Journal of the Air & Waste Management Association

ISSN: 1096-2247 (Print) 2162-2906 (Online) Journal homepage: https://www.tandfonline.com/loi/uawm20

Treatment of odor by a seashell biofilter at a wastewater treatment plant

Samantha Abraham, Scott Joslyn & I.H. (Mel) Suffet

To cite this article: Samantha Abraham, Scott Joslyn & I.H. (Mel) Suffet (2015) Treatment of odor by a seashell biofilter at a wastewater treatment plant, Journal of the Air & Waste Management Association, 65:10, 1217-1228, DOI: 10.1080/10962247.2015.1075918

To link to this article: https://doi.org/10.1080/10962247.2015.1075918

Published online: 09 Oct 2015.

Submit your article to this journal

Article views: 3516

View related articles

View Crossmark data

Citing articles: 8 View citing articles

Full Terms & Conditions of access and use can be found at
https://www.tandfonline.com/action/journalInformation?journalCode=uawm20

TECHNICAL PAPER

Treatment of odor by a seashell biofilter at a wastewater treatment plant

Samantha Abraham,[1,*] Scott Joslyn,[2] and I.H. (Mel) Suffet[1]

[1]*Department of Environmental Health Sciences, Fielding School of Public Health, University of California, Los Angeles, Los Angeles, CA, USA*

[2]*Nevada County Sanitation District No. 1, Nevada City, CA, USA*

[*]*Please address correspondence to: Samantha Abraham, Department of Environmental Health Sciences, Fielding School of Public Health, University of California, 650 Charles E. Young Drive S., Box 951772, 56-070 CHS, Los Angeles, CA 90095, USA; e-mail: samantha_m_abraham@yahoo.com*

Biofilters are becoming an increasingly popular treatment device for odors and other volatiles found at wastewater treatment plants. A seashell media based biofilter was installed in April 2011 at Lake Wildwood Wastewater Treatment Plant located in Penn Valley, California. It was sampled seasonally to examine its ability to treat odorous compounds found in the air above the anaerobic equalization basin at the front end of the plant and to examine the properties of the biofilter and its recirculating water system. The odor profile method sensory panels found mainly sulfide odors (rotten eggs and rotten vegetable) and some fecal odors. This proved to be a useful guidance tool for selecting the required types of chemical sampling. The predominant odorous compounds found were hydrogen sulfide, methyl mercaptan and dimethyl sulfide. These compounds were effectively removed by the biofilter at greater than 99% removal efficiency therein reducing the chemical concentrations to below their odor thresholds. Aldehydes found in the biofilter were below odor thresholds but served as indicators of biological activity. Gas chromatography with mass spectrometry and gas chromatography with sensory detection showed the presence of dimethyl disulfide and dimethyl trisulfide as well, but barely above their respective odor thresholds. The neutrality of the pH of the recirculating water was variable depending on conditions in the biofilter, but a local neutral pH was found in the shells themselves. Other measurements of the recirculating water indicated that the majority of the bio-activity takes place in the first stage of the biofilter. All measurements performed suggest that this seashell biofilter is successful at removing odors found at Lake Wildwood. This study is an initial examination into the mechanism of the removal of odorous compounds in a seashell biofilter.

Implications: This paper presents a thorough examination of a seashell media biofilter, a sustainable treatment technology used to remove reduced sulfide compounds. The durable performance of the seashell biofilter ensures that odors will be adequately controlled, preventing odor nuisance to surrounding residences, which is an emerging problem faced by waste management facilities. The odor profile method technique used in this study can be applied in many situations by waste management facilities and regulatory air management organizations for source tracking in relation to prevention and management of odor complaints, respectively.

Introduction

Odorous compounds and hazardous volatile organic compounds (VOCs) are released into the environment from natural biodegradation and anthropogenic sources, including composting, sludge drying, and wastewater treatment (Higgins et al., 2008; Jones, Martinez, and Rizwan, 2005). Due to recent population growth, waste treatment facilities are in close proximity to population centers, resulting in a large increase in odor nuisance complaints from the public (Curren, 2012; Hayes, Stevenson, and Stuetz, 2014; Lebrero et al., 2011; Leson and Winer, 1991; Muñoz et al., 2010).

As no instrument can properly simulate the human nose, trained panelists can conduct sensory measurements of nuisance odors. Muñoz et al. (2010) have reviewed recent advances of the air odor assessment, specifically the use of the individual odor character, defined by odor wheels, rather than total odor. Odor wheels have been developed to characterize the types of odor present from different odorous sources including wastewater treatment, composting, landfills, and, more recently, urban odors (Suffet and Rosenfeld, 2007). For this study, the researchers used the odor profile method (OPM) with odor wheels to define individual odor characters (Burlingame, 1999; Curren et al., 2014; Suffet et al., 2004) and the intensity approach used by Curren et al. (2014) (e.g., rotten vegetable, intensity 8 and rancid, intensity 6). OPM can provide insights into the proper type of chemical analyses needed to identify the compounds responsible for the total odor. Application of odor control technologies is best with an understanding of the compounds comprising the total odor. In order to improve the ability to assess odor emissions in terms of composition and concentration, gas chromatography–mass spectroscopy (GC-MS) was used to corroborate gas chromatography-sensory

Journal of the Air & Waste Management Association, 65(10):1217–1228, 2015. Copyright © 2015 A&WMA. ISSN: 1096-2247 print
DOI: 10.1080/10962247.2015.1075918 Submitted February 16, 2015; final version submitted June 25, 2015; accepted July 21, 2015.

detection (GC-Sniff) (Wright et al., 2005). Additionally, the ratio of a chemical concentration to its odor threshold concentration was used to help define odor nuisances, which can aid in the development of odor control strategies.

Traditionally, volatile compounds released from industry and community sources have been treated by activated carbon, incineration, and chemical scrubbers (Iranpour et al., 2005; Leson and Winer, 1991). In the last few decades, biofilters have become an increasingly popular treatment device for odorous compounds and other VOCs. Biofilters have many advantages, including lower operating cost, decreased energy consumption, and reduced by-product pollution (Iranpour et al., 2005; Leson and Winer, 1991). A biofilter consists of a bed of media with immobilized microbes that biodegrade pollutants passed through in an air stream. The pollutants partition into the biofilm, where the microorganisms, in a matrix of extrapolymeric substances, access the pollutants for biodegradation. The majority of biofilters operate aerobically and use organic media such as compost or peat (Delhomenie and Heitz, 2005).

Biofilters designed to treat hydrogen sulfide (H_2S) and other reduced sulfide compounds are known to accumulate sulfuric acid from biodegradation, resulting in a pH drop in the filter media that is often accompanied by a decrease in the removal efficiency of the biofilter (Carlson and Leiser, 1966; Jones, Martinez, and Rizwan, 2005; Smet et al., 1996). *Thiobacillus thioparus* has been shown to biodegrade H_2S to sulfuric acid (H_2SO_4) through elemental sulfur. It has also been demonstrated to break down dimethyl disulfide to methyl mercaptan, then H_2S, and finally to H_2SO_4. *Hyphomicrobium* species are known to biodegrade dimethyl sulfide to methyl mercaptan, followed by H_2S, and eventually to H_2SO_4 (De Bont, Van Dijken, and Harder, 1981; Jones, Martinez, and Rizwan, 2005; Oyarzun et al., 2003; Smith and Kelly, 1988). Systems degrading compounds other than H_2S have typically experienced rapid declines in efficiency when below neutral pH (Iranpour et al., 2005). This is thought to be in part because many of the microorganisms involved in the degradation of organic sulfides and VOCs are not tolerant of acid pH, unlike some of those that degrade hydrogen sulfide. Typical organisms found in sulfide removing biofilters include *Thiobacillus thioparus* (pH tolerance 4.5–7.8), *Thiobacillus thiooxidans* (pH tolerance 0.5–6.0), and *Hyphomicrobium* spp. (pH tolerance 6.0–9.5) (Bergey et al., 1974; Delhomenie and Heitz, 2005).

It is important to note that some of the organisms capable of degrading H_2S are also capable of degrading other reduced sulfides, but are not tolerant of acid pH, for example, *Hyphomicrobium* species (Iranpour et al., 2005; Smet, Langenhove, and Verstraete, 1993; Smet et al., 1996). Information on which species have been found to be acid tolerant is limited in the literature, as microbiological investigations are not commonly performed in biofilters, even after declining performance.

In order to control pH, some studies have demonstrated the use of organic media mixed with crushed calcium carbonate, the material that composes seashells, in order to neutralize the acid produced in sulfide oxidation, but few studies have examined the use of media made entirely from seashells (Delhomenie and Heitz, 2005). In 1994, Bord na Móna, marketed in the United States as Anua (Newbridge, Ireland), developed an innovative seashell media biofiltration system known as Mónashell. The use of seashells made of calcium carbonate as biofilter media theoretically allows for maintenance of a neutral pH due to their buffering capacity under aerobic conditions during the removal of odorous compounds such as reduced sulfides (Kouyoumjian and Saliba, 2006; Smet et al., 1996). The composition of oyster shells is 95% calcium carbonate with small percentages of aluminum, silicon, and manganese, and trace amounts of protein (Yoon et al., 2003). In addition to the assumed pH control, the seashells have a high porosity, which is associated with good biofilm growth and prevention of compaction and channeling compared to an organic medium like compost (Delhomenie and Heitz, 2005).

Bord na Móna Environmental Products U.S. (Naples, 2010) and Orange County (FL) Utilities (Van Durme, Koh, and Gay, 2010) have demonstrated that the Mónashell system is capable of treating high levels of H_2S. The performance of the seashell biofilter system treating emissions from a mixed sludge storage tank at a Greensboro, NC, water reclamation facility was reported by Naples (2010). Removal of greater than 99% H_2S was shown over an extended time period with concentrations as high as 376 ppm. Two grab samples showed that methyl mercaptan (MM) was removed at 100% and 89%, dimethyl sulfide (DMS) at 83% and 71%, and dimethyl disulfide (DMDS) at 100% and 50% without the use of nutrient or chemical addition for pH balancing. VOC sampling was performed and all compounds were found to be below or very close to their odor thresholds. The performance for reduced sulfides was mostly adequate in this performance validation. While odor analysis was conducted, results were not shown; thus, a picture of the performance can only be understood in the concept of removal efficiencies. This does not help one to understand how much nuisance would be posed to surrounding residences by incompletely removed compounds, some of which are above their respective odor thresholds. Additionally, many details of the operation, including size of the media bed, detention time, and flow rates of air and water, were not described. Van Durme, Koh, and Gay (2010) conducted pilot testing of a Mónashell System at the Orange County Utilities Stillwater Crossing Pump Station, Windermere, FL. The data showed that the pilot system provided more than 99% H_2S removal at inlet concentrations as high as 124.5 ppm during 8 months of testing. The pilot unit also provided odor removal efficiencies of more than 97% based on the method of detection threshold (DT) (EN13725). However, a look at outlet DT still showed that some odor was not being fully removed, but efforts were not made to attribute this odor to particular chemical compounds. Three grab samples at the end of 2, 3, and 5 months of operation showed more than 81% removal of MM. DMS at 140 ppbV showed a 94% removal rate at one time; however, when 23 ppbV and 33 ppbV of DMS were present in the inlet, 0% and 48% removal were observed, respectively. VOCs were once again found to be very close to or below their odor thresholds. A laboratory study of a similar style system with seashell media treating H_2S showed removal efficiencies of 75–90% (Massoudinejad et al., 2008).

While these studies from WEF Proceedings show seashell media to be quite effective, a further operational understanding and investigation of the seashell biofilter was necessary, as claims of pH neutrality and variations of H_2S over time (daily, weekly, and seasonally) were not fully defined and evaluated. Also, the odor character and intensity of the odorants, water quality changes in the recirculation system, and microbiological investigations had not been performed for the biofilter. Aldehyde and carboxylic acid analyses were also not performed, but these types of compounds are often found at wastewater treatment plants (Burlingame et al., 2004). This evaluation further differs from previous studies in that it demonstrated the use of a seashell biofilter at a different type of waste treatment process, rather than a mixed sludge storage tank (Naples, 2010) or a crossing pump station (Van Durme, Koh, and Gay, 2010).

The objectives of this study were to investigate the functioning of an operational, aerobic, theoretically neutral pH, seashell biofilter treating the air from an equalization (EQ) basin at a wastewater treatment plant (WWTP) to understand how it functioned to remove odorous compounds based on the results of chemical and physical analyses, and to examine whether the OPM accurately indicated the types of chemical compounds present in the odorous WWTP air.

Techniques used to evaluate the objectives included measurement of odor using the OPM, broad-spectrum GC-MS and GC-Sniff analysis, and quantitative chemical analysis of odiferous sulfides, carboxylic acids, and aldehydes. The water quality of the seashell biofilter was evaluated using pH, conductivity, hardness, alkalinity, and dissolved oxygen measurements.

Experimental methods

Site description

A small treatment plant, Lake Wildwood WWTP, located in Penn Valley, California, treating 1,325,000 L (350,000 gal) per day of dry-weather flow, was selected as an evaluation site for the seashell biofilter. The wastewater treatment plant follows a fairly traditional treatment scheme, although it includes an anoxic denitrification tank prior to aeration. The plant used chlorination of water before release until it switched to ultraviolet (UV) treatment in 2013. Lake Wildwood WWTP typically treats young, raw wastewater with a short time of transfer of less than a day from the wastewater sources to the plant. Volume during heavy rain flows can reach three times the dry weather flow. Due to the relatively rural location of the plant, it does not treat industrial waste. Thus, toxicity to microorganisms and the surrounding community due to trace organic volatiles is minimized.

At Lake Wildwood WWTP, raw wastewater is screened at the headworks and then flows to the pump mixed anaerobic EQ basin. Wastewater is pumped from the EQ basin to the anoxic denitrification tank. Pre-aeration occurs after denitrification, and then the flow is directed to and processed using an oxidation ditch. The flow from this stage goes to a clarifier with return activated sludge. After clarification, the wastewater is filtered and disinfected with chlorine gas. Following disinfection, water is dechlorinated using sulfur dioxide and released to the nearby river. A process flow diagram of Lake Wildwood WWTP is shown in Figure 1.

Air above the EQ basin is drawn through the biofilter, which treats the air before its release into the atmosphere. Samples were taken for chemical analysis at the inlet and outlet of the biofilter in March 2012, August 2012, and September 2013 to evaluate the long-term performance and stability of the system. The Mónashell biofilter (Bord na Móna, Newbridge, Ireland) is shown in the process flow diagram (Figure 1). The first stage of the biofilter contains oyster shells and the second stage contains mussel shells, both by-products of the shell fishing industry. The shells are placed into the biofilter. The two stages of the biofilter are thought to allow for separate microorganism communities to develop, which may enhance biodegradation of mixed waste steams. The airflow rate is 28,300 L/min (1000 ft^3/min) flowing up through the first stage and down through

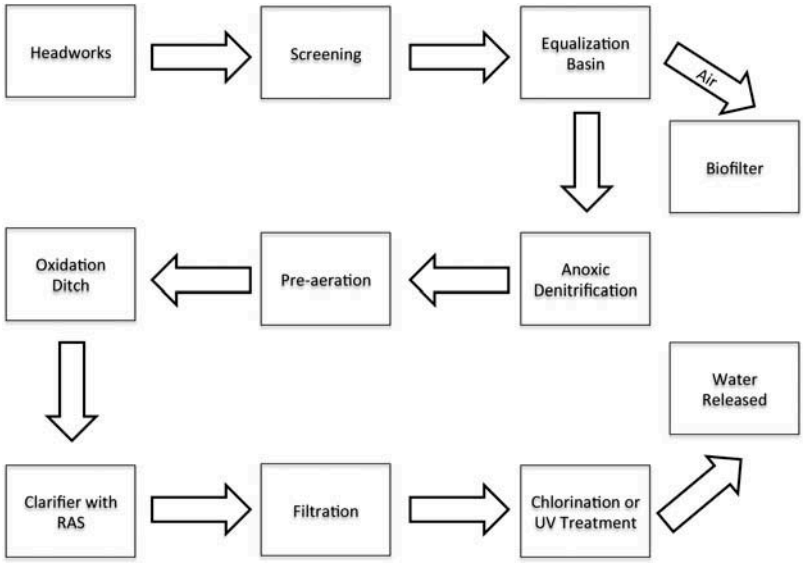

Figure 1. Process flow diagram of Lake Wildwood WWTP.

the second stage. The unit can treat flows ranging from 21,200 L/min to 34,000 L/min. The unit is 5 m by 2 m by 2 m (total length, width, and height). The first stage is 3 m long and the second stage is 2 m long. The bed depth is approximately 1.5 m, bearing in mind that the shells dissolve with time. Thus the total volume of biofilter media is 15 m^3. The water recirculation flow rate throughout the biofilter is 38–76 liters (10–20 gallons) per minute. The water recirculates constantly throughout the biofilter using a biotrickling style of operation where nutrients are contained in the water rather than the bed material (seashells). The empty bed residence times are 19 sec and 13 sec for the first and second stage, respectively. The source of the makeup water is the chlorinated plant water, which passes through a sodium sulfite feeder for dechlorination. When the plant switched to UV treatment in 2013, the sodium sulfite addition was discontinued.

The biofilter was seeded once only at the startup of the biofilter with nutrients and organisms supplied with the unit from Bord na Móna (Newbridge, Ireland) that were supplied with the unit. There was some carbon source provided in the initial nutrients, but it was expected that the biofilter community would perpetuate itself without additional nutrients. The expected carbon source for chemoheterotrophic organisms, such as *Hyphomicrobium* species, was organic reduced sulfide compounds that also serve as an energy source (Hwang et al., 1994). For chemoautotrophic organisms such as *Thiobacillus thioparus*, a carbon source would be provided by breakdown of calcium carbonate from the shells to produce carbon dioxide due to contact with sulfuric acid or potentially by fungal organisms growing in the community (Kouyoumjian and Saliba, 2006). The unit has been in operation since April 2011.

Odor profile method sampling

One-liter unconditioned Tedlar bags (SKC West, Fullerton, CA) and a foot pump apparatus (Columbia Analytical Service, Simi Valley, CA) were used to take samples at the inlet and outlet of the biofilter. Samples were taken on August 29, 2012, and September 4, 2013, and were shipped overnight to the University of California, Los Angeles (UCLA). Samples were analyzed using the OPM at UCLA on August 30, 2012, and September 5, 2013, respectively (Burlingame, 1999). The Wastewater Treatment Plant Wheel was used with the flavor profile analysis (FPA) scale from Standard Method 2170 to evaluate the intensity of samples (American Public Health Association [APHA], 2012b; Curren et al., 2014; Suffet et al., 1988, Suffet and Rosenfeld, 2007). The 7-point odor intensity scale includes the numbers 1, 2, 4 (weak), 6, 8 (moderate), 10, and 12 (strong).

Sulfide sampling

One-liter Tedlar bags (Zefon, Ocala, FL) and a foot pump apparatus (Columbia Analytical Services, Simi Valley, CA) were used to take samples from the biofilter. In March 2012, inlet samples were taken over the EQ basin and outlet samples were taken from a diluted outlet stream (10 times dilution). In August 2012 and September 2013, inlet samples were taken directly from the inlet of the biofilter and outlet samples were taken directly from the outlet of the biofilter. Samples were shipped to Columbia Analytical for processing within 24 hr of sampling. Samples were analyzed by ASTM Method D5504-01 using a gas chromatography–sulfur chemiluminescent detector. Chemical analysis looked for hydrogen sulfide, carbonyl sulfide, methyl mercaptan, ethyl mercaptan, dimethyl sulfide, carbon disulfide, isopropyl mercaptan, tert-butyl mercaptan, *n*-propyl mercaptan, ethyl methyl sulfide, thiophene, isobutyl mercaptan, diethyl sulfide, *n*-butyl mercaptan, dimethyl disulfide, 3-methylthiophene, tetrahydrothiophene, 2,5-dimethylthiophene, 2-ethylthiophene, and diethyl disulfide.

Additionally, this paper includes data measured using an OdaLog L2 Hydrogen Sulfide Gas Logger (Detection Instruments Corporation, Phoenix, AZ). The unit was used to measure the H_2S concentration in parts per million of the air above the EQ Basin (which is pumped to the inlet of the biofilter) and the outlet of the biofilter. The instrument range is 0–200 ppm with a resolution of 0.1 ppm and an accuracy of 1% of the scale (Detection Instruments Corporation, 2013). The instrument was calibrated and standardized using 50 ppm H_2S.

Aldehyde and carboxylic acid sampling

In March and August 2012, aldehyde sampling was performed. An air pump (model 224-PCXR4, SKC West, Fullerton, CA) was used at a rate of 1 L/min for 100 min through a sorbent tube of silica gel containing 2, 4-dinitrophenylhydrazine. Samples were capped and mailed to Columbia Analytical in a cooler, on ice, for processing. Samples were analyzed by U.S. Environmental Protection Agency (EPA) Method TO-11A using high-performance liquid chromatography (HPLC) with an ultraviolet–visible (UV-Vis) Detector for formaldehyde, acetaldehyde, proprionaldehyde, crotonaldehyde, butyraldehyde, benzaldehyde, isovaleraldehyde, valeraldehyde, *o*-tolualdehyde, *m*-tolualdehyde, *p*-tolualdehyde, *n*-hexaldehyde, and 2,5-dimethylbenzaldehyde.

In March 2012, sorbent tubes were also collected for carboxylic acid analysis. An air pump (model 224-PCXR4, SKC West, Fullerton, CA) was used at a rate of 1 L/min for 100 min through a sorbent tube of silica gel. Samples were shipped to Columbia Analytical and were analyzed by Columbia Analytical Method 102 on a GC-MS for acetic acid, propionic acid, 2-methylpropanoic acid, butanoic acid, 2-methylbutanoic acid, 3-methylbutanoic acid, pentanoic acid, 2-methylpentanoic acid, 3-methylpentanoic acid, 4-methylpentanoic acid, hexanoic acid, heptanoic acid, 2-ethylhexanoic acid, cyclohexanecarboxylic acid, octanoic acid, benzoic acid, and nonanoic acid. All chemicals were nondetect below 2.9 $\mu g/m^3$ except acetic acid at 23 $\mu g/m^3$, indicating that this analysis did not need to be performed in the future. In both March and August 2012, inlet samples were taken at the biofilter. In March, outlet samples were taken at a location of 10-fold dilution. In August, outlet samples were taken at the direct outlet of the biofilter.

GC-MS and GC-Sniff sampling

In August 2012, 1-L Tedlar bags and a foot pump apparatus (Columbia Analytical Services, Simi Valley, CA) were used to take samples at the inlet and outlet of the biofilter. Samples were shipped to UCLA and processed the next day. One hundred milliliters of sample was injected using a ground-glass syringe onto the gas adsorbent traps/heat desorption system (designed by Randy Cook, Lotus Instruments, Long Beach, CA) and purged to a Varian 450 GC (Varian, Inc., Palo Alto, CA) with two detectors: a Varian 220 MS and an SGE Brand Olfactory "Sniffer" (SGE Analytical Science, Austin, TX) using 99.9999% helium as the carrier gas at a flow rate of 1 mL/min. The sniff port consists of a glass nose cone at the base of the capillary column. Air from the column exits at atmospheric pressure and is mixed with a humidified breathable air stream created by bubbling air through water in a glass tube to prevent drying out of nasal membranes. The chemical traps contained five layers, consisting of 60/80 mesh glass beads, Carbopack C and B, Carbosieve 569, and 1003. The traps were maintained at 35ºC and following injection were rapidly heated to 220ºC over 0.1 min to desorb the samples. The columns used for both detectors were Restek Rtx-wax columns (length 60 m, f 0.25 mm ID, film thickness of 0.5 μm). The temperature program used was as follows: 40ºC hold for 10 min, a ramp of 6ºC /min to 160ºC with a hold of 13 min, and a ramp of 10ºC /min to 220ºC with a hold of 5 min. The ion trap MS monitored from 47 to 300 *m/z* for the first 17 min and then from 44 to 300 *m/z* for the remaining duration to avoid interferences. Samples were processed in a similar manner in September 2013 and the results of the analysis yielded similar results.

The relationship between the retention times on the GC-MS and the GC-Sniff was fitted to a cubic power and a linear equation. The use of these equations allowed for the establishment of the time of elution on the GC-MS based on the time of elution on the GC-Sniff. Quantification of DMS, DMDS, and dimethyl trisulfide (DMTS) was performed using standard curves on the GC-MS. VOC standards have been run on the GC-MS to examine the level of industrial chemicals in the biofilter at Lake Wildwood WWTP. These include toluene, xylenes (*ortho, meta*, and *para*), ethylbenzene, and dichloromethane.

Water sampling

On August 29, 2012, samples were removed from the recirculating tanks from both stages of the biofilter in 50-mL Nalgene bottles for future pH and conductivity analysis. A pH measurement was taken on the water inside the shells on August 29, 2012, using a variety of types of long- and short-range pH paper. pH and conductivity were measured using an Accumet AP85 (Fisher Scientific, Pittsburgh, PA) pH/conductivity meter. pH paper was used to measure the pH in the water pooled inside the seashells. Three types of paper were used: pH 4.0–7.0 ColorpHast pH Test Strips, pH 6.0–8.0 Hydrion pH Papers, and pH 2–10 Universal pH Paper. On September 4, 2013, pH and conductivity were measured live using the Accumet AP85. Dissolved oxygen measurements were taken on site using a Hach Dissolved Oxygen Meter (Loveland, CO). Samples were taken back to UCLA in 250-mL glass bottles for alkalinity and hardness measurements following Standard Method 2320 for low alkalinity and Standard Method 2340 by ethylenediamine tetraacetic acid (EDTA) titration, respectively (APHA, 2012a). The samples were kept on ice for transport and processed the following day.

Results

Odor profile method

Tables 1 and 2 show the data analyzed by OPM of samples taken from the inlet and outlet of the biofilter. Rotten eggs and

Table 1. Results of odor profile method from 8/30/12.

Inlet odor character	Odor intensity	Outlet odor character	Odor intensity
Rotten eggs	8 ± 1.63	Phenol	3 ± 3.46
		Plastic	3 ± 3.46
Odor note[a]: rotten vegetable		Odor note: sweet	

Note: [a]An "odor note" is an odor character reported by less than 50% of the panelists.

Table 2. Results of odor profile method from 9/5/13.

Inlet morning		Outlet morning		Inlet afternoon		Outlet afternoon	
Odor character	Odor intensity	Odor character	Odor intensity	Odor character	Odor intensity	Odor character	Odor intensity
Rotten vegetable	4.6 ± 3.0			Fecal	4.3 ± 2.1	Solvent	2.0 ± 2.0
Fecal	4.0 ± 3.1			Rotten vegetable	3.4 ± 3.4		
Odor notes[a]: rotten eggs, musty, glue, rubbery		Odor notes: glue, solvent, medicinal, rotten vegetable, rotten garlic, rubbery, fecal		Odor notes: rotten eggs, musty, sewery, garlic, putrid		Odor notes: glue, medicinal, fecal, rotten vegetable, rubbery, musty	

Note: [a]An "odor note" is an odor character reported by less than 50% of the panelists.

rotten vegetable were the primary characteristics of the inlet with intensities between 4 (weak) and 8 (moderate). These types of odors are usually the result of reduced sulfide compounds such as H_2S and MM, respectively. These constant odors could cause odor complaints depending on wind direction and off-site neighbors. The fecal odors found during the September sampling (Table 2) may have originated from indole or skatole, whose odors were previously characterized as fecal in the water of wastewater treatment plants, or from other unidentified compounds (Godayol et al., 2011; Hwang et al., 1995). In the literature, indole and skatole have not been detected in air samples from wastewater treatment plant samples due to the difficulty of analytical analysis for trace levels of these compounds.

The outlet results indicated mainly phenol/medicinal odors, which are characteristic of the use of Tedlar bags for sampling. Tedlar bags are known to have a high phenol background level (Trabue, Anhalt, and Zahn, 2006). In a set of analyses of Tedlar bags filled with pure air, odor characters of phenol/medicinal (4.3±1.9) and odor notes of solventy, rubbery, musty, and burnt were found with similar odor intensities to those found in the outlet bags in this study. These types of odors are not predominant at WWTPs; thus, the background odor did not interfere with the odor panel evaluation. As seen in this instance, the OPM provided clues as to what types of chemical analysis were necessary. It was seen here that the inlet samples needed sulfide analysis and indole and skatole measurements, and that the outlet samples contained significantly reduced odors that were more characteristic of a blank Tedlar bag.

Sulfide analyses

In March 2012, August 2012, and September 2013, samples were taken for sulfide analysis by Columbia Analytical Services using ASTM Method D5504-01 (Devos et al., 1990). All samplings found H_2S (rotten egg), MM (rotten vegetable), and DMS (rotten vegetable) present at the inlet of the biofilter in concentrations exceeding their odor thresholds (see Table 3). The relative strength of the compound can be compared to its odor threshold, which gives an indication of the nuisance of the different compounds. The relative strengths of the inlet concentration of the sulfide compounds divided by their respective odor thresholds are given in Table 3. H_2S and MM had a similar relative strength (940× to 38,000× and 1500× to 33,000×, respectively) indicating that both compounds present a probable nuisance. DMS had a much lower relative strength (9.2× to 30×), indicating that it was probably a lesser nuisance at the WWTP. However, DMS would have added to the MM rotten vegetable odor. Due to the limited bioreaction time, <1 day from the wastewater sources to the plant, other sulfide compounds were not detected. Biochemically, larger sulfide compounds are typically produced later in the sewage biodegradation process (Higgins et al., 2006, Lomans, Pol, and Camp, 2002). This sampling data is consistent with OPM data shown in Tables 1 and 2, which found rotten egg and rotten vegetable odors.

Morning samples showed higher concentrations than afternoon samples due to longer overnight storage times in the collection system. H_2S concentrations ranged from 470 ppb to 19 ppm in the inlet with removal efficiencies of between 99.93% and 100% based on the method detection limit of 5 ppb. The maximum elimination capacity measured for H_2S was 24 g/hr for the 15 m^3 biofilter. MM concentrations ranged from 30 ppb to 650 ppb in the inlet with a removal efficiency of 100% and elimination capacity of 1.5 g/hr. DMS concentrations of the inlet ranged from below the 5 ppb method detection limit to 29 ppb with removal efficiency of 100% and elimination capacity of 0.11 g/hr. Sampling difficulties on the morning of August 29, 2012, prevented the analysis of the outlet sample. The outlet afternoon sample of the same day showed H_2S at a level near its odor threshold depending upon the source of the odor threshold concentration (Rosenfeld et al., 2007), presenting a possible odor nuisance to nearby residents; however, air exiting the biofilter experiences a 10-fold dilution, which should have brought the concentration below odor threshold before release into the surrounding community. Samples were taken at the outlet of the biofilter rather than at the point of 10-fold dilution. The sulfide data show the seashell biofilter to be quite effective at removing all types of reduced sulfide compounds.

Data from the OdaLog unit were used to assess seasonal and daily trends in H_2S concentrations in the air above the EQ basin, which is pumped to the inlet of the biofilter. The Lake Wildwood manager considered these concentrations to be

Table 3. Results of sulfide analysis.

		3/27/12				8/29/12				9/4/13			
		In	Out	In	Out	In	Out	In	Out	In	Out	In	Out
Compound	OTC	p.m.		p.m.		a.m.		p.m.		a.m.		p.m.	
H_2S (ppbV)[a]	0.50	4100	<5.0	1500	<5.0	19,000	~	10,000	7.3	960	<5.0	470	<5.0
H_2S relative intensity		8200×		3000×		38,000×		20,000×	15x	1900×		940×	
MM (ppbV)[b]	0.020	160	<5.0	61	<5.0	650	~	460	<5.0	68	<5.0	30	<5.0
MM relative intensity		8000×		3100×		33,000×		23,000×		3400×		1500×	
DMS (ppbV)[c]	0.98	9.0	<5.0	<5.0	<5.0	29	~	24	<5.0	24	<5.0	25	<5.0
DMS relative intensity		9.2×				30×		24×		24×		26×	

Notes: [a]Hydrogen sulfide (rotten egg odor).
[b]Methyl mercaptan (rotten cabbage odor).
[c]Dimethyl sulfide (rotten cabbage odor).

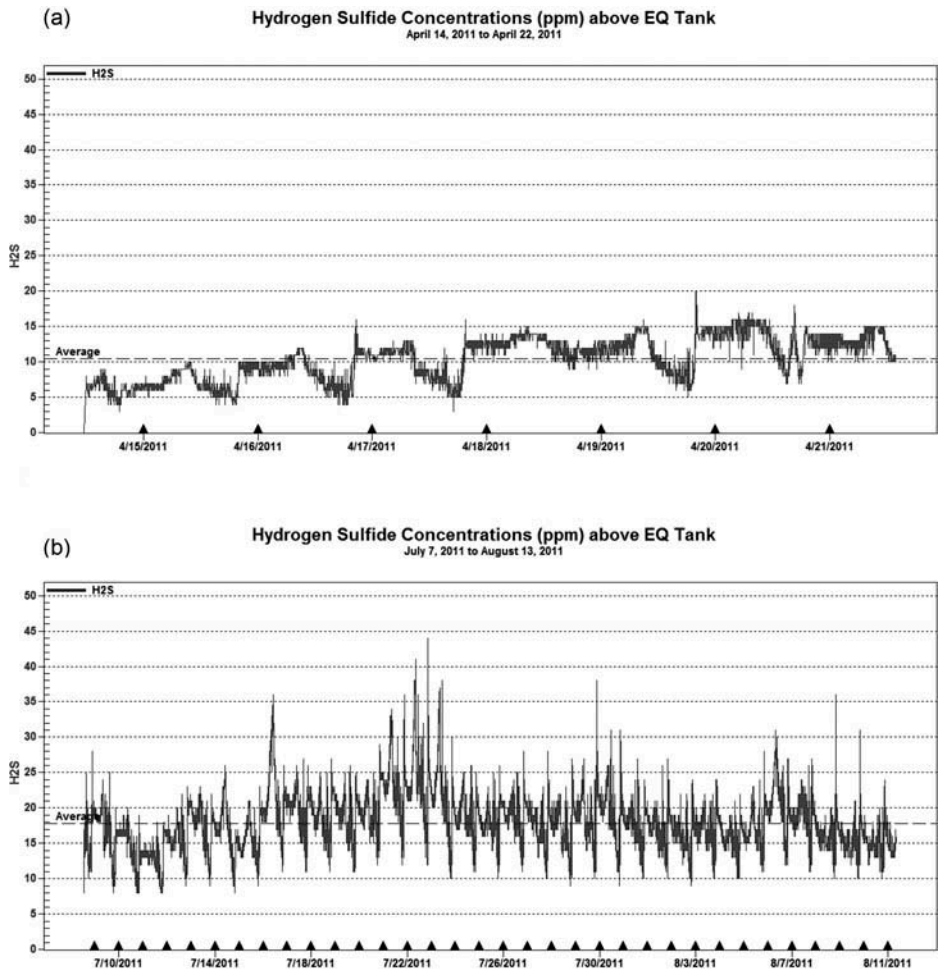

Figure 2. OdaLog H_2S concentrations from 2011 showing seasonal trends: (a) spring, (b) summer.

representative of odor trends at the WWTP. Figure 2 shows the seasonal differences in H_2S concentrations, which are applicable to changes in loading experienced by the biofilter. The OdaLog data show that average concentrations are higher in the summer within a given year (July and August 2011) compared to the spring (April 2011). Microbial activity is typically higher in warmer conditions; thus, more protein in wastewater is biodegraded, leading to the increased production of sulfur compounds (Liang, Das, and McClendon, 2003; Sekyiamah et al., 2008). This is consistent with sampling in March 2012 showing lower H_2S concentrations compared to August 2012 (see Table 3). The average during April was 10 ppm, compared to the average of 18 ppm in July through August. As seen in Figure 2, there were daily spikes in the H_2S concentration during the hours slightly after residents awoke in the morning. This spike was caused by low flow conditions, which caused longer overnight storage. A similar peak was observed following the residents' return to their homes in the evening, caused by higher flow. During the time period shown in Figure 2a (April 2011), the outlet of the biofilter was also measured. The range of the outlet was from 0 to 0.2 ppm (data not shown). The average value of the exit of the biofilter was 0.0 ppm during this time. One should bear in mind that the resolution of the OdaLog is 0.1 ppm; thus, exact determination of values close to 0 ppm is challenging.

Aldehyde and carboxylic acid analyses

Analysis for aldehydes was performed in both March 2012 and August 2012. The most commonly detected aldehydes were acetaldehyde, *n*-hexaldehyde, 2,5-dimethylbenzaldehyde, formaldehyde, and valeraldehyde (see Table 4). While all of these compounds are odorous, they were all found at levels well below their odor threshold concentrations in both the inlet and outlet of the biofilter (Devos et al., 1990). The presence of aldehydes in the inlet and outlet is indicative of biodegradation activity. Aldehydes are formed during lipolysis when lipids are broken down into fatty acids and then further biodegraded into aldehydes through biochemical reactions of organisms. Aldehydes can also be formed during proteolysis when proteins are degraded into amino acids, which go through alpha-keto acids into several compounds including aldehydes (McSweeney, 2004). A sampling difficulty with the aldehydes occurred in the morning in August and in March as a result of the dewpoint and rain, respectively. The excessive moisture caused dripping into the lines following the sorbent tube and

Table 4. Results of aldehyde analysis.

		3/27/12				8/28/12		8/29/12	
		In	Out	In	Out	In	Out	In	Out
Compound	OTC	a.m.		p.m.		p.m.		a.m.	
Acetaldehyde[a]	190	0.83	4.3	0.68	4.2	6.0	6.2	<0.56	4.5
Butyraldehyde[b]	9500	<0.34	<0.34	<0.34	<0.34	0.60	0.47	<0.34	<0.34
2,5-Dimethyl benzaldehyde[c]	Unknown	<0.18	0.22	<0.18	0.32	<0.18	0.2	<0.18	<0.18
Formaldehyde[d]	870	<0.81	<0.81	<0.81	<0.81	36	60	1.2	12
n-Hexaldehyde[e]	14	0.38	0.37	0.30	<0.25	1.7	1.1	<0.25	0.37
Isovaleraldehyde[d]	Unknown	<0.28	<0.28	<0.28	<0.28	0.51	0.54	<0.28	<0.28
Propionaldehyde[f]	9.3	<0.42	<0.42	<0.42	<0.42	1	0.92	<0.42	<0.42
Valeraldehyde[d]	28	<0.28	<0.28	<0.28	<0.28	0.42	0.68	<0.28	0.39

Notes: All values in ppbV.
[a]Sweet, fruity odor.
[b]Sweet odor.
[c]Almond odor.
[d]Pungent odor.
[e]Green odor.
[f]Sweet, ester odor.

decreased concentrations of aldehydes detected due to losses. Regardless, it appears that aldehydes were not a contributor to the odors found at Lake Wildwood WWTP. Carboxylic acid analysis was performed in March 2012, but all chemicals were below method detection limits; thus they were not pursued on future sampling trips. This data is consistent with the OPM data in Tables 1 and 2 where sweet odors characteristic of aldehydes and rancid odors typical of carboxylic acids were not observed.

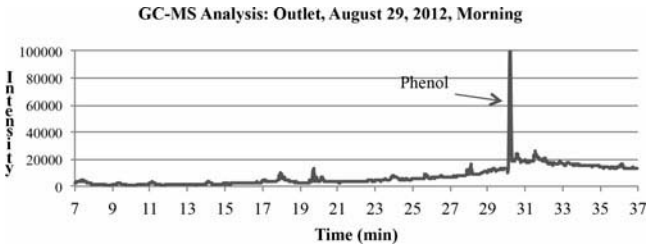

Figure 4. GC-MS chromatogram for outlet sample from morning of 8/29/2012.

GC-MS and GC-Sniff sampling

Analyses were also performed at UCLA using GC-MS and GC-Sniff to further evaluate the OPM analyses, that is, to look for sulfur odors and fecal odors and to try to identify them, to confirm the contract laboratory quantitative analysis, and to search for the presence of other volatile compounds. The GC-Sniff was performed to better define the odor profile of the biofilter influent and effluent and to help identify unknown peaks from the GC-MS. Odors are a clue to help understand the MS library search results. Figures 3 and 4 show the chromatograms from the GC-MS of an inlet and outlet sample taken from the biofilter on the morning of August 29, 2012.

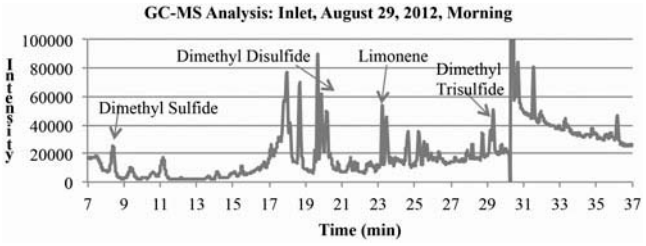

Figure 3. GC chromatogram for inlet sample from morning of 8/29/2012.

The x-axis of the figures shows the time of elution of the peaks of interest. It can be seen that the inlet sample has a many more peaks than the outlet sample, which is similar to a blank. Three notable peaks were quantified: DMS at 19.7 ppb (20×), DMDS at 5.6 ppb, (odor threshold = 2.2 ppb, 2.5×) and DMTS at 5.8 ppb (odor threshold = 1.2 ppb, 4.8×) (Nagata and Takeuchi, 2003). These compounds would contribute to the rotten vegetable odors detected but are not primary odorants, as their relative intensities were much less than H_2S or MM (see Table 3). There were some tentatively identified aldehydes and ketones found on the GC-MS in the inlet sample and some in the outlet sample that were in accordance with the quantitative aldehyde analyses. Regardless, these compounds appeared to be below their odor thresholds, as the GC-MS peaks were quite small and there were not odors of aldehyde or ketone character observed from the GC-Sniff. Fecal odors were not observed from the GC-Sniff, and indole and skatole were not detected on the GC-MS based upon retention times of standards. This indicates three possibilities: (1) that the fecal odors may be a result of an unknown fecal compound, or (2) that the analytical methods for analyzing indole and skatole are not suitable for analyzing the concentrations found in this

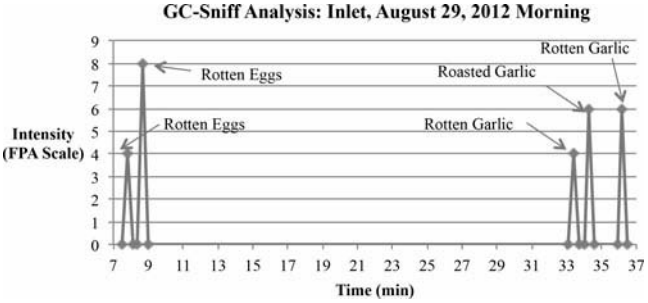

Figure 5. GC-Sniff Chromatogram for inlet sample from morning of 8/29/2012.

sampling, or (3) that odor panelists incorrectly named odors as fecal. The chemical cause of the fecal odor needs further investigation.

The GC-MS detected very low parts per billion levels of toluene, ethyl benzene, *o*-xylene, *m*-xylene, and *p*-xylene in the samples, but the concentration of these aromatics was only slightly above the background level found in a blank bag. While these type of VOCs are typical of those found at WWTPs, the concentrations were not of concern from a health perspective (Leson and Winer, 1991). No other VOCs were observed, confirming that the plant was not receiving industrial wastes.

Figure 5 shows the GC-Sniff results from an inlet sample from August 2012. All outlet samples had no odors on the GC-Sniff. The inlet sample had rotten egg, roasted garlic, and rotten garlic odors. The garlic odor peak at 34.3 min is believed to be associated with DMTS based on standards run. One of the sulfur odors at the beginning is most likely MM, which is too volatile to be detected using the GC-MS but can be sniffed. While rotten egg is not the odor descriptor commonly associated with MM, it is in the same family of compounds as rotten cabbage. The additional garlic odors toward the end of the run are most likely associated with higher molecular weight sulfide compounds. Generally, the GC-MS analysis provided important additional information such as the presence of DMDS and DMTS, as well as the presence of additional sulfide compounds discovered via the GC-Sniff. These compounds were apparently enhancing the rotten vegetable odors observed by OPM analysis (Tables 1 and 2). In September 2013, samples were also collected for GC-MS. GC-Sniff once again found the presence of DMTS but concentrations were below quantitation limits on GC-MS.

Water sampling

In August 2012 and September 2013, some water measurements were performed including pH, conductivity, dissolved oxygen, and alkalinity measurements (see Table 5). The biofilter has oyster shells for the first stage and mussel shells for the second stage. One aim of the sampling project was to examine claims of neutrality in seashell biofilters. Small amounts of water collect inside the shells in the biofilter. Measurement of the pH of this water was critical because biodegradation takes place at this location. However, the volume was too small to be measured using a pH probe, so the pH of this water was measured with pH paper. It was found to be between pH 6 and 6.5 in both stages of the biofilter, indicating neutrality locally at the sites of the biofilm growth on the shells. In August 2012, the pH of the oyster shell recirculating water was measured using a pH probe and found to have be 3.23. One explanation for the acidic pH was that insufficient flush water was being added to the recirculating water at the time. Another possible explanation was that the loading in August 2012 exceeded the buffering capabilities of the system.

Table 5. Results of water measurements for August 2012 and September 2013.

		8/29/12		9/4/13	
Measurement	Units	Oyster	Mussel	Oyster	Mussel
pH	pH	3.23	7.65	7.49	8.23
Conductivity	uS/cm	3850	2740	756	545
Dissolved Oxygen	mg/L	—	—	6.90	8.28
Alkalinity	mg/L as $CaCO_3$	—	—	40.4	134
Total hardness	mg/L as $CaCO_3$	—	—	213	130

The pH of the oyster side recirculating tank was later brought to neutrality, as shown in the September 2013 measurement of 7.49. A plant operator increased the amount of flush water added to between 7.6 and 38 liters per minute (2 to 10 gallons per minute) as a way of addressing the acidic pH of the system. While the flow rates of air and water remained the same in the biofilter unit, this additional makeup water was added to the recirculating tank (previously 3.8 liters per minute was added to maintain levels against evaporation). In September 2013, both recirculating tanks were in neutral pH range. The additional buffering capacity provided by the flush water may have neutralized the acid, or other variables in the system (such as loading) may have brought the system back to neutrality. The flush water was added starting in September 2012 and neutrality of the recirculating water has since been maintained. The pH in the mussel side was near neutrality during both samplings.

The neutral pH at the seashells themselves should enable the growth of neutral organisms in those biofilms. However, organisms carried off in the recirculating water may not be able to inhabit the acidic environment. Organisms growing toward the outside of the biofilm on the seashells may be impacted by recirculating water pH as well. It is important to control the pH of the recirculating water to ensure the continued health of microorganisms in the biofilter. Neutral pH shows potential for the continuous growth of sulfide removing organisms that have experienced performance declines with acid accumulation. All chemical analyses conducted indicate the seashell biofilter was efficient at removing odorous sulfur compounds following several years of operation.

The conductivity of the recirculating water of the first stage (oyster shells) was consistently higher than that of the second stage. Conductivity is influenced by dissolved anions and cations, including sulfate, calcium, and aluminum. Thus, the conductivity in the first stage could be impacted by additional biodegradation, which produces more sulfate ions and acid; additional acid would dissolve the shells and produce more calcium, or there could be differences in the solubility of the different types of shells. Additionally, seashells contain low

percentages of compounds such as aluminum oxide that could dissolve and disassociate, thereby affecting the conductivity of the recirculating water. It is likely that most of the biodegradation activity takes place in the first stage of the biofilter as it is larger and encountered first. However, there could be other factors influencing the location of the biodegradation activity, such as the organisms present in the community. Different organisms may be responsible for biodegrading hydrogen sulfide and dimethyl sulfide. The conductivity of the water was significantly reduced in September 2013, possibly as a result of additional flush water or reduced inlet loadings (see Table 5); however, the conductivity remained higher in the first stage.

The dissolved oxygen measurements in the recirculating water indicate that the biofilter is an aerobic environment. Dissolved oxygen levels of 6.9 and 8.28 mg/L are near oxygen saturation levels for water at room temperature. The alkalinity levels measured in September 2013 show that the mussel stage had nearly 3.5 times the alkalinity of the oyster stage as milligrams per liter of $CaCO_3$. As with the conductivity measurements, the reduced alkalinity of the oyster stage is indicative of a lower pH compared to the mussel stage. This may be a result of greater acid production as result of higher biological activity taking place and thus reduced buffering capacity of the recirculating water. The hardness measurements indicate that the water is very hard and hard in the oyster and mussel stages, respectively, based upon the U.S. Geological Survey scale of water hardness. This is in line with more biodegradation taking place in the first stage of the filter and hence more calcium being leached from the shells as the calcium carbonate buffers the sulfuric acid. All water sampling data indicate that greater biological activity takes place in the oyster stage of the biofilter.

Discussion

One objective of the study was to evaluate the ability of OPM to correlate properly with the chemical composition of the inlet and outlet airflows of the biofilter. While many studies at WWTPs have measured the chemicals thought to be contributing to total odor, few have done so in a comprehensive fashion with chemical analysis and odor panels characterizing individual odors. A thorough odor evaluation was undertaken to ensure that a proper evaluation of the biofilter took place. One can only measure removal efficiencies for a system if the compounds are evaluated. The composition of total odor is variable at different WWTPs, and this study utilized OPM to help determine the groups of chemical compounds that required analysis at a given location. It was confirmed that OPM correlated properly with the actual chemical composition of the incoming and outgoing biofilter air. The types of chemical compounds above the odor threshold were reduced sulfide compounds, as reported by the odor panels. Aldehydes and carboxylic acids were not detectable above odor thresholds, and panels did not report these types of odors.

The other objective was to understand how the seashell biofilter functions to remove odorous compounds based on the results of chemical and physical analyses. Chemical analyses indicated that the biofilter functioned consistently to remove greater than 99% of the main chemical compounds detected (H_2S, MM, and DMS). The inlet loading of the biofilter approached the elimination capacity of the system in August 2012, but all compounds except H_2S were still removed to below their odor thresholds. The outlet showed low levels of H_2S at that time, 15 times above the odor threshold, but this concentration was rapidly diluted exiting the biofilter and should not provide any nuisance to surrounding residences.

This type of biofilter would be suitable for treatment at other types of WWTP sites that are characterized by mainly reduced sulfide odors. Based upon a comprehensive study at two WWTPs, treatment processes such as headworks, primary clarifiers, and waste haulers would likely be properly treated from this type of unit (Vitko et al., 2014). Secondary treatment processes may have other types of odors, which might require different types of seeding organisms, or even other odor control technologies. The size of the unit and the airflow rate would need to be adjusted, depending upon the inlet loading of the biofilter. The unit was successful in treating a mixed sulfide waste stream and with an elimination capacity of 1.6 g S/m^3-hr for H_2S. This information can be used to figure out the media bed size needed, depending on the desired concentration of reduced sulfides to be treated.

The performance of the biofilter seemed to be somewhat independent of physical properties that were measured. The performance was consistent throughout samplings even though there were changes in the properties of the recirculating water. The conductivity of both stages was considerably higher in the recirculating water in August 2012 than in September 2013. The pH was acidic in the recirculating water of the oyster side in August 2012, though neutral on the shells themselves. The inlet loading at this time was considerably higher than during other measurements, which may have impacted these variables. Based upon these measurements, the plant operated began adjusting the flush water used in the system in September 2012. In September 2013, the pH was neutral in the recirculating water. These variables did not particularly impact the performance of the system; however, they would be expected to affect the microorganism community to some degree. A lthough the pH of the water inside the shells themselves maintained neutrality, the acidic pH of the recirculating water might be expected to impact the types of microorganisms that could grow on the outside of the biofilm layer. The mechanism of biodegradation is likely based upon oxidation of reduced sulfide compounds to sulfuric acid or elemental sulfur via a diverse community of microorganisms. As in most biofilters, the predominant organisms present likely depends upon the conditions in the biofilter, including the composition of the inlet air, the pH, the temperature, salinity, and other variables that can impact the growth rate of microorganisms. Regardless, the system was somehow able to adapt itself to continue performing adequately. The biofilter showed its ability to withstand changes.

The limited studies on biofilters containing seashell media have failed to explore physical variables such as pH. This is the first study to consider the impact of these variables on the performance of the biofilter. While these variables were not

found to greatly affect the functioning of the biofilter, it is valuable to examine these from time to time in order to have an understanding of potential variables that could eventually cause problems. While some of the biofiltration literature suggests investigation of these types of parameters and their consideration in biofilter design, this approach has not been always been used in the field (Devinny, Deshusses, and Webster, 1998). Having a baseline understanding of operational parameters is important for optimal performance.

Conclusion

The seashell biofilter at Lake Wildwood WWTP appears to be successful at removing odorous sulfur compounds to a level of greater than 99%. This reduction finds the odorous compounds below their odor thresholds. Sampling at Lake Wildwood in March 2012, August 2012, and September 2013 to explore the biofilter's ability to treat odors and other VOCs found that the predominant odorous compounds were H_2S (rotten eggs), MM (rotten vegetable), and DMS (rotten vegetable). GC-MS and GC-Sniff analysis indicated the presence of DMDS (rotten vegetable) and DMTS (rotten garlic) and the possibility of additional sulfides in the inlet air, which were eliminated in the outlet. Daily spikes in the H_2S concentration were observed during the hours slightly following when residents awoke in the morning and when they returned home for the evening. Seasonal differences in H_2S concentrations showed higher levels in the summer (18 ppm average) compared to the spring (10 ppm average).

This paper addresses new findings on sulfides and other odorants that have not previously been reported for seashell biofilters. The OPM indicated the presence of sulfides (rotten egg and rotten vegetable) in the biofilter inlet samples, while the outlet samples had mainly odors characteristic of a blank bag (phenol, plastic, glue, etc.). The chemical nature of the fecal odors in some inlet samples was not able to be defined by GC-MS or GC-Sniff systems used. Further work is needed to define the chemical nature of these odors. OPM measurements by odor panels were confirmed by the results of GC-MS, GC-Sniff, and specific chemical analyses.

The use of the OPM provides transparency of the appropriate chemical analyses required, unlike many other types of odor panel measurements. Were the biofilter not functioning to remove greater than 99% of odorous compounds, OPM would help identify which compounds were still causing a nuisance. H_2S and MM had a similar relative strength (940× to 38,000× and 1500× to 33,000×) indicating that both compounds present a probable nuisance. DMS had a much lower relative strength (9.2× to 30×), indicating that it is probably a lesser nuisance at the WWTP. However, GC-MS and GC-Sniff analyses provided important additional information that DMDS, DMTS, and the probable presence of additional sulfide compounds were apparently enhancing the rotten vegetable odors observed by the OPM.

Odorous aldehydes were all found at levels well below their odor threshold concentrations in both the inlet and outlet of the biofilter. The presence of aldehydes in the inlet and outlet is indicative of biodegradation activity of the microbes within the biofilter. Organic acids were not found above their method detection limits in the inlet or outlet of the biofilter. These data are consistent with the odor profile data where sweet odors characteristic of aldehydes and rancid odors typical of carboxylic acids were not observed. Very minimal concentrations of aromatics were detected, corresponding to the lack of industrial waste treated at the plant.

The pH of the biofilter was found to be neutral in the water within the seashells themselves. The seashells appear to be limited in their ability to correct the pH in the biofilter recirculating water. This biofilter may be able to support neutral pH organisms that have not been previously observed to inhabit other types of biofilters if the biofilter is well controlled. Control of the system may be dependent on the amount of flush water used, inlet loading, or other factors. Maintenance of a neutral pH is promising for organisms that have been observed in sulfide-removing biofilters that have experienced performance declines with acid accumulation, such as *Hyphomicrobium* species. The organisms found in the Lake Wildwood Biofilter will be discussed in future publications. Other water sampling indicated that more biological activity takes place in the first stage of the biofilter (oyster shells) and that the biofilter is an aerobic environment. All measurements performed indicate that the seashell biofilter was efficient at removing of odorous sulfur compounds following several years of operation and that removal efficiency seems to be independent of the pH of the system. Future microbiological investigations and laboratory-based studies hope to provide understanding of the biofilter communities and allow for a more efficient assessment of operating parameters that can be applied to optimize the function of seashell biofilters.

ORCID

Samantha Abraham http://orcid.org/0000-0003-0677-0412

References

American Public Health Association. 2012a. *Standard Methods for the Examination of Water and Wastewater*. 22nd ed. Washington, DC: American Public Health Association (APHA), the American Water Works Association (AWWA), and the Water Environment Federation (WEF).

American Public Health Association. 2012b. *Standard Methods for the Examination of Water and Wastewater: Method 2170*. 22nd ed. Washington, DC: American Public Health Association (APHA), the American Water Works Association (AWWA), and the Water Environment Federation (WEF).

Bergey, D.H., R.E. Buchanan, N.E. Gibbons, and American Society for Microbiology. 1974. *Bergey's manual of determinative bacteriology*, 8th ed. Baltimore, MD: Williams & Wilkins.

Burlingame, G., I. Suffet, D. Khiari, and A. Bruchet. 2004. Development of an odor wheel classification scheme for wastewater. *Water Sci. Technol*. 49(9):201–9.

Burlingame, G.A. 1999. Odor profiling of environmental odors. *Water Sci. Technol*. 40(6):31–38. doi:10.1016/S0273-1223(99)00534-X

Carlson, D.A., and C.P. Leiser. 1966. Soil beds for the control of sewage odors. *J. Water Pollut. Control Fed*. 38(5):829–40.

Curren, J. 2012. Characterization of odor nuisance. DrEnv, Environmental Science and Engineering, University of California, Los Angeles, Los Angeles, CA.

Curren, J., C.L. Snyder, S. Abraham, and I.H. Suffet. 2014. Comparison of two standard odor intensity evaluation methods for odor problems in air or water. *Water Sci. Technol*. 69(1):142–46. doi:10.2166/wst.2013.567

De Bont, J.A.M., J.P. Van Dijken, and W. Harder. 1981. Dimethyl sulphoxide and dimethyl sulphide as a carbon, sulphur and energy source for growth of *Hyphomicrobium* S. *J. Gen. Microbiol*. 127:315–23. doi:10.1099/00221287-127-2-315

Delhomenie, M.-C., and M. Heitz. 2005. Biofiltration of air: A review. *Crit. Rev. Biotechnol*. 25:53–72. doi:10.1080/07388550590935814

Detection Instruments Corporation. 2013. OdaLog Logger L2. Phoenix, AZ: Detection Instruments Corporation.

Devinny, J.S., M.A. Deshusses, and T.S. Webster. 1998. *Biofiltration for air pollution control*. Boca Raton, FL: CRC Press.

Devos, M., F. Patte, J. Rouault, P. Laffort, and L. J. Van Gemert. 1990. *Standardized Human Olfactory Thresholds*. New York, NY: Oxford University Press.

Godayol, A., M. Alonso, E. Besalú, J.M. Sanchez, and E. Anticó. 2011. Odour-causing organic compounds in wastewater treatment plants: Evaluation of headspace solid-phase microextraction as a concentration technique. *J. Chromatogr. A* 1218(30):4863–68. doi:10.1016/j.chroma.2011.02.017

Hayes, J.E., R.J. Stevenson, and R.M. Stuetz. 2014. The impact of malodour on communities: A review of assessment techniques. *Sci. Total Environ*. 500:395–407. doi:10.1016/j.scitotenv.2014.09.003

Higgins, M.J., Y.-C. Chen, J. Novak, D. Glindemann, R. Forbes, Z. Erdal, J. Witherspoon, D. McEwen, S. Murthy, J.R. Hargreaves, and G. Adams. 2008. A multi-plant study to understand the chemicals and process parameters associated with biosolids odors. *Environ. Eng. Appl. Res. Pract*. Winter:27–38.

Higgins, M.J., Y.-C. Chen, D.P. Yarosz, S.N. Murthy, N.A. Maas, D. Glindemann, and J.T. Novak. 2006. Cycling of volatile organic sulfur compounds in anaerobically digested biosolids and its implications for odors. *Water Environ. Res*. 78(3):243–52. doi:10.2175/106143005X90065

Hwang, Y., T. Matsuo, K. Hanaki, and N. Suzuki. 1994. Removal of odorous compounds in wastewater by using activated carbon, ozonation and aerated biofilter. *Wat. Res*. 28(11):2309–19. doi:10.1016/0043-1354(94)90046-9

Hwang, Y., T. Matsuo, K. Hanaki, and N. Suzuki. 1995. Identification and quantification of sulfur and nitrogen containing odorous compounds in wastewater. *Water Res*. 29(2):711–18. doi:10.1016/0043-1354(94)00145-W

Iranpour, R., H.J. Cox, M.A. Deshusses, and E.D. Schroder. 2005. Literature review of air pollution control biofilters and biotrickling filters for odor and volatile organic compound removal. *Environ. Prog*. 24(3):254–67. doi:10.1002/ep.10077

Jones, K. A. Martinez, and M. Rizwan. 2005. Sulfur toxicity and media capacity for H_2S removal in biofilters packed with a natural or a commercial granular medium. *J. Air Waste Manage. Assoc*. 55:415–20. doi:10.1080/10473289.2005.10464636

Kouyoumjian, H., and N.A. Saliba. 2006. Mass concentration and ion composition of coarse and fine particle in an urban area in Beirut: Effect of calcium carbonate on the absorption of nitric and sulfuric acids and the depletion of chloride. *Atmos. Chem. Phys*. 6:1865–1877.

Lebrero, R., L. Bouchy, R. Stuetz, and R. Munoz. 2011. Odor assessment and management in wastewater treatment plants: a review. *Crit. Rev. Environ. Sci. Technol*. 41(10). doi:10.1080/10643380903300000

Leson, G., and A.M. Winer. 1991. Biofiltration: An innovative air pollution control technology for VOC emissions. *J. Air Waste Manage. Assoc*. 41(8):1045–54. doi:10.1080/10473289.1991.10466898

Liang, C., K.C. Das, and R.W. McClendon. 2003. The influence of temperature and moisture contents regimes on the aerobic microbial activity of a biosolids composting blend. *Bioresource Technol*. 86(2):131–37. doi:10.1016/S0960-8524(02)00153-0

Lomans, B.P., A. Pol, and H.J.M. Op den Camp. 2002. Microbial cycling of volatile organic sulfur compounds in anoxic environments. *Water Sci. Technol*. 45(10):55–60.

Massoudinejad, M.R., M. Manshouri, M. Khatibi, A. Adibzadeh, and H. Amini. 2008. Hydrogen sulfide removal by *Thiobacillus thioparus* bacteria on seashell bed biofilters. *Pakistan J. Biol. Sci*. 11(6):920–24. doi:10.3923/pjbs.2008.920.924

McSweeney, P.L.H. 2004. Biochemistry of cheese ripening. *Int. J. Dairy Technol*. 57(2/3):127–44. doi:10.1111/j.1471-0307.2004.00147.x

Muñoz, R., E.C. Sivret, G. Parcsi, R. Lebrero, X. Wang, I. H. Suffet, and R.M. Stuetz. 2010. Monitoring techniques for odour abatement assessment. *Water Res*. 44(18):5129–49. doi:10.1016/j.watres.2010.06.013

Nagata, Y., and N. Takeuchi. 2003. Measurement of odor threshold by triangle odor bag method. *Odor Measurement Review*, 118–127. Tokyo, Japan: Office of Odor, Noise and Vibration Environmental Management Bureau, Ministry of the Environment, Government of Japan.

Naples, B.K. 2010. Performance validation of the first North American shell-based biological air treatment system. *Proc. Water Environ. Fed*. 2010 (3):841–54. doi:10.2175/193864710802768217

Oyarzun, P., F. Arancidbia, C. Canales, and A.E. German 2003. Biofiltration of high concentration of hydrogen sulphide using Thiobacillus thioparus. *Process Biochem*. 39:165–70. doi:10.1016/S0032-9592(03)00050-5

Rosenfeld, P., Clark, J., Hensley, A., and Suffet, I. 2007. The use of an odour wheel classification for the evaluation of human health risk criteria for compost facilities. *Water Science & Technology*. 55(5):345–357.

Sekyiamah, K., H. Kim, L.L. McConnell, A. Torrents, and M. Ramirez. 2008. Identification of seasonal variations in volatile sulfur compound formation and release from the secondary treatment system at a large wastewater treatment plant. *Water Environ. Res*. 80(12):2261–67. doi:10.2175/106143008X304677

Smet, E., G. Chasaya, H. Van Langenhove, and W. Verstraete. 1996. The effect of inoculation and the type of carrier material used on the biofiltration of methyl sulphides. *Appl Microbiol Biotechnol*. 45:293–98. doi:10.1007/s002530050686

Smet, E., H. Van Langenhove, and W. Verstraete. 1993. Long-term stability of a biofilter treating dimethyl sulphide. *Appl. Microbiol. Biotechnol*. 46:191–96. doi:10.1007/s002530050804

Smith, N.A., and D.P. Kelly. 1988. Mechanism of oxidation of dimethyl disulphide by *Thiobacillus thioparus* strain E6. *J. Gen. Microbiol*. 134:3031–39. doi:10.1099/00221287-134-11-3031

Suffet, I.H., B.M. Brady, J.H.M. Bartels, G. Burlingame, J. Mallivialle, and T. Yohe. 1988. Development of the flavor profile analysis method into a standard method for sensory analysis of water. *Water Sci. Technol*. 20 (8–9):1–9.

Suffet, I.H., G.A. Burlingame, P.E. Rosenfeld, and A. Bruchet. 2004. The value of an odor-quality-wheel classification scheme for wastewater treatment plants. *Water Sci. Technol*. 50(4):25–32.

Suffet, I.H., and P. Rosenfeld. 2007. The anatomy of odour wheels for odours of drinking water, wastewater, compost, and the urban environment. *Water Sci. Technol*. 55(5):335–44. doi:10.2166/wst.2007.196

Trabue, S.L., J.C. Anhalt, and J.A. Zahn. 2006. Bias of Tedlar bags in the measurement of agricultural odorants. *J. Environ. Qual*. 35(5):1668–77. doi:10.2134/jeq2005.0370

Van Durme, G., S.-H. Koh, and A. Gay. 2010. Performance validation of a shell-media biological odor control system. *Proc. Water Environ. Fed*. 2010 (15):2093–105. doi:10.2175/193864710798159228

Vitko, T., I.H. Suffet, Y. Zhou, and S. Abraham. 2014. 2014 OCSD Odor Control Master Plan. Odors and Air Pollutants. Miami, FL: OCSD.

Wright, D.W., D.K. Eaton, L.T. Nielsen, F.W. Kuhrt, J.A. Koziel, J.P. Spinhirne, and D.B. Parker. 2005. Multidimensional gas chromatography-olfactometry for the identification and prioritization of malodors from confined animal feeding operations. *J. Agric. Food Chem*. 53 (22):8663–72. doi:10.1021/jf050763b

Yoon, G.-L., B.-T. Kim, B.-O. Kim, and S.-H. Han. 2003. Chemical–mechanical characteristics of crushed oyster-shell. *Waste Manage*. 23(9):825–34. doi:10.1016/S0956-053X(02)00159-9

About the authors

Samantha Abraham is a public health microbiologist.

Scott Joslyn is the wastewater operations manager at Nevada County Sanitation District No. 1.

I. H. (Mel) Suffet is a professor at the Fielding School of Public Health in the University of California, Los Angeles.