Article (refereed) - postprint

Castro-Castellon, Ana Teresa; Hughes, Jocelyne Mary Rose; Read, Daniel Steven; Azimi, Yaldah; Chipps, Michael James; Hankins, Nicholas Peter. 2021. **The role of rhizofiltration and allelopathy on the removal of cyanobacteria in a continuous flow system.** *Environmental Science and Pollution Research*, 28. 27731-27741. https://doi.org/10.1007/s11356-021-12343-9

The role of rhizofiltration and allelopathy on the removal of cyanobacteria in a continuous flow system

Ana Teresa Castro-Castellon[1], Jocelyne Marie Ragody Hughes [2], Daniel Steven Read[3], Yaldah Azimi[1], Michael John Chipps[4], Nicholas Peter Hankins[1]*

[1] Department of Engineering Science, University of Oxford, Parks Road, Oxford, OX1 3PJ, UK.

[2]School of Geography and the Environment, University of Oxford, South Parks Road, Oxford, OX1 3QY, UK.

[3]Centre for Ecology & Hydrology, Wallingford, Oxford, OX10 8BB, UK

[4]Thames Water Innovation, Thames Water Utilities Ltd., Reading, UK

[*]Corresponding author contact details: a.castro-castellon@bathspa.ac.uk
Present address: Bath Spa University, Newton Park, Newton St Loe, Bath, BA2 9BN, UK
Tel.: +44-07729-731080

Abstract

A continuous flow filtration system was designed to identify and quantify the removal mechanisms of Cyanobacteria (*Microcystis aeruginosa*) by hydroponic biofilters of *Phalaris arundinacea* compared to synthetic filters. The filtration units were continuously fed under plug-flow conditions with *Microcystis* grown in photobioreactors. *Microcystis* cells decreased at the two flow rates studied (1.2 ±0.2 and 54 ±3 $cm^3\ min^{-1}$) and results suggested physical and chemical/biological removal mechanisms were involved. Physical interception and deposition was the main removal mechanism with packing density of the media driving the extent of cell removal at high flow, whilst physical and chemical/biological mechanisms were involved at low flow. At low flow, the biofilters decreased *Microcystis* cell numbers by 70% compared to the controls. The decrease in cell numbers in the biofilters was accompanied by a chlorotic process (loss of green colour), suggesting oxidative processes by the release of allelochemicals from the biofilters.

Keywords: biofilters, biofiltration, *Phalaris arundinacea,* allelopathy, cyanobacteria removal, *Microcystis* removal

1. Introduction

Eutrophication affects inland and marine waters worldwide since the boom of intensive agriculture practices after the Second World Word with typical manifestations of algal (algae and cyanobacteria) blooms (Le Moal *et al*, 2019; Qin *et al*., 2019). Water scarcity leading to shortages of potable water due to climate change is a reality (Flörke *et al*. 2018). Furthermore, toxic algal blooms frequency will continue to increase with rising temperatures due to climate change (Huisman *et al*., 2018). Not only do algal blooms threaten water quality with significant economic, biologic and public health consequences (Martinez-Hernandez *et al*., 2009; Hudnell, 2010; O'Neil *et al.,* 2014; Ger *et al.,* 2014; Otten and Pearl, 2015) but they can cause significant process disruption and reduce output from water treatment works (WTW) up to 25% by interfering/clogging downstream processes (coagulation, slow sand and rapid gravity filtration).

Green technologies are widely used for wastewater and water reuse but are less frequently used to remediate eutrophication in reservoirs used for recreational or drinking water purposes. Field scale biofilters based on plant rhizofiltration of high-surface-area are used to absorb nutrient or metals from waste waters (Dushenkov *et al.,* 1995; Enley and Raskin, 2000). The Living-Filter, a floating constructed treatment wetland, was used in a surface reservoir to successfully reduce algal biomass prior to the treatment works for production of potable water (Castro-Castellon *et al*., 2016).

Bench-scale rhizofiltration studies have been limited to nutrient/metal uptake. Marchand *et al.* (2014) combined plants and biofilms for metal removal using planted and unplanted Bio-rack™ to increase copper uptake rate. Kurzbaum *et al.* (2014) separated the role of the roots and the associated biofilm for the removal of pollutants in a hydroponic system, whilst Weiss

53 *et al.* (2014) used recirculating metal-rich flows to investigate the effect of water flux through
54 hydroponic roots on metal removal. Removal mechanisms of cyanobacteria by biofiltration
55 processes have not been studied before.

56 In this study, filtration units of hydroponically developed *Phalaris arundinacea* (biofilters)
57 and synthetic filters of plastic material were used in a novel set-up under continuous flow
58 conditions to study cyanobacteria (*Microcystis aeruginosa*) cell removal by deposition
59 throughout the filter media (depth filtration). The aim of this study was to determine the
60 interactions of *Microcystis* cells with each type of filter, and to understand how the inflow
61 associated with *Microcystis* cells is processed by the filter media. The outcome of this work
62 will provide insights to improve future designs of field scale Living-Filters promoting the use
63 of green technologies in eutrophic waters.

64 ## 2. Materials and methods

65 ### 2.1 Operational system

66 An experimental mesocosm was set up to test if hydroponically developed roots can be
67 used in an in-reservoir pre-treatment process for removing cyanobacteria from the inflow prior
68 to the water treatment works. The mesocosm system consisted of triplicate filter units with
69 three types of filter media: one biofilter (i.e. *Phalaris arundinacea* roots) and two synthetic
70 monofilament filters (i.e. plastic three dimensional mesh). In addition, control units with no
71 filter media were included. *Microcystis aeruginosa 1450/3* was obtained from Culture
72 Collection of Algae and Protozoa (CCAP) (hereafter *Microcystis*) was cultured in
73 photobioreactors and later mixed with dechlorinated water to constitute the feed, which was
74 pumped to the filter and control units. The schematic of the mesocosm set up is displayed in
75 Figure 1.

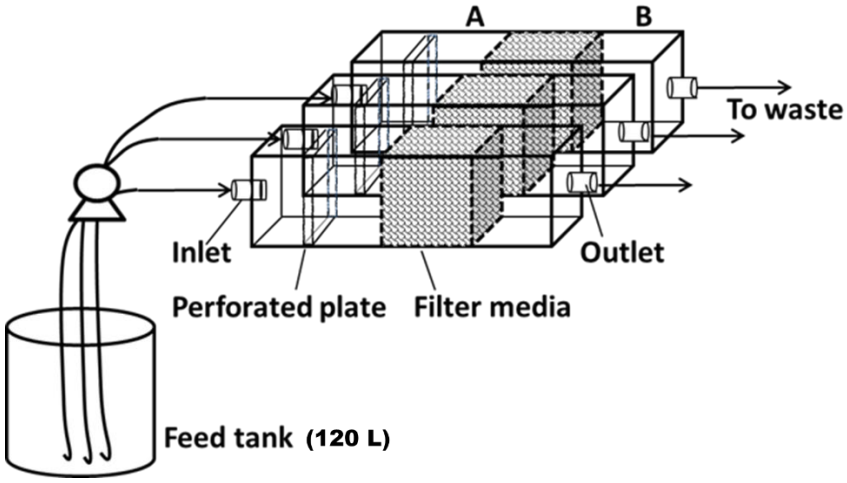


Figure 1. Schematic of the mesocosm set up showing the feed tank (120L) and only three filtration units (3 L each). Top unit with sampling points, where: A=upstream and B=downstream of the filter; bottom unit showing inlet and outlet of the units, the perforated plate and the position of the filter media.

The filtration units (0.32 m L x 0.104 m W x 0.11m H) were designed in collaboration with Tuan Ta Ltd., London, UK and made of acrylic transparent material with an internal perforated plate placed at 0.05 m from the inlet. The units were operated in plug-flow mode, with water supplied from an air-mixed 120 L high density polyethylene tank through nine lines of Marprene long life flexible tubing (1.6 x1.6 mm). The inflow was controlled by nine pump cassettes in three peristaltic pumps (Watson-Marlow Series 500 x2 and a Series 325).

2.2 Hydraulic configuration of the filtration units

Two flow velocities were chosen for running the experiments. A high flow rate (54±3.0 $cm^3\ min^{-1}$) with a filtration rate of ~ 0.29 m h^{-1} was chosen to resemble the range of filtration rates for slow sand filters (0.3 - 0.6 m h^{-1}) as slow sand filtration is an effective ecological process in supply water treatment (Campos *et al*. 2002). A low flow rate (1.3±0.2 $cm^3\ min^{-1}$) was chosen to investigate whether deposition mechanisms could take place in the filter media.

Laminar flow in the system becomes mixed in contact with the filter media. The hydraulic properties of the system are summarized in Table 1.

Table 1. Hydraulic properties of the filter units

Experimental flow	Cross Sectional Area (m^2)	Volume (m^3)	V=Q/A ($m\ h^{-1}$)	Q ($cm^3\ h^{-1}$)	HRT= Vol/Q (s)
Low	$1x10^{-2}$	$3.3x10^{-3}$	0.005	60	$2.008x10^{-5}$
High	$1x10^{-2}$	$3.3x10^{-3}$	0.29	$3.3x10^{-3}$	$3.3x10^{-3}$

V: velocity; Q: discharge; HRT: hydraulic retention time; Vol: volume

2.3 Fluid flow of *Microcystis* cells through biological and synthetic filter media

Three types of filter media were compared: the living roots of hydroponically grown *Phalaris arundinacea* and two synthetic monofilament fabrics with different packing density (fabric1and fabric 2). The packing density for the filter media was estimated from Equation (1):

Packing density = volume occupied by roots or fabric/ total volume (10 cm^3) (1)

Stainless steel cages (10 cm^3) were made to contain the biofilters and synthetic media, and empty cages were placed in the control units. The wire diameter was 1.1 mm with an aperture width of 14 mm.

2.3.1 Biofilters: hydroponic growth of Phalaris arundinacea

Seeds of *Phalaris arundinacea*, a species from the Poaceae family known as Reed Carnary Grass, were obtained from British Wildflower Plants, Norfolk, UK and cultivated in sterile coconut coir pellets. After two weeks, the roots of plantlets were rinsed, rolled up individually in strips of foam and transferred to 50 cm^3 Falcon™ tubes. To maximize the production of hairy roots, the plantlets were cultured hydroponically in Hoagland's solution at 50% concentration. The tubes were inserted with a hollow plastic straw to facilitate gas interchange and the supply of the solution, and were refilled every three/four days. The plants were placed

in a growing tent (1.5 m H x 1 m W x 1 m D) equipped with two blue 125 watts compact fluorescent lamps (CFL) and a Sun Mate Grow CFL reflector. The photoperiod was adjusted to a 10:14 hr light/darkness cycle (Conn *et al.,* 2006). The photosynthetic active radiation (PAR) of 40 µmol photons m^{-2} s^{-1} (30-45 µmol photons m^{-2} s^{-1} is recommended) was measured with a terrestrial quantum sensor LI-190SA and light meter LI-250A (Li-Cor International, Ltd., UK). The tubes were covered individually with aluminium foil to prevent light damage to the roots.

After 10 weeks, 48 plants (roots 8-10 cm length) in sets of 16 were placed on top of the stainless-steel cages and transferred to 2.1 L tanks. The root biovolume was estimated by rolling them into a plasticized sheet forming a cylinder, and the cylinder's volume was calculated and recorded (Faulwetter *et al.,* 2013).

2.3.2 Synthetic filter media: two types of monofilament nonwoven plastic material

The synthetic filtration media (polyamide nylon) consisted of two types of monofilament nonwoven plastic material of different packing density. These synthetic media were tested to compare their filtration removal efficiencies for *Microcystis* with that of the biofilters. The plastic monofilament layers are bonded by heat to create a sheet of fabric. Differences in flexibility that exist between fabric1 and fabric2 are based on the diameter of the filaments and the number of filament layers that create the sheet of fabric. The filament diameter of fabric1 and fabric 2 is 0.5 and 0.03 mm respectively. Fabric1 is a two layered filament sheet of 0.8 mm thickness and fabric2 is a multi-layered filament sheet of 1.8 mm thickness. Six stainless steel cubic cages (10 cm^3) were made to contain the fabrics, which were cut in squares of 10 cm^2 (10 cm x 10 cm). Three cages used for fabric1 were tightly filled with 15 pieces each and another three cages for fabric2 were filled with six squares each. The pieces were placed in the

cages with the largest surface area facing the direction of the flow. New cages were made and fabric cut for each flow experiment.

2.4 Photobioreactors for growing *Microcystis*

*Microcystis w*as cultured in BG11 (modified by Ripka 1979). Aseptic techniques and materials were used throughout the experiment. Four 150 cm^3 sterile flasks each with 50 cm^3 media were spiked with 1cm^3 of the *Microcystis* stock culture and kept on the bench under fluorescent laboratory lights providing photosynthetic active radiation (PAR) of 10 µmol photons m^{-2} s^{-1} (recommended by the CCAP) (Imai *et al.,* 2008). The flasks were shaken manually every three-four days and tested weekly for contamination prior to adding 10 cm^3 of fresh media, maintaining *Microcystis* at the exponential growth phase. An aliquot of 5 cm^3 of the *Microcystis* suspension was used as inoculum to grow larger quantities in photobioreactors. The photobioreactors consisted of 1 L Duran bottles, placed in an open water bath with a thermostat keeping the temperature at 22±1ºC. The bottle mouth was closed with a sterile foam plug covered with aluminium foil. To the initial volume of 300 cm^3, 200-250 cm^3 of BG11 was added weekly up to 1 L. Air was diffused into the solutions at a rate of 0.1 – 0.4 L min^{-1} with a daily cycle (12:12 on/off) divided into four intervals. An additional fluorescent lamp provided a range of 35 µmol photons m^{-2} s^{-1} to the closest photobioreactors and a minimum of 10 µmol photons m^{-2} s^{-1} to the furthest, and the photoreactors were rotated weekly.

Quality control was conducted by assessing microscopic morphology of *Microcystis* cells and conducting triplicates of cell counts from each bioreactor on a haemocytometer at x400 magnification using an inverted microscope (Imai *et al.,* 2008). *Microcystis* culture optical density (O.D) was measured in triplicates at $O.D_{620}$ and $O.D_{750}$ nm (Vezie *et al.,* 2002; Dagnino *et al*., 2006) in a Shimadzu 1800-UV spectrophotometer. To ensure measurable changes in the

bioreactors, a minimum $O.D_{620}$ was estimated to be 0.3 (~6.5 x 10^{-6} cells $cm^{3\ -1}$) with the resulting calibration Equation (2):

$$Y=1E+07x + 504384 \quad (2)$$

To ensure there was no bacterial growth monitoring changes at $O.D._{750nm}$ were conducted.

2.4.1 Microcystis as the particles in suspension

Microcystis cells are spherical particles with an average size of 3.2 µm± 0.8µm (*n*=30), obtained under light microscope (Eclipse E-200 Nikkon). Cells that were in division, representing approximately 20% of the cell population, were also included in the average size.

An increase in the concentration of *Microcystis* cells is expected upstream of the filter media (see Figure 1). The particle concentration in plug-flow mode can be expressed as a dimensionless parameter, which represents the ratio of the particle concentration upstream of the filter media relative to the inflow to the filtration unit following Equation (3),

$$\text{Cumulative mass fraction (CMF)} = C_m/C_f \quad (3)$$

Where: CMF is the cumulative mass fraction; C_m is the concentration of cells on the upstream side of the filter at sampling point A (figure 1) and C_f is the concentration of cells in the inflow upstream of the filter unit at the inlet (figure 1). This parameter will indicate the filtration mode of the filter media (US-EPA, 2005). Hence CMF =1 indicates an operating system in deposition mode. The cells are moving with the inflowing water at a steady-rate before passing through the filter media. CMF ≥1 indicates there is a scouring force applied tangentially upstream to the media, and CMF ≤1 suggests the system operates in deposition filtration mode.

2.4.2 Microcystis cell count with flow cytometry

Microcystis cell concentration was first estimated by manual counting on a haemocytometer at x400 magnification on an inverted microscope. The large number of samples generated and the poor count discrimination of low O.D readings (<0.015) led to the use of flow cytometry for subsequent cell counts using a BD Accuri C6 (BD Biosciences, UK).

The fluidic system in the instrument is designed so that the suspended cells in the sample are delivered one by one to a specific point with the illuminating beam. The velocity of the samples loaded into the channel was set at 35 µl min^{-1}. The instrument measures the light scattered by the cells at right angles to the laser beams (called side scatter, SS) and light scattered in a forward direction (forward scatter, FS). The size and shape of the cells affects the forward scatter whilst small structures (internal or external) of the cells affect more the SS. The forward scatter threshold was set up to 15000 events, this mean that some debris and instrument noise is ignored. The instrument is equipped with blue (488 nm) and red (638 nm) excitation lasers and four emission filters (Table 2). The data were displayed using density dots for two parameters using a bivariate histogram, or cytogram (C), where the dot density of a population of cells forms a specific shape called a region (Dubelaar and Jonker, 2000). A region can be drawn using a fluorescence parameter to define the population of interest (signature), or a region can be used to limit the cells that are drawn on a light scatterplot (gate).

Table 2. Naturally occurring fluorescent pigments in phytoplankton and their detection on the BD Accuri C6.

Pigments	Excitation	Emission	C6 Detector
Chlorophyll *a, b*	488	>640 nm	FL3 (670 LP)
Phycoerythrin	488	575 nm	FL2 (585 ±20)
Phycocyanin	640	650 nm	FL4 (675 ±12.5)
Allophycocyanin	640	646 nm	FL4 (675 ±12.5)

A manual gate was drawn around the cell population on a plot of chlorophyll-a fluorescence (FL3; 488 nm excitation, 640 nm emission) versus phycocyanin fluorescence (FL4; 640 nm excitation, 650 nm emission) and was used to discriminate and count the cells against volume calibrated fluidics.

2.5 Testing the concept of *Microcystis* removal by biofilter roots.

To test for *Microcystis* cell removal by the biofilters (roots of living plants), the mesocosm experiments were conducted under different continuous flow conditions. At high flow (54±3.0 cm^3 min^{-1}) with biofilters and synthetic Fabric 1 and 2. Fabric2 had higher packing density and was used as positive control with units as negative controls. At low flow (1.3±0.2 cm^3 min^{-1}) the design included biofilters, one type of synthetic fabric (fabric1) and negative controls. The two flow conditions and three filter media treatment were each run in triplicates (Figure 2).

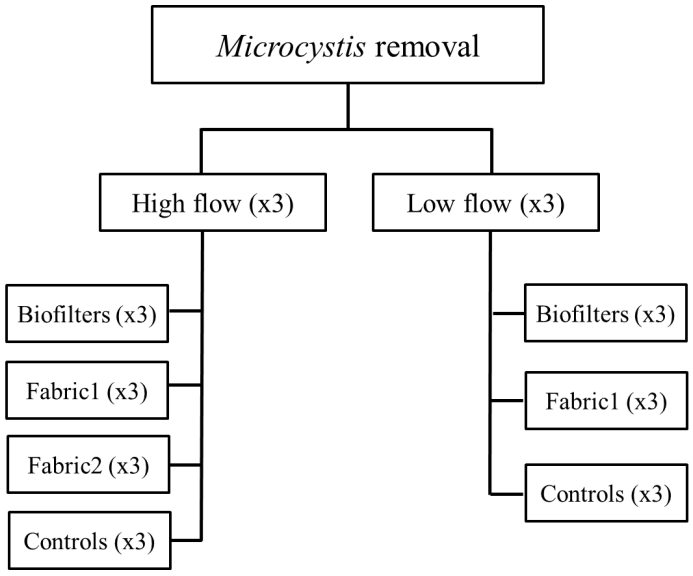


Figure 2. Schematic of the experimental mesocosm design at two flow conditions for biofilters, synthetic fabrics (fabric 1 and 2) and controls (x3=triplicates).

Prior to starting each of the experiments (high and low flow) the filtration units filled with dechlorinated water and were left to stabilize for 2-3h (high flow) and 48h (low flow). The

outflow was calibrated to 54±3.0 cm^3 min^{-1} and 1.3±0.2 cm^3 min^{-1} for high and low flow respectively. Replicates of the experiment were run for five, seven and 11 days, which corresponded to two, three and approximately five hydraulic retention times. A 12w LED submersible lamp with blue-red-white emission 400-700 nm, 800 lux providing approximately 20.6 µmol photons m^{-2} s^{-1} was introduced into the feed tank after being thoroughly disinfected with 1 ppm chlorine solution and rinsed with sterilized (autoclaved) deionized water for the 11 day-run experiments. The high flow experiments ran for five hours.

Daily sampling (1 cm^3) was carried out for the low flow experiment at the sampling points shown in Figure 1: feed tank (x3); sampling points A (x1), B (x3) (depths 1, 5 and 9 cm); and C (x1) for each filtration unit. Samples were taken every 55 min from the feed tank (x3), A and B (x1).

2.5.1 Removal efficiency

Removal efficiency as a percentage (RE%) was calculated for every paired inflow-outflow sample taken from the filtration units. This parameter is calculated with the assumption of a similar inlet and outlet flow rate, Equation (4)

$$\%RE = \frac{(C_0 - C)}{C_0} \times 100 \quad (4)$$

where: C_0 is the concentration of *Microcystis* cells at the inlet and C is at the outlet.

2.6 Data processing and statistical analysis

Derived variables (ratio, percentages and rates) and exploratory statistical analysis were undertaken using Excel (Microsoft © 2010) and SPSS v. 22 software. Data were transformed to meet the normal distribution and other parametric assumptions, and if assumptions could not be met, non-parametric tests were used. The non-parametric Friedman's test was applied to

find differences between treatments and the Wilcoxon-Sign-Rank test was applied to test for differences in the number of *Microcystis* cells in the unit's inflow and outflow.

2.7 Investigating chemical mechanisms: allelopathy

Further experimental work was required to understand what quantitative and qualitative mechanisms were taking place during the low flow experiments and determine if filtration alone or in combination with allelopathy was contributing to the removal of cells in the biofilters (Hilt and Gross, 2008; Rojo *et al*. 2013). Two composite root exudate samples (from three biofilters) were screened and analysed using gas chromatography coupled with mass spectrometry (GC-MS) to identify potential allelochemical compounds at Wessex Water Scientific Centre, Bath, UK; a United Kingdom Accredited Systems laboratory.

2.7.1 High pressure liquid chromatography: sample preparation and analysis

To identify allelochemicals in the root exudate and root extraction from biofilters a new method was developed using high pressure liquid chromatography (HPLC) at Wessex Water Scientific Centre. Six chemical compounds (anthraquinone, gallic acid, gramine, hordenine, 4-5 indole-aldehyde and stigmasterol) were purchased from Sigma-Aldrich. These compounds are known to be produced by *Phalaris arundinacea* and were selected based on their algaecide/algastatic properties against cyanobacteria and micro-algae (Hong et al., 2009; Xia et al., 2009; Shao et al., 2013). The compounds were used as standards, and all the stock solutions were prepared at 1 mg L^{-1} concentration in methanol. Individual (1:100) and a mix standard (6:100) in ultrapure water (UPW) solutions were prepared for extraction. Root exudate samples of 100 cm^3 taken from each biofilter were concentrated by a solid phase extraction (SPE) step using a Visiprep (Supelco™ – Sigma-Aldrich). Roots of three plants were cut coarsely with a blender, adding 100 cm^3 of 30% methanol in deionized water (MilliQPore system) and concentrated by SPE. All samples were eluted from the cartridges with 1 cm^3 of

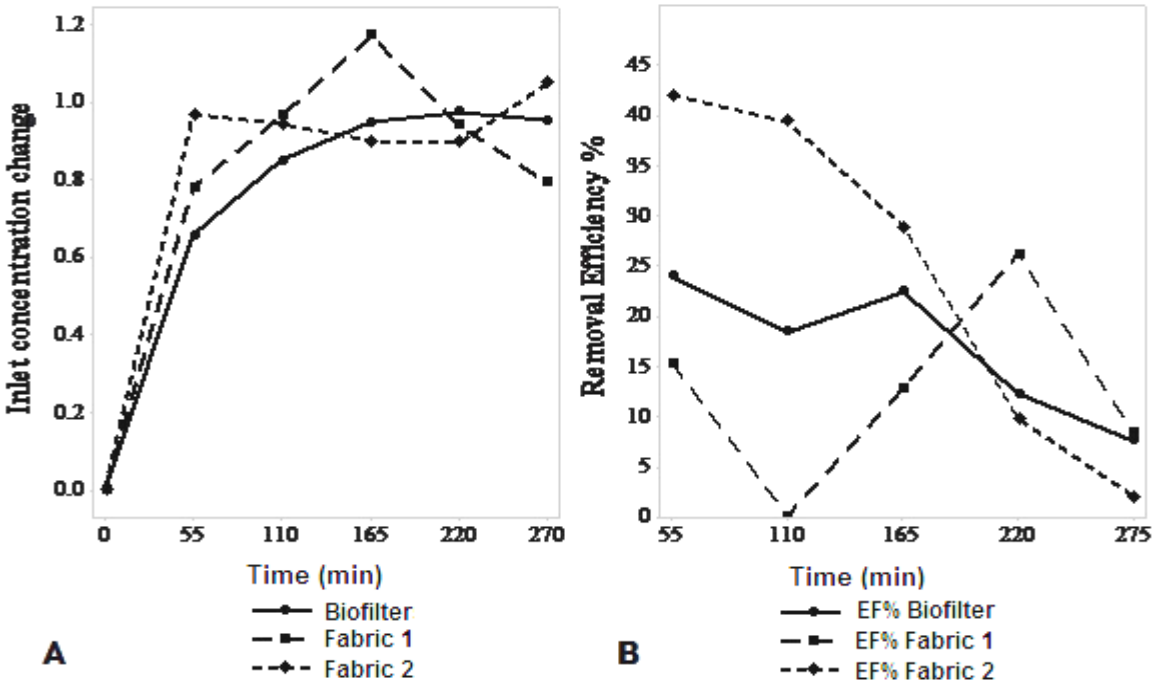
1% formic acid in methanol; the extracts were air dried (0.8 L min^{-1}) using a Visidry (Supelco – Sigma-Aldrich); and the dried residue was resuspended with 1 cm^3 10% acetonitrile: 0.1 % acetic acid in ultrapure water (UPW).

An Agilent 1200 LC series system was used with UV/Vis-DAD detector. A HPLC Agilent column (C18, 150 mm x 5 mm) was used to achieve separation with a gradient elution consisting of acidified (acetic acid) acetonitrile: acidified (acetic acid) water (95:5). Column temperature was maintained at 40°C and total run time was 45 min. The UV/Vis absorbance detector collected data at three wavelengths 205, 250 and 280 nm. Peak identity was validated through the use of retention times of external non-extracted and extracted standards with their respective spectrums All samples were kept at 5°C at all times.

3. Results

3.1 *Microcystis* cell removal during high flow rate experiment

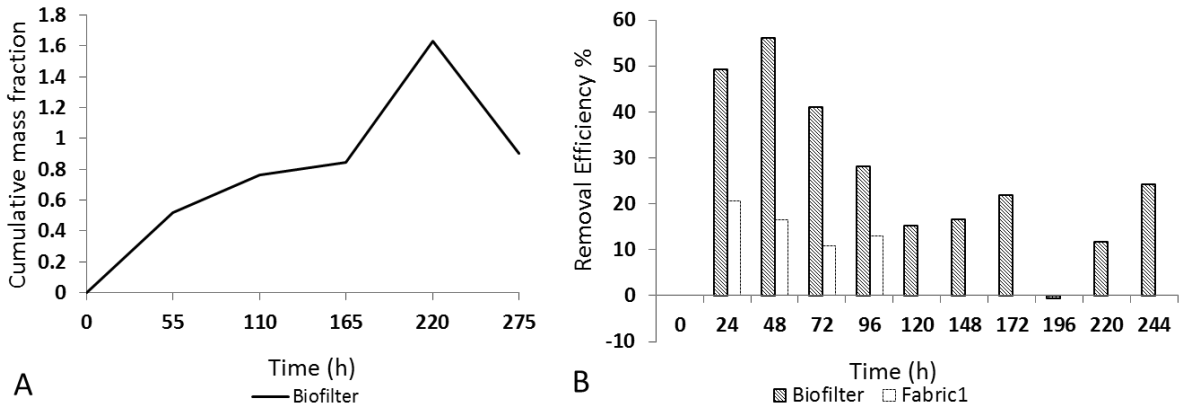
The packing densities of the synthetic filters with fibrous media were 0.03 for fabric1 and 0.07 for fabric2. The estimated packing density of the biofilters was 0.05. The cumulative mass fraction of biofilters and fabric2 was ≤1 with the inflowing cell concentration, suggesting the system operates in deposition filtration mode for both type of fibrous media (Figure 3-A). The biofilter removal efficiency (RE%) of *Microcystis* cells is presented in Figure 3-B.



Microcystis cells were effectively removed from the biofilters and fabric2 (higher packing density) as shown by the decrease in numbers when comparing cell numbers from the outflow to the inflow (Wilcoxon-Sign Test T=8, z=-4.34, p<0.5, r=-0.47 for biofilter and T=5, z=-0.46, p<0.5, r=-0.47 for fabric2.

3.2 *Microcystis* cell removal during low flow rate experiments

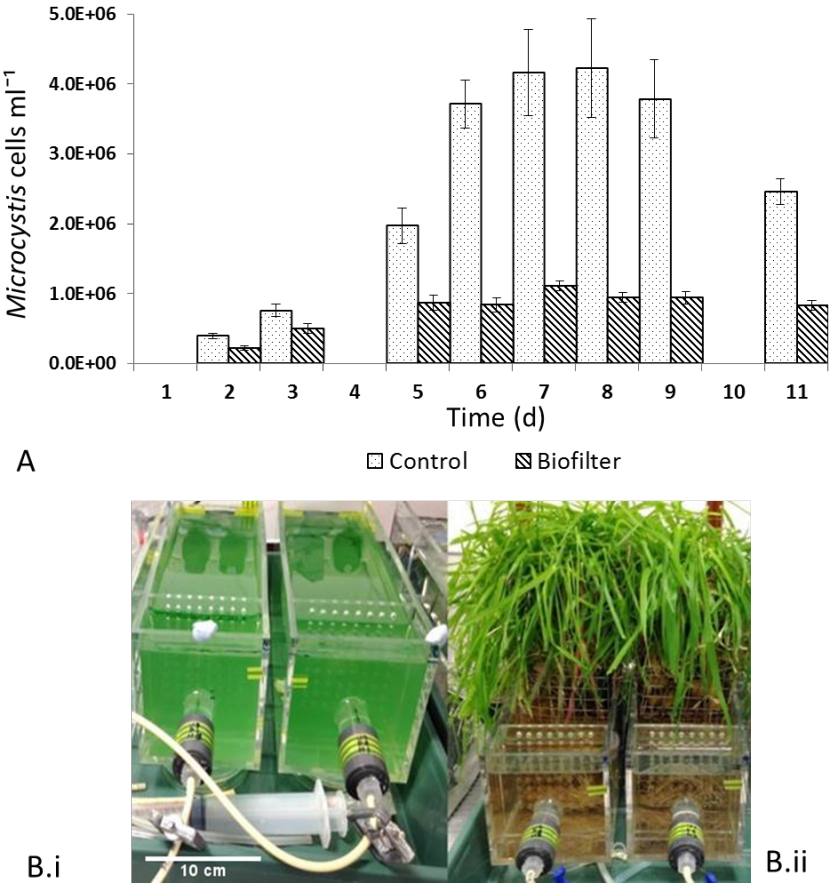
Results for *Microcystis* removal shown in Figure 4 correspond to the experimental run of five hydraulic retention time (5 HRT). The biofilter cumulative mass fraction was >1 at 4HRT before dropping rapidly (Figure 4A). Fabric1 results shown are from the run of 3HRT (Figure 4-B). The biofilters showed removal efficiency of 40- 55% within the first three days, dropping to 10-20%; after three days RE for fabric1 was 8-20% (Figure 4-B).



297 **Figure 4-A. The cumulative mass fraction for the biofilters (solid line as a function**
298 **of time (hours) equivalent to hydraulic retention time. 4-B Removal efficiency (%) of**
299 ***Microcystis* cells by the biofilter in relation to time (hours).**

300 Colour change in the units with biofilters, fabric and the controls were observed. Results
301 are shown for the biofilters and controls in Figure 5-A-B. The water in the biofilters is
302 colourless, indicating the visual absence of *Microcystis* cells; while the fabric units and controls
303 appear green, rich with cells. The same qualitative pattern was observed for all replicates of
304 the experiment at low flow (run at 2 HRT and 3 HRT (x2)).

305 Quantification of *Microcystis* cells (by flow cytometry) was four and a half times higher
306 in the controls and fabric1 (low packing density) when compared to the biofilters. The number
307 of cells from the feed tank increased every 48-72 hours (results not shown) suggesting cell
308 growth and conditions in the tank adequate for survival. By contrast the number of cells in the
309 biofilters remained at $1x10^6 \pm 2x10^5$ ml^{-1} from day five onwards (Figure 5-A). Comparative
310 results between controls and biofilters are considered only until day nine because a slight
311 decline of *Microcystis* cells was observed in the controls after this day.



313 **Figure 5-A. Quantitative changes in *Microcystis* average concentration (#cells ml^{-1})**
314 **for a mesocosm experiment run for 11 days or 5HRT. Controls (stipple), Biofilters**
315 **(striped), number of samples is n=3 for each day. Error bars represent standard errors.**
316 **5-B. Qualitative changes in colouration: green control units (B.i) indicating the presence**
317 **of *Microcystis* cells versus colourless biofiltration units (B.ii).**

318 Significant differences in cell numbers were found between the inflow and outflow in the
319 biofilters, with higher counts in the inflow than in the outflow (Wilcoxon sign-test T=5, z=-
320 5.164, p<0.05, r=-0.53). No significant differences in the number of *Microcystis* cells were
321 found in the controls.

3.3 Investigating chemical mechanisms of *Microcystis* removal by biofilters

3.3.1 Allelochemical presence and HPLC-UV/VIS-DAD

Allelochemicals (i.e. phenols) and other secondary metabolites (i.e. cholesterol) were found in the screened samples. Table 3 shows the identified compounds with the GC-MS screening from the root exudate samples.

Table 3. GC-MS screening results of water samples from the filter units

Chemical	Concentration (ng dm^{-3})
Stigmasterol	660
Phenols	620
Salicylates	440
Cholesterol	250
β-cyclocitral	220

Standards with their respective retention times from the developed method to identify allelochemicals by HPLC-UV/Vis-DAD are shown in Table 4.

Table 4. Limits of detection and quantification for the selected analytes.

Analytes from standard solution	Limit of Detection (µg L^{-1})	Limit of Quantification (µg L^{-1})
Hordenine	3.63	11.01
Gramine	13.46	40.78
Naringin	9.11	38.86
4-nitroindole-5-carboxaldehyde	14.83	44.93
Anthraquinone	29.20	88.48

Table 5 compiles the identified compounds: gramine and 4-nitroindol-5-carboxaldehyde. Hordenine could not be traced in the mixed standard sample.

Table 5. Allelochemicals quantified in root exudates and root extracts (n=3).

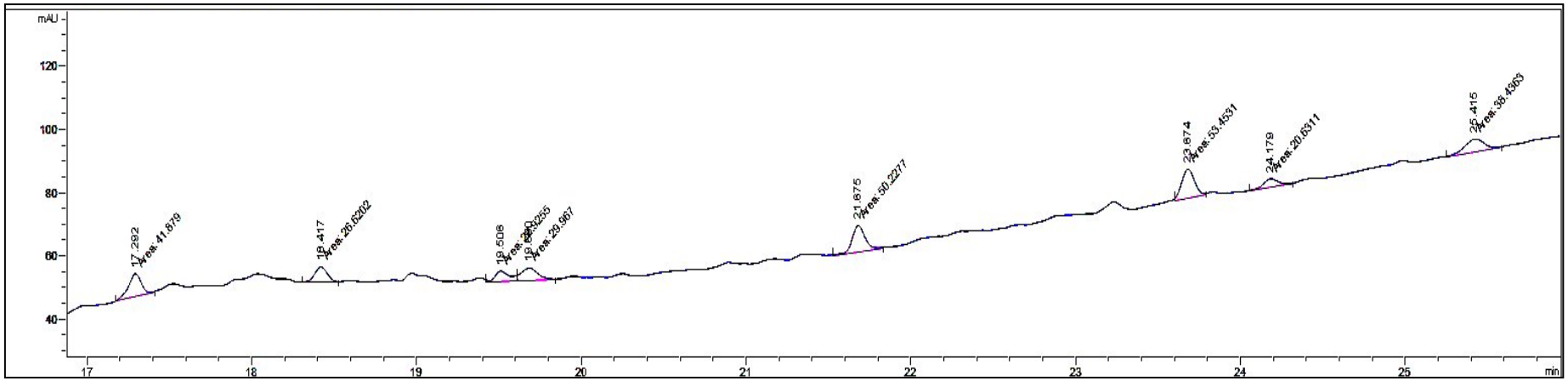
Allelochemicals	Root exudate mean±SE µg L^{-1}	Root extract mean±SE µg L^{-1}
Gramine		3447±1043
Naringin (ISTD 100 µg L-1)	109.3±9.9	4688±1433
4-Nitroindol-5-carboxaldehyde	96.13±10.1	176±10.1

335

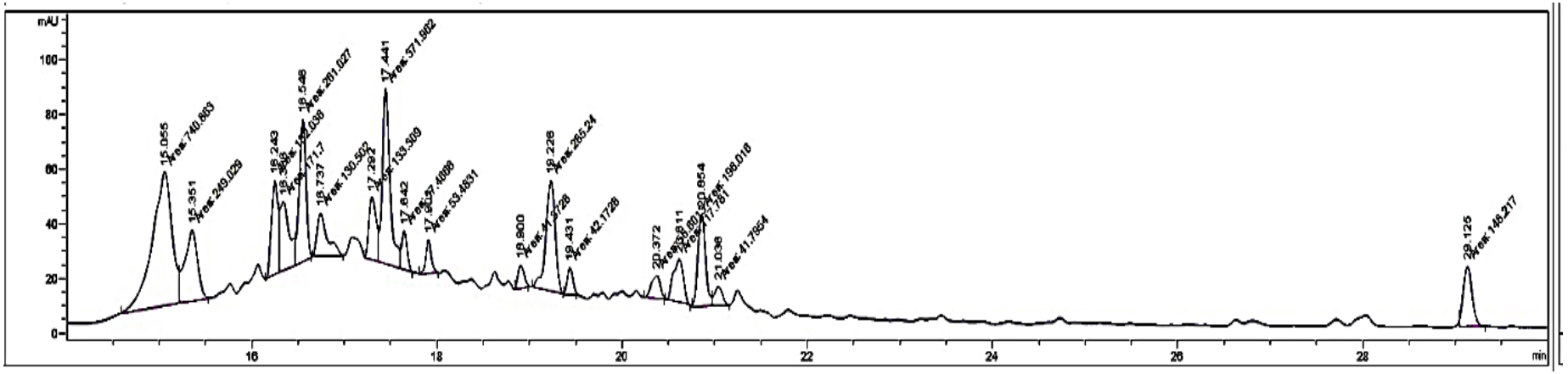
336 Figure 6 shows the chromatogram of a root exudate sample and root extracted sample.
337 There are a large number of unidentified compounds in the samples. Spectrums of the
338 unidentified peaks and their retention times might be compatible with phenolic compounds.

339

340


mAU
120
100
80
60
40
17.292
Area:41.879
19.417
Area:26.6202
19.506
Area:29.967
21.875
Area:50.2277
23.674
Area:53.4531
24.179
Area:20.6311
25.415
Area:38.4563
17
18
19
20
21
22
23
24
25
min

341


mAU
100
80
60
40
20
0
15.055
Area:740.863
15.351
Area:249.029
18.243
18.546
Area:261.027
18.737
Area:171.7
18.900
19.226
Area:265.24
19.431
Area:42.1726
17.441
Area:371.862
17.642
Area:153.309
Area:57.4609
Area:53.4531
Area:150.502
20.372
Area:198.016
Area:41.7854
28.125
Area:145.217
16
18
20
22
24
26
28
min

344

4. Discussion

Microcystis is a known bloom-forming cyanobacteria found worldwide in surface water reservoirs, and there has been increasing interest in eco-biological/rhizofiltration systems for its removal. The purpose of this study was to investigate whether *Microcystis* cells could be removed by the roots of *Phalaris arundinacea* under continuous flows and to establish the interactions between inflowing *Microcystis* cells with biofilters and synthetic filters. The distribution of the cells/ compounds generated in the experimental system will are known to be affected by hydrological processes (Alcocer *et al.,* 2012; Ruggeri and Sassi. 2013). Although it is assumed a system will reach steady-state in hydraulic-biochemically mediated processes, steady-state is rarely achieved in either plug-flow or continuously stirred systems (Poitier *et al.,* 2005). At water or sewage treatment plants, where is very difficult to predict the load and environmental changes, the system typically does not reach equilibrium (Davis and Conwell 2013). The system studied in this research did not reach steady-state within the experimental period.

4.1 *Microcystis* cells removal during the high flow experiment

Microcystis cell behaviour (as suspended particles) in relation to natural and synthetic media (natural and synthetic) suggests a deposition filtration mode or cumulative mass fraction (CMF=1) (Figure 3-A). Differences in the cumulative mass fraction at different retention times initially showed the cells through the biofilters reach CMF=1 at 220 min compared to fabric1, where CMF=1 at 55 min. For removal efficiency, the biofilters were capable of removing 20 - 25 % of the *Microcystis* cells during three retention times. The most effective filter media was fabric2 (synthetic filter with higher packing density than the biofilters and fabric1) with a maximum removal efficiency (%) slightly above 40% compared to 25% for the biofilters and

368 to17% for fabric1 at 55 min. These results show that there is a physical removal mechanism of
369 the *Microcystis* cells by the biofilters, which can be related to packing density.

370 **4.2 *Microcystis* cell removal during the low flow experiment**

371 In the higher contact time experiment (i.e. low flow) the biofilters showed removal
372 efficiencies twice as high as those observed under high flow conditions, suggesting that the
373 increased exposure to the hydroponic rhizofiltration may contribute to higher removal
374 efficiencies. At lower flowrates, the cell numbers in the feed tank doubled between 48-72
375 hours, following a growth curve. A Growth Model fitted to the cell number data in the feed
376 tank explained 95% of the variance of the data (results not shown). This growth could also have
377 affected results for the CMF (Figure 4-A), as an increasing cell concentration would directly
378 affect the ratios. The number of cells in the controls $\geq 3.0 \times 10^6$ cells ml^{-1}, was at least twice as
379 high as in the biofilters where they did not increase above 1.2×10^6 cells ml^{-1} (Figure 5-A).
380 Hence it was postulated that besides the physical removal chemical mechanisms may
381 contribute to the removal of Microcystis when exposed to biofilters i.e. rhizofiltration.

382 ***4.2.2 The role of the biofilters on Microcystis removal***

383 The most striking results found were the loss of colouration (bleaching) and the reduction
384 in cell number in the biofilters when compared to the control and fabric units. “Bleaching” or
385 chlorosis is defined in the literature as the change in pigmentation from blue-green to yellow-
386 green to yellow (or orange) (Collier and Grossman 1992; Baier *et al.,* 2014) in non-N_2 fixing
387 cyanobacteria (like *Microcystis*) under nitrogen starvation conditions. Chlorosis is a process
388 generally described after 72 hours of nitrogen starvation in cultures of cyanobacteria model
389 organisms such as *Synechocystis* and *Synechococcus* (Krasikov *et al.et al.,* 2012). However,
390 there is no reference in the literature to the chlorotic process as a complete lack of colouration
391 (clear as tap water) as observed in the biofilters (Figure 5). The chlorotic process of *Microcystis*

392 in the biofilters is observed after 72 hours, and at 96 hours there is a total absence of colouration
393 with no-absorbance spectrophotometric detection at $O.D_{620}$ and $O.D_{680}$ (results not shown).
394 Dagnino *et al.,* (2006) described a chlorotic process in nutrient-depleted *Microcystis* cultures
395 which after 3-5 days showed lack of colouration, but from blue-green to very pale blue-green
396 and decreasing cell density. They attributed this phenomenon to intercellular signalling present
397 in the medium of the nutrient-starved *Microcystis*. Intercellular signalling is a chemically
398 mediated process between microorganisms (e.g. cyanobacteria and algae/organisms of higher
399 taxa) known as allelopathy (Gross *et al.,* 2003). In this study, decrease in cell numbers was also
400 seen, measured and compared between the biofilters and the controls. These results suggest
401 that either nutrient competition between the roots in the biofilters and *Microcystis* cells or
402 allelochemical mechanisms with inhibiting/biocide effects (or both) induced chlorosis in the
403 *Microcystis* cells. It is important to point out that the *Microcystis* cells were continuously
404 injected to all the units in increasing concentration, but the only units with loss of colouration
405 or reduction in cell numbers were the biofilters.

406 **4.3 Investigating allelochemical in roots and root exudate**

407 The results of this study suggest the chlorotic effect and reduction in *Microcystis* cell
408 numbers in the biofilters could be triggered by allelopathy. Allelochemical compounds with
409 potentially algistatic/algaecide effects (Laue *et al.*, 2014) were identified in the root exudate at
410 nanogram concentrations (Table 2). These results mimic natural aquatic environments, where
411 the allelochemicals excreted by macrophytes are diluted in the surrounding water (Hilt and
412 Gross, 2008; Rojo *et al.,* 2013), but where they still have a suppression effect on phytoplankton
413 growth rates (Korner and Nicklisch, 2002; Mulderij *et al.,* 2007; Chang *et al.*, 2012). In water,
414 the distances between cells are crucial as the chemical information is transmitted by diffusion
415 and advective laminar flow (Wolfe 2000). By contrast, laboratory experiments are generally

carried out using *Microcystis* cells at concentrations of $1x10^5$-$1x\ 10^6$ µl in the presence of µg or mg concentrations of allelochemicals either extracted from the roots or supplied commercially to demonstrate their biocidal effect (Lurling *et al.,* 2013; Lurling and Van Oosterhout 2014). However, allelopathy might not be only produced by the roots as any bacteria and fungi present could be sources of allelopathic chemicals. Nevertheless, studies on the detection and identification of allelochemicals in root tissue or root exudate from *Phalaris* spp. let alone *Phalaris arundinacea*, have not been described in the literature.

The allelochemicals in the root exudate showed the presence of gramine and 4-nitroindol-5-carboxaldehyde with the targeted method developed for allelochemical detection (Figure 6). However, there were additional unidentified compounds present in the root exudate and a large number in macerated root samples, which could potentially be released by the roots into the water. It is envisaged in future experiments to increase the number of allelochemicals in the mix-standard to enable further identification. However, this does not mean that all compounds seen in the sample will have an algaecide/algistatic effect on *Microcystis*, and further investigation will be required to distinguish the impact of individual compounds on *Microcystis* cells.

5. Conclusions

Microcystis can be removed by the roots of *Phalaris arundinacea* under continuous flow conditions, but the mechanism could vary depending on residence time and packing density on synthetic/natural filter media. At higher residence times on natural filter media the decrease in *Microcystis* cell numbers is accompanied by a chlorotic process, indicating physicochemical mechanisms operate in natural filters compared to high flow where physical deposition was the main removal mechanism. This highlights the importance of creating pilot/field scale designs to allow low flow velocities and/or large retention times, and

increasing the probability of contact between the allelochemicals produced by the roots and the *Microcystis* cells.

Overall, at low residence time, it was shown that the packing density of the filter porous media plays the dominant role in *Microcystis aeruginosa 1450/3* removal in both biofilters and synthetic filter media. The packing density is a relevant factor to be considered in future Living-Filter systems, if these are to be used as a pre-treatment process for phytoplankton removal.

Physicochemical interactions between living roots and *Microcystis* in hydroponic and continuous flow conditions have not been described in the literature before. Findings from this experimental set up are more realistic at demonstrating these physicochemical interactions providing grounds for further experimentation, either with other phytoplankton or other plant species. Additionally, the biofiltration set-up used in this study can be used to investigate and optimize the removal of pharmaceuticals by living roots.

6. Acknowledgments

This project is fully sponsored by Thames Water Utilities Ltd. The views of the authors are not necessarily those of the sponsor. The authors thank specially Dr Paul Rutter for his support and Nick Sutherland for assisting with the experimental set-up logistics. Lady Margaret Hall, University of Oxford sponsored attendance to conferences where aspects of this work were presented. Thanks to Helen Shapland and Kevin O'Reilly, Trace Organics laboratory, Scientific Center, Wessex Water Ltd for allowing us the use of their HPLC. The authors are especially grateful to Toni Hall, Alexander T. Jennings and Gary Hunt for their assistance with developing the HPLC allelochemicals method.

7. References

Alcocer, D. J. R., Vallejos, G. G., & Champagne, P. (2012). Assessment of the plug flow and dead volume ratios in a sub-surface horizontal-flow packed-bed reactor as a representative model of a sub-surface horizontal constructed wetland. *Ecological Engineering*, 40, 18-26.

Baier, A., Winkler, W., Korte, T., Lockau, W., & Karradt, A. (2014). Degradation of Phycobilisomes in *Synechocystis* sp. PCC6803 Evidence for essential formation of a NblA1/NblA2 heterodimer and its codegradation by a Clp protease comples. *Journal of Biological Chemistry*, 289 (17), 11755-11766.

Benton, T. G., Solan, M., Travis, J. M. & Sait, S. M. 2007. Microcosm experiments can inform global ecological problems. *Trends Ecol Evol,* 22**,** 516-21.

Blom, J. F., Brutsch, T., Barbaras, D., Bethuel, Y., Locher, H. H., Hubschwerlen, C. & Gademann, K. 2006. Potent algicides based on the cyanobacterial alkaloid nostocarboline. *Organic Letters,* 8**,** 737-740.

Castro-Castellon, A.T., Chipps, M.J., Hankins, N.P. and Hughes, J.M.R., 2016. Lessons from the "Living-Filter": An in-reservoir floating treatment wetland for phytoplankton reduction prior to a water treatment works intake. *Ecological engineering*, *95*, pp.839-851.

Chang, D. W., Hsieh, M. L., Chen, Y. M., Lie, T. F. & Chang, J. S. 2011. Kinetics of cell lysis for *Microcystis aeruginosa* and *Nitzschia palea* in the exposure to beta-cyclocitral. *Journal of Hazardous Materials,* 185**,** 1214-20.

Chorus I. & Bartram J. 1999 Toxic Cyanobateria in Water: A guide to their public health consequences, monitoring and management. World Health Organization, 440 p., E&FN Spon, London.

Collier, J. L., & Grossman, A. R. 1992. Chlorosis induced by nutrient deprivation in *Synechococcus* sp. strain PCC 7942: not all bleaching is the same. *Journal of Bacteriology*, 174(14), 4718-4726.

Conn, S. J., Hocking, B., Dayod, M., Xu, B., Atham, A., Henderson, S., Aukett, L., Conn, V., Shearer, M. K., Fuentes, S., Tyerman, S. D. & Gilliham, M. 2013. Protocol: optimising hydroponic growth systems for nutritional and physiological analysis of Arabidopsis thaliana and other plants. *Plant Methods,* 9(1), 1-11.

Dagnino, D., De Abreu Meireles, D., & De Aquino Almeida, J. C. 2006. Growth of nutrient-replete *Microcystis* PCC 7806 cultures is inhibited by an extracellular signal produced by chlorotic cultures. *Environmental Microbiology*, 8(1), 30-36.

Dushenkov, V., Kumar, P. N., Motto, H. & Raskin, I. 1995. Rhizofiltration: the use of plants to remove heavy metals from aqueous streams. *Environmental Science & Technology,* 29**,** 1239-1245.

Raskin, I. & Ensley, B. D. 2000. *Phytoremediation of toxic metals: Using plants to clean up the environment*, 304 p. John Wiley & Sons. Chichester, West Sussex.

Fan, J., Ho, L., Hobson, P. & Brookes, J. 2013. Evaluating the effectiveness of copper sulphate, chlorine, potassium permanganate, hydrogen peroxide and ozone on cyanobacterial cell integrity. *Water Research,* 47**,** 5153-5164.

Faulwetter, J. L., Gagnon, V., Sundberg, C., Chazarenc, F., Burr, M. D., Brisson, J., Camper, A.K. & Stein, O. R. 2009. Microbial processes influencing performance of treatment wetlands: a review. *Ecological Engineering*, 35(6), 987-1004.

Flörke, M., Schneider, C. and McDonald, R.I., 2018. Water competition between cities and agriculture driven by climate change and urban growth.*Nature Sustainability*,*1*(1), p.51.

Ger, K. A., Hansson, L.-A. & Lürling, M. 2014. Understanding cyanobacteria-zooplankton interactions in a more eutrophic world. *Freshwater Biology,* 59**,** 1783-1798.

Gross, E. M. 2003. Allelopathy of aquatic autotrophs. *Critical Reviews in Plant Sciences,* 22**,** 313-339.

510 Headley, T. R. & Tanner, C. C. 2012. Constructed Wetlands With Floating Emergent
511 Macrophytes: An Innovative Stormwater Treatment Technology. *Critical Reviews in*
512 *Environmental Science and Technology,* 42**,** 2261-2310.
513 Hilt, S. & Gross, E. M. 2008. Can allelopathically active submerged macrophytes stabilise
514 clear-water states in shallow lakes? *Basic and Applied Ecology,* 9**,** 422-432.
515 Hong, Y., Hu, H. Y., Xie, X., Sakoda, A., Sagehashi, M. & Li, F. M. 2009. Gramine-induced
516 growth inhibition, oxidative damage and antioxidant responses in freshwater
517 cyanobacterium *Microcystis aeruginosa*. *Aquatic Toxicology,* 91**,** 262-9.
518 Hudnell K.H. 2010. The state of US freshwater harmful algal blooms assessments, policy and
519 legislation. *Toxicon*, 55 (5), SI, 1024-1034.
520 Huisman, J., Codd, G.A., Paerl, H.W., Ibelings, B.W., Verspagen, J.M. and Visser, P.M., 2018.
521 Cyanobacterial blooms. *Nature Reviews Microbiology*, *16*(8), p.471.
522 Imai, H., Chang, K. H., Kusaba, M. & Nakano, S. I. 2008. Temperature-dependent dominance
523 of *Microcystis* (Cyanophyceae) species: *M. aeruginosa* and *M. wesenbergii*. *Journal of*
524 *Plankton Research,* 31**,** 171-178.
525 Jancula, D. & Marsalek, B. 2011. Critical review of actually available chemical compounds for
526 prevention and management of cyanobacterial blooms. *Chemosphere,* 85**,** 1415-22.
527 Körner, S., & Nicklisch, A. 2002. Allelopathic growth inhibition of selected phytoplankton
528 species by submerged macrophytes1. *Journal of Phycology*, 38(5), 862-871.
529 Krasikov, V., Aguirre von Wobeser, E., Dekker, H. L., Huisman, J., & Matthijs, H. C. 2012.
530 Time-series resolution of gradual nitrogen starvation and its impact on photosynthesis in
531 the cyanobacterium *Synechocystis* PCC 6803. *Physiologia plantarum*, 145(3), 426-439.
532 Kurzbaum, E., Kirzhener, F. & Armon, R. 2012. Improvement of water quality using
533 constructed wetland systems. *Reviews on Environmental Health,* 27(1), 59-64.

534 Campos, L.C., Su, M. F. J., Graham, N.J.D. Smith, S.R. 2002. Biomass development in slow
535 sand filters. *Water Research* 36**,** 4543-4551.
536 Laue, P., Bahrs, H., Chakrabarti, S. & Steinberg, C. E. 2014. Natural xenobiotics to prevent
537 cyanobacterial and algal growth in freshwater: contrasting efficacy of tannic acid, gallic
538 acid, and gramine. *Chemosphere,* 104**,** 212-20.
539 Le Moal, M., Gascuel-Odoux, C., Ménesguen, A., Souchon, Y., Étrillard, C., Levain, A.,
540 Moatar, F., Pannard, A., Souchu, P., Lefebvre, A. and Pinay, G., 2019. Eutrophication: a
541 new wine in an old bottle? *Science of the Total Environment*, *651*, pp.1-11.
542 Lu, H., Yuan, Y., Campbell, D. E., Qin, P., & Cui, L. 2014. Integrated water quality, energy
543 and economic evaluation of three bioremediation treatment systems for eutrophic water.
544 *Ecological Engineering*, 69, 244-254.
545 Lürling, M. & Va Oosterhout, F. 2013. Controlling eutrophication by combined bloom
546 precipitation and sediment phosphorus inactivation. *Water Research,* 47**,** 6527-37.
547 Marchand, L., Nsanganwimana, F., Oustriere, N., Grebenshchykova, Z., Lizama-Allende, K.
548 & Mench, M. 2014. Copper removal from water using a bio-rack system either unplanted
549 or planted with *Phragmites australis*, *Juncus articulatus* and *Phalaris arundinacea*.
550 *Ecological Engineering,* 64**,** 291-300.
551 Martinez Hernandez J., Lopez-Rodas V. & Costas E. 2009. Microcystins from tap water could
552 be a risk factor for liver and colorectal cancer: a risk intensified by global change. *Medical*
553 *hypotheses,* 72 (5), 539-40.
554 Mei, X.-Q., Yang, Y., Tam, N. F.-Y., Wang, Y.-W. & Li, L. 2014. Roles of root porosity, radial
555 oxygen loss, Fe plaque formation on nutrient removal and tolerance of wetland plants to
556 domestic wastewater. *Water Research,* 50**,** 147-159.

557 Moss, B. R. 2009. Ecology of fresh waters: man and medium, past to future. 480 p. John Wiley
558 & Sons. Chichester, West Sussex.
559 Mulderij, G., Van Nes, E. H., & Van Donk, E. 2007. Macrophyte–phytoplankton interactions:
560 the relative importance of allelopathy versus other factors. *Ecological Modelling*, 204(1),
561 85-92.
562 Murray, D., Jefferson, B., Jarvis, P., & Parsons, S. A. 2010. Inhibition of three algae species
563 using chemicals released from barley straw. *Environmental technology*, 31(4), 455-466.
564 Nan, C., Zhang, H., Lin, S., Zhao, G. & Liu, X. 2008. Allelopathic effects of *Ulva lactuca* on
565 selected species of harmful bloom-forming microalgae in laboratory cultures. *Aquatic*
566 *Botany,* 89**,** 9-15.
567 O'Neil J.M., Davis T.W., Burford M.A., Gobler C.J. 2012. The rise of harmful cyanobacteria
568 blooms: The potential roles of eutrophication and climate change. *Harmful Algae*, 14, 313-
569 334.
570 Otten, T. G., & Paerl, H. W. 2015. Health effects of toxic cyanobacteria in US drinking and
571 recreational waters: our current understanding and proposed direction. *Current*
572 *Environmental Health Reports*, 2(1), 75-84.
573 Persson, F., Langmark J., Heinecke, G., Hedberg, T., Tobiason, J., Stenstrom, T. A. &
574 Hermansson, M. 2005. Characterisation of the behaviour of particles in biofilters for pre-
575 treatment of drinking water. *Water Research,* 39**,** 3791-800.
576 Pontier, O., Leclerc, J. P., & Pons, M. N. 2005. Influence of geometrical and operational
577 parameters on the axial dispersion in an aerated channel reactor. *Water research*, 39(18),
578 4454-4462.
579 Qin,B., Paerl, H.W., Brookes, J.D., Liu, J., Jeppesen, E., Zhu, G., Zhang, Y., H. Xu, K. Shi, J.
580 Deng. 2019. Why Lake Taihu continues to be plagued with cyanobacterial blooms through

581 10 years (2007-2017) efforts? *Science Bulletin* (doi:
582 https://doi.org/10.1016/j.scib.2019.02.008)
583 Rojo, C., Segura, M. & Rodrigo, M. A. 2013. The allelopathic capacity of submerged
584 macrophytes shapes the microalgal assemblages from a recently restored coastal wetland.
585 *Ecological Engineering,* 58**,** 149-155.
586 Ruggeri, B. & Sassi, G. 1993. On the modelling approaches of biomass behaviour in bioreactor.
587 *Chemical Engineering Communications*, 122, 1-56
588 U.S. EPA. 2005. Membrane Filtration Guidance Manual, EPA 815-R-06-009. USEPA,
589 Washington, DC, USA
590 Vanderstukken, M., Declerck, S. A. J., Decaestecker, E. & Muylaert, K. 2014. Long-term
591 allelopathic control of phytoplankton by the submerged macrophyte *Elodea nuttallii*.
592 *Freshwater Biology,* 59**,** 930-941.
593 Weiss, P., Westbrook, A., Weiss, J., Gulliver, J. & Bierboer, D. 2014. Effect of water velocity
594 on hydroponic phytoremediation of metals. *International Journal of Phytoremediation,* 16**,**
595 203-17.
596 Wolfe, G. V. 2000. The chemical defense ecology of marine unicellular plankton: constraints,
597 mechanisms, and impacts. *The Biological Bulletin*, 198(2), 225-244.