11-2015

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Energy Reports 1 (2015) 169–174

Contents lists available at ScienceDirect

Energy Reports

journal homepage: www.elsevier.com/locate/egyr

An experimental investigation of microalgal dewatering efficiency of belt filter system

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ARTICLE INFO

Article history:
Received 28 April 2015
Received in revised form
27 July 2015
Accepted 11 August 2015

Keywords:
Microalga
Belt filter
Dewatering efficiency
Harvesting

ABSTRACT

The objective of this study was to investigate the microalgal dewatering efficiency of a belt filter system for feed concentrations below 10 g dry wt./L. A prototype belt filtration system designed for 50 g dry wt./L microalgal feed concentration was used for this investigation. The highest concentration of microalgal suspension available for testing on the prototype belt filtration system was 6 g dry wt./L obtained from biomass settling tanks at the Lawrence, Kansas domestic wastewater treatment plant. For preparation of feed suspension with concentrations below 10 g dry wt./L, microalgal cultivation was followed by flocculation. A mixed laboratory culture of freshwater species dominated by three eukaryotic green microalgae (Chlorella vulgaris, Scenedesmus sp., and Kirchneriella sp.) was cultivated in wastewater effluent. This was followed by flocculation which resulted in a microalgal feed suspension concentration of 4 g dry wt./L. Belt dewatering tests were conducted on microalgal suspensions with feed concentrations of 4 g dry wt./L and 6 g dry wt./L. The maximum microalgal recovery with the belt dewatering system was 46% from the 4 g dry wt./L, and 84% from the 6 g dry wt./L suspensions respectively. The results of this study indicate that microalgal suspension concentrations as low as 6 g dry wt./L can be recovered with a belt filter system improving the overall dewatering efficiency of the system.

1. Introduction

Climate change policy and concerns regarding future energy security have stimulated an unprecedented increase in the production of bioenergy sources that have the potential to reduce future greenhouse gas emissions (Smith et al., 2012). Microalgae are of particular interest because many of the resources required for their mass cultivation can be provided by waste streams (e.g., municipal wastewater: Sturm and Lamer, 2011; carbon dioxide from industrial flue gas: Brentner et al., 2011), and because microalgal cells synthesize many different harvestable bioproducts having a wide variety of compositions and uses (Menetrez, 2012). In particular, microalgae possess many favorable characteristics as a biofuel feedstock, including rapid growth rates and high lipid contents (Chen et al., 2011), high areal energy (Chisti, 2007; Hu et al., 2008), and the ability to avoid undesirable 'food versus fuel' conflicts via the cultivation of microalgal biomass on marginal lands (Singh and Gu, 2010). Production to processing of microalgae is shown in Fig. 1. Nonetheless, profitable large-scale production has not yet been demonstrated (NRC, 2012).

The high operational costs associated with microalgal harvesting are a major challenge (Uduman et al., 2010) due to the very dilute nature of the microalgal suspension and their small cell size (Grima et al., 2003). An optimal harvesting method for microalgae should be independent of the microalgal species being cultivated, and also should have a low chemical and energy demand (Amaro et al., 2011). Centrifuge and belt filter are commonly used microalgal dewatering systems (Spellman, 1997). The primary difference between a centrifuge and the belt filter system is the principle of separation. A centrifuge applies centrifugal forces to the solution to aid the separation of solid and liquid. For a belt filter system, the principle of separation is gravity drainage followed by compression shear (Spellman, 1997). Centrifugation is a highly effective method for harvesting microalgae but it has a high energy demand and is expensive (Knuckey et al., 2006). Compared to a centrifuge, belt filter system has lower energy consumption (Grima et al., 2003) and operational costs (Spellman, 1997), has a continuous mode of operation and can be up-scaled. However, microalgal suspension with a concentration of 10–40 g dry wt./L is needed prior to dewatering on a belt filter (Grima et al., 2003; Sturm and Lamer, 2011).

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http://dx.doi.org/10.1016/j.egyr.2015.08.002

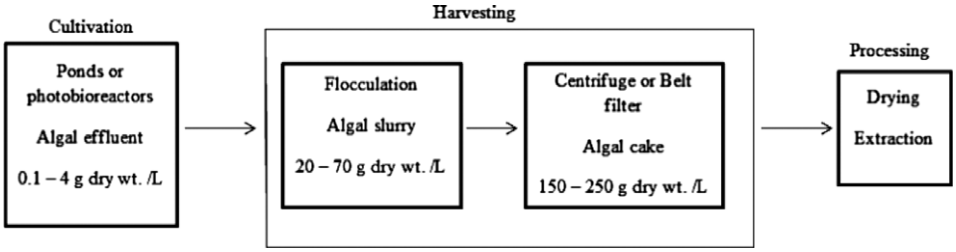

Fig. 1. Schematic of microalgal production and processing (Shelef et al., 1984).

The objective of this study was to further investigate the microalgal dewatering efficiency of a belt filter system for feed concentrations below 10 g dry wt./L.

To further investigate this, microalgal suspensions with feed concentrations of 4 g dry wt./L and 6 g dry wt./L were produced. A prototype belt filter dewatering system consisting of a filter section followed by two drying sections was designed and developed by the authors (Fig. 2(a) and (b)). A doctor blade was installed at the end of the drying section to scrape off the dried algal cake. Air drying was the chosen drying method, due to its low energy and cost requirements. The design was based on filtration tests conducted on 50 g dry wt./L microalgal suspension. The prototype is a 1% scale of a system proposed to process 60,000 gallons (or 227 124.71 L) of 50 g dry wt./L microalgal solution per day. The difference between a standard belt filter system and the prototype belt filter dewatering system developed is the dewatering mechanism. For a standard belt filter press, the principle dewatering mechanism is gravity drainage followed by compression shear. The principle dewatering mechanism of the prototype belt filter dewatering system is gravity drainage (Fig. 2(c)). Another system developed based on belt filter gravity drainage dewatering mechanism is Salsnes Water to Algae Treatment (SWAT) technology (Sahu et al., 2013). However, there are several differences between SWAT technology and the prototype belt filter dewatering system developed by the authors. Firstly, the filter section of the SWAT technology is enclosed in a chamber. Secondly, the belt movement in the filter sections of the prototype belt filter dewatering system and the SWAT technology are in opposite directions. Lastly, there is no drying unit in the SWAT technology.

To determine the filtration belt mesh needed for the prototype belt filter dewatering system developed, gravity filtration tests were conducted on microalgal samples at their stationary growth phase. These tests used a range of polyester mesh sizes from 10 to 200 μm. Based on the test results a 70 μm mesh size resulted in the highest microalgal recovery rate (Fig. 3). Using 70 μm polyester filter mesh, belt dewatering tests were conducted on microalgal suspensions with feed concentrations of 4 g dry wt./L and 6 g dry wt./L.

2. Materials and methods

2.1. Microalgal feed suspension preparation

2.1.1. Microalgal suspension with feed concentration of 4 g dry wt./L

A mixed culture of microalgal species dominated by three eukaryotic green algae (*Chlorella vulgaris, Scenedesmus sp.*, and *Kirchneriella sp.*) was cultivated in domestic wastewater effluent from the Lawrence, Kansas wastewater treatment plant. Flocculant type, dosage and pH that were the most efficient and cost-effective for the cultivated microalgal suspension were determined using jar tests. The results of the jar tests were then used to prepare sufficient volume of concentrated microalgal suspension for belt dewatering tests. A total of 54 l of 4 g dry wt./L microalgal suspension were produced.

Table 1
Optical density and biomass concentration measurements of microalgal culture over a cultivation period of 8 days.

Culture time (days)	$OD_{600\ nm}$	Biomass concentration (g dry wt./L)
2	5.4 ± 0.45	0.7 ± 0.09
4	8.2 ± 1.6	1.1 ± 0.3
6	11.3 ± 0.5	1.45 ± 0.1
8	12.5 ± 1.5	1.5 ± 0.3

2.1.1.1. Microalgae cultivation. Mixed-species microalgae were cultured in a 272 L glass photobioreactor with an operating volume of 208 L. This photobioreactor was initially filled with prechlorination wastewater effluent collected from the secondary treatment stage of the Lawrence, KS, wastewater treatment plant. Then an inoculum was added that was comprised of a natural mixed species assemblage of three eukaryotic green algae (*Chlorella vulgaris, Scenedesmus sp.*, and *Kirchneriella sp.*). 650 g of inorganic nitrogen (supplied as KNO3) and 160 g of inorganic phosphorus (supplied as KH2PO4) were added to the photobioreactor and replenished on a weekly basis to provide nutrients for the growing microalgal community. Light was provided by LED light panels (∼265 μmol/[m^2 s]) with a 12 h on, 12 h off light: dark cycle.

Because wastewater effluent typically contains insufficient inorganic carbon for optimal microalgal growth (Benemann et al., 2003), commercial-grade CO_2 was bubbled into the photobioreactor. The water column pH in the photobioreactor was controlled using a pH controller (Milwaukee Instruments, MC122) to regulate the flow of CO_2. For this experiment the pH of the photobioreactor was set at 6.5 and the room temperature was maintained at 23 ± 1 °C. To provide turbulent mixing, room air was bubbled into the tank at a rate of 4.6 L/min using four aerators placed at each of the four corners of the tank. This turbulent mixing helped to maintain the microalgal cells in suspension during cultivation. Microalgal biomass measurements were made at different stages of post-inoculation growth using a calibrated UV/Vis Spectrophotometer (Thermo Fisher Scientific Model G10S) followed by a standard total suspended solids test (Becker, 1994). Microalgal culture in the 272 L glass photobioreactor achieved a concentration of 1.5 ± 0.3 g dry wt./L at the stationary growth phase in 8 days (Table 1). Bench scale flocculation was conducted on the biomass samples taken from the photobioreactor.

2.1.1.2. Bench scale flocculation. Jar tests were conducted to determine the flocculation conditions (flocculant type, dosage and pH) that were the most efficient and cost-effective (Appendix). 52 L of microalgal culture harvested at their stationary growth phase concentration (1.5 ± 0.2 g dry wt./L) were pumped into a 56 L graduated cylinder equipped with a spigot to allow decantation of the flocculation product. Pre-test pH value of the microalgal suspension was adjusted to 6.5 using 0.1 M NaOH or 0.1 M HCl. Alum at a dosage of 200 mg/L was added to the microalgae suspension and mixed rapidly at 700 rpm for 60 s, followed by slow mixing at 60 rpm for 15 min using a 1.2 HP (895 W) variable speed mixer

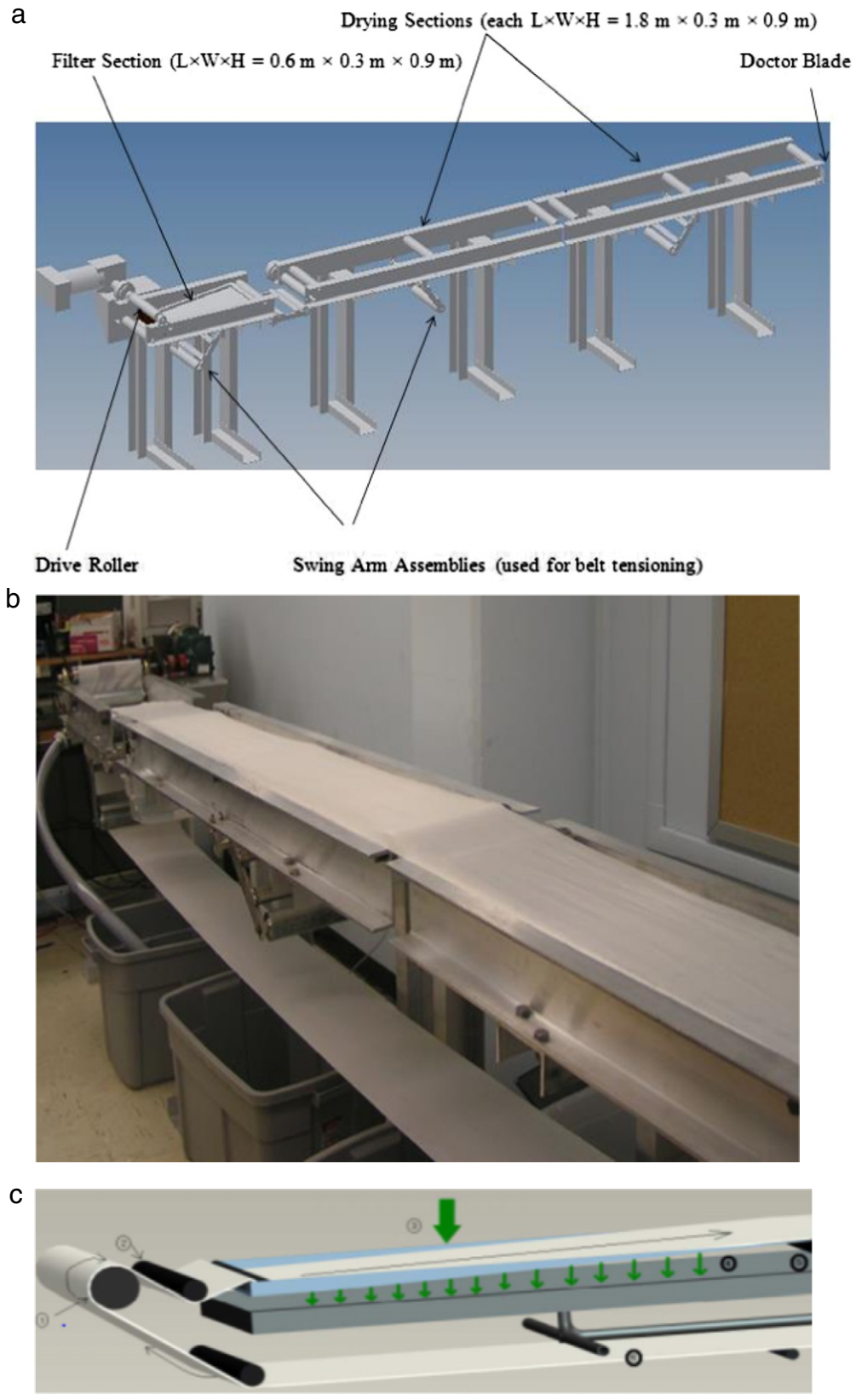

Fig. 2. Design and development of belt filter dewatering system. (a) 3D CAD drawing in Autodesk Inventor 2011. (b) Dewatering test set-up (c) Dewatering mechanism—gravity drainage.

with an axial-flow impeller. The flocculated microalgal suspension was then allowed to settle for 2 h, and at the end of the settling period, approximately 5 L of ∼4 g dry wt./L concentrated microalgal suspension were collected. This procedure was repeated multiple times until a total of 54 L of ∼4 g dry wt./L concentrated microalgal suspension were collected.

Table 2
Belt filter dewatering test performance. Belt dewatering tests were conducted using 70 μm mesh size filter on sample suspensions, 18 L of 4 g dry wt./L and 6 L of 6 g dry wt./L, to determine the percent of microalgae recovered (mean ± standard deviation, $n = 3$).

Number of successive filtrations (#)	4 g dry wt./L suspension Cumulative microalgae recovered (%)	6 g dry wt./L suspension Cumulative microalgae recovered (%)
1	23.26 ± 7.2	65 ± 6.5
2	31.7 ± 5.9	76 ± 5.7
3	36.6 ± 6.6	82 ± 5.8
4	40.5 ± 4.3	83.3 ± 3.6
5	43.7 ± 0.7	84.2 ± 1.7
6	46.1 ± 0.1	84.7 ± 0.06

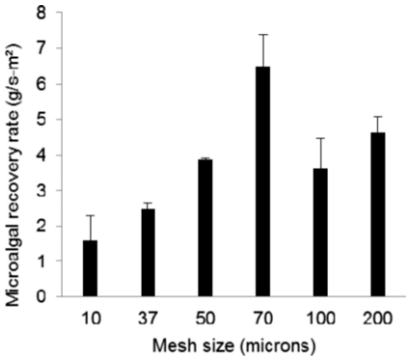

Fig. 3. Determination of mesh size with the highest microalgal recovery rate. Gravity filtration tests (Sigma–Aldrich vacuum filter assembly for 47 mm Whatman GF/C glass filter with glass support—Product # Z290432) were conducted on samples of microalgal culture at its stationary growth phase with an initial biomass concentration of 1.5 ± 0.3 g dry wt./L for a range of mesh sizes to measure microalgal recovery rate (g m^{-2} s^{-1}) = $W_{cake}/(FA \times FT)$, where W_{cake} is the mass of the wet microalgal cake collected on the filter in grams, FA is the filter area (7×10^{-3} m^2), and FT is the filtration time in seconds. Error bars represent the standard deviation ($n = 5$) of measured values of microalgal recovery rate.

2.1.2. Microalgal suspension with feed concentration of 6 g dry wt./L

The highest concentration of microalgal suspension available for testing on the prototype belt filtration system was 6 g dry wt./L. This was obtained from biomass settling tanks at the Lawrence, Kansas domestic wastewater treatment plant.

2.2. Belt dewatering test procedure

Three belt dewatering tests each were conducted for microalgal suspension with feed concentration of 4 g dry wt./L and 6 g dry wt./L. The belt filter mesh used in this testing was the 70 μm polyester mesh identified in the earlier filtration testing. The belt speed for the dewatering system was set at 0.7 mm per second. The depth of the microalgal suspension in the filter section was controlled by a level sensor driving a pumping system.

Microalgal suspension was pumped into the filter section through a manifold. Filtered microalgae were air dried on the belt for 8 h at room temperature at zero belt speed. Air drying was conducted to improve the accuracy of measurement of recovered microalgae. The dried microalgal cake was then scraped off manually and the harvested biomass weight was recorded.

The percent of dried microalgae recovered (PR) was calculated from the following equations:

$$PR(\%) = (M_D/M_I) \times 100 \quad (1)$$

$$M_I = (C_I \times V_I)/10^6 \quad (2)$$

where M_D is the recovered mass of the dried microalgae (in grams); M_I is the initial total suspended solids mass in the suspension (in grams); C_I is the initial concentration of the microalgal suspension (in milligrams dry wt./Liter); and V_I is the feed volume of microalgal suspension (Liters).

3. Results

The percent of microalgae recovered during belt filter testing is shown in Table 2. The 4 g dry wt./L microalgae suspension yielded a maximum of 46% recovered microalgae. The 6 g dry wt./L microalgae suspension yielded a maximum of 84% recovered microalgae. The authors assumed the percent of microalgae recovered to be independent of the microalgal species or chemical treatment, for a particular feed concentration. Biomass losses of microalgae embedded in the filter belt, and not recoverable, ranged from 3% to 7%. The need for multiple filtration passes of the microalgal suspension was primarily due to leakage in the filter test section of the belt filter system. The next step in this line of research would involve sealing the filter section of the system.

4. Discussion

The results of the study indicate that the system could effectively recover concentrations as low as 6 g dry wt./L thereby improving the overall dewatering efficiency of the system. To further improve the overall dewatering efficiency of the system, effect of machine parameters such as filter feed rate and belt speed must be further explored. Belt filter performance characteristics such as flow throughput and biomass recovery rate need to be investigated as it takes the time taken to recover the biomass into consideration.

5. Conclusions

The objective of this study was to investigate the microalgal dewatering efficiency of a belt filter system for feed concentrations below 10 g dry wt./L. The results of this study indicate that microalgal suspension with concentrations as low as 6 g dry wt./L can be effectively recovered improving the overall dewatering efficiency of the belt filter system. For microalgal suspension with concentration of 4 g dry wt./L, the percent of microalgae recovered dropped significantly. This could be partly attributed to the leakages in the filter section of the system. The next step in this line of research would involve sealing the filter section of the system.

Future studies must address several questions. Firstly, what is the effect of machine parameters such as filter feed rate and belt speed on the percent of microalgae recovered? Secondly, for a microalgal suspension with concentrations below 10 g dry wt./L, what is the belt filter performance in terms of flow throughput and microalgal recovery rate? Finally, the question that needs to be addressed is whether the belt filter system would satisfy the requirements of an optimal microalgal harvesting technique—system reliability independent of the properties of microalgal feed suspension, low chemical and energy demand. The results of this study provide researchers with data for achieving energy efficient harvesting and processing of microalgae.

Acknowledgments

This work was supported by the University of Kansas Transportation Research Institute (Grant # FED0067503). We thank Dr. Belinda Sturm for her guidance and support and Dr. Edward Peltier for his thoughtful reviews of early drafts of this manuscript. We also thank the staff at the Lawrence, Kansas wastewater treatment plant.

Appendix

A.1. Flocculation test procedure

Three different flocculants were chosen for testing—chitosan powder, aluminum potassium sulfate dodecahydrate and zetag 7650. Both chitosan powder and aluminum potassium sulfate dodecahydrate (an inorganic cationic flocculant), were obtained from Sigma–Aldrich Company Ltd. (Missouri, USA). Zetag 7650, a high molecular weight synthetic cationic polymer used for sludge dewatering (Danquah et al., 2009), was obtained from Southwest Engineers (Louisiana, USA).

Jar tests were conducted to determine the flocculation conditions (flocculant type, dosage and pH) that were the most efficient and cost-effective. Three flocculant mixtures were evaluated: (1) Aluminum sulfate (Alum) alone; (2) Alum combined with zetag 7650 (10:1 by mass); and (3) Alum combined with chitosan (10:1 by mass). Stock solutions for each of the three flocculants – chitosan (Divakaran and Pillai Sivasankara, 2002), alum, and zetag 7650 (Tillman, 1996) – were prepared at concentrations 10 g/L, 1 g/L and 1 g/L, respectively. Jar tests were then performed on a multi-jar magnetic stirrer using 500 mL samples of microalgal suspension. Flocculation was conducted for a range of pH and dosage values for each of the three flocculant mixtures (Figs. A.1 and A.2). Pre-test pH values of the microalgal samples were adjusted using 0.1 M NaOH or 0.1 M HCl. The desired flocculant mixture was added to the microalgae samples and mixed rapidly at 100 rpm for 60 s, followed by slow mixing at 60 rpm for 15 min. After flocculation, the suspension was allowed to settle for a period of 30 min. Flocculant performance was then evaluated as clarification efficiency (Eq. (A.1)).

$$\text{Clarification Efficiency}(\%) = (1 - OD_s/OD_f) \times 100 \qquad \text{(A.1)}$$

where OD_s is the optical density of the supernatant after flocculation of the microalgal suspension, and OD_f is the optical density of the feed sample. Optical density was measured at 600 nm for all samples, using a 1 cm path length cuvette.

The combined dosage and pH level that resulted in the highest clarification efficiency (Eq. (1)) was determined for the following two mixtures—(1) Alum and chitosan (10:1 by mass), and (2) Alum and zetag 7650 (10:1 by mass). The highest clarification efficiencies of the two above-mentioned flocculant mixtures were then compared to that of alum alone (Dosage = 10 mg/L; pH = 6.5). Because the clarification efficiencies of the three flocculant mixtures were essentially the same, the lowest cost flocculant was chosen to prepare the concentrated microalgal suspension for the belt dewatering tests.

A.2. Determination of optimum flocculant type, dosage and pH for stationary growth phase culture

Microalgal culture in the 272 L glass photobioreactor achieved a concentration of 1.5 ± 0.3 g dry wt./L at the stationary growth phase in 8 days. Studies on closed photobioreactors such as tubular or flat plate (systems) have reported biomass concentrations on the order of 2 g/L with a maximum of 5 g dry wt./L (Pulz, 2001).

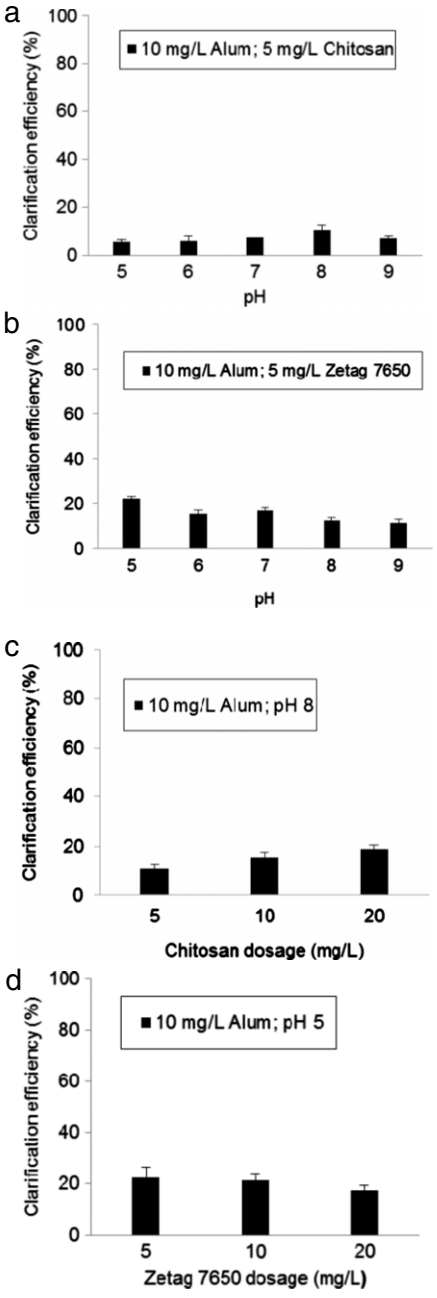

Fig. A.1. Determination of pH and dosage values that result in the highest clarification efficiency for the two flocculant mixtures—alum + chitosan and alum + zetag 7650. Jar tests were conducted on microalgal suspensions with an initial biomass concentration of 1.5 ± 0.3 g dry wt./L to measure clarification efficiency (mean ± standard deviation; $n = 3$). (a) A coagulation dose of 10 mg/L for alum and 5 mg/L for chitosan was used for the tested pH range. Clarification efficiency at pH value of 8 was significantly higher than all other clarification efficiencies. Four one-way ANOVA tests were conducted comparing clarification efficiency at pH value 8 with each of the other pH values and $p < 0.05$ for every test. (b) A coagulation dose of 10 mg/L for alum and 5 mg/L for zetag 7650 was used for the tested pH range. Clarification efficiency at pH value of 5 was significantly higher than all other clarification efficiencies. Four one-way ANOVA tests were conducted comparing clarification efficiency at pH value 5 with each of the other pH values and $p < 0.05$ for every test. (c) At a fixed alum dosage of 10 mg/L, a pH of 8 was used for a range of chitosan dosages. Clarification efficiency at chitosan dosage of 20 mg/L was significantly higher than all other clarification efficiencies. Two one-way ANOVA tests were conducted comparing clarification efficiency at 20 mg/L dosage with each of the other dosages and $p < 0.05$ for every test. (d) At a fixed alum dosage of 10 mg/L, a pH of 5 was used for a range of zetag 7650 dosages. There were no significant differences in clarification efficiencies at zetag 7650 dosages of 5 and 10 mg/L (One-way ANOVA, $p > 0.05$). Clarification efficiency at 5 mg/L dosage was significantly higher than the 20 mg/L dosage (One-way ANOVA, $p < 0.05$).

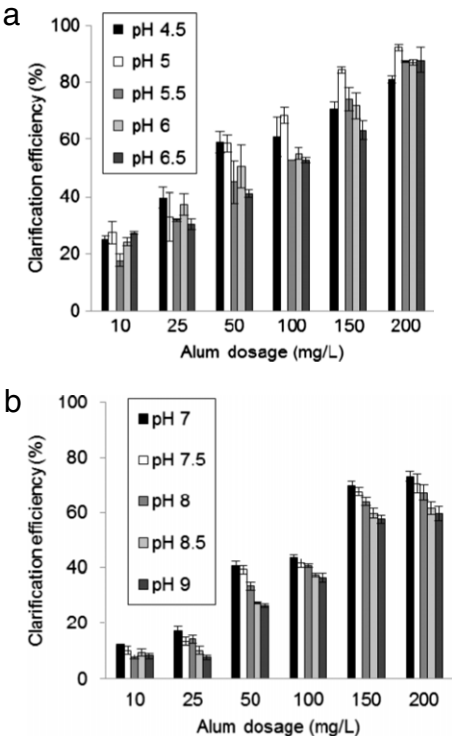

Fig. A.2. Determination of pH and dosage values that result in the highest clarification efficiency for alum. Jar tests were conducted on samples of the microalgal suspension at their stationary phase of growth with an initial biomass concentration of 1.5 ± 0.3 g dry wt./L to measure clarification efficiency (mean ± standard deviation; $n = 3$) of alum. (a) A pH range of 4.5–6.5 in 0.5 pH increments for the tested dosage range. (b) A pH range of 7–10 in 0.5 pH increments for the tested dosage range. For the alum dosage of 200 mg/L there were no significant differences in the clarification efficiency for a pre-test pH range from 5 to 6.5 (One-way ANOVA, $p > 0.05$). Clarification efficiencies were significantly lower for all other tested pH values at 200 mg/L dosage.

The lower biomass productivity in this study was probably due to reduced light intake caused by the photobioreactor structure.

At a fixed alum dosage (10 mg/L), flocculation performance of chitosan improved as the dosage was increased up to a maximum of 20 mg/L. With the alum dosage fixed at 10 mg/L, the flocculation efficiency of zetag 7650, starting at 5 mg/L, decreased with increasing zetag dosage. Danquah et al. (2009), who had similar results, suggested that over dosage of high molecular weight polymers led to a formation of elastic colloids reducing the effectiveness of the polymer as a flocculant. Clarification efficiencies for pH and dosage values for the two flocculant mixtures are listed in Fig. A.1.

A series of tests were conducted, using alum as the flocculant, for a range of pH and dosage values. The results of this testing showed an almost linear increase in clarification efficiency up to an alum dosage of 200 mg/L. At alum dosage of 200 mg/L the pre-test pH was varied from 4.5 to 9. For the alum dosage of 200 mg/L there were no significant differences in the clarification efficiency for a pre-test pH range from 4.5 to 6.5 (Fig. A.2(a), one-way ANOVA, $p > 0.05$). Above a pre-test pH of 6.5 the clarification efficiency significantly decreased (Fig. A.2(b)). The additions of alum, for all dosages tested, increased the pH of the microalgal suspension by 0.5 ± 0.1 pH units. For further testing a pre-test pH value of 6.5 was chosen to reduce the cost involved in lowering the pH of the microalgal solution from 7 to 6.5.

Comparing the highest clarification efficiencies of the two flocculant mixtures, alum + chitosan and alum + zetag 7650, with that of alum alone (Dosage = 10 mg/L; pH = 6.5) showed no significant improvement in clarification efficiency (Fig. A.3). Since alum was the most cost effective further testing was focused on this flocculant.

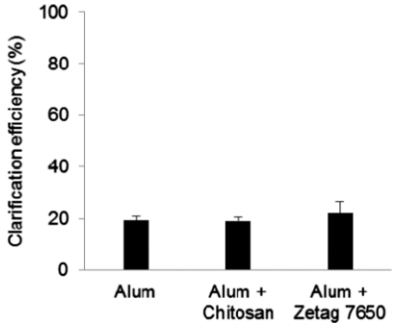

Fig. A.3. Determination of flocculant mixture – alum, alum + chitosan and alum + zetag 7650 – that results in the highest clarification efficiency. Comparing the highest clarification efficiencies of the two flocculant mixtures, alum + chitosan and alum + zetag 7650, with that of alum alone (Dosage = 10 mg/L; pH = 6.5) showed no significant improvement in clarification efficiency (One-way ANOVA, $p > 0.05$).

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