Additionally, predicting an air exchange rate for a typical building as done in the (Cummings and Waring, 2019) meta study, even for comparative purposes is far too simplistic, as each building would have differing ventilation types (mechanical or natural) and a wide array of other factors including form, shape, and wind direction to consider (Chartered Institution of Building Services, 2005; HM-Government, 2015). Therefore, in this study, more realistic comparisons have been used to estimate the impact of plants namely, for CO_2, comparing removal to the hourly grams of CO_2 produced by humans (Chapter 3), or

the time taken to reduce concentrations to a near-ambient concentration of 600 ppm (where the majority of associated health and cognitive issues are avoided; Chapter 4). With NO_2, we estimated the concentration removed per hour and compared this to a renowned clean air zone policy, whereby, measures are implemented to reduce ambient NO_2 concentrations (Chapter 5; Castro, Künzli and Götschi, 2017).

The present study investigated pollutant removal of individual potted plants only. But, as highlighted in the introduction to this discussion, current and previous work utilising plants in this format has value and can contribute alongside green wall research. This study found that concerning the pollutant CO_2 — along with a very high light level requiring additional supplementary lighting — a large number of plants are required to achieve measurable removal on a room scale. Specifically, leaf level extrapolations to room scale found 150 of the best performing plant — namely, *Spathiphyllum wallisii* 'Verdi' in 'wet' GM and at 'very high' light — would be required to offset a human's CO_2 contribution indoors (30 g/hour; Chapter 3). Chamber level estimations suggested that in a static room, 14 plants (2 m^2 of leaf area) would require 12 hours to reduce concentrations to a near-ambient 600ppm or approximately 75 plants to remove a human's CO_2 contribution indoors — both, unrealistic requirements for indoor environments (Chapter 4). Furthermore, similar results were found in literature whereby, plant numbers often into the hundreds were required for measurable room scale removal of CO_2 (Pennisi and van Iersel, 2012; Torpy, Irga and Burchett, 2014).

Thus, only the higher number and density of plants provided by green walls offers the potential of room scale concentration change. Especially with an active green wall, where Torpy *et al.* estimated that *Chlorophytum comosum* would be capable of removing 11 g of

CO_2 per hour in a 16 m^3 room (Torpy, Zavattaro and Irga, 2017). In comparison, this study estimated that the best performing species in traditional potted plant form and with a comparable light level and GM type to *Torpy et al.* removed only 0.75 g of CO_2 per hour and 3 g per m^3 of CO_2 per hour in a 15 m^3 room (leaf and chamber level respectively; Chapter 3 and 4).

However, the study investigating NO_2 removal (Chapter 5) showed more promise for individual potted plants. It concluded that a potted plant could remove 0.62 ppb per plant per hour in a 15 m^3 static room — at typical indoor light levels. Consequently, the placement of five houseplants within the room, could equate to a removal in such a small and unventilated indoor environment roughly equivalent to that of a successful clean air policy in Switzerland, which was suggested to have lowered the NO_2-related deaths in the region by 51 and the life-years lost by 550 years (Castro, Künzli and Götschi, 2017).

It can therefore be suggested that, for the studied pollutants CO_2 and NO_2 and many other pollutants already studied in literature (such as benzene and formaldehyde) potted plants do not provide enough leaf to measure significant room level concentration reductions. Many other indoor pollutants — including NO_2 until very recently (Pettit *et al.*, 2019) Chapter 5) — which are yet to be studied, may yield more significant results thus, disregarding plants as a passive removal technique altogether as suggested by (Cummings and Waring, 2019) is likely premature.

In terms of indoor environmental quality, a number of previous experiments have investigated plants for their ability to raise RH indoors via the natural process of transpiration. Results however, in 'real-life' environments have not clearly confirmed the

expected RH increases (Lohr and PearsonMims, 1996; Lim *et al.*, 2009; Jeong *et al.*, 2008; Pegas *et al.*, 2012). On review of these experiments, it can be suggested that if enough plants are present (> 5% of the floor area), humidity increases were measured, with species type and transpiration ability very likely playing a key role and an area where further research is required (Jeong *et al.*, 2008; Gubb *et al.*, 2018)

7.2 Do plants need to be fully physiologically active to positively influence the indoor environmental quality?

This thesis investigated if species need to be functioning optimally to effectively remove pollutants. This was achieved by measuring the removal ability of the studied species (Figure 2-1) under differing environmental conditions which are clearly known to influence physiological function (Sailsbury and Ross, 1991). Specifically considered for this study were moisture content of the GM, lighting conditions and GM type.

7.2.1 Substrate moisture content (SMC)

Soil water deficit is the main environmental factor which effects photosynthesis and respiration, it also causes stomatal closure and reduced mesophyll and stomatal conductance — and thus, is likely to effect pollutant removal especially if it is via the stomatal pathway (Lawlor and Cornic, 2002; Flexas et al., 2006). Additionally, anecdotally it is well known that plants may often be left to dry out, hence, how they perform under conditions of drought stress in terms of changes to CO_2 assimilation or respiration is important to quantify. Measurement of the SMC has been overlooked in all other experiments of this type in literature. Therefore, the author felt it important to measure SMC on all experiments in this study which utilised GM (Chapters 3 - 5).

However, in the leaf scale experiments (Chapter 3), only one studied species measured statistically significant differences between 'dry' and wet' GM (at both high and low light levels). This was mirrored in chamber level experiments (Chapter 4), where only infrequent statistically significant differences between 'dry' and wet' GM (at both high and low light levels) were measured. Therefore, it can be concluded that moisture content of the GM did not have the hypothesised effect namely, that drought stressed plants would not remove as much CO_2 as well watered ones for any of the studied species. It may be that because the measured CO_2 assimilation rate was generally low (below 3 µmol m^{-2} s^{-1}) — even at 'very high' light levels — in comparison to what would be measured for example, with an outdoor species in direct sunlight (e.g. upwards of 10 µmol m^{-2} s^{-1}) differences are hard to quantify. Thus, it can be suggested that the effect of a 'dry' GM and a reduced stomatal aperture on CO_2 assimilation is much less pronounced when the stomatal conductance is already low.

This was also mirrored in the NO_2 experiments presented in Chapter 5, whereby, no statistical differences were measured between 'dry' and 'wet' GM in terms of plants' ability to remove NO_2 and reduce its concentration in the experimental space. This suggests that if the stomatal pathway of removal for various airborne pollutants is not particularly active, moisture content makes little difference to removal albeit, if still within biological and practical horticultural ranges (15 – 20 %). This was supported by both experiments in this current study. Namely, the NO_2 experiments, where the GM and not the stomata was found to be the predominate pathway of removal (Chapter 5) and with CO_2 experiments, where measured assimilation and thus stomatal conductance was relatively low (Chapter 3 & 4).

7.2.2 Growing Media

Past experiments investigating the ability of plants to remove VOCs showed that the removal of these compounds by plant species was predominately associated with the microflora in the GM, with plants themselves only having an indirect effect to maintain and support GM microorganisms (Wood *et al.*, 2002; Orwell *et al.*, 2004; Kim *et al.*, 2008; Cruz *et al.*, 2014; Irga, Pettit and Torpy, 2018; Kim *et al.*, 2018). These microorganisms, especially those associated with the roots and GM — where metabolising ability has long been established, as opposed to the lesser researched phyllosphere — have been shown to degrade, detoxify or sequester an array of different pollutants including VOCs, PM and certain inorganic compounds (Weyens *et al.*, 2015). The author, therefore, felt the testing of more than one GM — preferably with as differing composition as possible and thus, likely supporting different microflora — was worthwhile because of their significance to other similar experiments (Wood *et al.*, 2002; Orwell *et al.*, 2004; Kim *et al.*, 2008; Cruz *et al.*, 2014; Irga, Pettit and Torpy, 2018; Kim *et al.*, 2018).

For experiments undertaken at the chamber scale (Chapter 4), two GM were selected — peat and peat-free — and measurements were contrasted for differences in CO_2 removal ability. Prior experiments of this type often utilised peat-free GM, hence, the intention was to contrast this with a peat-based GM (Torpy, Irga and Burchett, 2014; Torpy, Zavattaro and Irga, 2017).

The results suggested that the CO_2 removal ability of all studied plant species was influenced by the type of GM selected, with peat GM contributing to a greater reduction of CO_2 across most species in the experimental chamber at a high light level (~ 22 200 lux, Chapter 4). This

may be due to certain plants establishing and growing better in certain types of GM (Young *et al.*, 2014; Charoenkit and Yiemwattana, 2016; Pettit, Irga and Torpy, 2018) or the additional carbon present in peat, both explored in Chapter 4. Moreover, considering the NO_2 experimental results presented in Chapter 5, whereby, no statistical differences were measured between GM type (peat based and peat free), it lends further weight to the additional carbon hypothesis for CO_2 removal concerning peat.

7.2.3 Light Level

Plants utilise a wide spectrum of light which includes visible and UV in fundamental processes such as photosynthesis and depend on it for survival. In terms of removal of gaseous pollutants by plants, light generally increases both the efficiency and rate of sequestration of various compounds including formaldehyde (Xu, Wang and Hou, 2011; Cruz *et al.*, 2014) up to a plant's light saturation point, with the effect depending on the pathway of removal. Previous studies have shown that light has the most effect when the stomatal pathway is responsible for removal (Cruz *et al.*, 2014; Torpy, Irga and Burchett, 2014; Gubb *et al.*, 2018) — the removal of benzene and toluene via both cuticle and microorganism pathways showed little change with varying light levels in previous work (Godish and Guindon, 1989; Wood *et al.*, 2002; Orwell *et al.*, 2004; Cruz *et al.*, 2014).

In this study, all chapters which have involved plant-based experiments (Chapters 3, 4 & 5), have had light levels carefully controlled or measured throughout. With CO_2, light levels significantly influence stomatal opening and gas exchange — the main pathway for CO_2 assimilation (Pennisi and van Iersel, 2012; Torpy, Irga and Burchett, 2014; Torpy, Zavattaro and Irga, 2017; Gubb *et al.*, 2018). Additionally, with NO_2 removal experiments, stomata

were originally considered a likely removal pathway based on prior studies (Nowak *et al.*, 2014; Gourdji, 2018) and thus, the light level needed appropriate consideration. Alongside this, as NO_2 can also degrade photochemically under UV light, blank experiments (without GM or plant) were run in parallel with the experiment, confirming any measured removal was only from the plant or GM itself (Chapter 5).

Experiments found that for CO_2, light level played a very significant role in the amount assimilated by the studied plant species (Chapter 3 & 4). Little removal was measured at typical indoor light levels (0 – 500 lux), whereas, at levels where supplementary lighting would be required i.e. ~ 22200 lux significantly greater removal of CO_2 was measured. Results therefore, pointed towards a predominately stomatal removal pathway for CO_2, as previously predicted, with little CO_2 assimilation from other possible pathways (e.g. via the GM). This aligns with previous studies, (Pennisi and van Iersel, 2012; Torpy, Irga and Burchett, 2014; Torpy, Zavattaro and Irga, 2017; Gubb *et al.*, 2018) which have found along with large numbers of plants that a very high light level is required for any 'considerable' room-scale removal of CO_2.

With NO_2 removal experiments (Chapter 5), results suggested differences between the studied light levels (0 and 500 lux) were negligible, with the GM and not the stomata being the key contributing pathway of removal. It is likely that assimilation would further increase at higher light levels, however the aim of this study was to test typical indoor light levels — not requiring supplemental lighting. Consequently, the results from these experiments are much more positive than CO_2, whereby NO_2 removal appeared to take place passively, in

typical indoor environments and without supplementary lighting requiring additional cost and energy use.

7.3 Do prior high-concentration experiments yield meaningful results when considering realistic indoor concentrations of pollutants?

As highlighted in the introduction (Chapter 1), prior studies present two main sets of shortcomings. Namely, that they report on pollutants which have little practical relevance (e.g. are infrequently present indoors or in concentrations too low to cause damage to human health, for example, toluene, formaldehyde trimethylamine and various aldehydes and ketones) (De Kempeneer *et al.*, 2004; Tani *et al.*, 2007; Tani and Hewitt, 2009; Kim *et al.*, 2011; Siswanto, Chhon and Thiravetyan, 2016). Or – they measure removal ability with pollutant concentrations up to 1000-fold greater than what is typically measured *in situ* (Aydogan and Montoya, 2011; Cruz *et al.*, 2014; Torpy *et al.*, 2018). This renders a large number of the studies and subsequent results inaccurate and potentially misleading (Wolverton and Wolverton, 1993; Lim *et al.*, 2009; Aydogan and Montoya, 2011; Jin *et al.*, 2013). This is not to suggest that all studies fit into the above, many have significantly advanced the field (e.g. Orwell *et al.* and the first studies in 'real life' offices environments, Torpy/Pettit *et al.* with pioneering active green wall work and the first testing in a 'real-life' classroom and Irga *et al.* for their work on hydroculture, confirming the significant influence of GM) and along with many others, provided robust evidence for plants' contribution to indoor pollutant removal (Wood *et al.*, 2006; Irga, Torpy and Burchett, 2013; Torpy, Zavattaro and Irga, 2017; Torpy *et al.*, 2018).

This study attempted to address one of the above issues, by investigating the suitability of different techniques to determine low concentrations of VOCs accurately and reproducibly (Chapter 6). To achieve this, a comparison between the traditionally used gas-tight syringe (De Kempeneer *et al.*, 2004; Orwell *et al.*, 2004; Orwell *et al.*, 2006; Yoo *et al.*, 2006; Yang *et al.*, 2009; Irga, Torpy and Burchett, 2013; Boraphech *et al.*, 2016; Siswanto, Chhon and Thiravetyan, 2016) and a novel SPME sampling method was undertaken. In contrast, to previous studies which displayed promising results (Martos and Pawliszyn, 1997; Larroque, Desauziers and Mocho, 2006a; Essah and Sanders, 2010), we found that little potential was offered by the technique to measure the low VOC concentrations desired.

Results from the experiments suggested that although the SPME could provide improved reproducibility (compared to the gas tight syringe), measured linearity was poor — other studies measured curves with $R^2 > 0.99$ (Martos and Pawliszyn, 1997; Larroque, Desauziers and Mocho, 2006a; Essah and Sanders, 2010) suggesting that utilising the SPME technique generally was not the problem. It was hypothesised that a problem with the adsorption and desorption from the fibre itself was causing the variable peak areas for the same concentration.

As an acceptable linearity curve was produced with the gas tight syringe using Method 1, it is likely that the problem is not with the gas-handling line or other elements of the method (Section 6-2) but the SPME process itself. Two differing fibres were tested, all with varying compositions and designed specifically for VOCs. However, none were able to produce the desired results suggesting adsorption of pollutants onto the fibre was not the main issue. It can therefore be assumed that the transfer from the bulb into the analytical instrument

caused the problem namely, the desorption process. It is likely that moving to a different laboratory to use the analytical instrument influenced the amount of alpha-pinene on the fibre more so than if the concentration was kept within the gas tight syringe — thus, explaining the variable peak areas obtained in the same gas-handling line. While it is not the most satisfactory finding to demonstrate that a SPME method does not offer the anticipated improved performance, the lead supervisor's move from Reading to Birmingham meant a firm end point for this part of the research project outside the control of the candidate.

7.4 Do chamber or leaf level measurements align and allow for accurate extrapolation to room scale?

The experiments carried out in Chapters 3 & 4 were designed to separately investigate both leaf and chamber scale removal of CO_2. In horticultural settings, the infrared gas analyser (IRGA) is a common piece of equipment for measuring gas exchange and a variety of other parameters at the leaf scale. While leaf-level measurements provide important, intrinsic information about the plant activity and function, their viability for room-scale relevance has been questioned (Torpy, Zavattaro and Irga, 2017). At the chamber scale, results were hypothesised to provide more accurate estimations for how plants can influence 'room-scale' concentrations of CO_2.

The results from this study for both the leaf and chamber scale CO_2 experiments — at the lower light levels typically measured indoors (0 – 500 lux) — found nearly all studied species to be respiring. However, any subtle, intrinsic differences measured between plant species across both experiments were not consistent, with *Hedera helix* potted in the same GM respiring the most at chamber scale (in 'wet' substrate) and *Dracaena fragrans* 'Golden

Coast' at respiring the most at the leaf scale. It could be suggested however, that with such low respiration measured across plant species, any differences between scales are at best minimal, and likely negligible.

At higher light levels (~ 22 200 lux), a similar result was found whereby, the best performing species (i.e. those removing most CO_2) were not consistent between scales. Additionally, quantitatively, the differences between removal of the best performing species on extrapolation to room level was significant whereby, at a comparable light level and with the same GM type, leaf level experiments found that 0.75 g of CO_2 was removed per hour and chamber results estimated 3 g per m^3 of CO_2 would be removed per hour inside a 15 m^3 static room (Chapter 3 and 4). Moreover, with little alignment between leaf and chamber scale results, accurate extrapolation to the room scale will likely present further challenges. Where experiments have been undertaken in 'real life' environments with green walls or botanical systems the CADRs range is often large due to the use of different room sizes and layouts for experiments, alongside differing plant species, GM, and different units of measurement (Wang and Zhang, 2011; Pettit, Irga and Torpy, 2019).

A number of assumptions have to be made when extrapolating from the leaf or chamber to room scale. This includes the effect of pollutant diffusion and how this would differ between scales, alongside the differences in transport on individual pollutants (Gong et al., 2010; Soreanu and Dumont, 2020; Deng and Gong, 2021). Furthermore, mass transfer resistance from the bulk air to the boundary layer is considerably less in chambers with fans than 'real-life' environments and would likely affect removal ability (Soreanu and Dumont, 2020).

7.5 Does the species type influence IEQ improvements, and can this be linked to any particular trait?

Experiments investigating CO_2 removal found that at the higher light level (~ 22 200 lux) little statistical difference was measured between species at the chamber scale whereas, at the leaf scale, all three chamber-studied species (*Hedera helix*, *Spathiphyllum wallisii* 'Verdi' and *Dracaena fragrans* 'Golden Coast') measured statistical differences in CO_2 uptake. Consequently, it could be argued that in 'real life' environments as the scale becomes larger, differences between species look less likely to have an impact. This was mirrored with the NO_2 experiments presented in Chapter 5, where once an extrapolation to room level is made, differences between the species shrink further. However, concerning measured ET rate, the experiments in Chapter 3 align with the conclusions of (Jeong *et al.*, 2008) whereby, each individual species clearly possesses a differing transpiration ability.

In terms of identifying traits linked to more effective CO_2 removal, several species were found to be superior in removal terms between the chamber and leaf scale. In terms of chamber level experiments, *Dracaena fragrans* 'Golden Coast' was the best performing in the CO_2 removal experiments per m^2 of leaf area and the best potted plant, but not per m^2 of leaf area for NO_2 removal under certain, but not all environmental conditions tested. Furthermore, when extrapolated to room level, little difference was measured between any species or the GM itself in terms of NO_2 removal ability. With GM as efficient i.e. equal within error for *Spathiphyllum wallisii* 'Verdi' and *Dracaena fragrans* 'Golden Coast'.

Thus, defining a species as the best remover even for individual pollutants and determining which specific traits may be behind this ability was not possible from this particular study.

Experiments in literature have also failed to identify a clear explanation. However, differences in plants' capacity for CO_2 uptake were measured down to a cultivar level (Chapter 3), and for other pollutants in literature (Cruz *et al.*, 2014), suggesting that a particular family, or even species cannot be grouped together as particularly effective removers. It is clear that any trait causing differences in removal ability is measured at a much more intrinsic scale than was the focus of this current study and therefore, a more detailed investigation of a plant species and its physiological function during experiments would be necessary to draw any further conclusions. Moreover, it is likely that because the removal rates for various pollutants including CO_2 are often so low, any subtle, intrinsic differences measured between species are small and the environmental conditions within which the plant is maintained (i.e. light levels) seem to have a more significant influence on pollutant removal. Moreover, it should also be noted that recently, suggestions of species–substrate interaction, where different plant species may affect different substrates in different ways (namely through the chemical effects of root exudates) has been hypothesised but not confirmed. This potentially means that to define the 'best' remover a species-substrate system (not just species of plant) may be required.

However, synergy was measured between plant species concerning CO_2 removal and Evapo-transpiration (ET) rate whereby, species with the highest CO_2 removal at the leaf level possessed the highest ET rate (*Hedera helix* and *Spathiphyllum wallisii* 'Verdi'; Chapter 3). However, in plant physiology terms this is not a particularly surprising result (Sailsbury and Ross, 1991), as vigorous species would be expected to have a high gas exchange rate via the stomata.

8.0 Key Conclusions & Recommendations

Key conclusions and recommendations taken from this study are:

- For measurable CO_2 removal at the room scale, supplementary lighting is required (at ~ 22 200 lux) together with a high number of plants. At typical indoor light levels (0 – 500 lux), little potential is offered for CO_2 removal, however, respiration rates were equally found to be negligible in terms of increasing CO_2 concentrations at the room scale. The type of GM was found to have a significant influence, with peat GM contributing to a greater reduction of CO_2. Additionally, substrate moisture was deemed to have a negligible effect, especially when extrapolated to the room scale.
- Removal of NO_2 from a simulated polluted environment can be achieved at typical indoor light levels, with the GM and its moisture content playing an important role in removal, but the type of GM seemed to have little effect. Derivation of results to the room scale found that plant type also seems to have little effect on removal, but a smaller number of plants would be required for significant (in terms of health impact) room scale concentration reductions than with CO_2.
- Evapo-transpiration (ET) rates of the studied plants varied significantly between species, with physiologically more active species such as *Hedera helix* and *Spathiphyllum wallisii* 'Verdi' offering the most potential to raise RH indoors.
- As a method for measuring low VOC concentrations to improve the current experimental methodology, little potential was offered by SPME as a technique over what has been previously utilised, the gas-tight syringe.

8.1 Future Research Direction

With the rapid rise of COVID-19 across the globe, the quality of our indoor environments and how they can impact our health has risen in prevalence. With this, the importance of adequate ventilation indoors is clearly recognised and further enhanced with filtration techniques to reduce both airborne pathogen transmission and improve indoor air quality (Morawska *et al.*, 2020). These filtration techniques include well-established, engineered solutions such as portable air cleaners and particle or carbon filters on air handling and fan coil units — all of which, require extra energy (when compared to natural ventilation) (ECON19, 2003; Nassif, 2012; Ben-David and Waring, 2016). Thus, the balance between minimising drivers of climate change such as energy consumption and IAQ (i.e. high ventilation rate and low pollutant concentrations), must now be struck (Tompkins *et al.*, 2010; The Lancet, 2019). Anecdotally, this balance has been debated between built environment professionals for some time, but now has heightened awareness because of the current pandemic. Therefore, now more than ever, greater importance should be placed on passive solutions such as indoor plants.

Indoor plants have clearly shown the potential to remove various pollutants which are harmful to health and contributors to poor indoor air quality (Orwell *et al.*, 2004; Kim *et al.*, 2008; Treesubsuntorn and Thiravetyan, 2012; Pettit *et al.*, 2017; Torpy, Zavattaro and Irga, 2017; Pettit *et al.*, 2019). This study lends further support to this, in terms of the pollutants CO_2 and NO_2 (Chapters 3, 4 & 5). However, the best way to utilise plants in terms of form (i.e. green walls or potted plants), number, and environmental conditions is still not clear; turning plants' inherent ability to remove pollutants into measurable room-scale concentration reductions will require further studies, of a specific combination of all three

factors, that are likely different for each plant species and individual pollutant (Cruz *et al.*, 2014; Irga, Pettit and Torpy, 2018).

Results from recent research focusing on active green walls have clearly presented a significant breakthrough in the clean air delivery rate provided, in comparison to single potted plants (Torpy *et al.*, 2018; Pettit, Irga and Torpy, 2019; Cummings and Waring, 2019). Measured removal rates of various pollutants have been found an order of magnitude greater than traditional potted plants with CADRs comparable to portable air cleaners (EPA, 2018). However, only a small number of studies have taken place, measuring a very limited number of both pollutants and plant species with only one study in a 'real-life' setting (Pettit, Irga and Torpy, 2019). Pollutants measured with active green walls so far include PM, CO_2 and methyl ethyl ketone with only a handful of plant species investigated alongside variations in the environmental conditions namely, light level and GM type (Pettit *et al.*, 2017; Torpy, Zavattaro and Irga, 2017; Pettit, Irga and Torpy, 2018; Torpy *et al.*, 2018; Pettit *et al.*, 2019; Pettit, Irga and Torpy, 2019). Future work should focus on testing relevant indoor pollutants identified in Chapter 1, Figure 1-1, at typically measured indoor concentrations. Furthermore, although little difference was measured in this current study, moisture content of a GM, light level and GM type should all be thoroughly varied and investigated in experiments. The reason for this is three-fold: firstly, to seek to maximise potential removal ability, secondly, to simulate the complexity of differing indoor environments and thirdly, to provide information on the pathway of removal utilised by different plant species. Where possible, laboratory experiments should take place alongside experiments in 'real-life' environments to both contrast differences between scales under the same environmental conditions and potentially, present significant concentration

reductions which are hard to contest when measured in a 'real-life' environment. A further consideration for green walls, especially active, is their economic and environmental viability. Both their energy and resource use need thorough investigation along with the cost implications of initial purchase and ongoing maintenance and upkeep. Ultimately, determining whether with a holistic view, this increased removal rate over passive potted plants is worthwhile.

To build on the specific aims of this study set out in the introduction (Chapter 1), experiments should continue to identify a 'bank' of plant traits that link species to a superior removal ability. Results would suggest that this is likely to be pollutant-specific, therefore, once identified, an array of plants with the same trait could all be tested in an attempt to identify species that provide the highest removal ability for individual pollutants. On a wider scale, for landscape architects working in the built environment, this could allow for specific plants to be chosen to remove indoor pollutants which have been identified as a problem through air quality monitoring within a specific space. For example, in urban environments both PM and NO_2 are likely to be an issue but more specifically, if a material has been specified which has been found to off-gas and release benzene for example, retrospective mitigation through plant species type could be an option to reduce concentrations, if the material could not be replaced.

Furthermore, for experiments measuring plants' ability to influence RH indoors, the mixed results in the literature are likely explained by both plant species choice and ventilation rate differences between the studies. In future experiments, both need to be tightly controlled or at least measured especially if the experiments are undertaken in 'real-life' settings; this was

often not the case in prior studies (Lohr and PearsonMims, 1996; Wang and Zhang, 2011; Pegas *et al.*, 2012). Additionally, further experiments should be undertaken investigating green walls and the subsequent effect of the higher number of plant species on the RH. It could be postulated, that the likely increased RH measured in the surrounding area, may reduce plants' ability to remove pollutants at the same rate (Aphalo and Jarvis, 1991).

Moreover, to address the problems with low-VOC concentrations being tested, further experiments with the SPME alongside other sampling strategies should be investigated. Previous experiments have found positive results at low concentrations with proton-transfer-reaction mass spectrometry (PTR-MS) as opposed to the traditional gas chromatography mass spectrometry (GC-MS) instrument and this avenue of experimentation should be explored further (Tani *et al.*, 2007). Additionally, to remove the sampling and transfer issues, further testing with Teflon bags directly flowing the gas into the analytical instrument or onto a trap should be undertaken — however, literature studies using similar methods have often tested at concentrations much higher than what is found in typical indoor environments (Tholl *et al.*, 2005; Tani and Hewitt, 2009; Kim *et al.*, 2011; Kim *et al.*, 2016; Gong *et al.*, 2019).

Finally, research suggests that the conditions in which plants are kept in terms of temperature, humidity and moisture levels may affect the stomata and thus, removal ability within the same plant type (Mott and Parkhurst, 1991; Lawlor and Cornic, 2002; Mena-Petite *et al.*, 2003; Flexas *et al.*, 2006). To effectively measure this, plants should be grown from seed and separated by condition with sufficient variations from 'normal'. If differences are measured in removal ability, an effective supply chain monitoring system would be of great

importance. Additionally, long term removal studies over a period of a year or more would provide insight into whether the same plant is able to provide the same removal rate over longer periods of time, as it ages and its physiology changes — or if replacement over regular periods is required.

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Appendices

Appendix A Pollutants measured in home and office environments

Key:	
S:	Summer
W:	Winter
SP:	Spring
N:	Night
D:	Day
SM:	Smoking environment
NS:	Non-smoking environment
HT:	High traffic area
LT:	Low traffic area
H:	Home > 10 stories high
L:	Home < 10 stories high
HF:	5th - 7th Floor
LF:	1st-4th Floor

Home environments – Alcohols

Home Environment		**µgm-3**		
Alcohols	**Reference**	**Arithmetic Mean**	**Geometric Mean**	**Median**
1,2-Propanediol	(Jarnstrom et al. 2006) (Cheng et al. 2016)	24/30/14 2.4	N/A 0.5	N/A 0.8
1,3-Butanediol	(Jarnstrom et al. 2006)	11	N/A	N/A
1-Butanol	(Jarnstrom et al. 2006) (Geiss et al. 2011) (Zhu et al. 2013) (Guo et al. 2013) (Cheng et al. 2016)	18/15/8 2.5 2.26 N/A 1.6	N/A N/A 1.19 N/A 1.1	N/A 1.8 N/A 9.8/8.6 1.1
1-Butoxy-2-propanol	(Geiss et al. 2011) (Cheng et al. 2016)	12.5 1.4	N/A 0.1	N/A <0.03
1-Methoxy-2-Propanol	(Logue et al. 2011) (Billionnet et al. 2011) (Langer et al. 2016)	N/A N/A N/A	N/A N/A 1.8	1.9 0.90 0.9
1-Pentanol	(Jarnstrom et al. 2006) (Zhu et al. 2013)	3/5 0.71	N/A 0.25	N/A N/A
2-(2-Ethoxyethoxy) Ethanol	(Jarnstrom et al. 2006)	13/5	N/A	N/A
2-Butoxyethanol	(Jarnstrom et al. 2006) (Geiss et al. 2011) (Logue et al. 2011) (Billionnet et al. 2011) (Zhu et al. 2013) (Langer et al. 2016) (Cheng et al. 2016)	3/2 0.6 2.6 N/A 6.16 N/A 1.0	N/A N/A N/A N/A 3.02 1.3 0.4	N/A 0 1.4 0.75 N/A 1.6 0.5

2-ethoxyethanol	(Logue et al. 2011)	0.43	N/A	0.06
2-Ethyl-1-hexanol	(Cheng et al. 2016)	1.2	1	1.1
2-Ethylhexanol	(Jarnstrom et al. 2006)	1/2	N/A	N/A
2-methoxyethanol	(Logue et al. 2011)	0.12	N/A	0.12
2-Methyl-1-Propanol	(Jarnstrom et al. 2006) (Logue et al. 2011)	9/6/6 8.2	N/A N/A	N/A N/A
2-Methyl-2-Propanol	(Zhu et al. 2013)	0.66	0.2	N/A
2-Propanol	(Jarnstrom et al. 2006) (Logue et al. 2011) (Zhu et al. 2013) (Cheng et al. 2016)	38/14/6 18 7.31 12.5	N/A N/A 1.42 2.8	N/A 3.3 N/A 4.8
Benzyl Alcohol	(Jarnstrom et al. 2006)	1	N/A	N/A
Butanol	(Logue et al. 2011)	35	N/A	55
Butoxyethoxyethanol	(Jarnstrom et al. 2006)	20	N/A	N/A
Ethanol	(Logue et al. 2011) (Cheng et al. 2016)	860 >131	N/A 55.6	160 >62
Isobutanol	(Cheng et al. 2016)	1.3	0.8	0.9
Menthol	(Cheng et al. 2016)	0.9	0.4	0.4
Phenoxyethanol	(Jarnstrom et al. 2006)	3	N/A	N/A

Office environments – Alcohols

Office Environment **µgm-3**

Alcohols	Reference	Arithmetic Mean	Geometric Mean	Median
1,2-Propanediol	(Salonen et al. 2009)	7	N/A	3
1-Butanol	(Brickus et al. 1998) (Tham et al. 2004) (Zuraimi et al. 2006) (Salonen et al. 2009)	20.3 / 20.7 / 27.6 / N/A 0.3 15.8 2.2	N/A N/A N/A N/A	N/A N/A N/A 1.0
2-(2-Ethoxyethoxy) Ethanol	(Salonen et al. 2009)	7.7	N/A	0
2-Butoxyethanol	(Tham et al. 2004) (Mandin et al. 2017)	2.6 S:5.7 W:2.7	N/A N/A	N/A S:2.5 W:0.4
2-Ethylhexanol	(Tham et al. 2004) (Zuraimi et al. 2006) (Mandin et al. 2017)	17.2 1.5 S:4.7 W:3.9	N/A N/A N/A	N/A N/A S:3.8 W:2.3
2-Fenoxyethanol	(Salonen et al. 2009)	1.6	N/A	0
2-Propanol	(Tham et al. 2004)	25.8	N/A	N/A
Ethanol	(Zuraimi et al. 2006)	20.8	N/A	N/A

Home environments - Aldehydes and Ketones

Home Environment		**µgm-3**		
Aldehydes/Ketones	**Reference**	**Arithmetic Mean**	**Geometric Mean**	**Median**
1-Propanal	(Zhu et al. 2013) (Uchiyama et al., 2015)	0.33 W:4.1 S:7.4	0.09 N/A	N/A W:2.5 S:5.3
2- Nonenal	(Uchiyama et al., 2015)	W:0.6 S:0.3	N/A	W:0.1 S:0
2,5-dimethoxybenzaldehyde	(Uchiyama et al., 2015)	W:0.9 S:2.6	N/A	W:0.9 S:2.2
2-Butanone	(Jarnstrom et al. 2006) (Weisel et al. 2008) (Logue et al. 2011) (Takigawa et al. 2012) (Zhu et al. 2013) (Uchiyama et al., 2015) (Singleton et al. 2016) (Cheng et al. 2016)	15/4/6 6.87 7.4 N/A 3.23 W: 1.3 S:1.7 N/A 0.9	N/A N/A N/A N/A 1.14 N/A N/A 0.7	N/A 3.50 3.4 2.1 N/A W: 0.9 S:1.2 7.7 0.6
2-Furancarboxaldehyde	(Zhu et al. 2013)	2.99	1.84	N/A
2-Heptanone	(Jarnstrom et al. 2006)	3	N/A	N/A
2-Pentanone	(Zhu et al. 2013)	0.57	0.36	N/A
4-Methyl-2-Pentanone	(Zhu et al. 2013)	0.54	0.23	N/A
6-Methyl-5-hepten-one	(Jarnstrom et al. 2006) (Cheng et al. 2016)	2/2 1	N/A 0.9	N/A 0.9
Acetaldehyde	(Baez et al. 2003) (Clarisse et al. 2003) (Ohura et al. 2006) (Takigawa et al. 2010) (Billionnet et al. 2011) (Geiss et al. 2011) (Logue et al. 2011) (Takigawa et al. 2012) (Uchiyama et al., 2015) (Duan et al. 2016) (Langer et al. 2016) (Fan et al. 2016) (Cheng et al. 2016)	5/12 N/A N/A N/A N/A 12.8 22 N/A W:22 S:17 17.0 N/A W:15.6 S:15.3 7.6	N/A 10.1/10.0/10.2 S:9.39 W:16.7 N/A N/A N/A N/A N/A N/A N/A 11.6 N/A 6.8	N/A N/A N/A 24.3/22.2 11 11.2 13 20.8/15.7 W:15 S:13 15.3 11.5 N/A 7.1
Acetone	(Baez et al. 2003) (Weisel et al. 2008) (Takigawa et al. 2010) (Geiss et al. 2011) (Logue et al. 2011) (Takigawa et al. 2012) (Zhu et al. 2013) (Uchiyama et al., 2015) (Duan et al. 2016) (Singleton et al. 2016) (Fan et al. 2016) (Cheng et al. 2016)	27/12 87.1 N/A 11.6 40 N/A 9.00 W: 27 S:22 23.6 N/A W:49.5S:6.1 3.3	N/A N/A N/A N/A N/A N/A 3.78 N/A N/A N/A N/A 2.8	N/A 34.5 42.6/33.4 47.4 21 32.1/22.9 N/A W:14 S:14 21.7 17 N/A 2.8
Acetophenone	(Jarnstrom et al. 2006) (Cheng et al. 2016)	1/2 0.6	N/A 0.5	N/A 0.5
Acrolein	(Billionnet et al. 2011) (Logue et al. 2011) (Uchiyama et al., 2015)	N/A 2.3 W:0.8 S:0.9	N/A N/A N/A	1.0 0.84 W:0.5 S:0.8

Benzaldehyde	(Ohura et al. 2006) (Jarnstrom et al. 2006) (Takigawa et al. 2010) (Logue et al. 2011) (Takigawa et al. 2012) (Zhu et al. 2013) (Uchiyama et al., 2015) (Duan et al. 2016) (Cheng et al. 2016)	N/A 5/4/3 N/A 2.5 N/A 3.98 W:0.8 S:1.3 1.30 1.1	S:1.04 W:1.00 N/A N/A N/A N/A 2.76 N/A N/A 1.0	N/A N/A 6.2/3.6 2.9 3.7 N/A W:0.7 S:0.9 1.05 1.0
Butyraldehyde	(Baez et al. 2003) (Jarnstrom et al. 2006) (Ohura et al. 2006) (Takigawa et al. 2010) (Logue et al. 2011) (Takigawa et al. 2012) (Duan et al. 2016) (Cheng et al. 2016)	4.5/3.3 5/2 N/A N/A 7.1 N/A 3.73 0.2	N/A N/A S:0.53 W:0.70 N/A N/A N/A N/A 0.2	N/A N/A N/A 4.1/2.2 1.2 2.2 3.31 0.1
Crotonaldehyde	(Ohura et al. 2006) (Takigawa et al. 2010) (Logue et al. 2011) (Takigawa et al. 2012) (Uchiyama et al., 2015)	N/A N/A 4.7 N/A W:0.5 S:0.2	S:0.28 W:0.37 N/A N/A N/A N/A	N/A 7.6/3.8 0.45 3.5/1.6 W:0.4 S:0.0
Decanal	(Jarnstrom et al. 2006) (Logue et al. 2011) (Zhu et al. 2013) (Uchiyama et al., 2015) (Cheng et al. 2016)	3/5/5 1.8 1.45 W:1.4 S:3.7 1.1	N/A N/A 0.83 N/A 0.9	N/A 0.92 N/A W:1.3 S:3.5 0.9
diphenyl Methanone	(Jarnstrom et al. 2006)	1	N/A	N/A
Formaldehyde	(Baez et al. 2003) (Clarisse et al. 2003) (Gustafson et al. 2005) (Ohura et al. 2006) (Jarnstrom et al. 2006) (Takigawa et al. 2010) (Geiss et al. 2011) (Billionnet et al. 2011) (Logue et al. 2011) (Takigawa et al. 2012) (Molloy et al. 2012) (Guo et al. 2013) (Uchiyama et al., 2015) (Duan et al. 2016) (Langer et al. 2016) (Fan et al. 2016) (Cheng et al. 2016)	37/47 N/A 26/35 N/A 19/21/26 N/A 21.5 N/A 69 N/A 14.98 N/A W:13 S:34 40.2 N/A W:19.5 S:31.6 16.4	N/A 21.7/24.3/24.5 N/A S:18.7 W:12.4 N/A N/A N/A N/A N/A N/A N/A N/A N/A N/A 19.5 N/A 15.2	N/A N/A 23/29 N/A N/A 48.5/39.4 19.7 19.4 23 39.2/31.5 N/A 29/30.6 W:11 S:27 32.1 19.7 N/A 15.4
Geranyl acetone	(Jarnstrom et al. 2006)	1/2	N/A	N/A
Glyoxal	(Logue et al. 2011)	2.4	N/A	2.6
Heptanal	(Jarnstrom et al. 2006) (Uchiyama et al., 2015)	3/2/2 W:0.8 S:0.7	N/A N/A	N/A W:0.8 S:0

Hexanal	(Clarisse et al. 2003) (Ohura et al. 2006) (Jarnstrom et al. 2006) (Takigawa et al. 2010) (Geiss et al. 2011) (Billionnet et al. 2011) (Logue et al. 2011) (Takigawa et al. 2012) (Zhu et al. 2013) (Uchiyama et al., 2015) (Duan et al. 2016) (Cheng et al. 2016)	N/A N/A 21/10/11 N/A 32.3 N/A 5.9 N/A 20.61 W:3.2 S:7.0 8.80 2.0	20.5/23.8/25.5 S:2.02 W:2.36 N/A N/A N/A N/A N/A N/A 11.13 N/A N/A 1.7	N/A N/A N/A 10.5/9.6 24.4 13.0 8.4 9.5 N/A W:2.4 S:4.2 6.9 1.7
Isovaleraldehyde	(Logue et al. 2011) (Takigawa et al. 2012)	1.2 N/A	N/A N/A	0.97 2.8
m,p,o-Tolualdehyde	(Takigawa et al. 2010) (Takigawa et al. 2012) (Uchiyama et al., 2015)	N/A N/A W: 1.8 S:2.1	N/A N/A N/A	1.0/1.0 1.0/1.0 W:0.8 S:1.6
m/o-Tolualdehyde	(Duan et al. 2016)	0.98	N/A	
Methyl Isobutyl Ketone	(Jarnstrom et al. 2006) (Logue et al. 2011) (Cheng et al. 2016)	12 1.2 0.7	N/A N/A 0.5	N/A 0.3 0.5
Methylglyoxal	(Logue et al. 2011) (Cheng et al. 2016)	2.6 2.8	N/A 2.4	2.7 2.8
Nonanal	(Jarnstrom et al. 2006) (Zhu et al. 2013) (Uchiyama et al., 2015) (Cheng et al. 2016)	8/6/7 3.45 W:4.3 S:12 4.3	N/A 2.24 N/A 3.9	N/A N/A W:3.1 S:11 4.1
Octanal	(Jarnstrom et al. 2006) (Logue et al. 2011) (Zhu et al. 2013) (Uchiyama et al., 2015) (Cheng et al. 2016)	5/3/3 4.3 2.10 W:1.0 S:1.6 1.8	N/A N/A 1.22 N/A 1.7	N/A N/A N/A W:1.0 S:1.3 1.8
Propionaldehyde	(Baez et al. 2003) (Ohura et al. 2006) (Takigawa et al. 2010) (Geiss et al. 2011) (Takigawa et al. 2012) (Logue et al. 2011) (Duan et al. 2016) (Cheng et al. 2016)	2.6/3.5 N/A N/A 3.0 N/A 6.9 2.73 0.9	N/A S:1.21 W:1.87 N/A N/A N/A N/A N/A 0.8	N/A N/A 10.9/7.4 2.7 7.7 1.8 2.45 0.8
Valeraldehyde	(Clarisse et al. 2003) (Jarnstrom et al. 2006) (Ohura et al. 2006) (Logue et al. 2011) (Takigawa et al. 2012) (Uchiyama et al., 2015) (Duan et al. 2016) (Cheng et al. 2016)	N/A 10/3/6 N/A 1.3 N/A W:0.5 S:0.1 2.76 0.7	5.7/6.0/6.4 N/A S:0.97 W:0.77 N/A N/A N/A N/A 0.6	N/A N/A N/A 1.8 3.7 W:0 S:0 2.15 0.6
Vleraldehyde	(Uchiyama et al., 2015)	W:0.8 S:1.9	N/A	W:0.6 S:1.3

Office environments – Aldehydes and Ketones

Office Environment		**µgm-3**		
Aldehydes/Ketones	**Reference**	**Arithmetic Mean**	**Geometric Mean**	**Median**
2-Butanone	(Tham et al. 2004)	9.7	N/A	N/A
Acetaldehyde	(Baez et al. 2003) (Wingfors et al. 2009) (Mandin et al. 2017)	16/7 S:22 W:22 S:6.4 W:4.9	N/A N/A N/A	N/A N/A S:6.1 W:4.5
Acetone	(Baez et al. 2003) (Zuraimi et al. 2006)	13/76 17.4	N/A N/A	N/A N/A
Acetophenone	(Tham et al. 2004)	2.2	N/A	N/A
Acrolein	(Mandin et al. 2017)	S:2.5 W:1.3	N/A	S:2.4 W:1.0
Benzaldehyde	(Zuraimi et al. 2006) (Salonen et al. 2009) (Mandin et al. 2017)	4.1 1.5 S:1.0	N/A N/A N/A	N/A 2.0 S:0.9
Butyraldehyde	(Baez et al. 2003)	3.1/14	N/A	N/A
Formaldehyde	(Wingfors et al. 2009) (Mandin et al. 2017)	S: 46 W: 16 S:16 W:8.1	N/A N/A	N/A S:14 W:7.5
Glutaraldehyde	(Mandin et al. 2017)	S:1.3	N/A	S:1.1
Hexanal	(Tham et al. 2004) (Salonen et al. 2009) (Mandin et al. 2017)	0.5 2.2 S:11 W:5.0	N/A N/A N/A	N/A 0.9 S:10 W:4.4
Methyl Isobutyl Ketone	(Tham et al. 2004) (Jia et al. 2010)	6.0 N/A	N/A N/A	N/A 0.4
Nonanal	(Tham et al. 2004) (Salonen et al. 2009)	3.6 2.5	N/A N/A	N/A 2.0
Propionaldehyde	(Baez et al. 2003) (Mandin et al. 2017)	4.2/4.2 S:2.8 W:1.4	N/A N/A	N/A S:2.4 W:1.2

Home environments - Aliphatic compounds

Home Environment		**µgm-3**		
Alkanes / Alkenes	**Reference**	**Arithmetic Mean**	**Geometric Mean**	**Median**
1,3-Butadiene	(Logue et al. 2011)	0.46	N/A	0.16
1-Butene	(Duan et al. 2016)	1.11	N/A	0.81
2,3-Dimethylpentane	(Logue et al. 2011)	3.4	N/A	0.7
2,4-Dimethylpentane	(Logue et al. 2011)	2.9	N/A	0.6
2-Methyl Hexane	(Logue et al. 2011)	0.13	N/A	1.8
2-Methylbutane	(Cheng et al. 2016)	6.9	6	5.9
2-Methylpentane	(Logue et al. 2011) (Duan et al. 2016) (Cheng et al. 2016)	0.37 2.00 5.2	N/A N/A 1.6	1.6 1.58 1.2
3-Ethyl Hexane	(Logue et al. 2011)	0.13	N/A	N/A
3-Methylpentane	(Duan et al. 2016) (Cheng et al. 2016)	1.36 2.5	N/A 0.9	1.17 0.7
Butane	(Duan et al. 2016) (Cheng et al. 2016)	8.79 67.9	N/A 26.1	6.87 29.8

Decane	(Schlink et al. 2004) (Jarnstrom et al. 2006) (Gokhale et al. 2008) (Pekey and Arslanbas 2008) (Geiss et al. 2011) (Billionnet et al. 2011) (Logue et al. 2011) (Takigawa et al. 2012) (Zhu et al. 2013) (Uchiyama et al., 2015) (Duan et al. 2016) (Langer et al. 2016) (Cheng et al. 2016)	9.0 32 2.20 S:4.74 W:5.09 17.3 N/A 15 N/A 6.81 W:13 S:7.8 1.14 N/A 1.3	N/A N/A N/A N/A N/A 5.3 N/A N/A 1.38 N/A N/A 6.4 0.8	3.0 N/A N/A N/A 15.6 N/A 3.8 5.1 N/A W: 2.9 S:1.5 0.73 5.4 0.7
Dodecane	(Schlink et al. 2004) (Jarnstrom et al. 2006) (Gokhale et al. 2008) (Geiss et al. 2011) (Logue et al. 2011) (Zhu et al. 2013) (Cheng et al. 2016)	6.5 4/3/2 6.27 17.3 22 1.71 3.2	N/A N/A N/A N/A N/A 0.46 1.0	2.0 N/A N/A 15.6 2.5 N/A 0.9
Ethane	(Duan et al. 2016)	30.9	N/A	17.6
Ethene	(Duan et al. 2016)	12	N/A	8.4
Heptadecane	(Jarnstrom et al. 2006)	1/1	N/A	N/A
Heptane	(Schlink et al. 2004) (Jarnstrom et al. 2006) (Gokhale et al. 2008) (Pekey and Arslanbas 2008) (Weisel et al. 2008) (Geiss et al. 2011) (Logue et al. 2011) (Takigawa et al. 2012) (Zhu et al. 2013) (Uchiyama et al., 2015) (Singleton et al. 2016) (Cheng et al. 2016)	7.6 3/2/2 3.35 S:5.49 W:8.73 4.43 1.2 11 N/A 5.64 W:3.9 S:2.0 N/A 1.8	N/A N/A N/A N/A N/A N/A N/A N/A 1.27 N/A N/A 0.7	2.3 N/A N/A N/A 2.00 0.7 2.5 1.2 N/A W:1.1 S:0 3 0.6
Hexadecane	(Jarnstrom et al. 2006)	1/2/1	N/A	N/A
Hexane	(Schlink et al. 2004) (Gokhale et al. 2008) (Pekey and Arslanbas 2008) (Weisel et al. 2008) (Geiss et al. 2011) (Logue et al. 2011) (Zhu et al. 2013) (Guo et al. 2013) (Uchiyama et al., 2015) (Duan et al. 2016) (Singleton et al. 2016) (Cheng et al. 2016)	7.3 4.69 S:9.31 W:12.63 8.42 2.5 7..3 4.33 N/A W: 2.8 S:3.5 4.58 N/A 2.6	N/A N/A N/A N/A N/A N/A 1.21 N/A N/A N/A N/A 1.2	3.1 N/A N/A 2.80 1.4 3 N/A 1.5/1.59 W:1.6 S:1.6 1.94 2.3 0.9
i-Butane	(Logue et al. 2011) (Duan et al. 2016)	52 8.4	N/A N/A	23 5.94
i-Octane	(Logue et al. 2011)	5.5	N/A	0.6
i-Pentane	(Duan et al. 2016)	7.79	N/A	5.71

Nonane	(Schlink et al. 2004) (Jarnstrom et al. 2006) (Gokhale et al. 2008) (Pekey and Arslanbas 2008) (Takigawa et al. 2010) (Logue et al 2011) (Takigawa et al. 2012) (Uchiyama et al., 2015) (Cheng et al. 2016)	4 12/1/1 1.45 S:9.05 W:13.53 N/A 14 N/A W:11 S:6.6 1.2	N/A N/A N/A N/A N/A N/A N/A N/A 0.7	1.4 N/A N/A N/A 1.8 1.4 1.5 W:1.2 S:0 0.5
Octane	(Schlink et al. 2004) (Jarnstrom et al. 2006) (Gokhale et al. 2008) (Pekey and Arslanbas 2008) (Logue et al. 2011) (Takigawa et al. 2012) (Uchiyama et al., 2015) (Duan et al. 2016) (Cheng et al. 2016)	2.8 4/1 0.62 S:4.18 W:6.51 3.9 N/A W:2.8 S:2.0 1.39 1.3	N/A N/A N/A N/A N/A N/A N/A N/A 1.1	1.0 N/A N/A N/A 1.0 1.3 W:0.6 S:0 0.81 1.0
Pentadecane	(Jarnstrom et al. 2006)	2/2/1	N/A	N/A
Pentane	(Jarnstrom et al. 2006) (Zhu et al. 2013) (Duan et al. 2016) (Cheng et al. 2016)	30/14 7.96 3.79 3.7	N/A 3.17 N/A 3.0	N/A N/A 2.94 2.9
Propane	(Duan et al. 2016)	16	N/A	11.2
Propene	(Duan et al. 2016)	5.99	N/A	4.67
Tetradecane	(Jarnstrom et al. 2006)	5/2/1	N/A	N/A
Tridecane	(Schlink et al. 2004) (Jarnstrom et al. 2006) (Gokhale et al. 2008) (Logue et al. 2011) (Cheng et al. 2016)	3.1 4/3/2 1.05 3.3 0.6	N/A N/A N/A N/A 0.2	1.5 N/A N/A 1.7 0.2
Undecane	(Schlink et al. 2004) (Jarnstrom et al. 2006) (Gokhale et al. 2008) (Pekey and Arslanbas 2008) (Geiss et al. 2011) (Billionnet et al. 2011) (Logue et al. 2011) (Takigawa et al. 2012) (Zhu et al. 2013) (Uchiyama et al., 2015) (Cheng et al. 2016)	8.9 4/3/2 2.36 S:4.26 W:5.59 7.2 N/A 3.3 N/A 5.27 W:19 S:18 2.5	N/A N/A N/A N/A N/A 6.0 N/A N/A 1.52 N/A 0.8	3.0 N/A N/A N/A 2.3 N/A 1.7 4.3 N/A W:5 S:8.2 0.8

Office environments – Aliphatic compounds

Office Environment **µgm-3**

Alkanes / Alkenes	Reference	Arithmetic Mean	Geometric Mean	Median
1,3-Butadiene	(Kim et al. 2001)	0.3	N/A	N/A
2-Methylpentane	(Zuraimi et al. 2006)	17.8	N/A	N/A
3-Methylpentane	(Tham et al. 2004) (Zuraimi et al. 2006)	1.2 30.8	N/A N/A	N/A N/A

Decane	(Brickus et al. 1998) (Tham et al. 2004) (Zuraimi et al. 2006) (Tovalin-Ahumada and Whitehead 2007) (Jia et al. 2010)	14.8 / 16.3 / 53.3 / 13.0 0.8 8.6 10 N/A	N/A N/A N/A N/A N/A	N/A N/A N/A 7 2.8
Dodecane	(Tham et al. 2004) (Zuraimi et al. 2006) (Jia et al. 2010)	2.4 10.9 N/A	N/A N/A N/A	N/A N/A 0.3
Heptane	(Brickus et al. 1998) (Baez et al. 2003) (Tham et al. 2004) (Zuraimi et al. 2006) (Jia et al. 2010)	37.5 / 39.2 / 54.1 / 12.9 97/26 0.8 31.7 N/A	N/A N/A N/A N/A N/A	N/A N/A N/A N/A 2.0
Hexadecane	(Tham et al. 2004)	3	N/A	N/A
Hexane	(Brickus et al. 1998) (Tham et al. 2004) (Zuraimi et al. 2006) (Tovalin-Ahumada and Whitehead 2007) (Ongwandee et al. 2011) (Mandin et al. 2017)	91.1 / 119.6 / 125.5 / 60.7 5.5 52.5 14 7.46 S:1.9 W:1.5	N/A N/A N/A N/A N/A N/A	N/A N/A N/A 12 N/A S:1.4 W:1.2
Nonane	(Brickus et al. 1998) (Tovalin-Ahumada and Whitehead 2007) (Jia et al. 2010)	11 / 10.3 / 27.7 / 9.2 4 N/A	N/A N/A N/A	N/A 4 1.3
Octane	(Brickus et al. 1998) (Tham et al. 2004) (Jia et al. 2010)	5.4 / 16.9 / 32.1 / 8.3 0.6 N/A	N/A N/A N/A	N/A N/A 0.8
Pentadecane	(Jia et al. 2010)	N/A	N/A	1
Pentane	(Tham et al. 2004)	1.2	N/A	N/A
Tetradecane	(Tham et al. 2004) (Zuraimi et al. 2006) (Jia et al. 2010)	3.2 3.1 N/A	N/A N/A N/A	N/A N/A 1.1
Tridecane	(Jia et al. 2010)	N/A	N/A	0.4
Undecane	(Brickus et al. 1998) (Tham et al. 2004) (Zuraimi et al. 2006) (Jia et al. 2010)	12.9 / 13.4 / 40.7 / 14.7 1.2 13.8 N/A	N/A N/A N/A N/A	N/A N/A N/A 1.5

Home environments – Aromatic compounds

Home Environment **µgm-3**

Aromatics	Reference	Arithmetic Mean	Geometric Mean	Median
(1-methylethyl)Benzene	(Zhu et al. 2013)	0.25	0.09	N/A
2-methyl decalin	(Jarnstrom et al. 2006)	5/1	N/A	N/A
1,2,3-Trimethylbenzene	(Ohura et al. 2006) (Logue et al. 2011) (Zhu et al. 2013) (Uchiyama et al., 2015)	N/A 1.2 4.33 W: 1.8 S:0.9	S:0.42 W:1.40 N/A 1.58 N/A	N/A 0.4 N/A W:0.5 S:0

1,2,4-Trimethylbenzene	(Ohura et al. 2006) (Pekey and Arslanbas 2008) (Weisel et al. 2008) (Geiss et al. 2011) (Logue et al. 2011) (Billionnet et al. 2011) (Zhu et al. 2013) (Uchiyama et al., 2015) (Duan et al. 2016) (Cheng et al. 2016)	N/A S:2.35 W:4.20 4.28 2.7 4.2 N/A 1.37 W:6.5 S:4.0 1.99 2.3	S:1.57 W:4.82 N/A N/A N/A N/A N/A 0.51 N/A N/A 1.7	N/A N/A 2.50 1.1 2.8 4.0 N/A W:2.0 S:1.5 1.32 1.7
1,3,5 Trimethylbenzene	(Ohura et al. 2006) (Logue et al. 2011) (Uchiyama et al., 2015) (Cheng et al. 2016)	N/A 1.6 W:2.0 S:1.2 0.8	S:0.40 W:1.26 N/A N/A 0.6	N/A 0.6 W:0.7 S:0 0.6
2 - Ethyl Toluene	(Schlink et al. 2004) (Ohura et al. 2006) (Gokhale et al. 2008) (Logue et al. 2011) (Cheng et al. 2016)	1.6 N/A 0.42 1.5 0.6	N/A S:0.41 W:1.54 N/A N/A 0.5	0.6 N/A N/A 0.5 0.5
2,5-Dimethyl Furan	N/A	N/A	N/A	N/A
2-Pentyl furan	(Cheng et al. 2016)	0.4	0.4	0.4
3-Ethenylpyridine	(Logue et al. 2011)	0.28	N/A	N/A
3-Ethyl Toluene	(Schlink et al. 2004) (Ohura et al. 2006) (Gokhale et al. 2008) (Logue et al. 2011) (Cheng et al. 2016)	1.7 N/A 0.66 2 1.1	N/A S:0.81 W:2.66 N/A N/A 0.8	0.7 N/A N/A 0.66 0.8
3-Ethyl-o-xylene	(Cheng et al. 2016)	0.9	0.7	0.6
4 - Ethyl Toluene	(Schlink et al. 2004) (Ohura et al. 2006) (Gokhale et al. 2008) (Weisel et al. 2008) (Logue et al. 2011)	2.4 N/A 1.47 3.70 2.6	N/A S:0.44 W:1.48 N/A N/A N/A	1.1 N/A N/A <2.5 0.99
4-Ethyl-m-xylene	(Cheng et al. 2016)	0.7	0.6	0.7

Benzene	(Fischer et al. 2000) (Schneider et al. 2001) (Son et al. 2003) (Jo et al. 2003) (Jo et al. 2003) (Jo et al. 2004) (Adgate et al. 2004) (Adgate et al. 2004b) (Schlink et al. 2004) (Jarnstrom et al. 2006) (Ohura et al. 2006) (Gokhale et al. 2008) (Stranger et al. 2007) (Pekey and Arslanbas 2008) (Weisel et al. 2008) (Takigawa et al. 2010) (Geiss et al. 2011) (Billionnet et al. 2011) (Logue et al. 2011) (Molloy et al. 2012) (Takigawa et al. 2012) (Zhu et al. 2013) (Guo et al. 2013) (Du et al. 2014) (Kumar et al. 2014) (Uchiyama et al., 2015) (Duan et al. 2016) (Langer et al. 2016) (Singleton et al. 2016) (Hazrati et al. 2016) (Fan et al. 2016) (Cheng et al. 2016)	HT:7.7 LT:5.7 2.3/2.5 20.26/43.71 N/A N/A N/A N/A 4.6 3.2 3 N/A 2.4 2.20 S:8.88 W:13.06 4.07 N/A 2.8 N/A 2.5 4.15 N/A 1.93 N/A 18.5 7.9/8.2/7.3 W:2.3 S:1.3 7.35 N/A N/A 15.18 W:12.5 S:7.8 1.3	N/A N/A N/A N/A N/A SM: 11.0/11.5 NS:6.6/6.7 N/A N/A N/A N/A S:0.99 W:2.69 N/A N/A N/A N/A N/A N/A N/A N/A N/A 1.04 N/A 14.0 N/A N/A N/A 2.0 N/A N/A N/A 1.1	N/A S:1.2/0.9 W:2.5/2.9 23.83/36.90 N:H:11.6 L:13.6 D:H:5.3 L:6.3 N/A W:2.2 SP:2.1 3.3 2.3 N/A N/A N/A N/A 1.80 1.1/1.1 1.9 2.0 2.1 N/A 1.1/1.6 N/A 2.4/2.8 18.8 N/A W:1.7 S:1.3 5.95 2.0 3.0 N/A N/A 1.0
Butyl Benzene	(Logue et al. 2011)	0.62	N/A	0.08
Ethylbenzene	(Schneider et al. 2001) (Jo et al. 2003) (Jo et al. 2003) (Schlink et al. 2004) (Adgate et al. 2004) (Adgate et al. 2004b) (Jo et al. 2004) (Ohura et al. 2006) (Jarnstrom et al. 2006) (Stranger et al. 2007) (Gokhale et al. 2008) (Pekey and Arslanbas 2008) (Weisel et al. 2008) (Takigawa et al. 2010) (Geiss et al. 2011) (Billionnet et al. 2011) (Takigawa et al. 2012) (Zhu et al. 2013) (Du et al. 2014) (Kumar et al. 2014) (Uchiyama et al., 2015) (Duan et al. 2016) (Singleton et al. 2016) (Cheng et al. 2016)	2.2/2.8 N/A N/A 9.8 N/A 7.9 N/A N/A 29/2/3 0.62 5.46 S:12.30 W:27.46 3.72 N/A 1.5 N/A N/A 14.44 58.1 4.2/5.4/4.7 W:8.3 S:5.3 6.33 N/A 1.2	N/A N/A N/A N/A N/A N/A SM:12.7/12.2 NS:8.5/8.0 S:3.4 W:8.86 N/A N/A N/A N/A 3.30 N/A N/A N/A N/A 4.88 33.3 N/A N/A N/A N/A 1.0	0.7/1.67 N:H:20.1 L:17.2 D:H:14.2 L:13.1 4.6 W:3.7 SP:3.3 4.6 N/A N/A N/A N/A N/A N/A 3.30 3.2/2.8 1.1 5.4 2.7/3.3 N/A 46.0 N/A W:3.7 S:2.9 5.64 11 0.9

Isopropylbenzene	(Ohura et al. 2006) (Logue et al. 2011)	N/A 0.4	S:0.13 W:0.41 N/A	N/A 0.08
m/p-Xylene	(Schneider et al. 2001) (Jo et al. 2003) (Jo et al. 2003) (Schlink et al. 2004) (Adgate et al. 2004) (Adgate et al. 2004b) (Jo et al. 2004) (Ohura et al. 2006) (Stranger et al. 2007) (Gokhale et al. 2008) (Pekey and Arslanbas 2008) (Weisel et al. 2008) (Geiss et al. 2011) (Billionnet et al. 2011) (Logue et al. 2011) (Zhu et al. 2013) (Du et al. 2014) (Kumar et al. 2014) (Uchiyama et al., 2015) (Duan et al. 2016) (Singleton et al. 2016) (Cheng et al. 2016)	7.3/6.5 N/A N/A 9.8 N/A 7.9 N/A N/A 1.36 5.46 S:12.30 W:27.46 10.2 3.8 N/A 8.2 14.44 58.1 4.2/5.4/4.7 W:8.3 S:5.3 6.33 N/A 4.1	N/A N/A N/A N/A N/A N/A SM:12.7/12.2 NS:8.5/8.0 S:3.4 W:8.86 N/A N/A N/A N/A N/A N/A N/A 4.88 33.3 N/A N/A N/A N/A 3.0	S:1.6/2.1 W:6.2/6.1 N:H:20.1 L:17.2 D:H:14.2 L:13.1 4.6 W:3.7 SP:3.3 4.6 N/A N/A N/A N/A N/A N/A 3.8 2.8 5.4 4.3 N/A 46.0 N/A W:3.7 S:2.9 5.64 11 2.6
Methyl Benzene	(Jarnstrom et al. 2006)	20/05/2011	N/A	N/A
Methyl salicylate	(Cheng et al. 2016)	0.9	0.1	<0.03
o-Xylene	(Schneider et al. 2001) (Son et al. 2003) (Jo et al. 2003) (Jo et al. 2003) (Jo et al. 2004) (Schlink et al. 2004) (Adgate et al. 2004) (Adgate et al. 2004b) (Ohura et al. 2006) (Stranger et al. 2007) (Gokhale et al. 2008) (Pekey and Arslanbas 2008) (Weisel et al. 2008) (Geiss et al. 2011) (Billionnet et al. 2011) (Logue et al. 2011) (Zhu et al. 2013) (Guo et al. 2013) (Du et al. 2014) (Kumar et al. 2014) (Uchiyama et al., 2015) (Duan et al. 2016) (Singleton et al. 2016) (Cheng et al. 2016)	1.9/1.9 9.32/33.45 N/A N/A N/A 2.7 N/A 2.9 N/A 0.68 1.78 S:5.73 W:16.24 3.92 1.8 N/A 1.7 4.33 N/A 40.8 1.8/2.3/1.9 W:3.4 S:2.6 2.32 N/A 2.2	N/A N/A N/A N/A SM: 8.3/8.1 NS:6.4/4.6 N/A N/A N/A S:1.10 W:3.40 N/A N/A N/A N/A N/A N/A N/A 1.41 N/A 25.2 N/A N/A N/A N/A 1.8	0.79/1.20 8.28/9.29 N:H:6.4 L:5.2 D:H:4.8 L:4.6 N/A 1.4 W:1.2 SP:1.1 2.1 N/A N/A N/A N/A 2.20 1.2 2.2 1.3 N/A 1.8/1.7 33.9 N/A W: 1.5 S:1.4 2.17 4.5 1.7
p-Cymene	(Cheng et al. 2016)	2.3	1.5	1.4
p-Cymenene	(Cheng et al. 2016)	0.7	0.5	0.5
Propylbenzene	(Ohura et al. 2006) (Logue et al. 2011) (Cheng et al. 2016)	N/A 1.1 0.6	S:0.43 W:0.90 N/A 0.4	N/A 0.5 0.4
Sec-Butyl Benzene	(Logue et al. 2011)	0.52	N/A	N/A

Styrene	(Schlink et al. 2004) (Adgate et al. 2004) (Adgate et al. 2004b) (Jarnstrom et al. 2006) (Gokhale et al. 2008) (Pekey and Arslanbas 2008) (Geiss et al. 2011) (Billionnet et al. 2011) (Logue et al. 2011) (Zhu et al. 2013) (Duan et al. 2016) (Cheng et al. 2016)	1.4 N/A 1.2 3/2/2 1.61 S:9.39 W:11.65 0.4 N/A 5.9 1.13 1.85 0.5	N/A N/A N/A N/A N/A N/A N/A N/A N/A 0.72 N/A 0.4	0.6 W:0.7 SP:0.8 0.9 N/A N/A N/A 0 0.92 0.7 N/A 1.19 0.4
Tert-Butyl Benzene	(Logue et al. 2011)	0.94	N/A	N/A
Tetrahydrofuran	(Logue et al. 2011) (Zhu et al. 2013)	1.7 0.34	N/A 0.05	N/A N/A
Toluene	(Schneider et al. 2001) (Son et al. 2003) (Jo et al. 2003) (Jo et al. 2003) (Jo et al. 2004) (Schlink et al. 2004) (Adgate et al. 2004) (Adgate et al. 2004b) (Ohura et al. 2006) (Stranger et al. 2007) (Gokhale et al. 2008) (Pekey and Arslanbas 2008) (Weisel et al. 2008) (Takigawa et al. 2010) (Geiss et al. 2011) (Billionnet et al. 2011) (Logue et al. 2011) (Takigawa et al. 2012) (Molloy et al. 2012) (Zhu et al. 2013) (Guo et al. 2013) (Du et al. 2014) (Kumar et al. 2014) (Uchiyama et al., 2015) (Duan et al. 2016) (Singleton et al. 2016) (Hazrati et al. 2016) (Fan et al. 2016) (Cheng et al. 2016)	32.6/53.2 17.31/170.67 N/A N/A N/A 29.5 N/A 23.4 N/A 4.25 21.73 S:44.19 W:72.44 25.1 N/A 0.4 N/A 15 N/A 33.16 17.80 N/A 173.2 30.7/32.9/28.8 W:11 S:12 23.5 N/A 69.70 W:10.4 S:9.2 10.7	N/A N/A N/A N/A SM: 290/59.1 NS:293/43.3 N/A N/A N/A S:11.5 W:25.9 N/A N/A N/A N/A N/A N/A N/A N/A N/A N/A N/A N/A N/A 7.94 N/A 95.8 N/A N/A N/A N/A N/A N/A N/A 7.4	S:14.6/27.1 W:33.8/43.3 13.90/54.44 N:H:44.5 L:57.4 D:H:30.4 L:40.2 N/A 18.3 W:8.2 SP:8.9 16.2 N/A N/A N/A N/A 13 12.9/12.9 0 11.9 18 11.8/10.9 N/A N/A 14.1/12.0 181.0 N/A W: 6.8 S:6.4 14.4 15 N/A N/A 6.1
Trimethylbenzene	(Takigawa et al. 2010) (Takigawa et al. 2012)	N/A N/A	N/A N/A	2.9/2.7 2.6/3.6
Xylenes m,p,o	(Jarnstrom et al. 2006)	110/28/8	N/A	N/A

Office environments – Aromatic compounds

Office Environment **µgm-3**

Aromatics	Reference	Arithmetic Mean	Geometric Mean	Median
(1-methylethyl)Benzene	(Jia et al. 2010)	N/A	N/A	0.1
1,2,3-Trimethylbenzene	(Tovalin-Ahumada and Whitehead 2007) (Jia et al. 2010)	2 N/A	N/A N/A	2 0.5
1,2,4-Trimethylbenzene	(Baek et al. 1997) (Chao et al. 2001) (Kim et al. 2001) (Tham et al. 2004) (Jia et al. 2010) (Tovalin-Ahumada and Whitehead 2007)	9.5 2.2 1.5 93.4 N/A 11	N/A 1.1 N/A N/A N/A N/A	6.8 1.5 N/A N/A 1.2 10
1,3,5 Trimethylbenzene	(Baek et al. 1997) (Chao et al. 2001) (Kim et al. 2001) (Tham et al. 2004) (Tovalin-Ahumada and Whitehead 2007) (Jia et al. 2010)	4.1 8.8 0.3 875.3 3 N/A	N/A 1.7 N/A N/A N/A N/A	3.1 1.6 N/A N/A 3 0.5
2 - Ethyl Toluene	(Tham et al. 2004) (Tovalin-Ahumada and Whitehead 2007) (Jia et al. 2010)	1.8 3 N/A	N/A N/A N/A	N/A 3 0.5
4 - Ethyl Toluene	(Jia et al. 2010)	N/A	N/A	1
Benzene	(Baek et al. 1997) (Brickus et al. 1998) (Chao et al. 2001) (Kim et al. 2001) (Tham et al. 2004) (Guo et al. 2004) (Zuraimi et al. 2006) (Tovalin-Ahumada and Whitehead 2007) (Srivastava and Devotta 2007) (Pekey and Arslanbas 2008) (Jia et al. 2010) (Ongwandee et al. 2011) (Mandin et al. 2017)	8.2 18.4 / 31.1 / 34.5 / 15.9 8.1 5.9 99.0 0.52 14.6 12 44.92 N/A N/A 8.08 S:1.4 W:2.1	N/A N/A 3.6 N/A N/A N/A N/A N/A N/A N/A N/A N/A N/A	7.9 N/A 2.9 N/A N/A N/A N/A 10 N/A S: 9.82 W: 10.63 0.4 N/A S:1.0 W:1.7
Cymene	(Salonen et al. 2009)	4.9	N/A	2
Ethyl Toluene	(Brickus et al. 1998)	19.7 / 14.4 / 15.3 / 21.1	N/A	N/A
Ethylbenzene	(Baek et al. 1997) (Brickus et al. 1998) (Chao et al. 2001) (Kim et al. 2001) (Tham et al. 2004) (Tovalin-Ahumada and Whitehead 2007) (Srivastava and Devotta 2007) (Pekey and Arslanbas 2008) (Jia et al. 2010) (Ongwandee et al. 2011) (Mandin et al. 2017)	5.1 13.1 / 13.6 / 21.2 / 9.3 7.3 2.4 1103.2 11 0.06 N/A N/A 12.1 S:1.8 W:1.3	N/A N/A 4.0 N/A N/A N/A N/A N/A N/A N/A N/A	4.0 N/A 2.5 N/A N/A 11 N/A S: 9.16 W:16.71 1.9 N/A S:1.1 W:1.0
m/p-Ethyl Toluene	(Chao et al. 2001)	14.2	2.1	0.5

m/p-Xylene	(Baek et al. 1997) (Chao et al. 2001) (Kim et al. 2001) (Tham et al. 2004) (Zuraimi et al. 2006) (Tovalin-Ahumada and Whitehead 2007) (Pekey and Arslanbas 2008) (Salonen et al. 2009) (Jia et al. 2010) (Ongwandee et al. 2011)	14.4 18.9 7.7 751.5 22.2 29 N/A 4.2 N/A 12.2	N/A 11.6 N/A N/A N/A N/A N/A N/A N/A N/A	11.6 11.1 N/A N/A N/A 27 S: 15.05 W: 35.21 2.0 7.5 N/A
Methyl Benzoate	(Chao et al. 2001)	3	1	1.4
o-Xylene	(Baek et al. 1997) (Chao et al. 2001) (Kim et al. 2001) (Tham et al. 2004) (Zuraimi et al. 2006) (Tovalin-Ahumada and Whitehead 2007) (Pekey and Arslanbas 2008) (Jia et al. 2010) (Ongwandee et al. 2011)	9.1 5.5 1.8 452.1 10.2 11 N/A N/A 9.63	N/A 3.2 N/A N/A N/A N/A N/A N/A N/A	7.0 2.7 N/A N/A N/A 10 S: 12.33 W:22.52 2.4 N/A
Phenol	(Jia et al. 2010)	N/A	N/A	0.5
Styrene	(Baek et al. 1997) (Chao et al. 2001) (Kim et al. 2001) (Tham et al. 2004) (Guo et al. 2004) (Tovalin-Ahumada and Whitehead 2007) (Pekey and Arslanbas 2008) (Jia et al. 2010) (Ongwandee et al. 2011) (Mandin et al. 2017)	3.9 5.1 0.6 159.9 0.15 2 N/A N/A 3.22 S:1.0 W:0.8	N/A 1.3 N/A N/A N/A 2 N/A N/A N/A N/A	3.7 2.7 N/A N/A N/A 2 S: 7.38 W: 17.81 0.3 N/A S:0.9 W:0.5
Toluene	(Baek et al. 1997) (Brickus et al. 1998) (Chao et al. 2001) (Kim et al. 2001) (Tham et al. 2004) (Zuraimi et al. 2006) (Tovalin-Ahumada and Whitehead 2007) (Srivastava and Devotta 2007) (Pekey and Arslanbas 2008) (Salonen et al. 2009) (Jia et al. 2010) (Ongwandee et al. 2011) (Mandin et al. 2017)	42.3 116.1/ 293.7/ 320.5/102.0 52.8 22.0 375.9 35.1 223 0.82 N/A 6.5 N/A 110 S:8.1 W:6.1	N/A N/A 31.4 N/A N/A N/A N/A N/A N/A N/A N/A N/A N/A	31.0 N/A 21.3 N/A N/A N/A 88 N/A S: 50.79 W: 66.26 3.0 6.0 N/A S:4.7 W:3.1
Xylene (m,p,o)	(Mandin et al. 2017)	S: 3.8 W:3.3	N/A	S:2.5 W:2.2

Home environments – Cycloalkanes/Cycloalkenes

Home Environment		**µgm-3**		
Cycloalkanes / Cycloalkenes	**Reference**	**Arithmetic Mean**	**Geometric Mean**	**Median**
Butylcyclohexane	(Jarnstrom et al. 2006)	7	N/A	N/A
Cycloheptane	(Logue et al. 2011)	1.2	N/A	0.5

Cyclohexane	(Schlink et al. 2004) (Gokhale et al. 2008) (Pekey and Arslanbas 2008) (Weisel et al. 2008) (Logue et al. 2011) (Zhu et al. 2013) (Singleton et al. 2016)	5.5 0.75 S:6.58 W:9.29 2.90 5.2 1.95 N/A	N/A N/A N/A N/A N/A 0.50 N/A	1.6 N/A N/A 1.70 1.6 N/A 1.7
Cyclohexanone	(Jarnstrom et al. 2006) (Zhu et al. 2013) (Cheng et al. 2016)	2/2/2 0.98 0.6	N/A 0.38 0.3	N/A N/A 0.4
Cyclopentane	(Duan et al. 2016)	1.13	N/A	0.61
Cyclopropylbenzene	(Logue et al. 2011)	3.6	N/A	N/A
Decamethylcyclopentasiloxane	(Zhu et al. 2013)	41.32	13.11	N/A
Ethylcyclohexane	(Logue et al. 2011)	1.1	N/A	1.5
Methylcyclohexane	(Schlink et al. 2004) (Geiss et al. 2011) (Logue et al. 2011) (Cheng et al. 2016)	5.3 0.9 5.2 1.1	N/A N/A N/A 0.6	1.7 0.5 1.7 0.5
Methylcyclopentane	(Schlink et al. 2004) (Gokhale et al. 2008) (Logue et al. 2011) (Cheng et al. 2016)	12 1.40 1.8 0.8	N/A N/A N/A 0.5	0.9 N/A 0.8 0.4
Methylcyclopropene	(Duan et al. 2016)	1.15	N/A	0.98

Office environments - Cycloalkanes/Cycloalkenes

Office Environment		**µgm-3**		
Cycloalkanes / Cycloalkenes	**Reference**	**Arithmetic Mean**	**Geometric Mean**	**Median**
Methylcyclohexane	(Brickus et al. 1998) (Tham et al. 2004) (Zuraimi et al. 2006) (Jia et al. 2010)	25 / 31.9 / 36.1 / 8.4 1.6 26.4 N/A	N/A N/A N/A N/A	N/A N/A N/A 0.2
Methylcyclopentane	(Brickus et al. 1998) (Tham et al. 2004) (Zuraimi et al. 2006) (Tovalin-Ahumada and Whitehead 2007)	58.2/ 42.8 / 8.4 / 17.3 3.9 57.5 7	N/A N/A N/A N/A	N/A N/A N/A 6

Home environments – Halogenated compounds

Home Environment		**(µgm-3)**		
	Reference	**Arithmetic Mean**	**Geometric Mean**	**Median**
1,1,1-Trichloroethane	(Adgate et al. 2004b) (Ohura et al. 2006) (Logue et al. 2011) (Guo et al. 2013) (Kumar et al. 2014) (Uchiyama et al., 2015)	13.1 N/A 2.4 N/A 1.3/1.7/1.2 W:0.1 S:0.2	N/A S:0.29 W:0.35 N/A N/A N/A N/A	2 N/A 0.3 0.6/0.6 N/A W:0 S:0
1,1,2,2-Tetrachloroethane	(Logue et al. 2011)	0.42	N/A	0.01

1,1,2-Trichloroethane	(Logue et al. 2011)	0.46	N/A	N/A
1,1,2-Trichloro-trifluoroethane	(Logue et al. 2011)	0.82	N/A	0.5
1,1-Dichloroethane	(Logue et al. 2011)	0.38	N/A	N/A
1,1-Dichloroethene	(Logue et al. 2011)	1.2	N/A	N/A
1,1-Dichloropropene	(Logue et al. 2011)	4.8	N/A	N/A
1,2,3-Trichlorobenzene	(Logue et al. 2011)	1.4	N/A	0.4
1,2,4-Trichlorobenzene	(Logue et al. 2011)	1.4	N/A	N/A
1,2-Dibromoethane	(Logue et al. 2011)	0.14	N/A	0.01
1,2-Dichlorobenzene	(Logue et al. 2011)	0.37	N/A	0.01
1,2-Dichloroethane	(Logue et al. 2011) (Guo et al. 2013) (Duan et al. 2016) (Singleton et al. 2016)	0.34 N/A 2.49 N/A	N/A N/A N/A N/A	0.1 1.0/1.1 2.27 0.07
1,2-Dichloropropane	(Logue et al. 2011) (Duan et al. 2016)	0.55 2.06	N/A N/A	0.02 1.94
1,2-Dichlorotetrafluoroethane	(Logue et al. 2011)	0.98	N/A	N/A
1,3-Dichlorobenzene	(Logue et al. 2011)	0.65	N/A	0.15
1,4-Dichlorobenzene	(Adgate et al. 2004) (Adgate et al. 2004b) (Ohura et al. 2006) (Weisel et al. 2008) (Takigawa et al. 2010) (Logue et al. 2011) (Billionnet et al. 2011) (Takigawa et al. 2012) (Zhu et al. 2013) (Chin et al. 2013) (Kumar et al. 2014) (Uchiyama et al., 2015) (Langer et al. 2016) (Cheng et al. 2016)	NA 1.8 N/A 8.41 N/A 50 N/A N/A 5.52 21 3.7/3.3/1.6 W:31 S:120 N/A 0.2	N/A N/A S:41.2 W:42.8 N/A N/A N/A N/A N/A 0.21 N/A N/A N/A 5.5 0.2	W: 0.7 SP: 0.9 0.5 N/A 3.00 1.1/2.0 2.8 4.1 1.7/3.2 N/A 0.36 N/A W:1.4 S:4.3 4.1 0.2
2,5-Dibenzylfluorescein	(Duan et al. 2016)	1.43	N/A	1.34
2-Chlorotoluene	(Logue et al. 2011)	0.42	N/A	N/A
3-Chlorotoluene	(Logue et al. 2011)	3.6	N/A	N/A
4-Chlorotoluene	(Logue et al. 2011)	4.8	N/A	N/A
Benzyl Chloride	(Logue et al. 2011)	0.5	N/A	N/A
Bromobenzene	(Logue et al. 2011)	0.44	N/A	N/A
Bromodichloromethane	(Ohura et al. 2006) (Logue et al. 2011) (Zhu et al. 2013)	N/A 0.49 0.07	S:0.03 W:0.04 N/A 0.03	N/A 0.2 N/A
Bromoform	(Logue et al. 2011)	0.39	N/A	N/A
Bromomethane	(Logue et al. 2011)	0.33	N/A	N/A
Carbon Tetrachloride	(Adgate et al. 2004) (Ohura et al. 2006) (Logue et al. 2011) (Zhu et al. 2013) (Guo et al. 2013) (Kumar et al. 2014) (Singleton et al. 2016)	N/A N/A 0.68 0.32 N/A 0.7/1.0/1.0 N/A	N/A S:0.53 W:0.75 N/A 0.29 N/A N/A N/A	W:0.6 SP:0.5 N/A 0.57 N/A 0.7/0.9 N/A 0.24

Chlorobenzene	(Schlink et al. 2004) (Logue et al 2011)	0.8 0.68	N/A N/A	0.1 0.14
Chlorodibromomethane	(Ohura et al. 2006)	N/A	S:0.03 W:0.03	N/A
Chlorodifluoromethane	(Duan et al. 2016)	4.14	N/A	3.59
Chloroethane	(Logue et al. 2011)	0.26	N/A	N/A
Chloroform	(Adgate et al. 2004) (Adgate et al. 2004b) (Ohura et al. 2006) (Weisel et al. 2008) (Logue et al 2011) (Zhu et al. 2013) (Kumar et al. 2014) (Duan et al. 2016) (Singleton et al. 2016)	N/A 2.4 N/A 2.05 1.5 0.62 2.0/1.7/1.0 1.15 N/A	N/A N/A S:0.25 W:0.92 N/A N/A 0.29 N/A N/A N/A	W:0.8 SP:1.5 1.7 N/A 2.40 0.92 N/A N/A 0.85 0.48
cis-1,2-dichloroethane	(Logue et al. 2011)	0.54	N/A	N/A
cis-1,2-dichloropropane	(Logue et al. 2011)	0.55	N/A	N/A
Dibromochloromethane	(Logue et al. 2011) (Uchiyama et al., 2015)	0.44 W:0.1 S:0	N/A N/A	0.08 W:0 S:0
Dichlorodifluoromethane	(Weisel et al. 2008) (Logue et al. 2011) (Duan et al. 2016)	7.73 0.77 3.25	N/A N/A N/A	3.30 N/A 3.04
Dichlorofluoromethane	(Logue et al. 2011)	7.8	N/A	3.3
Dichloromethane	(Adgate et al. 2004) (Weisel et al. 2008) (Logue et al. 2011) (Kumar et al. 2014) (Duan et al. 2016)	N/A 5.28 8.2 4.4/3.7/2.9 12.5	N/A N/A N/A N/A N/A	W: 0.4 SP: 0.3 <1.7 1.1 N/A 4.05
HCL	(Uchiyama et al., 2015)	W:2.7 S:1.9	N/A	W:0.9 S:1.3
Hexachlorobutadiene	(Logue et al. 2011)	1.7	N/A	N/A
Methyl Chloride	(Weisel et al. 2008) (Duan et al. 2016)	1.49 2.64	N/A N/A	1.40 2.51
Perchloroethylene	(Zhu et al. 2013) (Langer et al. 2016)	N/A 1.94	1.2 0.31	1.4 N/A
Poly(dichlorophosphazene)	(Duan et al. 2016)	23.7	N/A	4.97
Tetrachloroethylene	(Schlink et al. 2004) (Adgate et al. 2004) (Adgate et al. 2004b) (Ohura et al. 2006) (Billionnet et al. 2011) (Logue et al. 2011) (Kumar et al. 2014) (Uchiyama et al., 2015) (Cheng et al. 2016)	5.3 N/A 2.3 N/A N/A 2.9 4.2/2.9/1.9 W:0.4 S:0.2 0.8	N/A N/A N/A 0.16 N/A N/A N/A N/A 0.5	1.4 W:0.5 SP:0.4 1.4 N/A 1.3 1.4 N/A W:0 S:0 0.5
Trans-1,2-dichloroethene	(Logue et al. 2011)	4.5	N/A	N/A
Trans-1,3-dichloropropene	(Logue et al. 2011)	2.2	N/A	N/A

Trichloroethylene	(Schlink et al. 2004) (Adgate et al. 2004) (Adgate et al. 2004b) (Ohura et al. 2006) (Billionnet et al. 2011) (Logue et al. 2011) (Kumar et al. 2014) (Zhu et al. 2013) (Uchiyama et al., 2015) (Cheng et al. 2016)	1.3 N/A 0.7 N/A N/A 2.3 2.6/2.2/2.2 0.21 W:0.1 S:0 0.6	N/A N/A N/A S:0.22 W:0.36 N/A N/A N/A 0.04 N/A 0.3	0.3 W:0.3 SP:0.2 0.5 N/A 0.50 0.5 N/A N/A W:0 S:0 0.3
Trichlorofluoromethane	(Weisel et al. 2008) (Logue et al. 2011) (Duan et al. 2016)	4.73 6.5 4.09	N/A N/A N/A	2.80 2.9 2.18
Trichloromethane	(Uchiyama et al., 2015)	W: 0.7 S:0.7	N/A	W:0.4 S:0
Vinyl chloride	(Logue et al. 2011)	0.16	N/A	N/A

Office environments – Halogenated compounds

Office Environment		**µgm-3**		
Halogenated Hydrocarbons	**Reference**	**Arithmetic Mean**	**Geometric Mean**	**Median**
1,1,1-Trichloroethane	(Brickus et al. 1998) (Chao et al. 2001) (Tham et al. 2004) (Zuraimi et al. 2006)	4.4 / 2.6 / 4.7 / 5.2 17.1 162.5 17.3	N/A 2.7 N/A N/A	N/A 0.8 N/A N/A
1,1,2,2-Tetrachloroethane	(Chao et al. 2001)	1	0.7	0.7
1,1,2-Trichloroethane	(Chao et al. 2001)	0.7	0.6	0.6
1,1,2-Trichloro-trifluoroethane	(Chao et al. 2001)	0.9	0.8	0.8
1,1-Dichloroethane	(Chao et al. 2001)	1.8	0.6	0.4
1,1-Dichloroethene	(Chao et al. 2001)	0.6	0.5	0.4
1,2,4-Trichlorobenzene	(Chao et al. 2001)	24.5	0.1	5
1,2-Dibromoethane	(Chao et al. 2001)	1.1	0.8	0.8
1,2-Dichlorobenzene	(Chao et al. 2001)	2.3	1	0.6
1,2-Dichloroethane	(Chao et al. 2001) (Ongwandee et al. 2011)	0.6 0.17	0.4 N/A	0.4 N/A
1,2-Dichloroethene	(Chao et al. 2001)	0.4	0.4	0.4
1,2-Dichloropropane	(Ongwandee et al. 2011)	0.08	N/A	N/A
1,2-Dichlorotetra-fluoroethane	(Chao et al. 2001)	0.7	0.6	0.7
1,3-Dichlorobenzene	(Chao et al. 2001)	2.2	1	0.6
1,4-Dichlorobenzene	(Chao et al. 2001) (Kim et al. 2001) (Tham et al. 2004)	10.2 0.1 128.0	2.9 N/A N/A	2.0 N/A N/A
Bromomethane	(Chao et al. 2001)	1.1	0.7	0.7
Carbon Tetrachloride	(Tovalin-Ahumada and Whitehead 2007) (Srivastava and Devotta 2007) (Jia et al. 2010)	1 57.50 N/A	N/A N/A N/A	0 N/A 0.2
Chlorobenzene	(Chao et al. 2001)	1.4	0.7	0.5

Chloroform	(Brickus et al. 1998) (Chao et al. 2001) (Guo et al. 2004) (Srivastava and Devotta 2007) (Ongwandee et al. 2011)	3.9 / 4.8 / 6.2 / 2.0 13.5 0.6 24.17 1.1	N/A 4.1 N/A N/A N/A	N/A 2.2 N/A N/A N/A
DiBromochloromethane	(Chao et al. 2001)	42.9	2	2.4
Dichloromethane	(Chao et al. 2001) (Guo et al. 2004)	50.2 0.03	9.1 N/A	4.3 N/A
Ethyl chloride	(Chao et al. 2001)	0.4	0.3	0.3
Perchloroethylene	(Tovalin-Ahumada and Whitehead 2007)	45	N/A	44
Tetrachloroethylene	(Chao et al. 2001) (Tham et al. 2004) (Guo et al. 2004) (Jia et al. 2010) (Ongwandee et al. 2011) (Mandin et al. 2017)	5.2 2321.0 0.10 N/A 0.92 W:8.2	1.9 N/A N/A N/A N/A N/A	1.8 N/A N/A 0.1 N/A N/A
Trichloroethylene	(Chao et al. 2001) (Guo et al. 2004) (Srivastava and Devotta 2007) (Ongwandee et al. 2011)	5.6 0.01 0.08 0.56	2.1 N/A N/A N/A	1.7 N/A N/A N/A
Trichlorofluoromethane	(Chao et al. 2001)	2.8	1.3	1.7
Vinyl chloride	(Chao et al. 2001)	0.3	0.3	0.3

Home environments – Other pollutants

Home Environment **µgm-3**

Others	Reference	Arithmetic Mean	Geometric Mean	Median
2-Ethyl hexanoic acid	(Jarnstrom et al. 2006) (Cheng et al. 2016)	2 0.4	N/A 0.1	N/A 0.2
3-Methyl butyl acetate	(Cheng et al. 2016)	0.8	0.5	0.5
Acetic Acid	(Jarnstrom et al. 2006) (Logue et al. 2011) (Uchiyama et al., 2015) (Cheng et al. 2016)	3/8 15 W:93 S:130 10.5	N/A N/A N/A 5.0	N/A 9.4 W:84 S:97 7.1
Acrylonitrile	(Logue et al. 2011)	0.27	N/A	0.06
Ammonia	(Jarnstrom et al. 2006) (Logue et al. 2011)	42/42/43 28	N/A N/A	N/A N/A
Benzoic Acid	(Jarnstrom et al. 2006)	3/3/3	N/A	N/A
Butyl Acetate	(Jarnstrom et al. 2006) (Takigawa et al. 2010) (Logue et al. 2011) (Takigawa et al. 2012) (Guo et al. 2013) (Uchiyama et al., 2015) (Cheng et al. 2016)	5/6/3 N/A 21 N/A N/A W: 4.2 S:6.2 1.5	N/A N/A N/A N/A N/A N/A 1.0	N/A 2.5/2.6 N/A 2.6/2.9 3.7/.36 W:1.4 S:0 1.0
Carbon disulfide	(Logue et al. 2011)	0.34	N/A	0.13
CO	(Logue et al. 2011)	810	N/A	710
Ethanol, 2,2-butoxyethoxyacetate	(Jarnstrom et al. 2006)	40/4	N/A	N/A

Ethyl Acetate	(Jarnstrom et al. 2006) (Logue et al 2011) (Takigawa et al. 2012) (Uchiyama et al., 2015) (Cheng et al. 2016)	3/4 18 N/A W:5.7 S:8.5 4.5	N/A N/A N/A N/A 2.8	N/A 1 5.7 W:1.2 S:0 2.9
Ethylmethacrylate	(Logue et al. 2011)	0.2	N/A	N/A
Formic Acid	(Uchiyama et al., 2015)	W:54 S:28	N/A	W:21 S:24
Hexanoic acid	(Cheng et al. 2016)	1.1	0.5	1.1
Methyl methacrylate	(Logue et al. 2011)	0.27	N/A	0.005
Methyl phenylacetate	(Cheng et al. 2016)	0.6	0.4	0.5
Methyl t-butyl ether	(Jo et al. 2003) (Jo et al. 2003) (Weisel et al. 2008) (Logue et al. 2011)	N/A N/A 19.3 12	N/A N/A N/A N/A	N:H:5.7 L:6.8 D:H:4.3 L:5.5 3.45 6
Nitrous Acid	(Logue et al. 2011)	5.3	N/A	6
NO	(Lawrence et al. 2005)	284.38 / 481.25/ 471.25	N/A	N/A
NO2	(Weschler et al. 1992) (Mosqueron et al. 2002) (Lawrence et al. 2005) (Stranger et al. 2007) (Logue et al. 2011) (Molloy et al. 2012) (Guo et al. 2013) (Abdul-Wahab et al. 2015) (Uchiyama et al., 2015) (Romagnoli et al. 2016)	23.99 / 43.77 / 39.52 35.1 433.81/479.4/ 489.74 33 13.1 15.79 N/A 75.2 / 18.8 W:220 S:13 42.2	N/A N/A N/A N/A N/A N/A N/A N/A N/A N/A	N/A 32.5 N/A N/A 16.5 N/A 20.5/40 N/A W:56 S:9.8 N/A
NOx	(Lawrence et al. 2005) (Romagnoli et al. 2016)	718.19 / 960.65 / 960.99 79.7	N/A N/A	N/A N/A
Octamethylcyclotetrasiloxane	(Zhu et al. 2013)	6.7	2.98	N/A
Ozone	(Logue et al. 2011) (Molloy et al. 2012) (Abdul-Wahab et al. 2015) (Uchiyama et al., 2015) (Romagnoli et al. 2016) (Bräuner et al. 2016)	17.2 1.4 60/ 80 / 100 W:1.7 S:10 3.1 2.66	N/A N/A N/A N/A N/A N/A	3.1 N/A N/A W:1.1 S:7.2 N/A N/A
PM10	(Fischer et al. 2000) (Stranger et al. 2007)	HT:37 LT:22 SM:49 NS:34	N/A N/A	HT: 28 LT: 21 N/A
PM2.5	(Fischer et al. 2000) (Kinney et al. 2002) (Mosqueron et al. 2002) (Stranger et al. 2007) (Logue et al. 2011)	HT:27 LT:12 20.9 24.7 SM:47 NS:30 15.9	N/A N/A N/A N/A N/A	HT:18 LT:12 N/A 22.5 N/A 15.7
SO2	(Logue et al. 2011) (Uchiyama et al., 2015)	15 W:2.2 S:0.6	N/A N/A	N/A W:0.7 S:0.5

Office environments – Other compounds

Office Environment **µgm-3**

Others	Reference	Arithmetic Mean	Geometric Mean	Median
Acetic Acid	(Salonen et al. 2009)	7.1	N/A	0

Butyl Acetate	(Tham et al. 2004)	0.7	N/A	N/A
Ethyl Acetate	(Brickus et al. 1998) (Salonen et al. 2009)	31.7/27.5/34.1/23.0 2.1	N/A N/A	N/A 0
Methyl t-butyl ether	(Tovalin-Ahumada and Whitehead 2007)	39	N/A	33
NO2	(Baek et al. 1997) (Mosqueron et al. 2002) (Wingfors et al. 2009) (Mandin et al. 2017)	62.04 44.9 S: 16 W: 9.7 S:16 W:18	N/A N/A N/A N/A	45.12 44.0 N/A S:16 W:18
Ozone	(Mandin et al. 2017)	S:9.0 W:3.9	N/A	S:5.6
PM10	(Horemans and Van Grieken 2010)	SM:67/26 NS:20/12	N/A	SM:63/26 NS:20/12
PM2.5	(Mosqueron et al. 2002) (Horemans and Van Grieken 2010)	34.5 SM:54/18 NS:15/11	N/A N/A	26.1 SM:57/20 NS:15.5/10

Home environments – Polyaromatic hydrocarbons (PAHs)

Home Environment **ngm-3**

PAHs	Reference	Arithmetic Mean	Geometric Mean	Median
1-Methylanthracene	(Logue et al. 2011)	3.5	N/A	3.1
1-Methylphenanthrene	(Logue et al. 2011)	2	N/A	2
2-Methylanthracene	(Logue et al. 2011)	0.74	N/A	0.49
3,6-Dimethylphenanthrene	(Logue et al. 2011)	0.95	N/A	0.87
9,10-Dimethylanthracene	(Logue et al. 2011)	0.13	N/A	0.044
9-Methylanthracene	(Logue et al. 2011)	0.17	N/A	0.11
Acenaphthylene	(Li et al. 2005) (Zhu et al 2009)	N/A S:69.9 W:40.8	0.06/0.05/0.09 N/A	N/A N/A
Acenaphthene	(Zhu et al 2009)	S:247 W:37.8	N/A	N/A
Anthracene	(Li et al. 2005) (Zhu et al 2009) (Logue et al. 2011)	N/A S:12.9 W:6.74 1	0.06/0.05/0.10 N/A N/A	N/A N/A 0.97
Benzo(a)fluorene	(Logue et al. 2011)	0.13	N/A	0.12
Benzo[a]fluoranthene	(Li et al. 2005)	N/A	0.71/0.210.91	N/A
Benzo(b)fluorene	(Logue et al. 2011)	0.049	N/A	0.033
Benzo(b)naphtho(2,1,d)thiopene	(Logue et al. 2011)	0.024	N/A	0.022
Benzo(b+k)fluoranthracene	(Logue et al. 2011)	0.24	N/A	0.15
Benzo[a]anthracene	(Li et al. 2005) (Zhu et al 2009) (Logue et al. 2011) (Romagnoli et al. 2016)	N/A S: 34.4 W: 11.7 0.065 0.02	1.29/0.65/0.91 N/A N/A N/A	N/A N/A 0.040 N/A
Benzo[a]pyrene	(Fischer et al. 2000) (Naumova et al. 2002) (Li et al. 2005) (Zhu et al 2009) (Logue et al. 2011) (Romagnoli et al. 2016)	HT: 0.49 LT: 0.17 N/A N/A S: 0.611 W: 4.48 0.091 0.15	N/A 0.078 / 0.02 / 0.055 4.19/1.25/4.92 N/A N/A N/A	HT: 0.38 LT: 0.16 N/A N/A N/A 0.09 N/A

Benzo[b]fluoranthene	(Li et al. 2005) (Zhu et al 2009) (Logue et al. 2011) (Romagnoli et al. 2016)	N/A S: 1.38 W: 5.63 0.14 0.24	7.10/1.98/5.76 N/A N/A N/A	N/A N/A 0.20 N/A
Benzo[e]pyrene	(Li et al. 2005) (Logue et al. 2011) (Romagnoli et al. 2016)	N/A 0.075 0.17	5.91/1.88/6.33 N/A N/A	N/A 0.12 N/A
Benzo[ghi]perylene	(Li et al. 2005) (Zhu et al 2009) (Logue et al. 2011) (Romagnoli et al. 2016)	N/A S: 1.19 W: 4.34 0.17 0.27	9.14/2.82/10.79 N/A N/A N/A	N/A N/A 0.19 N/A
Benzo[j]fluoranthene	(Romagnoli et al. 2016)	0.16	N/A	N/A
Benzo[k]fluoranthene	(Li et al. 2005) (Zhu et al 2009) (Logue et al. 2011) (Romagnoli et al. 2016)	N/A S: 0.678 W: 2.41 0.12 0.10	4.77/1.46/5.37 N/A N/A N/A	N/A N/A 0.05 N/A
Chrysene	(Li et al. 2005) (Zhu et al 2009) (Logue et al. 2011) (Romagnoli et al. 2016)	N/A S: 7.2 W: 6.10 0.15 0.10	2.94/1.38/1.84 N/A N/A N/A	N/A N/A 0.21 N/A
Coronene	(Li et al. 2005) (Logue et al. 2011)	N/A 0.2	3.78/1.28/3.12 N/A	N/A 0.15
Cyclopenta[c,d]pyrene	(Logue et al. 2011)	0.073	N/A	0.04
Dibenzo[ac+ah]anthracene	(Logue et al. 2011)	0.014	N/A	0.0088
Dibenzo[ah]anthracene	(Li et al. 2005) (Zhu et al 2009) (Logue et al. 2011) (Romagnoli et al. 2016)	N/A S: 0.310 W:0.696 0.019 0.06	2.53/0.79/2.71 N/A N/A N/A	N/A N/A 0.07 N/A
Fluoranthene	(Li et al. 2005) (Zhu et al 2009) (Logue et al. 2011)	N/A S:17.0 W:12.2 1.4	0.86/0.49/0.49 N/A N/A	N/A N/A 0.45
Fluorene	(Li et al. 2005) (Zhu et al 2009)	N/A S:229 W:48.9	0.16/0.12/0.24 N/A	N/A N/A
Indeno[1,2,3-cd]pyrene	(Li et al. 2005) (Zhu et al 2009) (Logue et al. 2011) (Romagnoli et al. 2016)	N/A S: 0.652 W: 4.89 0.16 0.24	6.52/2.29/5.78 N/A N/A N/A	N/A N/A 0.24 N/A
Naphthalene	(Schlink et al. 2004) (Li et al. 2005) (Logue et al. 2011) (Zhu et al. 2013) (Cheng et al. 2016)	800 N/A 1200 2620 1500	N/A 2.11/2.24/1.02 N/A 850 700	300 N/A 310 720 500
Perylene	(Li et al. 2005) (Logue et al. 2011) (Romagnoli et al. 2016)	N/A 0.021 0.04	0.78/0.23/0.93 N/A N/A	N/A 0.011 N/A
Phenanthrene	(Naumova et al. 2002) (Li et al. 2005) (Zhu et al 2009) (Logue et al. 2011)	N/A N/A S: 464 W: 124 18	21/27/33 0.53/0.35/0.67 N/A N/A	N/A N/A N/A 13
Pyrene	(Li et al. 2005) (Zhu et al 2009) (Logue et al. 2011)	N/A S:36.1 W:16.4 1.3	0.94/0.58/0.61 N/A N/A	N/A N/A 1.5
Retene	(Logue et al. 2011)	0.72	N/A	0.71

Office environments - Polyaromatic hydrocarbons (PAHs)

Office Environment **ngm-3**

PAHs	Reference	Arithmetic Mean	Geometric Mean	Median
Acenaphthene	(Tsai et al. 2002) (Liu et al. 2010)	87.42 70	N/A N/A	N/A N/A
Acenaphthylene	(Tsai et al. 2002) (Liu et al. 2010)	93.98 180	N/A N/A	N/A N/A
Anthracene	(Tsai et al. 2002) (Liu et al. 2010)	0.99 70	N/A N/A	N/A N/A
Benzo(b+j+k)fluoranthene	(Romagnoli et al. 2014)	W: 2.22/2.57 SP:2.2/0.42	N/A	N/A
Benzo[a]anthracene	(Tsai et al. 2002) (Romagnoli et al. 2014)	0.12 W: 0.30/0.72 SP: 0.05/0.08	N/A N/A	N/A N/A
Benzo[a]pyrene	(Tsai et al. 2002) (Wingfors et al. 2009) (Romagnoli et al. 2014)	0 S: 0.09 W: 7.4 W: 1.4 / 1.75 SP: 0.11 / 0.19	N/A N/A N/A	N/A N/A N/A
Benzo[b]chrycene	(Tsai et al. 2002)	0.043	N/A	N/A
Benzo[b]fluoranthene	(Tsai et al. 2002) (Liu et al. 2010)	0.09 100	N/A N/A	N/A N/A
Benzo[e]pyrene	(Tsai et al. 2002)	0.04	N/A	N/A
Benzo[ghi]perylene	(Tsai et al. 2002) (Romagnoli et al. 2014)	0.09 W: 1.47/1.57 SP: 0.2/0.27	N/A N/A	N/A N/A
Benzo[k]fluoranthene	(Tsai et al. 2002)	0.07	N/A	N/A
Chrysene	(Tsai et al. 2002)	0.26	N/A	N/A
Coronene	(Tsai et al. 2002)	0.06	N/A	N/A
Cyclopenta[c,d]pyrene	(Tsai et al. 2002)	0.14	N/A	N/A
Dibenzo[ah]anthracene	(Tsai et al. 2002) (Romagnoli et al. 2014)	0.028 W: 0.27/0.24 SP: 0.02/0.04	N/A N/A	N/A N/A
Fluoranthene	(Tsai et al. 2002) (Liu et al. 2010)	8.24 100	N/A N/A	N/A N/A
Fluorene	(Tsai et al. 2002) (Liu et al. 2010)	4.510 800	N/A N/A	N/A N/A
Indeno[1,2,3-cd]pyrene	(Tsai et al. 2002) (Liu et al. 2010) (Romagnoli et al. 2014)	0.28 10 W: 1.46/2.04 SP: 0.13/0.27	N/A N/A N/A	N/A N/A N/A
Naphthalene	(Kim et al. 2001) (Tsai et al. 2002) (Tham et al. 2004) (Zuraimi et al. 2006) (Srivastava and Devotta 2007) (Liu et al. 2010) (Jia et al. 2010)	1700 1.17548 55000 6500 20 15870 N/A	N/A N/A N/A N/A N/A N/A N/A	N/A N/A N/A N/A N/A N/A 500
Perylene	(Tsai et al. 2002)	0.03	N/A	N/A
Phenanthrene	(Tsai et al. 2002) (Liu et al. 2010)	2.483 170	N/A N/A	N/A N/A
Pyrene	(Tsai et al. 2002) (Liu et al. 2010)	7.05 10	N/A N/A	N/A N/A

Home environments – SVOCs

Home Environment			**ngm-3**	
SVOCs	**Reference**	**Arithmetic Mean**	**Geometric Mean**	**Median**
2,2',5,5'-Tetrachlorobiphenyl	(Hazrati et al. 2006)	0.146/0.150/0.037/0.077	N/A	N/A
2,4,4/ 2,4,5-Trichlorobiphenyl	(Hazrati et al. 2006)	0.433/0.438/0.097/0.258	N/A	N/A
2-Ethyl hexanol	(Logue et al. 2011)	3700	N/A	N/A
4-Nonylphenol	(Logue et al. 2011)	N/A	N/A	110
4-tertbutylphenol	(Logue et al. 2011)	N/A	N/A	16
Atrazine	(Logue et al. 2011)	N/A	N/A	0.32
Azinphos methyl	(Logue et al. 2011)	N/A	N/A	0.4
BDE 100	(Logue et al. 2011)	0.029	N./A	0.011
BDE 153	(Logue et al. 2011)	0.0032	N./A	0.001
BDE 154	(Logue et al. 2011)	0.002	N./A	0.0006
BDE 17	(Logue et al. 2011)	0.024	N./A	0.016
BDE 209	(Han et al. 2016)	S:0.213 W:0.067	S:0.138 W:0.056	S:0.130 W:0.046
BDE 28	(Logue et al. 2011)	0.05	N./A	0.032
BDE 47	(Hazrati et al. 2006) (Logue et al. 2011)	0.004/0.003/0.019/0.061 0.37	N/A N/A	N/A 0.14
BDE 66	(Logue et al. 2011)	0.058	N./A	0.045
BDE 71	(Logue et al. 2011)	0.019	N./A	0.0092
BDE 75	(Logue et al. 2011)	0.0099	N./A	0.0012
BDE 85	(Logue et al. 2011)	0.001	N./A	0.0001
BDE 99	(Hazrati et al. 2006) (Logue et al. 2011)	0.008/0.007/0.019/0.053 0.35	N/A N/A	N/A 0.041
Bis(2etylhexyl) phthalate	(Logue et al. 2011)	N/A	N/A	77
Butyl benzyl phthalate (BBP)	(Fromme et al. 2004)	N/A	N/A	18
Cischlordane	(Logue et al. 2011)	2.1	N/A	0.5
Cisnonachlor	(Logue et al. 2011)	0.11	N/A	0.041
DEHA	(Logue et al. 2011)	N/A	N/A	9
Di(2-ethylhexyl) phthalate (DEHP)	(Fromme et al. 2004) (Bu et al. 2016)	N/A 270	N/A N/A	156 N/A
Di(isobutyl) phthalate (DiBP)	(Logue et al. 2011) (Bu et al. 2016)	N/A 320	N/A N/A	11 N/A
Di(nbutyl) phthalate (DnBP)	(Bu et al. 2016)	210	N/A	N/A
Dibenzothiopene	(Logue et al. 2011)	3.6	N/A	3
Diethyl phthalate (DEP)	(Fromme et al. 2004) (Logue et al. 2011) (Bu et al. 2016)	N/A N/A 170	N/A N/A N/A	643 590 N/A
Dimethyl phthalate (DMP)	(Fromme et al. 2004) (Bu et al. 2016)	N/A 910	N/A N/A	436 N/A
Dimethylpropyl phthalate (DMPP)	(Fromme et al. 2004)	N/A	N/A	459
Di-n-butyl phthalate (DBP)	(Fromme et al. 2004) (Logue et al. 2011)	N/A N/A	N/A N/A	1083 220

Methyl Benzoate	(Schlink et al. 2004) (Logue et al. 2011)	600 640	N/A N/A	0 50
Methylparaben	(Logue et al. 2011)	N/A	N/A	2.9
Nonylphenol monoethyoxylate	(Logue et al. 2011)	N/A	N/A	17
Octylphenol monoethyoxylate	(Logue et al. 2011)	N/A	N/A	8.6
O-phenylphenol	(Logue et al. 2011)	N/A	N/A	71
Oxychlorodane	(Logue et al. 2011)	0.0039	N/A	0.0018
PCB 70/76	(Hazrati et al. 2006)	0.058/0.062/0.010/0.022	N/A	N/A
PCP	(Logue et al. 2011)	N/A	N/A	1.6
Phenol	(Jarnstrom et al. 2006) (Logue et al. 2011) (Cheng et al. 2016)	3000/2000 360 500	N/A N/A 0.3	N/A 420 300
Transchlordane	(Logue et al. 2011)	3.5	N/A	0.82
Transnonachlor	(Logue et al. 2011)	1.4	N/A	0.38
Transpermethrin	(Logue et al. 2011)	N/A	N/A	0.9

Office environments – SVOCs

Office Environment **ngm-3**

SVOCs	Reference	Arithmetic Mean	Geometric Mean	Median
2,4,4/ 2,4,5-Trichlorobiphenyl	(Hazrati et al. 2006)	0.217/0.247	N/A	N/A
2,2',5,5'-Tetrachlorobiphenyl	(Hazrati et al. 2006)	0.100/0.098	N/A	N/A
Biphenyl	(Wingfors et al. 2009)	W:28 S: 2.1	N/A	N/A
Butyl benzyl phthalate (BBP)	(Song et al. 2015)	419.61	N/A	N/A
BDE 209	(Han et al. 2016)	S:0.138 W:0.075	S:0.107 W:0.049	S:0.090 W:0.047
BDE 47	(Hazrati et al. 2006)	0.568/0.069	N/A	N/A
BDE 99	(Hazrati et al. 2006)	0.633/0.081	N/A	N/A
Di(2-ethylhexyl) phthalate (DEHP)	(Song et al. 2015)	591.22	N/A	N/A
Diethyl phthalate (DEP)	(Song et al. 2015)	657.96	N/A	N/A
Di-n-butyl phthalate (DBP)	(Song et al. 2015)	658.78	N/A	N/A
PCB 70/76	(Hazrati et al. 2006)	0.030/0.027	N/A	N/A

Home environments – Terpenes

Home Environment **µgm-3**

Terpenes / Terpenoids	Reference	Arithmetic Mean	Geometric Mean	Median
2-Carene	(Schlink et al. 2004) (Logue et al. 2011)	0.5 0.29	N/A N/A	0.0 0.03

3-Carene	(Schlink et al. 2004) (Jarnstrom et al. 2006) (Gokhale et al. 2008) (Logue et al. 2011) (Cheng et al. 2016)	7.3 18/18/19 4.88 8.5 0.4	N/A N/A N/A N/A 0.2	2.5 N/A N/A 3.7 0.3
Alpha-pinene	(Schlink et al. 2004) (Adgate et al. 2004) (Jarnstrom et al. 2006) (Gokhale et al. 2008) (Takigawa et al. 2010) (Geiss et al. 2011) (Logue et al. 2011) (Takigawa et al. 2012) (Zhu et al. 2013) (Uchiyama et al., 2015) (Cheng et al. 2016)	23.3 N/A 61/37/35 6.4 N/A 14.5 37 N/A 13.10 W: 5.6 S:30 5.8	N/A N/A N/A N/A N/A N/A N/A N/A 5.62 N/A 3.5	9.8 W:2.4 SP:2.4 N/A N/A 7.0/7.6 6.1 12 7.4/8.2 N/A W:1.3 S:3.7 2.9
Beta-Pinene	(Schlink et al. 2004) (Adgate et al. 2004) (Jarnstrom et al. 2006) (Gokhale et al. 2008) (Logue et al. 2011) (Cheng et al. 2016)	2.7 N/A 18/18/19 0.99 3.4 3.9	N/A N/A N/A N/A N/A 2.5	1.3 W:2.5 SP:1.5 N/A N/A 1.2 2.2
Camphene	(Zhu et al. 2013)	0.77	0.46	N/A
Camphor	(Cheng et al. 2016)	1.7	0.6	0.4
Eucalyptol	(Cheng et al. 2016)	5.6	2	1.8
Isoprene	(Logue et al. 2011) (Zhu et al. 2013) (Duan et al. 2016) (Cheng et al. 2016)	4 5.83 4.16 2.0	N/A 3.06 N/A 1.6	2 N/A 3.38 1.6
Limonene	(Schlink et al. 2004) (Adgate et al. 2004) (Jarnstrom et al. 2006) (Gokhale et al. 2008) (Takigawa et al. 2010) (Geiss et al. 2011) (Logue et al. 2011) (Takigawa et al. 2012) (Zhu et al. 2013) (Uchiyama et al., 2015) (Cheng et al. 2016)	32.9 N/A 18/18/19 14.58 N/A 29.2 34 N/A 37.89 W:24 S:17 11.3	N/A N/A N/A N/A N/A N/A N/A N/A 21.30 N/A 7.1	16.0 W: 28.6 SP: 21.2 N/A N/A 9.0/9.0 9.5 18 8.7/9.3 N/A W:13 S:7.3 6.5

Office environments – Terpenes

Office Environment **µgm-3**

Terpenes	Reference	Arithmetic Mean	Geometric Mean	Median
Alpha-pinene	(Tham et al. 2004) (Tovalin-Ahumada and Whitehead 2007) (Salonen et al. 2009) (Jia et al. 2010) (Mandin et al. 2017)	1.3 15 2.0 N/A S:4.2 W:6.3	N/A N/A N/A N/A N/A	N/A 5 0.6 0.8 S:3.0 W:4.0
Camphene	(Tovalin-Ahumada and Whitehead 2007) (Srivastava and Devotta 2007) (Jia et al. 2010)	1 57.50 N/A	N/A N/A N/A	0 N/A 0.2

Isoprene	(Zuraimi et al. 2006)	9	N/A	N/A
Limonene	(Brickus et al. 1998) (Tham et al. 2004) (Zuraimi et al. 2006) (Tovalin-Ahumada and Whitehead 2007) (Salonen et al. 2009) (Jia et al. 2010) (Ongwandee et al. 2011) (Mandin et al. 2017)	52.0 / 21.5 / 16.7 / 5.6 8.6 34.6 11 4.0 N/A 60.6 S:4.7 W:19	N/A N/A N/A N/A N/A N/A N/A N/A	N/A N/A N/A 7 0.6 3.0 N/A S:3.9 W:13

Appendix B Relevant health guidelines or standards for indoor pollutants

Guidelines or standards from international and national organisations

		Ambient Air	Indoor Air	
	Averaging Period	**EU Ambient Air Quality Directive (µg m^{-3})**	**WHO IAQ Guidelines (µg m^{-3})**	**Japanese Ministry of Health, Labour and Welfare (µg m^{-3})**
1,2-Dichlorobenzene	Eight Hour	N/A	N/A	N/A
1,4-Dichlororbenzene	Long Term	N/A	N/A	240
Acetaldehyde	Long Term	N/A	N/A	48
Benzene	Eight Hour Annual	N/A Limit Value : 5	No safe level of exposure recommended (suggests no difference between ambient and indoor guidelines)	N/A N/A
Carbon Tetrachloride	Eight Hour	N/A	N/A	N/A
Chloroform	Eight Hour	N/A	N/A	N/A
DBP	Long Term	N/A	N/A	220
DEHP	Long Term	N/A	N/A	120
Ethylbenzene	Eight Hour Long Term	N/A N/A	N/A N/A	N/A 3800
Formaldehyde	30 Minute Eight Hour	N/A N/A	100 N/A	100 N/A
Naphthalene	Annual	N/A	10	N/A
NO_2	Eight Hour One Hour Annual	N/A Limit Value: 200 Limit Value: 40	N/A 200 40	N/A N/A N/A
Ozone	Eight Hour	120	N/A	N/A
B(a)P	Annual	0.001	All exposures considered relevent to health (B(a)P used as marker for all PAH)	N/A
PM_{10}	Eight Hour One Day Calender Year	N/A Limit Value: 50 Limit Value: 40	N/A N/A N/A	N/A N/A N/A
$PM_{2.5}$	One Day Calender Year	Limit Value: N/A Limit Value: 25	N/A N/A	N/A N/A
Styrene	Long Term	N/A	N/A	220
Tetrachloroethylene	Eight Hour Annual	N/A N/A	N/A 250	N/A N/A
Tetradecane	Long Term	N/A	N/A	330
Toluene	Eight Hour Long Term	N/A N/A	N/A N/A	N/A 260
Trichloroethylene	Eight Hour	N/A	N/A	N/A
TVOC	Eight Hour Long Term	N/A N/A	N/A N/A	N/A 400

Xylene	Eight Hour	N/A	N/A	N/A
	Long Term	N/A	N/A	870

Dose response assessments

	OEHHA (µg m^{-3})				US EPA (µg m^{-3})	
	Acute REL (1hr)	**REL (8 hr)**	**Non-cancer - Chronic REL**	**Cancer**	**Non cancer - Chronic REL**	**Cancer**
Acetaldehyde	470	300	140	3.70	9	
Acrolein	2.5	0.7	0.35		0.02	
Ammonia	3200		200			
Acrylonitrile			5	0.03	2	
Benzene	27	3	3		30	
Benzyl Chloride	240			0.20		
Bis(2etylhexyl) phthalate					10	
Butadiene. 1,3-	660	9	2		2	
Carbon disulfide	6200		800		700	
Carbon tetrachloride	1900		40	0.24	100	
Chlorobenzene			1000		1000	
Chloroethane			30000			
Chloroform	150		300	1.89	98	1.89
Dibromoethane, 1,2-			0.8			
Dichloroethane, 1,1-					500	6.25
Dichloroethane, 1,2-			400	0.48		
Dichloroethene, 1,1-			70			
Ethyl benzene			2000	4.00	1000	
Ethyl chloride					10000	
Formaldehyde	55	9	9	1.57	9.8	
Hexachlorobutadiene					90	0.45
Hexane			7000		700	
Isopropylbenzene (Cumene)					400	
Methyl Chloride					90	
Methylene chloride (Dichloromethane)	14000		400	10.00	600	
MIBK					3000	
MTBE			8000		3000	
Naphthalene			9		3	
NO2	470					
Ozone	180					
Phenol	5800		200		200	

Propionaldehyde				8	
Styrene	21000	900		1000	
Tetrachloroethene	20000	35	1.69	40	
Toluene	37000	300		5000	
Trichlorobenzene, 1,2,4-				200	
Trichloroethane, 1,1,1-	68000	1000		5000	
Trichloroethane, 1,1,2-				400	0.63
Trichloroethene		600		2	
Vinyl chloride	180000		0.13	100	
Xylenes (m,o,p)	22000	700		100	

Appendix C Summary of Plant-pollutant VOC removal experiments

Species	Chemical	Reference	Family
Adenium obesum	Triethylamine	Siswanto et al. (2016)	*Apocynaceae*
Adiantum capillusveneris L.	Formaldehyde	Kim et al. (2010)	*Pteridaceae*
Aechmea fasciata	Formaldehyde	Wolverton and Wolverton (1993)	*Bromeliaceae*
Agave potatorum	Formaldehyde	Zhou et al. (2011)	*Agavaceae*
Aglaonema 'Silver Queen' (Species not Given)	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Araceae*
Aglaonema 'Silver Queen' (Species not Given)	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Araceae*
Aglaonema commutatum 'Golden Jewelry'	Formaldehyde	Zhou et al. (2011)	*Araceae*
Aglaonema commutatum 'Red Narrow'	Formaldehyde	Zhou et al. (2011)	*Araceae*
Aglaonema commutatum 'Silver Queen'	Formaldehyde	Zhou et al. (2011)	*Araceae*
Aglaonema commutatum 'Treubii'	Formaldehyde	Zhou et al. (2011)	*Araceae*
Aglaonema commutatum 'White Rajah'	Formaldehyde	Zhou et al. (2011)	*Araceae*
Aglaonema modestum	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Araceae*
Aglaonema modestum	Formaldehyde	Kim et al. (2010)	*Araceae*
Alocasia macrorrhiza	Formaldehyde	Zhou et al. (2011)	*Araceae*
Aloe aristata	Formaldehyde	Zhou et al. (2011)	*Asphodelaceae*
Aloe barbadensis	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Asphodelaceae*
Aloe nobilis	Formaldehyde	Zhou et al. (2011)	*Asphodelaceae*
Aloe vera	Formaldehyde	Xu et al. (2011)	*Asphodelaceae*
Aloe vera	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Asphodelaceae*
Aloysia triphylla	Toluene	Kim et al. (2011b)	*Verbenaceae*
Anthurium andraeanum	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Araceae*
Anthurium andraeanum	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Araceae*
Anthurium andraeanum	Triethylamine	Siswanto et al. (2016)	*Araceae*
Anthurium andraeanum Linden	Formaldehyde	Kim et al. (2010)	*Araceae*

Arachniodes aristata (G. Forst.) Tindale	Formaldehyde	Kim et al. (2010)	*Dryopteridaceae*
Araucaria heterophylla Franco	Formaldehyde	Kim et al. (2010)	*Araucariaceae*
Ardisia crenata	Toluene	Kim et al. (2011b)	*Primulaceae (formerly Myrsinaceae)*
Ardisia crenata	Toluene	Kim et al. (2012)	*Primulaceae (formerly Myrsinaceae)*
Ardisia crenata Sims	Formaldehyde	Kim et al. (2010)	*Primulaceae (formerly Myrsinaceae)*
Ardisia japonica	Formaldehyde	Kim and Kim (2008)	*Primulaceae (formerly Myrsinaceae)*
Ardisia japonica	Toluene	Kim et al. (2011b)	*Primulaceae (formerly Myrsinaceae)*
Ardisia japonica	Toluene	Kim et al. (2012)	*Primulaceae (formerly Myrsinaceae)*
Ardisia pusilla	Toluene	Kim et al. (2011b)	*Primulaceae (formerly Myrsinaceae)*
*Ardisia pusilla*DC	Formaldehyde	Kim et al. (2010)	*Primulaceae (formerly Myrsinaceae)*
Asparagus densiflorus	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Asparagaceae*
Asparagus setaceus	Formaldehyde	Zhou et al. (2011)	*Asparagaceae*
Aspidistra elatior	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Asparagaceae*
Asplenium nidus	Triethylamine	Siswanto et al. (2016)	*Aspleniaceae*
Asplenium nidus L. 'Avis'	Formaldehyde	Kim et al. (2010)	
Azalea indica	Toluene	De Kempeneer et al. (2004)	*Ericaceae*
Begonia maculata	Toluene	Kim et al. (2011b)	*Begoniaceae*
Begonia maculata	Toluene	Kim et al. (2012)	
Botrychium ternatum(Thunb.) Swartz.	Formaldehyde	Kim et al. (2010)	*Ophioglossaceae*
Bougainvillea spectabilis	Triethylamine	Siswanto et al. (2016)	*Nyctaginaceae*
Calathea makoyana E. Morr.	Formaldehyde	Kim et al. (2010)	*Marantaceae*
Calathea ornata	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Marantaceae*
Calathea roseopicta	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Marantaceae*
Camellia japonica L.	Formaldehyde	Kim et al. (2010)	*Theaceae*
Camellia sinensis Kuntz	Formaldehyde	Kim et al. (2010)	

Carica papaya	Triethylamine	Siswanto et al. (2016)	*Caricaceae*
Chamaecyparis obtusa Endl	Formaldehyde	Kim et al. (2010)	*Cupressaceae*
Chamaedorea elegans	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Arecaceae*
Chamaedorea seifrizii	Benzene	Treesubsuntorn and Thiravetyan (2012)	*Arecaceae*
Chamaedorea seifrizii	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Arecaceae*
Chlorophytum bichetii Baker	Formaldehyde	Kim et al. (2010)	*Asparagaceae*
Chlorophytum comosum	Benzene pentane toluene	Cornejo et al. (1999)	*Asparagaceae*
Chlorophytum comosum	Formaldehyde	Godish and Guindon (1989)	*Asparagaceae*
Chlorophytum comosum	Formaldehyde	Xu et al. (2011)	*Asparagaceae*
Chlorophytum comosum	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Asparagaceae*
Chlorophytum comosum	Formaldehyde	Zhou et al. (2011)	*Asparagaceae*
Chlorophytum comosum	Formaldehyde	Su et al. (2015)	*Asparagaceae*
Chlorophytum comosum 'Vittatum'	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Asparagaceae*
Chlorophytum elatum	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Asparagaceae*
Chrysalidocarpus lutescens H. Wendl	Formaldehyde	Kim et al. (2010)	*Arecaceae*
Chrysanthemum morifolium	Formaldehyde	Aydogan and Montoya (2011)	*Asteraceae*
Chrysanthemum morifolium	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Asteraceae*
Chrysanthemum morifolium	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Asteraceae*
Cinnamomum camphora	Toluene	Kim et al. (2011b)	*Lauraceae*
Cissus rhombifolia	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Vitaceae*
Cissus rhombifolia	Benzene toluene	Yoo et al. (2006)	*Vitaceae*
Citrus medica 'Sarcodactylis'	Benzene	Liu et al. (2007)	*Rutaceae*
Clivia miniata Regal	Formaldehyde	Kim et al. (2010)	*Amaryllidaceae*
Codiaeum variegatum	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Euphorbiaceae*
Coniogramme japonica (Thunb.) Diels	Formaldehyde	Kim et al. (2010)	*Pteridaceae*
Cordyline fruticosa	Formaldehyde	Zhou et al. (2011)	*Asparagaceae*
Crassula portulacea	Benzene	Liu et al. (2007)	*Crassulaceae*

Cupressus macrocarpa Hartweg 'Gold Crest'	Formaldehyde	Kim et al. (2010)	*Cupressaceae*
Cycas revoluta Thunb	Formaldehyde	Kim et al. (2010)	*Cycadaceae*
Cyclamen persicum	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Primulaceae*
Cymbidium 'Golden Elf'	Benzene	Liu et al. (2007)	*Orchidaceae*
Cymbidium Meglee 'Ms Taipei'	Formaldehyde	Kim and Lee (2008)	*Orchidaceae*
Cymbidium (Species not Given)	Formaldehyde	Kim and Lee (2008)	*Orchidaceae*
Cyrtomium caryotideum Nakai *'coreanum'*	Formaldehyde	Kim et al. (2010)	*Dryopteridaceae*
Cyrtomium falcatum (L.f.) Presl.	Formaldehyde	Kim et al. (2010)	*Dryopteridaceae*
Davallia mariesii	Toluene	Kim et al. (2011b)	*Davalliaceae*
Davallia mariesii Moore ex Baker	Formaldehyde	Kim et al. (2010)	*Davalliaceae*
Dendranthema morifolium	Benzene	Liu et al. (2007)	*Asteraceae*
Dendrobium phalaenopsis	Formaldehyde	Kim and Lee (2008)	*Orchidaceae*
Dendrobium(Species not Given)	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Orchidaceae*
Dendropanax morbifera Nakai	Formaldehyde	Kim et al. (2010)	*Araliaceae*
Dieffenbachia 'Exotica Compacta'	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Araceae*
Dieffenbachia amoena 'Camilla'	Formaldehyde	Zhou et al. (2011)	*Araceae*
Dieffenbachia amoena 'Green Magic'	Formaldehyde	Zhou et al. (2011)	*Araceae*
Dieffenbachia amoena 'Marianne'	Formaldehyde	Kim et al. (2010)	*Araceae*
Dieffenbachia amoena 'Tropic Snow'	Benzene	Liu et al. (2007)	*Araceae*
Dieffenbachia amoena 'Tropic Snow'	Benzene toluene	Porter (1994)	*Araceae*
Dieffenbachia camille	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Araceae*
Dieffenbachia compacta	Formaldehyde	Aydogan and Montoya (2011)	*Araceae*
Dieffenbachia maculata	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Araceae*
Dieffenbachia seguine	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Araceae*
Dizygotheca elegantissima R. Vig. & G *(Syn. Schefflera elegantissima; Plerandra elegantissima)*	Formaldehyde	Kim et al. (2010)	*Araliaceae*
Dracaena angustifolia	Formaldehyde	Zhou et al. (2011)	*Asparagaceae*
Dracaena concinna Kunth	Formaldehyde	Kim et al. (2010)	*Asparagaceae*
Dracaena deremensi	Benzene pentane toluene	Cornejo et al. (1999)	*Asparagaceae*
Dracaena deremensis 'Compacta'	Formaldehyde	Zhou et al. (2011)	*Asparagaceae*
Dracaena deremensis 'Janet Craig'	Toluene, m-xylene	Orwell et al. (2006)	*Asparagaceae*
Dracaena deremensis 'Janet Craig'	Benzene n-hexane	Wood et al. (2002)	*Asparagaceae*

Dracaena deremensis 'Janet Craig'	Benzene	Orwell et al. (2004)	*Asparagaceae*
Dracaena deremensis 'Janet Craig'	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Asparagaceae*
Dracaena deremensis 'Janet Craig'	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Asparagaceae*
Dracaena deremensis 'Variegata'	Benzene	Liu et al. (2007)	*Asparagaceae*
Dracaena deremensis 'Warneckii'	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Asparagaceae*
Dracaena deremensis 'Warneckii'	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Asparagaceae*
Dracaena deremensis N.E. Br. *'Warneckii'*	Formaldehyde	Kim et al. (2010)	*Asparagaceae*
Dracaena fragrans	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Asparagaceae*
Dracaena fragrans	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Asparagaceae*
Dracaena fragrans 'Massangeana'	Formaldehyde	Zhou et al. (2011)	*Asparagaceae*
Dracaena fragrans Ker. *'Massangeana'*	Formaldehyde	Kim et al. (2010)	*Asparagaceae*
Dracaena marginata	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Asparagaceae*
Dracaena marginata	Benzene	Orwell et al. (2004)	*Asparagaceae*
Dracaena marginata	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Asparagaceae*
Dracaena massangeana	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Asparagaceae*
Dracaena reflexa	Formaldehyde	Zhou et al. (2011)	*Asparagaceae*
Dracaena sanderiana	Benzene	Treesubsuntorn and Thiravetyan (2012)	*Asparagaceae*
Dracaena sanderiana	Formaldehyde	Zhou et al. (2011)	*Asparagaceae*
Dryopteris nipponensis Koidz.	Formaldehyde	Kim et al. (2010)	*Dryopteridaceae*
Duranta erecta	Triethylamine	Siswanto et al. (2016)	*Verbenaceae*
Elaeocarpus sylvestris Hara *'ellipticus'*	Formaldehyde	Kim et al. (2010)	*Elaeocarpaceae*
Epipremnum aureum	Formaldehyde	Aydogan and Montoya (2011)	*Araceae*
Epipremnum aureum	Formaldehyde acetone benzene toluene xylene styrene	Baosheng et al. (2009)	*Araceae*
Epipremnum aureum	Formaldehyde	Kim and Kim (2008)	*Araceae*
Epipremnum aureum	Formaldehyde	Kim et al. (2009)	*Araceae*
Epipremnum aureum	Benzene	Orwell et al. (2004)	*Araceae*

Epipremnum aureum	Acetone benzene ethyl alcohol formaldehyde toluene, trichloroethylene xylene	Oyabu et al. (2001)	*Araceae*
Epipremnum aureum	Formaldehyde	Oyabu et al. (2003a)	*Araceae*
Epipremnum aureum	Acetone formaldehyde	Oyabu et al. (2003b)	*Araceae*
Epipremnum aureum	Gasoline	Oyabu et al. (2003c)	*Araceae*
Epipremnum aureum	Formaldehyde toluene xylene	Oyabu et al. (2005)	*Araceae*
Epipremnum aureum	Formaldehyde toluene xylene	Sawada and Oyabu (2008)	*Araceae*
Epipremnum aureum	Acetone benzaldehyde iso-butyraldehyde n-butyraldehyde crotonaldehyde diethyl ketone, methacrolein methyl ethyl ketone methyl isobutyl ketone methyl isopropyl ketone methyl n-propyl ketone, propionaldehyde iso-valeraldehyde, n-valeraldehyde	Tani and Hewitt (2009)	*Araceae*
Epipremnum aureum	Methyl isobutyl ketone	Tani et al. (2007)	*Araceae*
Epipremnum aureum	Benzene	Treesubsuntorn and Thiravetyan (2012)	*Araceae*
Epipremnum aureum	Formaldehyde	Xu et al. (2011)	*Araceae*
Epipremnum aureum	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Araceae*
Epipremnum aureum Bunt	Formaldehyde	Kim et al. (2010)	*Araceae*
Eugenia myrtifolia 'Compacta'	Formaldehyde	Kim et al. (2010)	*Myrtaceae*
Euphorbia milii	Triethylamine	Siswanto et al. (2016)	*Euphorbiaceae*
Euphorbia pulcherrima	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Euphorbiaceae*
Eurya emarginata	Toluene	Kim et al. (2011b)	*Theaceae*
Eurya emarginata (Thunb.) Makino	Formaldehyde	Kim et al. (2010)	*Theaceae*
Farfugium japonicum	Toluene	Kim et al. (2011b)	*Asteraceae*
Fatsia japonica	Formaldehyde	Kim et al. (2008)	*Araliaceae*
Fatsia japonica	Formaldehyde	Lim et al. (2009)	*Araliaceae*
Fatsia japonica Decne. et Planch.	Formaldehyde	Kim et al. (2010)	*Araliaceae*

Ficus benghalensis	Xylene Toluene	Kim et al. (2016)	*Moraceae*
Ficus benjamina	Formaldehyde	Kim and Lee (2008)	*Moraceae*
Ficus benjamina	Formaldehyde	Kim et al. (2008)	*Moraceae*
Ficus benjamina	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Moraceae*
Ficus benjamina	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Moraceae*
Ficus benjamina L.	Formaldehyde	Kim et al. (2010)	*Moraceae*
Ficus benjamina	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Moraceae*
Ficus elastica	Benzene toluene m/p-xylene o-xylene	Chun et al. (2010)	*Moraceae*
Ficus elastica	Benzene pentane toluene	Cornejo et al. (1999)	*Moraceae*
Ficus elastica	Formaldehyde	Oyabu et al. (2003a)	*Moraceae*
Ficus elastica	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Moraceae*
Ficus elastica Roxb. ex Horne.	Formaldehyde	Kim et al. (2010)	*Moraceae*
Ficus microcarpa 'Fuyuensis'	Benzene	Liu et al. (2007)	*Moraceae*
Ficus sabre	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Moraceae*
Fittonia argyroneura	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Acanthaceae*
Fittonia verschaffelti	Toluene	Kim et al. (2011b)	*Acanthaceae*
Gardenia jasminoides	Formaldehyde	Kim et al. (2009)	*Rubiaceae*
Gardenia jasminoides Ellis	Formaldehyde	Kim et al. (2010)	*Rubiaceae*
Gasteria gracilis	Formaldehyde	Zhou et al. (2011)	*Asphodelaceae*
Gerbera jamesonii	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Asteraceae*
Guzmania 'Cherry'	Xylene	Wolverton and Wolverton (1993)	*Bromeliaceae*
Guzmania (Species not given)	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Bromeliaceae*
Haemaria discolor Lindl	Formaldehyde	Kim et al. (2010)	*Orchidaceae*

Hedera helix	Formaldehyde	Aydogan and Montoya (2011)	*Araliaceae*
Hedera helix	Formaldehyde	Jin et al. (2013)	*Araliaceae*
Hedera helix	Toluene	Kim et al. (2011b)	*Araliaceae*
Hedera helix	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Araliaceae*
Hedera helix	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Araliaceae*
Hedera helix	Benzene toluene	Yoo et al. (2006)	*Araliaceae*
Hedera helix L.	Formaldehyde	Kim et al. (2010)	*Araliaceae*
Hedera helix	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Araliaceae*
Hemigraphis alternata	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Acanthaceae*
Hibiscus rosa-sinensis	Triethylamine	Siswanto et al. (2016)	*Malvaceae*
Homalomena (Species not given)	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Araceae*
Howea belmoreana	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Arecaceae*
Howea belmoreana Becc.	Formaldehyde	Kim et al. (2010)	*Arecaceae*
Howea forsteriana	Benzene n-hexane	Wood et al. (2002)	*Arecaceae*
Howea forsteriana	Benzene	Orwell et al. (2004)	*Arecaceae*
Hoya carnosa	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Apocynaceae*
Hoya cornosa (L.f.) R.Br	Formaldehyde	Kim et al. (2010)	
Hydrangea macrophylla	Benzene	Liu et al. (2007)	*Hydrangeaceae*
Ilex cornuta	Toluene	Kim et al. (2011b)	*Aquifoliaceae*
Ilex crenata Thunb.	Formaldehyde	Kim et al. (2010)	
Ipomoea batatas	Formaldehyde	Wolverton and Mcdonald (1982)	*Convolvulaceae*
Ixora barbata 'Craib'	Benzene	Treesubsuntorn and Thiravetyan (2012)	*Rubiaceae*
Ixora chinensis	Triethylamine	Siswanto et al. (2016)	
Jasminum polyanthum Franchet	Formaldehyde	Kim et al. (2010)	*Oleaceae*
Jasminum sambac (L.) Aiton	Formaldehyde	Kim et al. (2010)	
Kalanchoë	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Crassulaceae*

Kalanchoë blossfeldiana	Benzene pentane toluene	Cornejo et al. (1999)	*Crassulaceae*
Laurus nobilis L.	Formaldehyde	Kim et al. (2010)	*Lauraceae*
Lavandula (Species not given)	Formaldehyde	Kim et al. (2010)	*Lamiaceae*
Ligustrum japonicum	Toluene	Kim et al. (2011b)	*Ligustrum*
Ligustrum japonicum Thunb.	Formaldehyde	Kim et al. (2010)	*Ligustrum*
Liriope spicata	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Asparagaceae*
Maranta leuconeura	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Marantaceae*
Melissa officinalis	Formaldehyde	Jin et al. (2013)	*Lamiaceae*
Melissa officinalis	Toluene	Kim et al. (2011b)	*Lamiaceae*
Mentha suaveolens 'applemint'	Formaldehyde	Kim et al. (2010)	*Lamiaceae*
Mentha piperita	Toluene	Kim et al. (2011b)	*Lamiaceae*
Mentha piperita 'Citrata'	Toluene	Kim et al. (2011b)	*Lamiaceae*
Mentha suaveolens	Toluene	Kim et al. (2011b)	*Lamiaceae*
Mentha suaveolens 'Variegata'	Toluene	Kim et al. (2011b)	*Lamiaceae*
Microlepia strigosa (Thunb.) Presl.	Formaldehyde	Kim et al. (2010)	*Dennstaedtiaceae*
Monstera acuminaa	Benzene	Treesubsuntorn and Thiravetyan (2012)	*Araceae*
Morus alba	Triethylamine	Siswanto et al. (2016)	*Moraceae*
Musa oriana	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Musaceae*
Nandina domestica Thunb.	Formaldehyde	Kim et al. (2010)	*Berberidaceae*
Neoregelia cv	Xylene	Wolverton and Wolverton (1993)	*Bromeliaceae*
Nephrolepis exaltata	Formaldehyde	Hasegawa et al. (2004)	*Nephrolepidaceae*
Nephrolepis exaltata	Formaldehyde	Oyabu et al. (2003a)	*Nephrolepidaceae*
Nephrolepis exaltata 'Bostoniensis'	Benzene	Liu et al. (2007)	*Nephrolepidaceae*
Nephrolepis exaltata 'Bostoniensis'	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Nephrolepidaceae*
Nephrolepis obliterata	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Nephrolepidaceae*
Nerium indicum	Formaldehyde	Kondo et al. (1995)	*Apocynaceae*
Nerium indicum	Trichloroethylene tetrachloroethylene	Kondo et al. (2005)	*Apocynaceae*

Nicotiana tabacum	1,1,1-Trichloroethane benzene bromodichloromethane carbon tetrachloride chloroform perchloroethylene toluene trichloroethylene vinyl chloride	James et al. (2008)	*Solanaceae*
Nicotiana tabacum	Formaldehyde styrene toluene xylene	Sawada et al. (2007)	*Solanaceae*
Oncidium (Species not given)	Formaldehyde	Kim and Lee (2008)	*Orchidaceae*
Osmunda japonica Thunb.	Formaldehyde	Kim et al. (2010)	*Osmundaceae*
Pachira aquatica Aubl.	Formaldehyde	Kim et al. (2010)	*Malvaceae*
Pachira aquatica	Benzene toluene m/p-xylene o-xylene	Chun et al. (2010)	*Malvaceae*
Pelargonium domesticum	Benzene pentane toluene	Cornejo et al. (1999)	*Geraniaceae*
Pelargonium graveolens	Toluene	Kim et al. (2011b)	*Geraniaceae*
Pelargonium graveolens	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Geraniaceae*
Pelargonium(Species not given)	Formaldehyde	Kim et al. (2010)	*Geraniaceae*
Peperomia clusiifolia	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Piperaceae*
Peperomia clusiifolia Hook	Formaldehyde	Kim et al. (2010)	
Phalaenopsis (Species not given)	Formaldehyde	Kim and Lee (2008)	*Orchidaceae*
Phalaenopsis (Species not given)	Formaldehyde Xylene	Wolverton and Wolverton (1993)	
Philodendron domesticum	Benzene	Treesubsuntorn and Thiravetyan (2012)	*Araceae*
Philodendron domesticum	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Araceae*
Philodendron martianum 'Congo'	Formaldehyde	Zhou et al. (2011)	*Araceae*
Philodendron oxycardium	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Araceae*
Philodendron hederaceum var. oxycardium	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Araceae*

Philodendron selloum	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Araceae*
Philodendron selloum	Formaldehyde	Zhou et al. (2011)	*Araceae*
Philodendron selloum C. Koch	Formaldehyde	Kim et al. (2010)	*Araceae*
Philodendron sodiroi 'Wendimbe'	Formaldehyde	Zhou et al. (2011)	*Araceae*
Philodendron (Species not given)*'Sunlight'*	Toluene	Kim et al. (2011b)	*Araceae*
Phoenix roebelenii	Formaldehyde acetone benzene toluene xylene styrene	Baosheng et al. (2009)	*Arecaceae*
Phoenix roebelenii	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Arecaceae*
Phoenix roebelenii O'Brien	Formaldehyde	Kim et al. (2010)	*Arecaceae*
Pinus densiflora	Toluene	Kim et al. (2011b)	*Pinaceae*
Pittosporum tobira	Toluene	Kim et al. (2011b)	*Pittosporaceae*
Pittosporum tobira Ait	Formaldehyde	Kim et al. (2010)	*Pittosporaceae*
Plectranthus tomentosus	Toluene	Kim et al. (2011b)	*Lamiaceae*
Polypodium formosanum Baker	Formaldehyde	Kim et al. (2010)	*Polypodiaceae*
Polyscias balfouriana Bailey	Formaldehyde	Kim et al. (2010)	*Araliaceae*
Polyscias fruticosa	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Araliaceae*
Polystichum tripteron (Kunze.) Presl.	Formaldehyde	Kim et al. (2010)	*Dryopteridaceae*
Primula sinensis	Benzene pentane toluene	Cornejo et al. (1999)	*Primulaceae*
Pseuderanthemum atropurpureum	Triethylamine	Siswanto et al. (2016)	*Acanthaceae*
Psidium guajava 'Safeda'	Formaldehyde	Kim et al. (2010)	*Myrtaceae*
Pteris dispar kunze.	Formaldehyde	Kim et al. (2010)	*Pteridaceae*
Pteris ensiformis Burm. *'victoriae'*	Formaldehyde	Kim et al. (2010)	*Pteridaceae*
Pteris multifida Poir.	Formaldehyde	Kim et al. (2010)	*Pteridaceae*
Quercus acuta Thunb.	Formaldehyde	Kim et al. (2010)	*Fagaceae*
Quercus glauca Thunb.	Formaldehyde	Kim et al. (2010)	*Fagaceae*
Raphiolepis umbellata Makino	Formaldehyde	Kim et al. (2010)	*Rosaceae*
Rhapis excelsa	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Arecaceae*
Rhapis excelsa Wendl.	Formaldehyde	Kim et al. (2010)	*Arecaceae*
Rhododendron fauriei	Toluene	Kim et al. (2011b)	*Ericaceae*
Rhododendron indicum	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Ericaceae*
Rosmarinus officinalis	Formaldehyde	Kim et al. (2009)	*Lamiaceae*

Rosmarinus officinalis	Toluene	Kim et al. (2011b)	*Lamiaceae*
Rosmarinus officinalis L.	Formaldehyde	Kim et al. (2010)	*Lamiaceae*
Saintpaulia ionantha H. Wendl	Formaldehyde	Kim et al. (2010)	*Gesneriaceae*
Salvia elegans	Toluene	Kim et al. (2011b)	*Lamiaceae*
Sanchezia speciosa	Triethylamine	Siswanto et al. (2016)	*Acanthaceae*
Sansevieria laurentii	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Asparagaceae*
Sansevieria trifasciata	Formaldehyde	Kim and Lee (2008)	*Asparagaceae*
Sansevieria trifasciata	Formaldehyde	Oyabu et al. (2003a)	*Asparagaceae*
Sansevieria trifasciata	Benzene	Treesubsuntorn and Thiravetyan (2012)	*Asparagaceae*
Sansevieria trifasciata	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Asparagaceae*
Sansevieria trifasciata	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Asparagaceae*
Sansevieria trifasciata 'Hahnii'	Formaldehyde	Zhou et al. (2011)	*Asparagaceae*
Sansevieria trifasciata 'Laurentii'	Formaldehyde	Zhou et al. (2011)	*Asparagaceae*
Sansevieria trifasciata Prain	Formaldehyde	Kim et al. (2010)	*Asparagaceae*
Saxifraga stolonifera	Benzene pentane toluene	Cornejo et al. (1999)	*Saxifragaceae*
Schefflera actinophylla	Xylene Toluene	Kim et al. (2016)	*Araliaceae*
Schefflera actinophylla 'Amate'	Benzene	Orwell et al. (2004)	*Araliaceae*
Schefflera arboricola	Formaldehyde	Hasegawa et al. (2003)	*Araliaceae*
Schefflera arboricola	Formaldehyde	Hasegawa et al. (2004)	*Araliaceae*
Schefflera arboricola Hayata *'Hong Kong'*	Formaldehyde	Kim et al. (2010)	*Araliaceae*
Schefflera arboricola	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Araliaceae*
Schefflera elegantissima	Toluene	Kim et al. (2011b)	*Araliaceae*
Schefflera elegantissima	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Araliaceae*
Scindapsus aureus	Benzene	Treesubsuntorn and Thiravetyan (2012)	*Araceae*
Scindapsus aureus	Formaldehyde	Wolverton and Mcdonald (1982)	*Araceae*
Scindapsus aureus	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Araceae*
Scindapsus aureus	Formaldehyde	Zhou et al. (2011)	*Araceae*

Scindapsus pictus 'Argyraeus'	Formaldehyde	Zhou et al. (2011)	*Araceae*
Sedirea japonica	Formaldehyde	Kim and Lee (2008)	*Orchidaceae*
Selaginella tamariscina Spring	Formaldehyde	Kim et al. (2010)	*Selaginellaceae*
Senecio cruentus	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Asteraceae*
Serissa foetida (L.F) Lam.	Formaldehyde	Kim et al. (2010)	*Rubiaceae*
Soleirolia soleirolii	Toluene	Kim et al. (2011b)	*Urticaceae*
Spathiphyllum (No species given) *'Clevelandii'*	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Araceae*
Spathiphyllum (No species given) *'Mauna Loa'*	Benzene formaldehyde trichloroethylene	Wolverton et al. (1989)	*Araceae*
Spathiphyllum (No species given) *'Sweet Chico'*	Toluene, m-xylene	Orwell et al. (2006)	*Araceae*
Spathiphyllum cannifolium	Triethylamine	Siswanto et al. (2016)	*Araceae*
Spathiphyllum clevelandi	Acetone benzaldehyde iso-butyraldehyde n-butyraldehyde crotonaldehyde diethyl ketone, methacrolein methyl ethyl ketone methyl isobutyl ketone methyl isopropyl ketone methyl n-propyl ketone, propionaldehyde iso-valeraldehyde, n-valeraldehyde	Tani and Hewitt (2009)	*Araceae*
Spathiphyllum floribundum 'Clevelandii'	Formaldehyde	Zhou et al. (2011)	*Araceae*
Spathiphyllum floribundum 'Petite'	Benzene	Orwell et al. (2004)	*Araceae*
Spathiphyllum floribundum 'Sensation'	Benzene	Orwell et al. (2004)	*Araceae*
Spathiphyllum (No species given)	Formaldehyde	Kim and Kim (2008)	*Araceae*
Spathiphyllum supreme	Benzene	Liu et al. (2007)	*Araceae*
Spathiphyllum wallisi	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Araceae*
Spathiphyllum wallisii	Benzene toluene	Yoo et al. (2006)	*Araceae*
Spathiphyllum wallisii 'Petite'	Benzene n-hexane	Wood et al. (2002)	*Araceae*
Spathiphyllum wallisii 'Regal'	Formaldehyde	Kim et al. (2010)	*Araceae*
Stauntonia hexaphylla (Thunb.) Dence.	Formaldehyde	Kim et al. (2010)	*Lardizabalaceae*
Stephanotis floribunda	Formaldehyde	Kim and Kim (2008)	*Apocynaceae*
Syngonium podophyllum	Benzene toluene m/p-xylene o-xylene	Chun et al. (2010)	*Araceae*

Syngonium podophyllum	Formaldehyde	Kim and Kim (2008)	*Araceae*
Syngonium podophyllum	Formaldehyde	Wolverton and Mcdonald (1982)	*Araceae*
Syngonium podophyllum	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Araceae*
Syngonium podophyllum	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Araceae*
Syngonium podophyllum	Benzene toluene	Yoo et al. (2006)	*Araceae*
Syngonium podophyllum	Formaldehyde	Zhou et al. (2011)	*Araceae*
Syngonium podophyllum 'White Butterfly'	Benzene	Irga et al. (2013)	*Araceae*
Syngonium podophyllum Schott	Formaldehyde	Kim et al. (2010)	*Araceae*
Thelypteris acuminata (Houtt.) Morton	Formaldehyde	Kim et al. (2010)	*Thelypteridaceae*
Thelypteris decursivepinnata Ching	Formaldehyde	Kim et al. (2010)	*Thelypteridaceae*
Thelypteris esquirolii K. Iwats. *'glabrata'*	Formaldehyde	Kim et al. (2010)	*Thelypteridaceae*
Thelypteris torresiana K. Iwats. *'calvata'*	Formaldehyde	Kim et al. (2010)	*Thelypteridaceae*
Tillandsia cyanea Linden ex C. Koch	Formaldehyde	Kim et al. (2010)	*Bromeliaceae*
Trachelospermum asiaticum Nakai	Formaldehyde	Kim et al. (2010)	*Apocynaceae*
Tradescantia fluminensis	Benzene pentane toluene	Cornejo et al. (1999)	*Commelinaceae*
Tradescantia pallida	Benzene octane α-pinene toluene trichloroethylene	Yang et al. (2009)	*Commelinaceae*
Tulip 'Yellow Present'	Formaldehyde Xylene	Wolverton and Wolverton (1993)	*Liliaceae*
Viburnum awabuki K. Koch	Formaldehyde	Kim et al. (2010)	*Adoxaceae*
Zamia pumila L.	Formaldehyde	Kim et al. (2010)	*Zamiaceae*
Zamioculcas zamiifolia	Benzene ethylbenzene toluene xylene	Sriprapat and Thiravetyan (2013)	*Araceae*
Zamioculcas zamiifolia	Formaldehyde	Zhou et al. (2011)	*Araceae*
Zamioculcas zamiifolia	Formaldehyde	Kim et al. (2010)	*Araceae*

Appendix D 'Can houseplants improve indoor air quality by removing CO_2 and increasing relative humidity?

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Can houseplants improve indoor air quality by removing CO_2 and increasing relative humidity?

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Abstract

High indoor CO_2 concentrations and low relative humidity (RH) create an array of well-documented human health issues. Therefore, assessing houseplants' potential as a low-cost approach to CO_2 removal and increasing RH is important. We investigated how environmental factors such as 'dry' (< 0.20 m^3 of water per m^3 of substrate, m^3 m^{-3}) or 'wet' (> 0.30 m^3 m^{-3}) growing substrates, and indoor light levels ('low' 10 μmol m^{-2} s^{-1}, 'high' 50 μmol m^{-2} s^{-1}, and 'very high' 300 μmol m^{-2} s^{-1}) influence the plants' net CO_2 assimilation ('A') and water vapour loss. Seven common houseplant taxa—representing a variety of leaf types and sizes—were studied for their ability to assimilate CO_2 across a range of indoor light levels. Additionally, to assess the plants' potential contribution to RH increase, the plants' evapo-transpiration (ET) was measured. At typical 'low' indoor light levels, 'A' rates were generally low (< 3.9 mg h^{-1}). Differences between 'dry' and 'wet' plants at typical indoor light levels were negligible in terms of room-level impact. Light compensation points (i.e. the light level where the CO_2 assimilation equals zero) were in the typical indoor light range (1–50 μmol m^{-2} s^{-1}) only for two studied *Spathiphyllum wallisii* cultivars and *Hedera helix*; these plants would thus provide the best CO_2 removal indoors. Additionally, increasing indoor light levels to 300 μmol m^{-2} s^{-1} would, in most species, significantly increase their potential to assimilate CO_2. Species which assimilated the most CO_2 also contributed most to increasing RH.

Keywords *Dracaena* · Drought · *Hedera* · Indoor light · Indoor air quality · *Spathiphyllum*

Abbreviations

RH	Relative humidity (%)
DLI	Daily light integral (mol m^{-2} d^{-1})
SMC	Substrate moisture content (m^3 m^{-3})
LCP	Light compensation point (μmol m^{-2} s^{-1})
ET	Evapo-transpiration (g)
PPM	Uptake or emission of CO_2 by potted-plant microcosm
LA	Leaf area (m^2)
ETLA	Evapo-transpiration per unit leaf area (g cm^{-2})

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Introduction

Indoor CO_2 concentrations are primarily dependent on the occupancy level and outdoor air supply rate (Zhang et al. 2017). Humans produce and exhale CO_2; therefore, a greater occupancy coupled with lower ventilation rates—intended to reduce energy consumption—gives rise to higher and often harmful CO_2 concentrations indoors (Satish et al. 2012). Additionally, even when ventilation by ambient air is employed, the problems may be exacerbated in the future: ambient CO_2 concentrations increased by 40% over the last century, to 400 ppm—with a rise to 670 ppm expected by 2100 (Hersoug et al. 2012).

The American Society of Heating, Refrigerating and Air-conditioning Engineers (ASHRAE) recommends a maximum

indoor CO_2 concentration of 1000 ppm (Torpy et al. 2017). Concentrations indoors (e.g. in fully occupied offices or meeting rooms) often reach 2000 to 2500 ppm but can rise as high as 5000 ppm (Zhang et al. 2017). Although discrepancies in the maximum safe exposure concentration are commonplace in literature, prior research suggests typical indoor CO_2 concentrations will continue to present unwanted health issues (Zhang et al. 2017). These include mucus membrane symptoms (i.e. sore/dry throat, dry eyes and sneezing) and respiratory problems (i.e. tight chest, wheezing/coughing and shortness of breath) (Seppanen et al. 1999; Erdmann and Apte 2004). Elevated CO_2 can also reduce the cognitive performance of students in schools, while long-term, regular exposure has been linked to increased absenteeism, weight gain, and obesity (Hersoug et al. 2012; Satish et al. 2012; Gaihre et al. 2014; Nieuwenhuis et al. 2014; Vehvilainen et al. 2016; Zhang et al. 2017).

An additional challenge in indoor environments is low relative humidity (RH). An RH below 30% has been shown to cause eye irritation and skin dryness, with an RH below 10% causing dryness of the nasal mucus membrane. Low RH can also increase the likelihood of influenza transmission, enhance indoor ozone concentration, and produce static electricity (Arundel et al. 1986; Berglund 1998; Sunwoo et al. 2006; Lowen et al. 2007; Abusharha and Pearce 2013; Zhang and Yoshino 2010). However, high RH (> 60%) too can cause issues by encouraging fungal/mould growth and contributing to the deterioration of building materials (Berglund 1998; Bin 2002; Zhang and Yoshino 2010; Frankel et al. 2012). The majority of adverse health effects concerning RH can be avoided by maintaining indoor levels between 40 and 60% (Arundel et al. 1986).

Various techniques are used in the built environment to control and regulate CO_2 levels. They include highly engineered approaches to ventilation (Hesaraki et al. 2015; Mateus and da Graca 2017) as well as low-tech approaches which can include the use of plants (Raji et al. 2015; Charoenkit and Yiemwattana 2016). A number of studies investigate a houseplants' potential to sequester CO_2 from indoor environments (Oh et al. 2011; Pennisi and van Iersel 2012; Torpy et al. 2014). Studies vary in scale and focus—from those focusing on individual plants in experimental chambers to room scale studies in situ.

A range of studies investigated houseplants' ability to sequester CO_2 in home, school, and office environments. Various combinations of houseplants were found to generally reduce room CO_2 concentrations and increase RH; however, studies rarely specify exact plant numbers and plant types. Plant species commonly used include *Dracaena deremensis*, *Dracaena marginata*, *Ficus benjamina*, *Hedera helix*, and *Spathiphyllum clevelandii* (Raza et al. 1991; Lohr and PearsonMims 1996; Jeong et al. 2008; Lim et al. 2009; Oh et al. 2011; Pegas et al. 2012).

Light levels and substrate moisture are the key factors influencing gas exchange between the plant and the environment, with 'low' light and 'dry' substrate both reducing houseplants' ability to sequester CO_2 and contribute to RH increases indoors via transpiration (Lawlor and Cornic 2002; Flexas et al. 2006; Torpy et al. 2017). In indoor environments, light levels are typically at least 100-fold lower compared to outdoors (on a clear summer day for example) and are maintained in the range of approximately 1–50 μmol m^{-2} s^{-1} (Thimijan and Heins 1983; Boyce and Raynham 2009; Lai et al. 2009; Hawkins 2011). Research suggests however that having higher indoor light levels (approximately 30–50 μmol m^{-2} s^{-1}) would greatly increase occupant comfort (Lai et al. 2009; Huang et al. 2012). As previously proposed, indoor light is the most limiting factor for CO_2 assimilation (Pennisi and van Iersel 2012).

The positive contribution of plants to the reduction of CO_2 levels and RH increases indoors are based on the premise that plants function optimally and are sequestering CO_2/releasing water vapour at their maximum capacity. However, the main challenges for maintaining plant function in the indoor environment are 'low' indoor light levels and issues arising from plants' (mis) management, most frequently plants' being under or over watered without the correct nutrients (RHS 2017). A few studies addressed these questions in part by investigating a wide range of light levels and their effect on CO_2 assimilation (Pennisi and van Iersel 2012; Torpy et al. 2014). However, no study to our knowledge investigated the effect of differing substrate moisture content (SMC)—namely, investigating the effect of 'wet' (> 0.30 m^3 m^{-3}) and 'dry' (< 0.20 m^3 m^{-3}) SMC conditions. Additionally, previous studies have not specifically focused on plants' cultivar-level differences; this may be of interest as for many houseplant species, there is a range of cultivars available, which may potentially offer augmented service compared to straight species if they are larger in size or more physiologically active.

Pennisi and van Iersel (2012) investigated the CO_2 assimilation of 17 houseplant species in both a simulated controlled environment utilising light levels of 10, 20, and 30 μmol m^{-2} s^{-1} and a public office building in Atlanta (USA). In the public office, the amount of CO_2 assimilated by plants varied depending on plant size. In the controlled environment, most species exhibited positive carbon assimilation over a 10-week period. The study found that in both environments, larger, woody plants (such as *Ficus benjamina*) assimilated more CO_2 than herbaceous species.

Torpy et al. (2014) investigated the CO_2 assimilation of eight common indoor plant species by producing light response curves and light compensation points (LCPs) using an infrared gas analyser. The results indicated that at least some CO_2 sequestration could be expected from the studied species under current indoor lighting systems and plants could be effectively utilised in the built environment to sequester CO_2 given a moderate increase in the targeted lighting levels.

Our research aims to improve the understanding of which taxa (i.e. plant species and cultivars) as well as which light and substrate moisture conditions are best placed to regulate indoor CO_2 and RH. Specifically, the aims of the study were to determine:

1. The impact of drying substrate on CO_2 removal capacity by different taxa
2. The impact of light levels on net CO_2 assimilation of taxa (i.e. to test the potential to improve the performance by supplementing indoor light levels)
3. The evapo-transpiration (ET) rates of each taxon and their potential contribution to increasing indoor RH.

Material and methods

Plant material

Five common houseplant species, including two cultivars, were selected for the study to represent a range of leaf types (succulent and herbaceous), plant sizes, and plant metabolisms often found in indoor environments (Table 1). Selected plants were 2 years old at the time of purchase in July 2016 from the RHS plant centre (Wisley, Surrey, UK), ranging between 10 and 60 cm in height, depending on the taxon. Within the species, plant height and stature were uniform (data not shown). Plants were maintained in Sylvamix growing medium (6:2:2 sylvafibre:growbark pine:coir; Melcourt, Tetbury, Gloucestershire, UK) in 3-L containers, with a slow release fertiliser feed (Osmocote, Marysville, OH, USA). For 3 months prior to experimentation, plants were kept at ambient temperatures (17–22 °C) and 'low' light levels (10 μmol m^{-2} s^{-1}) in an indoor office environment within the Crops Laboratory in the Glasshouse Complex of the School of Agriculture, Policy and Development, at the University of Reading (UK).

Net leaf-level CO_2 assimilation at 'low' and 'high' indoor light levels under 'dry' and 'wet' conditions

Experiments were conducted on five plants per taxon. Measurements of the net CO_2 assimilation rate (μmol m^{-2} s^{-1}) were made using a LCPro infrared gas analyser (ADC Bioscientific, Hoddesdon, Hertfordshire, UK) on three young, fully expanded leaves per plant (with consistent leaf selection, i.e. third fully expanded leaf from the plant tip (Fig. 1)) under office conditions (16.6–21.8 °C, RH > 35%) at 'low' and 'high' indoor light levels (Hawkins 2011; Huang et al. 2012). 'Low' 10 μmol m^{-2} s^{-1} lighting was achieved in the usual lighting conditions of the room (eight fluorescent lights, Osram, Munich, Germany lighting a floor area of 20 m^2). To achieve 'high' 50 μmol m^{-2} s^{-1} during measurements, the photosynthetic photon flux density (i.e. light level, μmol m^{-2} s^{-1}) was supplemented at the leaf by an external halogen source (50 W, 12 V). Each light increment was administered for 7 min and the net CO_2 assimilation rate recorded at the end of the seven-minute period.

Substrate moisture content (SMC) based on volume of water per volume of substrate was measured daily for each plant, in two locations per container using a SM300 capacitance-type probe connected to a HH2 Moisture Meter (Delta-T Devices, Cambridge, Cambridgeshire, UK; 0–100% range and an accuracy of ± 2.5%). At the start of the experiment, substrate moisture was at the container capacity (SMC > 30%, 0.3 m^3 m^{-3}) and plants were thus considered optimally watered (Vaz Monteiro et al. 2016). Measurements were also made on 'dry' plants (SMC < 20%, 0.2 m^3 m^{-3}). Measurements were made over approximately 1 month.

Calculation of the respiration of the potted-plant microcosm

To ensure that CO_2 removal by the aboveground parts of the plant (i.e. leaves and stem) was not cancelled out by respiration of the potted-plant microcosm (PPM) (i.e. substrate and non-photosynthetic plant parts), the PPM was investigated for CO_2 contributions at both 'high' and 'low' light and under 'wet' and 'dry' SMC conditions ($n = 3$). The PPM respiration values were then subtracted from all the leaf CO_2 assimilation values made, to obtain the overall contribution of the plant and substrate.

Measurements of the PPM respiration were made utilising a 150 L (45 × 45 × 75 cm, 0.15 m^3) Perspex chamber (The plastic people, Leeds, West Yorkshire, UK) sealed with Swagelok's (Swagelok, Bristol, South Gloucestershire, UK). Enclosed inside the Perspex chamber was a HOBO MX1102 CO_2 logger (Onset Computer Corporation, Bourne, MA, U.S.A), a 12 V DC brushless fan (RS Components, Corby, Northants, UK), and a calibrated (20–90% RH, 0–40 °C) Tinytag RH/temperature logger (Gemini data loggers, Chichester, West Sussex, UK). The external RH/temperature surrounding the chamber was also monitored with another, identical Tinytag logger. Inside the chamber 'low' light levels were achieved as described in "Net leaf-level CO2 assimilation at 'low' and 'high' indoor light levels under 'dry' and 'wet' conditions" section; 'high' levels were generated by two LED lights (V-TAC Europe Ltd., Sofia, Bulgaria) and measured with a calibrated light sensor (Skye instruments, Llandrindod Wells, Wales, UK). Bare substrate was prepared for the experiment as explained in "Net leaf-level CO2 assimilation at 'low' and 'high' indoor light levels under 'dry' and 'wet' conditions" section. Experiments were undertaken for 2 h, with the chamber analysed for leakage prior, during and after experimentation; leakage was found to be < 2% of the

Table 1 Characteristics of the houseplant taxa (i.e. plant species and cultivars) chosen for experiments. Leaf area ($n = 2$) and plant height ($n = 5$) are means ± SEM. Species' Latin name is given in italic and cultivar, where applicable, follows

Species/cultivars	Family	Metabolism	Leaf area (cm^2)	Plant height (cm)
Dracaena fragrans 'Lemon Lime'	*Asparagaceae*	C3	1742 ± 91	51 ± 1
Dracaena fragrans 'Golden Coast'	*Asparagaceae*	C3	1438 ± 10	60 ± 1
Guzmania 'Indian Night'	*Bromeliaceae*	C3/CAM	1230 ± 6	32 ± 1
Hedera helix	*Araliaceae*	C3	1509 ± 243	9 ± 0
Spathiphyllum wallisii 'Bellini'	*Araceae*	C3	1766 ± 189	35 ± 1
Spathiphyllum wallisii 'Verdi'	*Araceae*	C3	5451 ± 1104	36 ± 1
Zamioculcas zamiifolia	*Araceae*	CAM	1388 ± 88	57 ± 1

starting concentration over a 2-h test period. Measurements were made over approximately 1 week.

Data obtained in "Net leaf-level CO2 assimilation at 'low' and 'high' indoor light levels under 'dry' and 'wet' conditions" section was normalised by leaf area by multiplying CO_2 assimilation (mg m^{-2} h^{-1}) with leaf area (m^2), providing CO_2 assimilation in mg h^{-1} $plant^{-1}$ for each taxon. Data were also corrected for PPM respiration and leakage by calculation of an average conversion value (mg h^{-1}) for both 'wet' and 'dry' SMC conditions.

Generating light response curves

To generate light response curves, measurements of the net photosynthetic rate (μmol m^{-2} s^{-1}) were made as explained in "Net leaf-level CO2 assimilation at 'low' and 'high' indoor light levels under 'dry' and 'wet' conditions" section on four plants per taxon and two leaves per plant. Environmental conditions within the leaf cuvette were temperature controlled at 25 °C, ambient CO_2 concentration (~ 400–450 ppm) and an ambient RH of 35–45%. Plants were prepared for the experiment as explained in "Net leaf-level CO2 assimilation at 'low' and 'high' indoor light levels under 'dry' and 'wet' conditions" section, achieving a SMC > 0.30 m^3 m^{-3} and were considered optimally watered on the commencement of each experiment (Vaz Monteiro et al. 2016). SMC was maintained at this level for the duration of the experiment.

To generate the light response curve, the light was supplemented in the following set increments: 0, 50, 300, and 1200 μmol m^{-2} s^{-1} as described in "Net leaf-level CO2 assimilation at 'low' and 'high' indoor light levels under 'dry' and 'wet' conditions" section. An increment of 0 μmol m^{-2} s^{-1} was chosen to investigate each species CO_2 assimilation in the dark; 50 μmol m^{-2} s^{-1} the highest indoor light level; 300 μmol m^{-2} s^{-1} was chosen to represent the highest feasible light level which could be engineered (with supplementary artificial lighting) in an indoor environment; 1200 μmol m^{-2} s^{-1} (a sunny day in a UK climate) was chosen to present information on a plant's maximal capacity for net CO_2 assimilation. Measurements were made over approximately 1 week.

The light response curves were based on an equation proposed by Prioul and Chartier (1977) and were produced using the model by Lobo et al. (2013). Light compensation points, LCPs (which represent the light level where the CO_2 assimilation is equal to zero) (Torpy et al. 2014), were calculated with the same model (Lobo et al. 2013) for all taxa apart from *Guzmania* 'Indian night', which was omitted due to very low assimilation rates and therefore, unreproducible results.

Plants' water use/evapo-transpiration (ET) experiments

Water use/ET of the plant taxa were inferred by consecutive plant/pot weight measurements using a precision balance (CBK 32, Adam Equipment, Milton Keynes, Buckinghamshire, UK) under indoor office conditions (RH > 35% and at 'low' light levels, 10 μmol m^{-2} s^{-1}. Plants were prepared for the experiment as explained in "Net leaf-level CO2 assimilation at 'low' and 'high' indoor light levels under 'dry' and 'wet' conditions" section, starting the experiment with SMC at full water-holding capacity and were not watered for the duration of the experiment. Measurements were made at 0 h and then every 24 h over a 3-week period on a whole 'plant – substrate system' (i.e. potted plant, with uncovered substrate) enabling the calculation of the water loss at each time-point. We were interested in total potential RH contribution of the plant along with substrate, mimicking a real-life scenario of an indoor plant. Each plant was removed from the experiment when its SMC dropped < 20% (0.2 m^3 m^{-3}). Destructive measurements of LA were made using a LA meter (Delta-T Devices, Cambridge, Cambridgeshire, UK) on two plants per taxon, at the end of the experiment. While we appreciate that measuring the leaf area at the end of the experiment may lead to under/over-estimating assimilation measured earlier in the experiment, we were limited by the number of experimental plants we could destructively harvest. Given that this approach was applied to all taxa that the leaf areas were assessed within 2 months

of the assimilation experiments and that plants did not increase in size significantly over this period (as evidenced by height measurements which we made at the start and the end of the experiment), we believe that the risk of the error is small and evenly spread. SMC was measured daily as explained in "Net leaf-level CO2 assimilation at 'low' and 'high' indoor light levels under 'dry' and 'wet' conditions" section. Water use/ET per unit leaf area (ETLA, expressed in g cm^{-2}) was calculated by dividing the ET (i.e. water loss) from a plant in a 24-h period by the mean leaf area.

Statistical analysis

Experimental data (gas exchange parameters and water loss/ET) were analysed using GENSTAT (16th Edition, VSN International, Hemel Hempstead, Hertfordshire, UK). An analysis of variance (ANOVA) was performed to compare means for each measured parameter between different taxa and/or over time. Values were presented as means with associated standard errors of the mean (SEM) and Tukey's 95% confidence intervals for multiple comparisons. Data on plants' water loss were log-transformed and Tukey's 95% confidence intervals were used to compare between taxa in the text ("Plants' water use/evapo-transpiration experiments" section).

Results

Net leaf-level CO_2 assimilation at 'low' and 'high' indoor light levels under 'dry' and 'wet' conditions

At 'low' indoor light, 'dry' *Spathiphyllum wallisii* 'Verdi' was statistically significantly respiring the most (− 87.6 mg h^{-1}, $p < 0.001$) and was therefore the only taxon to measure significant differences between 'dry' and 'wet' substrate. In 'dry' substrate, statistically significant differences in CO_2 assimilation were measured between the cultivars of *Spathiphyllum wallisii* 'Bellini' and 'Verdi' (− 19.6 and − 60.7 mg h^{-1}, respectively; $p < 0.001$). In 'wet' substrate, there were no significant differences in CO_2 between any studied taxa (Table 2).

At 'high' indoor light, only *Spathiphyllum wallisii* 'Verdi' measured statistically significant differences between 'dry' and 'wet' substrate (− 60.7 and 60.0 mg h^{-1}, respectively; $p < 0.001$; Table 2). No statistically significant differences in CO_2 assimilation were measured between cultivars under the same SMC conditions; significant differences were measured with *Spathiphyllum wallisii cvs* 'Bellini' and 'Verdi' between 'dry' (− 19.6 and − 60.7 mg h^{-1}, respectively) and 'wet' (11.7 and 60.0 mg h^{-1}, respectively) SMC conditions ($p < 0.001$, Table 2).

Generating light response curves and light compensation points

Light compensation points (LCPs), which represent the light level where the CO_2 assimilation is equal to zero, were calculated for each species (Table 3). Of the studied species, *Spathiphyllum wallisii* 'Verdi' and *Hedera helix* had the lowest LCPs of 20 and 31 μmol m^{-2} s^{-1} respectively. The highest LCP was recorded for *Dracaena fragrans* 'Golden Coast' (96 μmol m^{-2} s^{-1}), with both *Dracaena fragrans* 'Lemon Lime' and *Zamioculcas zamiifolia* also having LCP values outside of the light level typically experienced in indoor environments (93 and 65 μmol m^{-2} s^{-1} respectively, Table 3).

At 0 μmol m^{-2} s^{-1}, *Hedera helix* was statistically significantly respiring the most (− 1.2 μmol m^{-2} s^{-1}, $p < 0.001$; Fig. 2), no significant differences were measured in net assimilation between other studied taxa.

At 300 μmol m^{-2} s^{-1}, all taxa were assimilating CO_2. Net assimilation was highest in *Hedera helix* (7.7 μmol m^{-2} s^{-1}) and was statistically significantly different to all other taxa ($p < 0.001$). *Spathiphyllum wallisii* 'Bellini' and *S. wallisii* 'Verdi' (2.4 and 2.4 μmol m^{-2} s^{-1} respectively) measured a net assimilation that was statistically significantly higher than three other studied taxa (*Dracaena fragrans* 'Lemon Lime', *Dracaena fragrans* 'Golden Coast', and *Guzmania* 'Indian Night', $p < 0.001$; Fig. 2). At this highest indoor photosynthetic photon flux density, there were no cultivar-level differences within the same species in net assimilation.

At 1200 μmol m^{-2} s^{-1}, all taxa were assimilating CO_2. Net assimilation was highest in *Hedera helix* (10.7 μmol m^{-2} s^{-1}) and was statistically significantly higher than all other taxa ($p < 0.001$). *Spathiphyllum wallisii* 'Bellini' (2.7 μmol m^{-2} s^{-1}) measured a net assimilation that was statistically significantly higher than three other studied taxa (*Dracaena fragrans* 'Lemon Lime', *Dracaena fragrans* 'Golden Coast', and *Guzmania* 'Indian Night', $p < 0.001$; Fig. 2). Again, no net assimilation was statistically significantly different between cultivars of the same species.

Plants' water use/evapo-transpiration experiments

In terms of ET per plant per day, when well-watered, the ET was statistically significantly higher for *Hedera helix* (70.5 g) and *Spathiphyllum wallisii* 'Verdi' (71.0 g) compared to all the other taxa ($p < 0.001$). ET per plant was also statistically significantly different between the taxa *Guzmania* 'Indian Night' (28.0 g) and *Dracaena fragrans* 'Lemon Lime' (44.3 g, $p < 0.001$); ET per plant at 24 h was statistically significantly different between *Spathiphyllum wallisii* cultivars ($p < 0.001$; Fig. 3a).

In terms of ET per leaf area per day, when well-watered, the ET was statistically significantly higher for *Hedera helix* (0.047 gcm^{-2}) in comparison to other taxa ($p < 0.001$). ET

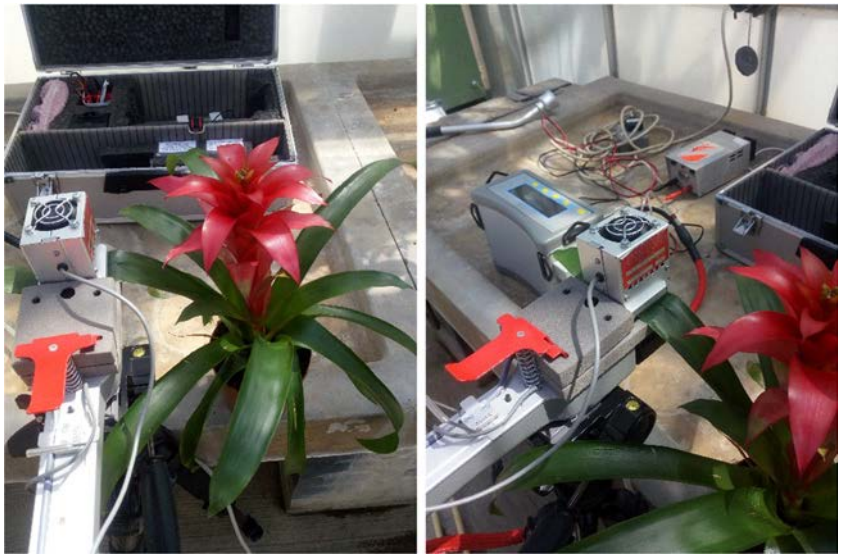

Fig. 1 Images of the experimental setup for leaf CO_2 assimilation measurements, equipment pictured includes infrared gas analyser, leaf cuvette, and external halogen light source

per leaf area was statistically significantly lower for *Spathiphyllum wallisii* 'Verdi' (0.013 g cm^{-2}), in comparison to the other taxa tested ($p < 0.001$); no ET per leaf area was statistically significantly different between any other taxa. The ET per leaf area was statistically significantly different between one pair of cultivars: *Spathiphyllum wallisii* 'Bellini' and *Spathiphyllum wallisii* 'Verdi' (0.02 g cm^{-2} and 0.013 g cm^{-2}, respectively; $p < 0.001$; Fig. 3b).

At the time when SMC decreased to 20%, ET reduction ranged between 7% (*Spathiphyllum wallisii* 'Verdi') and 63% (*Guzmania* 'Indian Night') (data not shown). The time taken for the SMC to decrease to < 20% ranged between 10 days (*Dracaena fragrans* 'Golden Coast' and *Spathiphyllum*) and 23 days (*Zamioculcas zamiifolia*) across studied taxa.

Discussion

The current work presents the first insight into leaf-level CO_2 assimilation—from plants in both 'dry' and 'wet' substrate—and potential RH increases for a range of common houseplant taxa (i.e. species and cultivars), differing in structure and physiological function.

In this study, we demonstrate that little potential is offered by the studied houseplants alone to reduce CO_2 concentrations in 'low' light indoor environments—with only three taxa's light compensation points falling within the typical indoor light level range (0–50 μmol m^{-2} s^{-1}; Table 3). However, our findings demonstrate that although respiration was generally occurring in houseplants grown in 'dry' substrate, the net CO_2 exchange recorded was extremely low and thus likely to have little or no negative impact on the CO_2 levels at a room scale. Our results suggest that increasing light levels to a technically feasible 300 μmol m^{-2} s^{-1} (e.g. through use of supplementary lighting) would provide a significant increase in CO_2 assimilation in most of the studied taxa. The study also indicates that the best performing taxa for CO_2 assimilation will also contribute the most to raising RH indoors.

From the results of this study, we estimated the mass (in grams) of CO_2 removed per hour, per plant, and per m^2 of each taxon. In home and office environments, each person contributes 30 g (CO_2)/h and 36 g (CO_2)/h, respectively (Persily and de Jonge 2017) and these different values are consequences of the level of individual's activity in various environments. Using both these values, we calculated the number of plants required to remove 10% of a single person's CO_2 contribution at the 'very high' (300 μmol m^{-2} s^{-1}) indoor light level (Supplementary Table 1). The plant numbers range from 15 (for more active plants like *Hedera* and *Spathiphyllum*) to > 100 for physiologically less active plants, highlighting how correct plant choice can result in a different air quality outcome. Of the taxa we investigated, *Guzmania*, *Dracaena*, and *Zamioculcas* would be better placed to provide services other than CO_2 reduction (e.g. pollutant sequestration (Yang et al. 2009; Kim et al. 2010). *Hedera* and *Spathyphyllum* would have more effect on room-level CO_2 exchange, and in numbers which can be realistically installed in small living walls. Estimates of the number of plants required to remove the CO_2 generated by human contributions were also made by Pennisi and van Iersel (2012) and Torpy et al. (2014). However, widely different estimates of the CO_2 generated per person were used by each study, making direct comparisons difficult.

In typical indoor environments with 'low' light levels, only one taxon, in 'wet' substrate conditions, was assimilating CO_2 (*Spathiphyllum wallisii* 'Verdi') and would contribute to CO_2 concentration reduction (3.9 mg h^{-1}, respectively; Table 2). Additionally, only three taxa were found to possess light compensation points that fall within the range of typical indoor light levels (i.e. *Hedera helix and Spathiphyllum wallisii*

Table 2 Net leaf-level CO_2 assimilation of each species at 'low' and 'high' indoor light (< 10 and 50 μmol m^{-2} s^{-1}) in 'wet' (> 0.30 m^3 m^{-3}) and 'dry' (< 0.20 m^3 m^{-3}) conditions. Data are a mean of five plants of each species, three young, fully expanded leaves per plant ± SEM (n = 15). Data are adjusted to account for PPM respiration and chamber leakage and is normalised by leaf area (Table 1). Different letters next to means correspond to statistically significant differences between means based on Tukey's 95% confidence intervals. (−) values signify respiration (i.e. the release of CO_2)

Taxa	Net CO_2 assimilation per plant (mg h^{-1})	
	'Wet' (> 0.30 m^3 m^{-3})	'Dry' (< 0.20 m^3 m^{-3})
'Low' light (< 10 μmol m^{-2} s^{-1})		
Dracaena fragrans 'Lemon Lime'	$-17.4^{b} \pm 2.1$	$-35.7^{b} \pm 4.9$
Dracaena fragrans 'Golden Coast'	$-28.4^{b} \pm 3.0$	$-25.3^{b} \pm 2.2$
Guzmania 'Indian Night'	$-14.3^{b} \pm 1.1$	$-23.8^{b} \pm 1.0$
Hedera helix	$-9.5^{b} \pm 2.2$	$-27.3^{b} \pm 1.0$
Spathiphyllum wallisii 'Bellini'	$-14.8^{b} \pm 4.5$	$-22.7^{b} \pm 2.5$
Spathiphyllum wallisii 'Verdi'	$3.9^{b} \pm 5.2$	$-87.6^{a} \pm 33.3$
Zamioculcas zamiifolia	$-17.5^{b} \pm 2.0$	$-23.9^{b} \pm 1.8$
'High' light (50 μmol m^{-2} s^{-1})		
Dracaena fragrans 'Lemon Lime'	$-5.5^{abc} \pm 6.0$	$-41.97^{ab} \pm 11.3$
Dracaena fragrans 'Golden Coast'	$-21.8^{ab} \pm 4.7$	$-24.0^{ab} \pm 4.7$
Guzmania 'Indian Night'	$-11.5^{ab} \pm 6.7$	$-19.6^{ab} \pm 1.3$
Hedera helix	$-6.6^{abc} \pm 8.2$	$9.4^{bc} \pm 4.7$
Spathiphyllum wallisii 'Bellini'	$11.7^{bc} \pm 9.3$	$-19.6^{ab} \pm 3.8$
Spathiphyllum wallisii 'Verdi'	$60.0^{c} \pm 31.3$	$-60.7^{a} \pm 24.5$
Zamioculcas zamiifolia	$-12.2^{ab} \pm 2.8$	$-20.9^{ab} \pm 0.8$

'Verdi' and 'Bellini'). Both *Hedera helix and Spathiphyllum wallisii* would require an unrealistic number of plants to see any significant CO_2 concentration reduction (data not shown); at typical 'low' indoor light levels, the study indicates that a plants' potential benefits psychologically or in productivity terms (Thomsen et al. 2011; Raanaas et al. 2011; Nieuwenhuis et al. 2014) would be more important than their contribution to indoor CO_2 removal. Furthermore, as suggested in Torpy et al. (2014), plants should not be expected to completely replace ventilation systems, but to act as a supplement in reducing the energy load required.

In typical 'low' light indoor environments, when grown in 'dry' substrate, all studied taxa were respiring. The results also indicated that in the range of typically observed indoor light levels, six of the studied species (*Dracaena fragrans* cvs 'Lemon Lime' and 'Golden Coast', *Guzmania* 'Indian Night', *Hedera helix, Spathiphyllum wallisii* 'Bellini' and *Zamioculcas zamiifolia*) were respiring in both 'dry' and 'wet' SMC conditions (Table 2).The (mis) management and under watering of houseplants is anecdotally a common problem; therefore, determining if a 'dry' houseplant is releasing significant amounts of CO_2 into an indoor environment and detrimentally impacting health is important; our results, however, suggest this is not the case. In 'dry' SMC conditions, in typical office light, *Spathiphyllum wallisii* 'Verdi' was releasing the most CO_2 into the indoor environment out of all studied taxa at 0.0876 g h^{-1}. In comparison, a single person, in an office environment would release 36 g/hour into the indoor environment (Persily and de Jonge 2017). This confirms that in typical office light conditions—even for plants growing in drying substrate—the contribution of plants to room-level CO_2 is negligible.

At a 'high' indoor light level (50 μmol m^{-2} s^{-1}), a greater net CO_2 assimilation was generally measured for all taxa, but no statistically significant differences were found between cultivars of the same species in 'dry' or 'wet' conditions. Although measurements were only made under 'wet' SMC conditions, this trend for the lack of cultivar differences continued at higher light levels of 300 and 1200 μmol m^{-2} s^{-1} suggesting that cultivar level differences were not pronounced in this study.

Our study suggests that for most studied taxa, light saturation occurs at around 300 μmol m^{-2} s^{-1} and further increases beyond this show little difference in assimilation terms (Fig. 2). As discussed in Torpy et al. (2014), targeted indoor lighting could be used to maximise a houseplants CO_2 assimilation potential. Extensive research has been undertaken into various light systems for plant cultivation and development on indoor living walls but not specifically with potted houseplants or concerning CO_2 assimilation (Yeh and Chung 2009; Egea et al. 2014). Our findings support the notion that increased light levels maximise plant gas exchange and we suggest future research should

Table 3 Light compensation points (LCPs) are means of eight leaves per species ± SEM for each of the studied species

Taxa	LCP (μmol m^{-2} s^{-1})
Dracaena fragrans 'Lemon Lime'	92.9 ± 7.1
Dracaena fragrans 'Golden Coast'	95.6 ± 13.2
Guzmania 'Indian Night'	N. A
Hedera helix	30.9 ± 3.9
Spathiphyllum wallisii 'Bellini'	31.9 ± 11.7
Spathiphyllum wallisii 'Verdi'	20.1 ± 9.8
Zamioculcas zamiifolia	64.7 ± 15.7

investigate the suitability of testing targeted lighting installations in indoor environments. Light compensation points calculated in our study are generally higher, but comparable with other indoor species previously tested (Burton et al. 2007; Pennisi and van Iersel 2012; Torpy et al. 2014; Torpy et al. 2017; Tan et al. 2017).

Earlier attempts at estimating the CO_2 removal of houseplants (Pennisi and van Iersel 2012) did not take into account ambient CO_2 concentrations or consider the effects of substrate moisture on CO_2 assimilation. A more robust study by Torpy et al. (2014) investigated several factors which could influence assimilation including different acclimatisation treatments, the respiration of the 'potted-plant microcosm', but again did not consider impact of substrate moisture conditions. Other studies did not specify the exact number or type of houseplant (Lim et al. 2009; Pegas et al. 2012) which contributed to any CO_2 concentration reduction or, only considered a single light level (Oh et al. 2011).

The results from the ET experiment indicate that the best performing species in CO_2 assimilation terms (*Hedera helix* and *Spathiphyllum wallisii* 'Verdi') both have the highest ET rates per plant. However, the comparative water use per area results show *Spathiphyllum wallisii* 'Verdi' having the lowest ET *per leaf area*; this species is, therefore, inherently more water use efficient and only uses more water *per plant* due to its large size. We found a difference between the *Spathiphyllum wallisii* cultivar pair in terms of water use per plant and per area, with no difference per plant or per area measured for the *Dracaena fragrans* pair. This confirms that our hypothesis that inherent physiological differences can be measured in water use terms down to a cultivar level. The results also suggest that certain species (i.e. *Spathiphyllum wallisii* 'Verdi') do not restrict their water loss under water stress conditions (SMC < 20%). *Spathiphyllum wallisii* 'Verdi' would therefore, in a drying substrate, continue to contribute the most to RH increases. To achieve the optimal function for the studied taxa, which would then support biggest improvements in IAQ—based on results from "Plants' water use/evapo-transpiration experiments" section and authors' experience—we suggest a watering regime of 200 ml per week for all studied species other than *Spathiphyllum wallisii* 'Verdi' and *Hedera helix*, where 250 ml is recommended twice a week. We also suggest that future studies should evaluate the CO_2 assimilation ability of other more physiologically active, vigorous species (i.e. *Osmunda japonica*, *Selaginella tamariscina*, and *Hemigraphis alternata*), which also performed well in pollutant sequestration experiments (Yang et al. 2009; Kim et al. 2010) under 'high' indoor light levels (300 μmol m^{-2} s^{-1}).

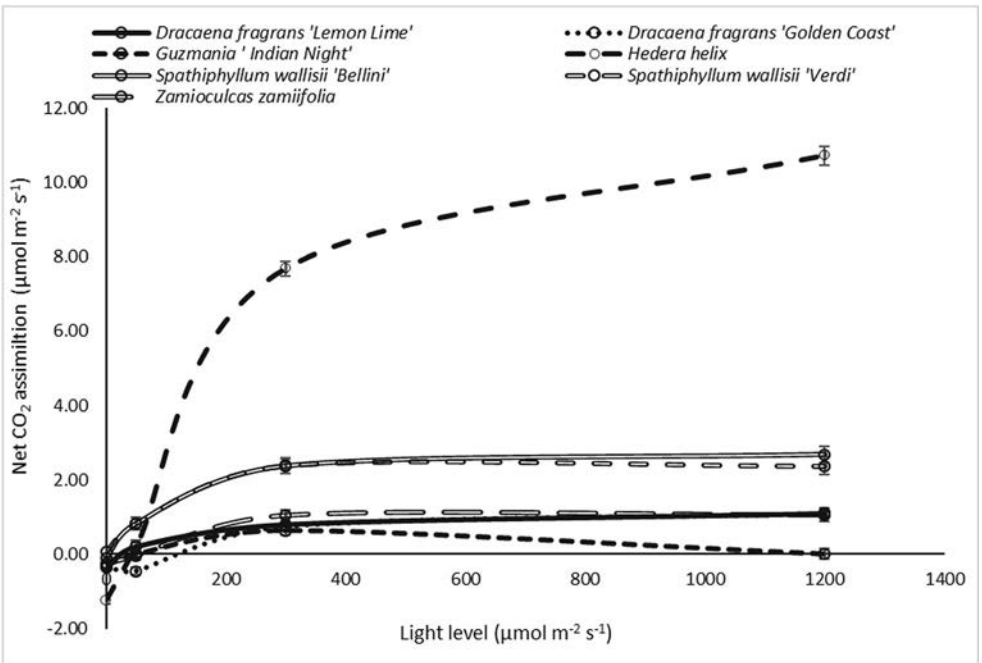

Fig. 2 Net CO_2 assimilation across three light levels (0, 50, 300, 1200 μmol m^{-2} s^{-1}); data are a mean of four containers of each species and two young fully expanded leaves per plant ($n = 8$). Tukey's 95% confidence intervals are used for species comparison in text; error bars represent SEM

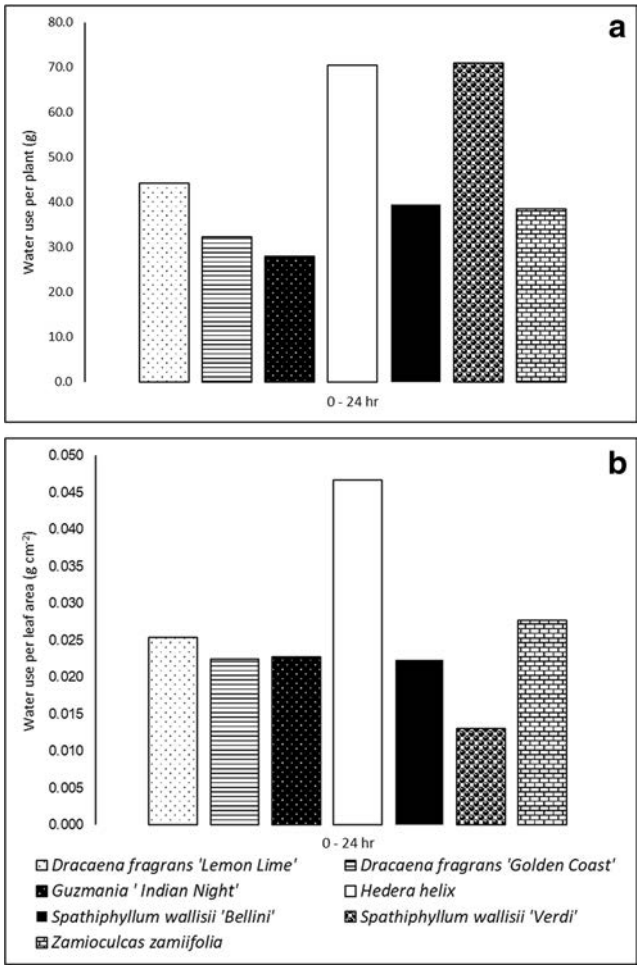

Fig. 3 Water use per plant (**a**) and per leaf area (**b**) per day; data are a mean of four containers of each species ($n = 4$). ANOVA was performed on the log-transformed data only (data not shown); Tukey's 95% confidence intervals generated in the analysis of the transformed data are used for species comparison in text

From the results of the ET experiment, we estimated the contribution of studied taxa to raising RH indoors. Calculations of the amount of water vapour in the air were made through the equation: RH (%) = 100 * actual vapour density (g m^{-3})/saturation vapour density (g m^{-3}) (using a saturation vapour density of 19.1 g m^{-3} at 22 °C) (Galindo et al. 2005). A RH of 40–60% is considered optimal in terms of human health (Arundel et al. 1986); we therefore calculated the number of plants—per taxon—required to raise RH from 40 to 60% in a static 100 m^3 office (Supplementary Table 2). Calculations assume that 100% of the water vapour 'lost' by taxa (Fig. 3a) was released into the surrounding environment. The results do not take into account the impact of ventilation, occupancy, or the feedback effect of taxa (i.e. as RH increases plants release less water vapour into the indoor environment). These calculations are intended to act as a guide on how the studied taxa could influence RH indoors. Our results indicate that five *Spathiphyllum wallisii* 'Verdi' or *Hedera helix* plants growing in an unmulched (i.e. uncovered) growing medium—over a 24-h period—could raise the RH from 40 to 60% (Supplementary Table 2). It also suggests that less physiologically active plants (such as *Guzmania*, *Dracaena*, and *Zamioculcas*) could be used in larger numbers (10+) as part of installations such as indoor living walls within even smaller offices, without a risk of office RH raising above 60%. Conversely, *Hedera* and large *Spathiphyllum* cultivars would be suitable in smaller numbers (5 or below) or in larger rooms with greater overall volume where their RH-influencing effect would be diluted.

Conclusions

The results indicate that net CO_2 assimilation of all studied plants was generally 'low', with *Spathiphyllum* cultivars and *Hedera helix* removing most CO_2.

While CO_2 assimilation of plants in 'wet' substrate was higher than in 'dry' conditions, in practical terms however (i.e. when considering the plant's potential to influence indoor CO_2 levels), net CO_2 assimilation differences between 'dry' and 'wet' plants at 'high' and 'low' indoor light levels were negligible for the taxa studied. Light compensation points were in the typical indoor light range for both *Spathiphyllum wallsii* 'Verdi' and *Hedera helix*, suggesting that these plants would be best suited to provide most CO_2 removal in a typical indoor setting. Additionally, both these taxa, per plant, had the highest transpiration rates, suggesting the highest potential for influencing the RH. Finally, our study indicates that increasing indoor light levels to 300 μmol m^{-2} s^{-1} would, in most taxa, have a significant impact on the potential for houseplants to assimilate CO_2 and increase RH in indoor environments.

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Compliance with ethical standards

Conflict of interest The authors declare that they have no conflict of interest.

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Appendix E 'Interaction between plant species and substrate type in the removal of CO_2 indoors'

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Interaction between plant species and substrate type in the removal of CO_2 indoors

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Abstract

Elevated indoor concentrations of carbon dioxide (CO_2) cause health issues, increase workplace absenteeism, and reduce cognitive performance. Plants can be part of the solution, reducing indoor CO_2 and acting as a low-cost supplement to building ventilation systems. Our earlier work on a selection of structurally and functionally different indoor plants identified a range of leaf-level CO_2 removal rates, when plants were grown in one type of substrate. The work presented here brings the research much closer to real indoor environments by investigating CO_2 removal at a whole-plant level and in different substrates. Specifically, we measured how the change of growing substrate affects plants' capacity to reduce CO_2 concentrations. *Spathiphyllum wallisii* 'Verdi', *Dracaena fragrans* 'Golden Coast', and *Hedera helix*, representing a range of leaf types and sizes and potted in two different substrates, were tested. Potted plants were studied in a 0.15-m^3 chamber under 'very high' (22,000 lx), 'low' (~ 500 lx), and 'no' light (0 lx) in 'wet' (> 30%) and 'dry' (< 20%) substrate. At 'no' and 'low' indoor light, houseplants increased the CO_2 concentration in both substrates; respiration rates, however, were deemed negligible in terms of the contribution to a room-level concentration, as they added ~ 0.6% of a human's contribution. In 'very high' light, *D. fragrans*, in substrate 2, showed potential to reduce CO_2 to a near-ambient (600 ppm) concentration in a shorter timeframe (12 h, e.g. overnight) and *S. wallisii* over a longer period (36 h, e.g. weekend).

Keywords Indoor air quality · Houseplants · Indoor light · *Dracaena* · *Spathiphyllum* · *Hedera*

Abbreviations

ASHRAE	The American Society of Heating Refrigeration and Air-Conditioning Engineers
SMC	Substrate moisture content (m^3 m^{-3})
VOCs	Volatile organic compounds
ANOVA	Analysis of variance
SEM	Standard error of the mean

Highlights

- Substrate type has a significant impact on the ability of indoor plants to remove CO_2.
- Plants were unable to reduce the 1000 ppm CO_2 at typical indoor light levels.
- Plants were able to remove 1000 ppm CO_2 at a light level of 22,200 lx.
- Respiration was deemed negligible in comparison to human contributions.

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Introduction

Elevated indoor concentrations of CO_2 (> 600 ppm) are harmful to human health, increase absenteeism, and reduce cognitive performance (Seppanen et al. 1999; Erdmann and Apte 2004; Shendell et al. 2004; Shaughnessy et al. 2006; Gaihre et al. 2014; Zhang et al. 2017). Traditional building ventilation systems are designed to keep CO_2 concentrations near-ambient with outdoor air infiltration, albeit increasing building energy consumption (Perez-Lombard et al. 2008). Indoor plants can act as a simple low-cost form of ventilation, reducing indoor ventilation requirements (by ~ 6%) with CO_2 removal and consequently providing a reduction in building energy consumption, but only under certain environmental conditions, i.e. a very high light level (~ 22,000 lx)—as confirmed by several previous studies (Torpy et al. 2014, 2017; Gubb et al. 2018).

Numerous health guidelines exist for maximum safe CO_2 concentrations, the lowest of these being 1000 ppm produced by the American Society of Heating, Refrigeration and Air-Conditioning Engineers (ASHRAE)—a concentration often exceeded indoors (Shendell et al. 2004; Gaihre et al. 2014; Torpy et al. 2014, 2017). Concentrations indoors are typically less than 2000–2500 pm but can rise as high as 5000 ppm, with the main source of CO_2 indoors being humans themselves (Zhang et al. 2017).

Elevated CO_2 concentrations (> 600 ppm) can cause an array of health issues including eye irritation, mucus membrane symptoms (i.e. sore/dry throat, dry eyes, and sneezing), and respiratory problems (i.e. tight chest, wheezing/coughing and shortness of breath) (Seppanen et al. 1999; Erdmann and Apte 2004; Tsai et al. 2012). Additionally, elevated concentrations have been associated with declines in cognitive function (at ~ 950 ppm); absenteeism, with increases of 100 ppm associated with a reduced annual attendance of half a day per annum and reductions in cognitive performance, with concentrations of 600–1000 ppm found to significantly reduce decision making ability (Shaughnessy et al. 2006; Satish et al. 2012; Gaihre et al. 2014; Vehvilainen et al. 2016; Allen et al. 2016).

Several studies have shown that light levels significantly influence a plant's ability to remove CO_2 via their impact on stomata as the main pathway for CO_2 uptake (Pennisi and van Iersel 2012; Torpy et al. 2014, 2017; Gubb et al. 2018). Indoors, the light level is typically between 0 and 500 lx but can be as high as 3000 lx in certain workplace environments (Boyce and Raynham 2009; Lai et al. 2009; Hawkins 2011; Huang et al. 2012). Often, supplementary lighting is required to support specific plant installations such as a green wall, where higher light levels are utilised above the installation and not throughout the entire room—this supplementary light can be engineered at least as high as 22,200 lx (Gubb et al. 2018). Plants' under- or over-watering also affects the plant's ability to remove CO_2 (Sailsbury and Ross 1991), but our previous work showed that indoor light level was the primary driver of CO_2 uptake and the soil drying had a smaller impact (Gubb et al. 2018).

Plants remove airborne pollutants via four different pathways: the aboveground plant part (by photosynthesis, deposition, and/or diffusion through the waxy layer), the roots (by deposition and/or direct uptake), and two of which directly involve the substrate—namely, sorption by the substrate itself, along with breakdown by the microbial activity within the substrate (Cruz et al. 2014). It can therefore be expected that both the type and condition (wet/dry) of the substrate will affect plants' CO_2 removal ability. Experiments investigating the ability of plants to remove volatile organic compounds (VOCs) have found that the removal of VOCs is predominately associated with the microflora in the substrate and plants themselves are only utilised indirectly to maintain and support substrate microorganisms (Wood et al. 2002; Orwell et al. 2004; Kim et al. 2008; 2018; Cruz et al. 2014; Irga et al. 2018); these microorganisms—especially those associated with the root system—have been shown to metabolise an array of different pollutants (Weyens et al. 2015).

Various substrates are available in the UK for growing indoor plants, including various types of peat and peat-free (Barrett et al. 2016). Peat—an organic material—is a limited resource, hence attempts by the UK government for voluntary phasing out of peat by 2030 (Defra 2018). Despite this, peat-based substrates are still commonly used across the UK because of their uniformity, providing easier water management (Schmilewski 2008; Alexander et al. 2013). Peat has been shown to have higher water-holding capacity compared with some alternatives such as coir, sand, and wood fibres (Schmilewski 2008). As several studies have linked soil moisture to microbial respiration, an investigation into substrates moisture content is of significance to CO_2 removal (Cook et al. 1985; Manzoni 2012). Furthermore, with different substrate types able to support different microorganisms (Zhang et al. 2013), it was hypothesised that differences in removal would be measured between our chosen substrates. Therefore, two different substrates (peat-free and peat)—referred to as Substrate 1 and Substrate 2, respectively, within this paper—were chosen for this experiment to determine to what extent they affected plants' ability to remove CO_2 within test chambers. We hypothesised that growing the same taxa in differing substrates might provide differing CO_2 removal abilities.

If houseplants are to reduce elevated CO_2 concentrations, they must be functioning optimally, i.e. experience appropriate light levels, feeding, and watering (i.e. substrate moisture content, SMC). A few studies have investigated these issues in part, testing various plants potted in different peat-free substrates (Irga et al. 2013; Torpy et al. 2014, 2017; Gubb et al. 2018).

Torpy et al. (2014) determined the light response curves of eight common plants potted a peat-free substrate consisting of composted hardwood, sawdust, composted bark fines, and coarse river sand (2:2:1). These authors suggested that in typical 'low' indoor light, some CO_2 removal could be expected, but moderately increasing light levels would mean the studied plants could be effectively utilised in a built environment setting. Torpy et al. (2017) also investigated the ability of two taxa (*Chlorophytum comosum* and *Epipremnum aureum*) potted in a peat-free substrate comprising coconut fibre—as part of an active green wall—to remove 1000 ppmv of CO_2 at light levels of 50 and 250 μmol m^{-2} s^{-1}. The study found removal was much more effective at 250 μmol m^{-2} s^{-1} and found that removal from a 5-m^2 wall of *C. comosum* could balance the respiratory emissions of a full-time occupant.

Our research aims to test which houseplants together with the substrate they are grown in (from now on referred to as houseplants or taxa) can best reduce a CO_2 concentration of

1000 ppm under differing environmental and growing conditions. Specifically, we tested the selected taxa:

- Under three light levels: 'very high' (~ 22,000 lx), typical 'low' light (~ 500 lx), and 'no' indoor light (0 lx);
- In 'wet' (SMC > 30%, 0.3 m^3 m^{-3}) and 'dry' (SMC < 20%, 0.2 m^3 m^{-3}) substrate moisture conditions;
- With two different substrate types.

Zero lux (0 μmol m^{-2} s^{-1}) was chosen to investigate CO_2 assimilation/respiration in the dark; ~ 500 lx (~ 7 μmol m^{-2} s^{-1}) was chosen to represent typical office conditions; 22,000 lx (~ 300 μmol m^{-2} s^{-1}) was chosen to represent the highest technically feasible light level which could be engineered indoors (with supplementary artificial lighting) (Torpy et al. 2017).

This experiment was undertaken on a whole-plant/substrate scale as opposed to leaf-level experiments investigated in prior work (Gubb et al. 2018). It was hypothesised that experiments on this larger scale would provide more accurate estimations for how plants can influence 'room-scale' concentrations of CO_2. Additionally, this study looks to highlight if substrate type can make a difference to the CO_2 removal ability of taxa and justify the need for further research with a more extensive range of appropriate substrates in subsequent studies.

Material and methods

Plant material

Three common houseplant taxa (*Dracaena fragrans* 'Golden Coast', *Hedera helix*, and *Spathiphyllum wallisii* 'Verdi'), which were shown in our previous study to have a range of CO_2 removal capacities, were selected for this study. They represented a range of leaf types (succulent and herbaceous) and plant sizes (Table 1). Plants were maintained either in 'Substrate 1': peat-free substrate, i.e. Sylvamix growing medium (Melcourt, Tetbury, Gloucestershire, UK; 6:2:2 sylvafibre:growbark pine:coir; air-filled porosity, 21%; moisture content by weight, 60%) or in 'Substrate 2': peat substrate, i.e. Clover professional pot bedding substrate (Clover, Dungannon, Co. Tyrone, UK, 100% Irish Moss Peat; air-filled porosity, 13%; moisture content by weight, 65%). Plants were maintained in 3-L containers, with a slow-release fertiliser feed (6–9 months, Osmocote; Marysville, OH, USA). Plants were purchased in Summer 2016 (apart from *Dracaena fragrans* 'Golden Coast' in Substrate 2, which was purchased in Spring 2018). Prior to experimentation (for > 90 days), plants were kept at room temperatures (17–22 °C) and 'low' light levels (~ 500 lx) in an indoor office environment within the Crops Laboratory in the Glasshouse Complex of the School of Agriculture, Policy and Development, at the University of Reading (UK). *Hedera helix* could not be successfully grown in Substrate 2 and was omitted from the study in this substrate after several failed attempts.

CO_2 chamber experiments

Experiments were carried out in an experimental laboratory with a non-bypass fume hood at the University of Reading (UK). The experimental setup (Fig. 1) consisted of a ~ 150-L (45 × 45 × 75 cm, 0.15 m^3) Perspex chamber (The Plastic People, Leeds, West Yorkshire, UK) connected to a CO_2 cylinder (CO_2 > 99% purity; Air Liquide, Coleshill, West Midlands, U.K) with a combination of Teflon tubing (¼-in. diameter) and Swagelok's (Swagelok, Bristol, South Gloucestershire, UK). Enclosed inside the Perspex chamber were a HOBO MX1102 CO_2 logger (Onset Computer Corporation, Bourne, MA, USA), a 12 V DC brushless fan (RS Components, Corby, Northants, UK), 500 g of silica gel (Sigma–Aldrich Company Ltd., Gillingham, Dorset, UK), and a calibrated (20–90% RH, 0–40 °C) Tinytag RH/temperature logger (Gemini Data Loggers, Chichester, West Sussex, UK). The external RH/temperature surrounding the chamber was also monitored with another identical Tinytag logger. Inside the chamber 'no' (0 lx, 0 μmol m^{-2} s^{-1}) light was achieved by undertaking at experiments at night; 'low' (~ 500 lx, ~ 7 μmol m^{-2} s^{-1}) light levels were achieved in the

Table 1 Characteristics of the houseplant taxa chosen for experiments in both substrates. Leaf area (n = 3) and plant height (n = 5) are means ± SEM. Species' botanical Latin name is given in italic and cultivar, where applicable, follows

Taxa (Substrate 1)	Family	Metabolism	Leaf area (cm^2)	Plant height (cm)
Dracaena fragrans 'Golden Coast'	*Asparagaceae*	C3	4057 ± 337	83 ± 1
Hedera helix	*Araliaceae*	C3	1542 ± 122	8 ± 1
Spathiphyllum wallisii 'Verdi'	*Araceae*	C3	6033 ± 128	38 ± 1
Taxa (Substrate 2)	Family	Metabolism	Leaf area (cm^2)	Plant height (cm)
Dracaena fragrans 'Golden Coast'	*Asparagaceae*	C3	1417 ± 112	48 ± 1
Spathiphyllum wallisii 'Verdi'	*Araceae*	C3	2591 ± 442	42 ± 2

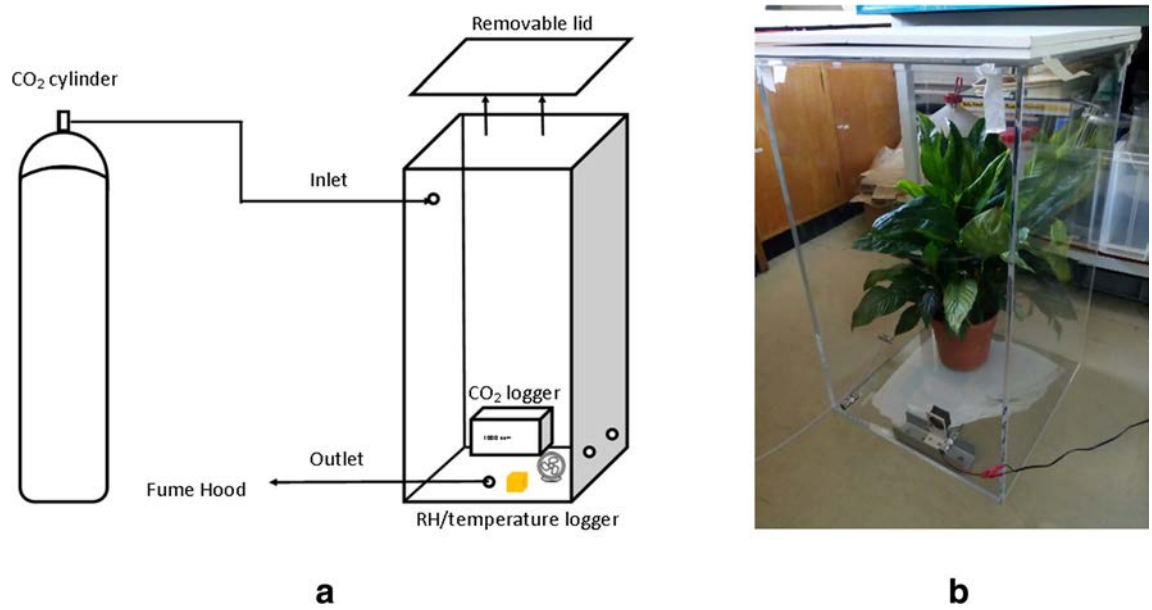

Fig. 1 Schematic diagram (**a**) and image (**b**) of the CO_2 chamber experimental setup

usual lighting conditions of the room (four fluorescent ceiling lights, Osram, Munich, Germany lighting a floor area of 11 m^2); 'very high' levels were achieved with two LED lights (V-TAC Europe Ltd., Sofia, Bulgaria) which were positioned on stands externally, one at an ~30-cm height above the chamber and another ~30 cm from the side of the chamber. Colour temperature of those lights was 6000 K and both lights combined produced a 'very high' (~22,000 lx, ~300 μmol m^{-2} s^{-1}) light level inside the chamber—all three levels were measured with a calibrated light sensor (SKP 200, Skye Instruments, Llandrindod Wells, Wales, UK). This 'very high' light level approximately corresponds to the light saturation for the studied species on a light response curve (Gubb et al. 2018) and was chosen to represent the highest feasible light level which could be engineered (with supplementary artificial lighting) in an indoor environment.

Measurements of the ability of studied taxa to reduce CO_2 concentrations of 1000 ppm (ASHRAE recommended maximum 8-h exposure guideline taken from Torpy et al. 2014) were undertaken on either three ('no' and 'low' light) or five ('very high' light) plants per taxon. Taxa were prepared for experiments with substrate moisture at the container capacity (SMC > 30%) and plants were thus considered optimally watered on the commencement of each experiment (Vaz Monteiro et al. 2016). Measurements were also made on each houseplants 'dry' substrate (SMC < 20%) after a period of drying—the length of which was dependent on the type of plant and its inherent evapotranspiration rate (Gubb et al. 2018). To ascertain when each taxon was 'dry', SMC was measured prior to experimentation for each plant, in two locations per container using an SM300 capacitance-type probe connected to an HH2 Moisture Meter (Delta-T Devices, Cambridge, Cambridgeshire, UK; 0–100% range and an accuracy of ± 2.5%). Experiments were made on one whole 'plant – substrate system' (i.e. potted plant, with uncovered substrate) enclosed inside the Perspex chamber at a CO_2 concentration of 1000 ppm (± 10%). Experiments were for a duration of 1 h with the CO_2 concentration logged every second. Appropriate 'control' measurements were run at all three light levels on both the empty chamber and pot with a substrate, but no plant (in both 'wet' and 'dry' SMC). The number of runs with only substrate and pot was either three for 'no' and 'low' light or five for 'very high' light.

Experimental parameters for each lighting treatment were as follows: 'no' light, ambient (CO_2 < 500 ppm; temperature 17–26 °C; RH 23–64%) and inside chamber (temperature 17–26 °C; RH 31–90%, average 61%); 'low' light, ambient (CO_2 < 500 ppm; temperature 13–23 °C; RH 24–61%) and inside chamber (temperature 13–24 °C; RH 36–90%, average 68%); and high light, ambient (CO_2 < 500 ppm; temperature 15–22 °C; RH 21–60%) and inside chamber (temperature 15–24 °C; RH 32–90%, average 64%). The chamber was also analysed for leakage prior, during, and after experimentation; leakage was found to be < 5% of the starting concentration over the test period. All results were corrected for leakage. This was achieved—for 'no' and 'low' light—by adding the average CO_2 concentration lost through leakage (ppm) to the amount of CO_2 respired by each taxon (ppm)—correcting for the fact that each taxon would have measured a greater concentration of CO_2 if the chamber was airtight. The opposite was done for 'very high' light, correcting for the fact that each taxon would have removed more CO_2 if the chamber was airtight.

Based on the findings of our previous leaf-level work with the same taxa (Gubb et al. 2018), we hypothesised that at 'no' and 'low' indoor light levels, taxa would increase CO_2 concentrations within the enclosure. The CO_2 concentration (ppm h^{-1}) removed by each taxon was calculated with the data measured directly every second by the appropriate logger and divided by the leaf area in meter squared presented in Table 1 to give a unit of ppm m^{-2} h^{-1}.

Statistical analysis

Experimental data (CO_2 concentrations) were analysed using GENSTAT (17th Edition, VSN International, Hemel Hempstead, Hertfordshire, UK). An analysis of variance (ANOVA) was performed to compare means for each measured parameter between different taxa and/or over time. Variance levels were checked for homogeneity and values were presented as means with either associated least significant differences (lsd) at a 5% significance level, standard error of the mean (SEM), or as Tukey's 95% confidence intervals for multiple comparisons. Where a lsd or Tukey's confidence interval has been used for data comparison, the associated p value is presented. Where this is not displayed, SEM has been used.

Results

CO_2 chamber experiments—'no' light

At 'no' indoor light, no taxa reduced CO_2 from the initial 1000 ppm concentration, and the CO_2 concentration inside the chamber increased with all treatments; no statistically significant differences in concentration were measured within taxon between 'dry' or 'wet' conditions (Table 2). Additionally, statistical differences were measured between the Substrates 1 and 2 for *Dracaena fragrans* 'Golden Coast' in both 'dry' (331 and 138 ppm m^{-2} h^{-1}, respectively; Table 2) and 'wet' conditions (332 and 151 ppm m^{-2} h^{-1}, respectively; Table 2).

CO_2 chamber experiments—'low' light

At 'low' indoor light, *Spathiphyllum wallisii* 'Verdi' potted in Substrate 2 reduced the concentration of CO_2 from the initial 1000 ppm concentration ('dry' and 'wet', 43 and 1 ppm m^{-2} h^{-1}, respectively; Table 3). All other plant/substrate combinations increased the CO_2 concentration. Statistically significant differences were measured within taxon between 'dry' and 'wet' conditions for *Hedera helix* in Substrate 1 (379 and 518 ppm m^{-2} h^{-1}, respectively; Table 3). Additionally, statistical differences in removal were measured between the two substrates for *Spathiphyllum wallisii* 'Verdi' in 'wet' conditions (227 and −1 ppm m^{-2} h^{-1}, respectively; $p = 0.03$; Table 3) but not in 'dry' (192 and −43 ppm m^{-2} h^{-1}, respectively; $p = 0.126$; Table 3), and for *Dracaena fragrans* 'Golden Coast' in 'dry' conditions (147 and 7 ppm m^{-2} h^{-1}, respectively, Table 3).

CO_2 chamber experiments—'very high' light

At 'very high' indoor light, all treatments reduced the concentration of CO_2 from the initial 1000 ppm. Significant differences were measured in CO_2 reduction between all taxa, under both 'dry' and 'wet' conditions and between the two substrates (Fig. 2). The range of removal rates was the smallest at 15 min and the largest at 60 min in both 'wet' and 'dry' conditions. After 15 min, no statistically significant differences in CO_2 reduction were measured within the same taxon in either substrate between 'dry' and 'wet' conditions. After 60 min, statistically significant differences were measured in both *Spathiphyllum* and *Dracaena* potted in Substrate 2 between 'dry' and 'wet' conditions but not in Substrate 1 (Fig. 2).

In 'wet' conditions after 15 min, no statistically significant differences were measured between any studied taxa in either

Table 2 Mean CO_2 increase in the chamber per meter squared of leaf area for each taxon potted in the two substrates at 'no' (0 lx, 0 μmol m^{-2} s^{-1}) indoor light in 'wet' (SMC > 30%, 0.3 m^3 m^{-3}) and 'dry' (SMC < 20%, 0.20 m^3 m^{-3}) conditions. Data are a mean of three plants per taxon ± SEM

Taxa (Substrate 1)	Mean CO_2 increase at 'no' light (ppm m^{-2} h^{-1})	
	'Wet' (> 30% SMC)	'Dry' (< 20% SMC)
Dracaena fragrans 'Golden Coast'	332 ± 24	331 ± 18
Hedera helix	745 ± 189	408 ± 148
Spathiphyllum wallisii 'Verdi'	177 ± 30	155 ± 15
Taxa (Substrate 2)	Mean CO_2 increase at 'no' light (ppm m^{-2} h^{-1})	
	'Wet' (> 30% SMC)	'Dry' (< 20% SMC)
Dracaena fragrans 'Golden Coast'	151 ± 78	138 ± 67
Spathiphyllum wallisii 'Verdi'	228 ± 42	185 ± 18

Table 3 Mean CO_2 increases in the chamber per meter squared of leaf area for each taxon potted in the two substrates at 'low' (~ 500 lx, ~ 7 μmol m^{-2} s^{-1}) indoor light in 'wet' (SMC > 30%, 0.3 m^3 m^{-3}) and 'dry' (SMC < 20%, 0.20 m^3 m^{-3}) conditions. Data are a mean of three plants per taxon ± SEM, (−) values signify CO_2 assimilation (i.e. CO_2 uptake by the plant thus its removal from the chamber)

Taxa (Substrate 1)	Mean CO_2 increase at 'low' light (ppm m^{-2} h^{-1})	
	'Wet' (> 30% SMC)	'Dry' (< 20% SMC)
Dracaena fragrans 'Golden Coast'	142 ± 8	147 ± 13
Hedera helix	518 ± 42	379 ± 54
Spathiphyllum wallisii 'Verdi'	227 ± 57	192 ± 104
Taxa (Substrate 2)	Mean CO_2 increase at 'low' light (ppm m^{-2} h^{-1})	
	'Wet' (> 30% SMC)	'Dry' (< 20% SMC)
Dracaena fragrans 'Golden Coast'	66 ± 68	7 ± 52
Spathiphyllum wallisii 'Verdi'	− 1 ± 38	− 43 ± 64

substrate (Fig. 2; $p = 0.550$). After 60 min, *Dracaena fragrans* 'Golden Coast' in Substrate 2 reduced statistically the largest amount of CO_2 from the initial 1000 ppm concentration (1420 ppm m^{-2} h^{-1}; $p < 0.001$). No statistically significant differences in CO_2 removal were measured between *Spathiphyllum wallisii* 'Verdi' (623 ppm m^{-2} h^{-1}) in

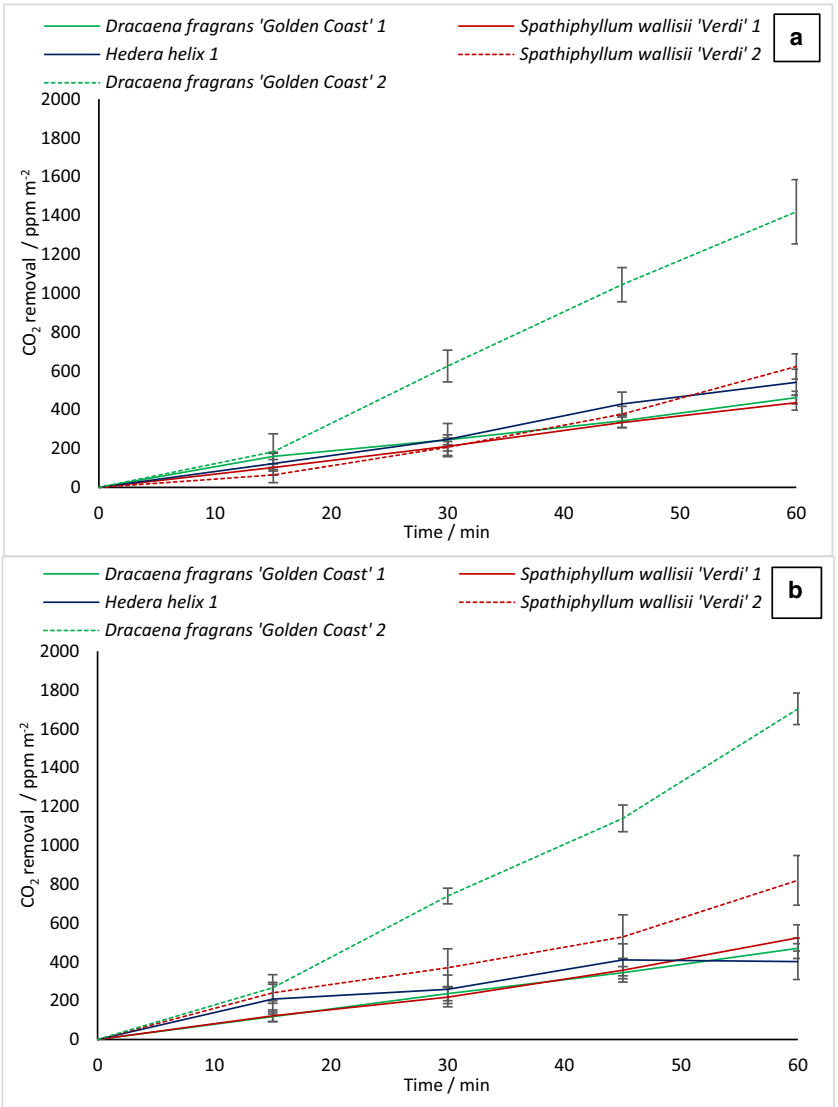

Fig. 2 Mean CO_2 removal by each taxon in Substrates 1 and 2 at 'very high' indoor light (~ 22,000 lx, ~ 300 μmol m^{-2} s^{-1}) per meter squared of leaf area in 'wet' (SMC > 30%, 0.3 m^3 m^{-3}) (**a**), and 'dry' (SMC < 20%, 0.20 m^3 m^{-3}) (**b**) conditions over a 60-min period. Data are a mean of five plants per taxa—error bars represent SEM

Substrate 2 and any of the taxa potted in Substrate 1—*Hedera helix*, *Spathiphyllum wallisii* 'Verdi' and *Dracaena fragrans* 'Golden Coast' (541, 436 and 463 ppm m^{-2} h^{-1}, respectively; $p < 0.001$; Fig. 2).

In 'dry' conditions after 15 min, no statistically significant differences were measured between any studied taxa in either substrate (Fig. 2; $p = 0.221$). After 60 min, *Dracaena fragrans* 'Golden Coast' in Substrate 2 reduced statistically the largest amount of CO_2 from the initial 1000 ppm concentration (1703 ppm m^{-2} h^{-1}; $p < 0.001$). A statistically significant difference was measured between *Spathiphyllum wallisii* 'Verdi' (820 ppm m^{-2} h^{-1}) in Substrate 2 and *Hedera helix* in Substrate 1 (401 ppm m^{-2} h^{-1}; $p < 0.001$). No statistically significant differences were measured between other studied taxa, i.e. *Spathiphyllum wallisii* 'Verdi' and *Dracaena fragrans* 'Golden Coast' (524 and 470 ppm m^{-2} h^{-1}, respectively; $p < 0.001$; Fig. 2).

Discussion

This work investigates how potting common houseplants in two differing substrates influenced their ability to reduce a harmful CO_2 concentration of 1000 ppm at a whole-plant/substrate scale.

In this study, we demonstrated that at 'low' light in 'dry' substrate conditions assimilation occurred with *Spathiphyllum wallisii* 'Verdi' potted in Substrate 2 (− 43 ppm m^{-2} h^{-1}) but not in Substrate 1 (192 ppm m^{-2} h^{-1}), contrary to the initial hypothesis where an increase in CO_2 concentration was expected from all studied taxa (Gubb et al. 2018). Similarly, the study found that *Dracaena fragrans* 'Golden Coast' was the most effective taxon at reducing high concentrations of CO_2 at 'very high' indoor light levels when potted in Substrate 2 (1703 ppm m^{-2} h^{-1}). When the same taxon was maintained in Substrate 1, CO_2 removal was statistically significantly lower (470 ppm m^{-2} h^{-1}). Although less strongly, there was a suggestion in our measurements that *S. wallisii* 'Verdi' in high light removed more CO_2 by the end of a 60-min period, when potted in Substrate 2 compared with Substrate 1.

These measurements suggest that differing substrate types may be able to influence CO_2 assimilation. A taxon may grow more effectively and be more physiologically active in a particular substrate, facilitating a stronger CO_2 removal ability. Peat has long been cited as a substrate which supports good plant growth, having good air-filled porosity, high water-holding capacity, and a relatively pest- and pathogen-free environment (Schmilewski 2008). Moreover, peat contains a carbon concentration in the range of 30–70 kg/m^3 (18–60%) whereas, for other mineral soils, this concentration is typically < 20% (Agus et al. 2011); this additional carbon might be a possible reason for greater CO_2 sequestration in our Substrate 2. Alternatively, the substrate and plant combined may support differing microorganisms, which in turn could provide a superior removal ability (Zhang et al. 2013). This, however, would need to be explored further by evaluation of the differing microorganisms in both substrates and additional inoculation experiments with the microorganism species in question (De Kempeneer et al. 2004). Moreover, studies have also found differences in CO_2 removal between species grown in traditional potting mix and hydroculture (Irga et al. 2013). Clearly, the substrate type is of importance in terms of CO_2 removal, and this should be further investigated in subsequent studies. Additionally, this needs to be kept in context of the fact that the overall capacity of individual plants to remove CO_2 indoors is small (Pennisi and van Iersel 2012; Irga et al. 2013; Torpy et al. 2014, 2017; Gubb et al. 2018). Furthermore, while we have expressed our CO_2 removal data per unit leaf area (thus taking differences in plant size into the account), we cannot exclude the possible impact of age differences between the plants. We made every effort to source the plants simultaneously, but their lifecycle and management prior to reaching us were beyond our control. Moreover, the authors acknowledge that photosynthetic activity can be reduced at high RH (Sailsbury and Ross 1991), and therefore, the results may have underestimated the CO_2 removal in some treatments.

At 'no' and 'low' light levels typically experienced in indoor environments (Hawkins 2011), most of the studied taxa would increase the concentration of CO_2 in indoor environments as measured in our earlier leaf-level work (Gubb et al. 2018). However, *Hedera*, the taxon which potted in a Substrate 1 respired most, increased the CO_2 concentration by 115 ppm h^{-1} (i.e. 0.2 g m^{-3} h^{-1}); comparatively, each person contributes 36 g h^{-1} of CO_2 in an office environment (Persily and de Jonge 2017). The contribution of plants to CO_2 concentration increases can therefore be considered negligible in comparison with human contributions indoors at ~ 0.6% of a human contribution, in agreement with prior experiments (Gubb et al. 2018).

Our study clearly suggests that increasing the lighting levels indoors—made possible with targeted lighting installations—would allow taxa to significantly reduce CO_2 concentration. This agrees with other similar studies, which show that light is the limiting factor for CO_2 reduction indoors (Pennisi and van Iersel 2012; Gubb et al. 2018) and that houseplants can be expected to aid ventilation systems—by providing additional CO_2 removal—but not replace them completely (Torpy et al. 2014).

The results of the current study allow us to estimate the number of houseplants required to reduce CO_2 concentrations to a safe acceptable indoor level—literature suggests that concentrations of 600 ppm and below cause fewer health issues than elevated CO_2 concentrations (Seppanen et al. 1999; Erdmann and Apte 2004; Allen et al. 2016). Therefore, for a small office of 15 m^3 (11 m^3 is the minimum space required

per person; HSE 1992), we calculated the time required for a 'dry' *Dracaena fragrans* 'Golden Coast' potted in Substrate 2 (as this plant/substrate combination led to most CO_2 removal under our experimental conditions) to remove 400 ppm of CO_2 (i.e. reduce CO_2 concentration from 1000 to 600 ppm), at a 'very high' light level assuming a sealed environment with no other sources of CO_2 (Eq. 1).

$$\text{Time per meter squared of leaf area (h)} = \text{concentration of } CO_2 \text{ to remove (ppm)} / \text{rate of } CO_2 \text{ removal } \left(\text{ppm m}^{-2}\ \text{h}^{-1}\right) \times 1/100 \quad (1)$$

Taking into account volumetric loading differences (Girman 1992) between the test chamber (0.15 m^3) and the small office (15 m^3), the rate of CO_2 removal is reduced by a factor of 100. Consequently, from the results in Fig. 2, we estimate 2 m^2 of *Dracaena fragrans* 'Golden Coast' (equating to 14 plants) in 'dry' conditions would require 12 h to remove 400 ppm of CO_2 in the office as per the above-stipulated conditions.

Differences in removal between 'dry' and 'wet' conditions across taxa at all light levels and substrates were deemed negligible in agreement with Gubb et al. (2018). This indicates that if plants are left to dry out—anecdotally a common occurrence—the impact on a room-scale CO_2 flux is small, although, on a leaf level, there are differences in CO_2 assimilation. Additionally, at 'no' and 'low' light levels, most taxa (i.e. the overall system) were respiring. Our study suggests that although at typical 'no' indoor light, all studied taxa added CO_2 to the indoor environment; the highest increase was approximately half the CO_2 concentration removed at 'very high' light levels. This current work therefore confirms that placing a number of the studied houseplants in a typical home/office environment would not significantly damage health by increasing CO_2 concentrations indoors under either 'wet' or 'dry' substrate conditions.

Even at 'very high' light levels, both *Spathiphyllum wallisii* 'Verdi' and *Hedera helix* would require an unrealistic number of plants in both substrates to reduce CO_2 concentrations from 1000 ppm to a near-ambient level. This is in contrast with plants' pronounced benefits in health and productivity terms (Park and Mattson 2008, 2009; Shibata and Suzuki 2002, 2004).

Our findings support the notion that the light level significantly impacts CO_2 removal, as suggested in previous studies (Pennisi and van Iersel 2012; Torpy et al. 2014, 2017; Gubb et al. 2018). Other previous work had also determined that unrealistic numbers of plants (> 200) are required to remove a significant amount of CO_2 in indoor environments (Pennisi and van Iersel 2012; Torpy et al. 2014). These studies, however, did not take into account substrate moisture differences or ambient CO_2 concentrations (Pennisi and van Iersel 2012). Other studies did not specify which or how many taxa provided any CO_2 removal (Lim et al. 2009; Pegas et al. 2012) or only considered one light level (Oh et al. 2011).

Torpy et al. (2017) estimated that a 2-m^2 active green wall of *Chlorophytum comosum* (where the substrate is actively ventilated by pushing air through it) in peat-free substrate would be capable of removing 11 g of CO_2 per hour in a 16-m^3 room. Our previous work estimated that 2 m^2 (of leaf area) of *Spathiphyllum wallisii* 'Verdi' in unventilated peat-free substrate removed 0.75 g of CO_2 per hour at a comparable light level (Gubb et al. 2018). This current work estimated that 2 m^2 (of leaf area) of *Dracaena fragrans* 'Golden Coast' at a light level comparable with both of the previous removes 3 g/m^3 of CO_2 per hour in a 15-m^3 room, clearly highlighting the benefits of 'active' walls (i.e. substrate ventilation) opposed to traditional 'passive' houseplants.

We support the notion that any future work should focus on green walls (Pettit et al. 2017; Torpy et al. 2017) (especially 'active' walls) which yield more effective removal due to an increased leaf area of taxa and increased substrate airflow. Additionally, taxa which have performed well in removing other indoor pollutants at high indoor light levels, i.e. *Osmunda japonica* (Kim et al. 2010), should be further examined. Furthermore, more substrate types should also be investigated. This study has shown that the ability of plants to remove CO_2 at typical indoor light levels may be maximised with certain substrate types and moisture conditions, therefore lower—more realistic—numbers of plants may be required to reduce harmful concentrations of CO_2. Additionally, as 'active' walls—which are clearly superior removers—place extra emphasis on the substrate, removal differences between substrate types will likely be further highlighted.

Conclusion

The study confirmed that growing the same taxa in differing substrates significantly influenced removal ability in most of the studied species—highlighting the key role substrate types play. The results from the current work indicates that 2 m^2 of *Dracaena fragrans* 'Golden Coast' would require 12 h at a 'very high' light level (~ 22,000 lx) in 'dry' conditions to reduce 1000 ppm of CO_2—the ASHRAE recommended maximum 8-h exposure guideline—to a 600 ppm concentration in a 15-m^3 closed environment (i.e. small office) with no other sources of CO_2. Other studied taxa (*Spathiphyllum wallisii* 'Verdi' and *Hedera helix*) were found to require an unrealistic number of plants at the same 'very high' light level.

At typical 'no' and 'low' indoor light levels, most studied houseplants increased CO_2 concentrations albeit for the highest respiring taxa at approximately half the concentration removed at 'very high' light. Therefore, none of the studied houseplants would significantly elevate CO_2 concentrations indoors and, thus, cause detrimental health effects.

Differences between 'dry' and 'wet' substrates in their capacity for CO_2 removal at either 'no', 'low', or 'very high' light can be considered negligible. Our findings support the notion that raising the light level indoors is paramount for studied taxa to remove CO_2.

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Appendix F 'Can plants be considered a building service?'

Review

BUILDING SERVICES ENGINEERING RESEARCH & TECHNOLOGY

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Can plants be considered a building service?

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Abstract

Plants are utilised in many forms within indoor environments, from simple houseplants to complex and species-rich green walls. Plants offer multi-faceted services indoors including pollutant removal and reductions in building energy consumption. This review firstly identifies – by critical assessment of the literature – pollutants which are currently measured at harmful concentrations indoors – classifying them as '2019's priority pollutants' and providing thorough health assessments of each. Secondly, the authors present which indoor plants have been shown to effectively remove '2019's priority pollutants' and direct future research onto any that have not been investigated. Thirdly, the authors consolidate the current research presenting why plants should be considered a building service.

Practical application: Plants are commonly used inside indoor environments. However, the benefits they bring are often overstated. This review paper looks to consolidate the current academic research on the various services plants can provide indoors including pollutant removal and relative humidity regulation. The authors hope that the paper can be used to inform and educate building service engineers and alike on the current state of play concerning indoor plants.

Keywords

Building service, indoor air quality, indoor plants, pollutants

Introduction

Plants deliver an array of multi-faceted benefits indoors providing improvements in human health (via pollutant removal) and building energy consumption (via removal of CO_2 and relative humidity (RH) regulation which in turn, reduces the ventilation requirements).[1,2]

Numerous airborne pollutants are present in indoor environments: these include volatile organic compounds (VOCs), inorganic gases/vapours (CO_2, NO_2) and particulate matter (PM).[3] The main sources of indoor pollutants are human activities indoors, construction materials and the infiltration of outdoor-produced particles and pollutants.[4–6] Typical activities such as cooking, cleaning and painting produce numerous indoor pollutants.[6,7] Additionally, both the closure of windows and a push for more tightly sealed buildings – in an attempt

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to reduce energy consumption – leads to an accumulation of indoor pollutants.[5,8]

In western Europe, people are commonly exposed to more than 20 h per day of indoor air.[9] Thus, quantifying the concentration of indoor pollutants with relevant safe exposure guidelines/standards is imperative – a guideline is based upon scientific evidence or expert opinion and a standard contains enforceable regulations adopted by regulatory authorities.[10] Such health guidelines produced by organisations such as the World Health Organisation (WHO) and US Environmental Protection Agency (EPA) contain only a limited number of indoor pollutants.[6,11]

Indoor pollutants cause an array of acute and long-term (chronic) health problems, contribute to poor indoor air quality (IAQ) and are the probable cause of sick building syndrome (SBS), a phenomenon describing health issues experienced by the occupants of a building, caused by spending time within the building but, where no specific cause can be found.[6,12] Moreover, indoor pollutants also react with ozone and produce radicals and secondary organic aerosol (SOA) – both, considered harmful to health.[13,14]

Indoor pollutants all possess varying toxicity and prevalence indoors. Prolonged exposure to an indoor pollutant, at a concentration greater than the recommended guideline, can cause varying symptoms from mild sensory irritation (i.e. alpha-pinene) to significant respiratory problems (i.e. NO_2) and cancer (i.e. benzene).[6,15] Indoor plants have been shown to remove a wide variety of organic and inorganic pollutants,[1,16] PM,[17–19] and ozone.[20,21] Additionally, houseplants have also been shown to help alleviate the symptoms of SBS.[22–24]

Additionally, high indoor concentrations of CO_2 are harmful to human health, increase absenteeism and reduce cognitive performance.[25–30] HVAC systems are therefore designed to keep concentrations low with ventilation, increasing energy consumption.[31] Indoor plants can act as a simple low-cost form of ventilation, contributing to CO_2 removal indoors and reducing the requirement of traditional heating, ventilation and air conditioning (HVAC) systems by $\sim 6\%$.[1]

Indoor plants can also reduce energy consumption by increasing RH. HVAC systems attempt to keep RH in the range of 40–60% — where the majority of adverse health effects can be avoided.[32] Both too high (>60%) and too low (<40%) an RH can cause an array of health and building issues.[32] High RH encourages fungal and mould growth and contributes to the deterioration of building materials,[33–36] whereas low RH can cause dryness of the eyes, skin and mucus membrane, enhance indoor ozone, increase the likelihood of influenza transmission, and produce static electricity.[32,33,35,37–39]

Our review aims to improve current understanding on which indoor pollutants – and at what concentrations – are harmful to health. The review updates the previous 'Logue priority pollutants'[40] by designating '2019's priority pollutants' highlighting future research needs to these pollutants, at realistic indoor concentrations. The review also assesses relevant literature for how indoor plants remove pollutants and reduce energy consumption in buildings. We conclude by presenting why indoor plants should be considered a building service.

Method

A systematic review of the literature was conducted to determine the indoor pollutants measured in home environments (up to and including 2018). Key word searches in Science direct, and Google Scholar were conducted using combinations of the following keywords: 'indoor plants', 'houseplants', 'home' and 'pollutants'. Once key pollutants were established, focused searches were conducted with specific pollutants, regions, and types of home. From these, 120 home environments were deemed suitable and used to compile both a common range and the arithmetic mean concentration of over 100 pollutants (some of which are presented in Table 1 and Figure 1).

Indoor pollutants

Which indoor pollutants are harmful to human health? – identifying '2019's priority pollutants'

A couple of review articles have previously compiled data on the concentration and identity of indoor pollutants measured in various indoor environments.[40,41] Cometto-Muniz et al.[41] compiled data from indoor and outdoor environments but used only a small number of studies and did not consider a comparison to relevant health guidelines.

Logue et al.[40] compared indoor pollutant concentrations to relevant health guidelines produced by the EPA and California Office of Environmental Health Hazard Assessment (OEHHA) for 67 home environments between 1998 and 2010. Logue et al. identified nine 'priority' indoor pollutants (i.e. Logue's priority pollutants, Table 1) considered to be harmful – all were chosen on the basis of the measured concentration data exceeding health guidelines and the number of homes impacted. We have included the range of concentrations at which each pollutant was measured indoors (The 'Method' section, Table 1), to direct future experiments to relevant concentrations and pollutants.

Since 2010, an assessment of 'Logue's priority pollutants' and their mean concentrations in indoor environments has not been carried out. We have therefore compiled data (section 'Method') from home environments post-2011, to determine if concentrations of these nine pollutants have changed since the Logue et al. study was undertaken. Furthermore, we compare the post-2011 results to up to date chronic health guidelines produced by the WHO and USEPA (Figure 1). Any pollutants with an average long-term concentration greater than the appropriate guideline will be designated a '2019 priority pollutant'.

The data collected in Figure 1 suggest that the mean concentrations of four indoor pollutants have increased in studies post-2010 namely, benzene, naphthalene, nitrogen dioxide and $PM_{2.5}$. Reductions in concentrations of acetaldehyde, acrolein, dichlorobenzene – 1,4, and formaldehyde were measured; perhaps, due to a large body of research focusing on lowering pollutant emissions from building materials.[52–54] Additionally, no guidelines are provided by the WHO or USEPA for $PM_{2.5}$ indoors; however, ambient EU guidelines[55] are higher than the average concentrations measured (Figure 1).

Acetaldehyde, benzene, formaldehyde, and NO_2 are the indoor pollutants commonly measured at concentrations greater than the appropriate guidelines (Figure 1) – causing long term health issues and thus, are classified as '2019's priority pollutants'.

Table 1. The identity of 'Logue's priority pollutants'[40] and the range of concentrations which they have been measured at in home environments in literature, section 'Method'.

Priority Pollutants identified by Logue et al.[54]	Indoor concentration range ($\mu g\ m^{-3}$)	Reference
Acetaldehyde	5.0–22.0	42,43
Acrolein	0.8–2.3	40,43
Benzene	1.9–43.7	44,45
Butadiene–1,3	0.5[a]	40
Dichlorobenzene–1,4	1.8–120.0	43,46
Formaldehyde	13.0–69.0	40, 43
Naphthalene	0.8–2.6	47,48
Nitrogen dioxide (NO_2)	13.1–489.7	43,49
$PM_{2.5}$	6.5–55.7	50,51

[a]Only one appropriate measurement.

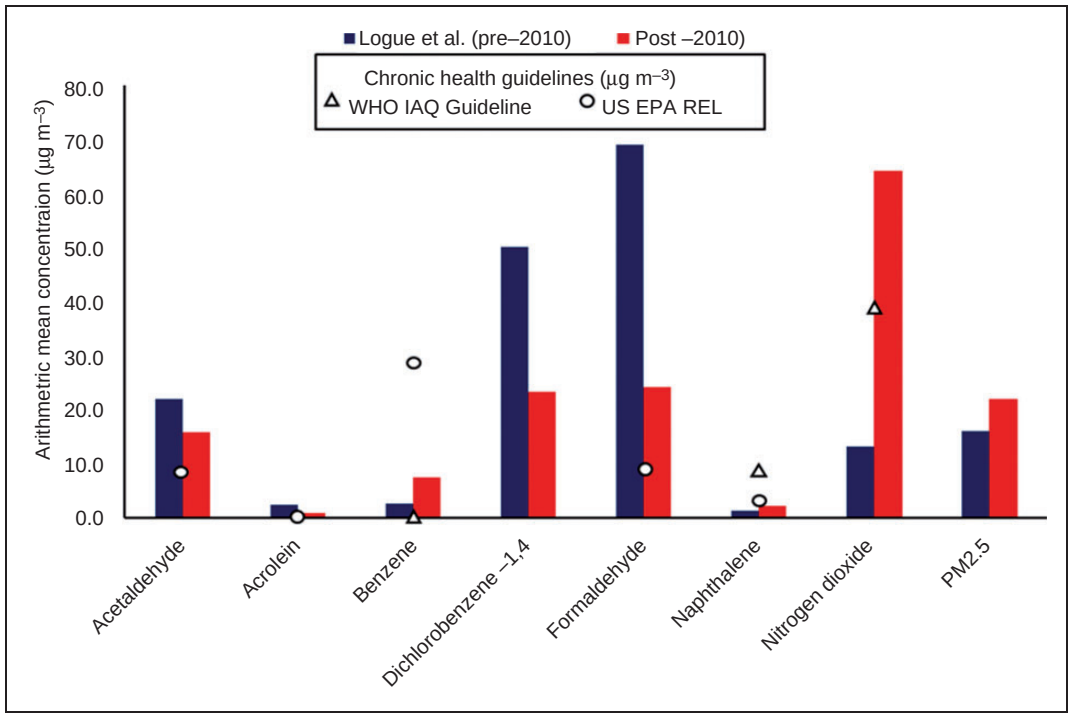

Figure 1. Arithmetic mean concentrations of 'Logue's priority pollutants'[40] both, pre and post 2010 (i.e. date of Logue et al. study). Triangles and circles on the figure represent the appropriate chronic health guideline produced by the WHO and the US EPA.[6,11] Butadiene-1,3 has been omitted from the figure, as no data was found post-2010 in home environments. Data post 2010 is taken from literature.[43,44,50,51,56–70]

Health issues caused by '2019's priority pollutants'

Table 2 presents both the indoor sources and the main health issues associated with exposure to 2019's priority pollutants. Table 2 does not consider outdoor sources, but infiltration is a significant contributor to indoor concentrations – especially NO_2.[6]

Indoor plants and '2019's priority pollutants'

Forty studies have investigated numerous indoor plants for their ability to remove the '2019 priority pollutants' benzene (>45 species/cultivars) and formaldehyde (>150 species/cultivars). The most robust, well cited and informative studies from the above have been selected and are presented in Table 3.

To the author's knowledge, no studies have investigated the potential of indoor plants to sequester either acetaldehyde or NO_2, although the removal of NO_2 by outdoor plants has been thoroughly studied with promising results.[71,72] Of 'Logue's priority pollutants' (Table 1) only $PM_{2.5}$ has been tested with indoor plants – highlighting the lack of 'relevant' pollutants studied. A $PM_{2.5}$ study has also been included in Table 3.

Plants as a building service

CO_2 removal

The main sources of CO_2 indoors are human respiratory emissions and the outdoor air supply rate – which is expected to increase

Table 2. The indoor sources and health issues associated with 2019's priority pollutants.[6]

2019's Priority pollutant	Indoor sources	Main health concerns from exposure
Acetaldehyde	Deodorants, foods and alcoholic drinks.	Carcinogen and an irritant to eyes and airways.
Benzene	Building materials, furniture, heating and cooking systems, cleaning, painting and consumer product use.	Blood dyscrasias and leukaemia, lung cancer, all haematological cancers and multiple myeloma.
Formaldehyde	Building materials, furniture, consumer product use and combustion processes (i.e. heating, cooking and smoking).	Myeloid leukaemia and all airway cancers.
NO_2	Combustion processes (i.e. heating appliances, fireplaces and stoves).	Respiratory illnesses, airway inflammation and decreases in immune defence.

significantly in the next 100 years.[30] Several health guidelines exist for maximum safe CO_2 concentrations, with the lowest 8 h guideline being recommended by ASHRAE at 1000 ppm.[1,77] Above this concentration a large number of health and respiratory issues can occur. Concentrations indoors are typically less than 2000–2500 pm, but have been measured as high as 5000 ppm[30]; 1000 ppm, however, is often exceeded in indoors; one study found 45% of 436 studied classrooms to exceed 1000 ppm over a 5-min measurement period and another measured a median concentration of 1086 ppm over the school day in 60 classrooms.[28,78]

Although guidelines recommend indoor CO_2 concentrations of no greater than 1000 ppm, significant health issues are measured at even lower concentrations. Concentrations of CO_2 > 800 ppm are more likely to cause eye irritation, and upper respiratory symptoms[79] and CO_2 concentrations of ~ 950 ppm reduce cognitive function.[29] Additionally, it has been determined that an increase in 100 ppm of CO_2 in a school environment is associated with a reduced annual attendance of half a day per annum.[28]

A number of studies have focused on indoor plants and their ability to reduce CO_2 concentrations,[77,80] with several specifically focusing on houseplants.[1,24,81–84] Studies vary in scale and focus, but most utilise experimental chambers enclosing a single or small number of houseplant taxa. Studies generally find that significant reductions can occur, with the correct environmental conditions namely, the light level, which needs to be significantly increased from room-level and is made achievable with supplementary lighting.[2,83]

We found that increasing the light level to 22,000 lux – made achievable with supplementary LED lighting – increased the CO_2 removal by 50-fold in some species. Moreover, we estimated that 15 *Spathiphyllum wallsii* 'Verdi' – a number which could be realistically installed in a small green wall – could offset 10% of a human's respiratory contribution.[2] Additionally, a similar study by Torpy et al.[77] found that a 5 m^2 green wall containing *Chlorophytum comosum* could balance the respiratory emissions of a full-time occupant using a similar lighting level.

RH regulation

An additional challenge along with high CO_2 concentrations in indoor environments is extreme RH (i.e. low $< 40\%$ and high $> 60\%$). Both cause an array of previously described issues mainly concerning human and building health. A number of studies have investigated the effect of indoor plants on RH indoors with mixed results.[24,84–86]

Table 3. Selected studies[18,73–76] showing plants removing '2019's priority pollutants' benzene and formaldehyde.

Priority pollutant	Plant species tested	Result	Reference
Benzene	*Dracaena marginata* *Dracaena 'Janet Craig''* *Epipremnum aureum* *Howea forsteriana* *Schefflera* 'Amate' *Spathiphyllum 'Petite'* *Spathiphyllum 'Sensation'*	The authors found varying removal rates from 12 (*Howea forsteriana*) to 27 (*Dracaena 'Janet* Craig') ppm d^{-1} (40 to 88 mg m^{-3} d^{-1}). These rates were maintained in the dark, rose linearly with a concentration increase but, could mostly be contributed to the growing substrate and not the plant.	73
Benzene	*Syngonium podophyllum*	The study compared plants grown in traditional substrate and hydroculture (i.e. no growing substrate) and concluded that indoor plants potted in traditional substrate (1444 μg m^{-3}, rate for 50% benzene removal) possessed a higher removal rate than hydroculture potted plants (739 μg m^{-3}, rate for 50% benzene removal) but, both treatments removed significant amounts of benzene.	74
Formaldehyde	*Fatsia japonica* *Ficus benjamina*	Comparison of the aboveground plant parts and the root zone to remove formaldehyde in the day and night. *Fatsia japonica* removed Formaldehyde faster than *Ficus benjamina* (50% decay in 96 and 123 min respectively). Both plants removed Formaldehyde in a 1:1 ratio (aboveground parts: root zone) in the day, and 1:11 in the night. The root zone was found to remove formaldehyde primarily through the microorganisms and roots (90%) and a small amount through growing medium absorption (10%).	75
Formaldehyde	*Chlorphytum comosum* *Aloe vera* *Epipremnum aureum*	All plant-substrate systems removed formaldehyde, with *Chlorphytum comosum* the most effective. The authors also determined that microorganisms in the substrate accounted for ~50% of the formaldehyde removal in all the plant-substrate systems.	76
PM2.5	*Chlorophytum orchidastrum* *Ficus lyrata* *Nematanthus glabra* *Nephrolepis cordifolia duffii* *Nephropelis exaltata bostoniensis* *Schefflera amate* *Schefflera arboricola*	Investigated the single pass removal efficiency (SPRE) of plants in an active green wall for $PM_{2.5}$ removal. The authors found that all studied plants removed PM with fern species recording the highest removal efficiencies. *Nephropelis exaltata bostoniensis* removed the most $PM_{2.5}$ with an SPRE of >70%.	18

Indoor plants have been shown to increase[2,85,86] decrease[24] and have no statistically significant effect[84] on RH indoors. Through transpiration houseplants release water vapour into an indoor environment and would be expected to increase RH indoors. Both plant species choice and ventilation rate would significantly influence results and most likely explain the mixed results in literature. However, correct employment of indoor plants with species consideration could help reduce the energy consumption of HVAC systems.

Our research suggests that less physiologically active plants (such as *Guzmania* sp., *Dracaena fragrans* and succulents such as *Zamioculcas zamiifolia*) could be used in larger numbers (10+) as part of installations such as indoor living walls within even smaller offices, without a risk of office RH raising above 60%. Conversely, *Hedera helix* (Ivy) and *Spathiphyllum* (peace lily's) would be suitable in smaller numbers (5 or below) or in larger rooms with greater overall volume where their RH-influencing effect would be diluted.[2]

Conclusions and future direction

A significant body of research has been undertaken on the ability of plants to remove indoor pollutants (i.e. VOCs); however, most focus on pollutants that are detected infrequently indoors or at concentrations too low to damage human health; experiments also commonly test pollutant concentrations that are not measured in real life (in situ).[87–89] This review highlights both the range of concentrations present in situ and which indoor pollutants can be considered unsafe – to help direct future research.

Experiments suggest that the growing substrate and the microorganisms within are predominately involved in the removal of pollutants and plants themselves are only utilised in-directly to maintain and support substrate microorganisms.[90,91] Results generally suggest that the plant-substrate system can remove a wide variety of pollutants, but with a lack of testing at in situ concentrations extrapolation of the results to room level lacks accuracy. Further experiments should focus on the untested '2019 priority pollutants' Acetaldehyde and NO_2 identified in this review – preferably at in situ concentrations.

Certain houseplants can remove CO_2 – and at significant quantities that would affect room level concentrations – but only with the correct environmental conditions (i.e. light level). Studies often suggest that a greater number of potted plants are required to measure concentration reductions then would be feasible indoors[1,2,83] and thus, the density provided by green walls would be more suitable. Studies are now beginning to investigate green walls[77] and additionally, how the substrate may influence removal – as measured with VOCs.

RH literature has produced conflicting results, anecdotally plants would be expected to increase RH indoors as found in literature;[2,85,86] however, this is not always the case.[24,84] We suggest a 'standard' method be devised controlling both chamber/room size and ventilation rate facilitating effective comparison between different plant species. Moreover, the performance of different plant species may also explain some of the inconsistency's seen in literature – this could be addressed by further replication and more studies investigating different plant species.

We believe that plants should not be considered as a single entity expected to provide all the above described benefits – differences between species provide large performance variability, so we recommend consulting literature for suitability concerning a given benefit. Furthermore, although some benefits of indoor plants are less clear, we believe that when considered as a whole – with all the benefits combined – plants should be considered as a building service alongside traditional ventilation systems.

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