Bioresource Technology 176 (2015) 1–7

Contents lists available at ScienceDirect

Bioresource Technology

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

Evaluation of operating conditions for sustainable harvesting of microalgal biomass applying electrochemical method using non sacrificial electrodes

Rohit Misra, Abhishek Guldhe, Poonam Singh, Ismail Rawat, Thor Axel Stenström, Faizal Bux *

Institute for Water and Wastewater Technology, Durban University of Technology, P.O. Box 1334, Durban 4000, South Africa

HIGHLIGHTS

- Non-sacrificial electrodes were applied for electrochemical harvesting of microalgae.
- Anode depletion and metallic contamination of biomass can be completely avoided.
- Addition of electrolyte is beneficial for the electrochemical harvesting process.
- ECH process has no adverse effect on the lipid extraction process.
- Electrolyte addition improves lipid extraction.

ARTICLE INFO

Article history:
Received 18 September 2014
Received in revised form 2 November 2014
Accepted 4 November 2014
Available online 11 November 2014

Keywords:
Microalgae
Non-sacrificial carbon electrode
Biodiesel
Electrochemical harvesting
Lipid

ABSTRACT

The efficient harvesting of microalgae is considered to be one of the challenging steps of algal biofuel production and a key factor limiting the commercial use of microalgae. To overcome the limitation of metallic electrodes depletion, the application of non-sacrificial electrode was investigated for the electrochemical harvesting (ECH) of microalgae. The effect of applied current, addition of electrolyte and initial pH were parameters investigated. The highest recovery efficiency of 83% was obtained for *Scenedesmus obliquus* at 1.5 A, initial pH 9 and 6 g L^{-1} NaCl with power consumption of 3.84 kWh kg^{-1}. Recovery efficiency of ECH process was comparable to literature reported centrifugation, filtration and chemical flocculation techniques but with a much lower power consumption. The ECH process with addition of electrolyte enhanced the lipid extraction by 22% without any adverse effects. The ECH process with non sacrificial carbon electrodes could be a possible harvesting step at commercial scale microalgal biomass production.

1. Introduction

The recent up-surge in global energy demands has re-invigorated researchers to look into novel resources and ways of acquiring renewable energy from these sources without a net carbon emission into the ecosystem. Microalgae hold promise as a sustainable source of biofuels based on their rapid growth rates and reportedly high concentration of lipids (Ramanna et al., 2014; Sostaric et al., 2012). Microalgae, like any other photosynthetic plants, utilize atmospheric carbon dioxide to synthesize carbohydrates (Singh et al., 2014). If 63.6 million acres of land were used to cultivate algae at a conservative rate of 10 g m^{-2} day^{-1}, then 2 billion tons of carbon dioxide could be captured in the biomass in 1 year (Pienkos and Darzins, 2009). Microalgal biomass finds its application in renewable biofuels production such as biodiesel, biomethane, bioethanol. Also microalgae can be utilized as fish and animal feed, and for production of pharmaceuticals and nutraceuticals (Singh et al., 2014). Economical viability of overall process is the major bottleneck for its successful commercialization. Thus an integrated biorefinery approach where along with biofuel production, value added products from microalgae such as pigments, nutraceuticals, therapeutic chemicals etc. are co-produced, is gaining interest (Sostaric et al., 2012).

Microalgal biodiesel synthesis is a multistep process consisting of cultivation for microalgal biomass production, biomass harvesting, extraction of lipids, and conversion of lipids to biodiesel (Guldhe et al., 2014a). Each step has challenges which need to be alleviated for sustainable and economical biodiesel production. Ensuring high lipid accumulation, effective harvesting and

* Corresponding author. Tel.: +27 313732346; fax: +27 313732777.
E-mail address: faizalb@dut.ac.za (F. Bux).

http://dx.doi.org/10.1016/j.biortech.2014.11.014
0960-8524/

extraction and addressing mass transfer limitations in conversion, exploring and effective extraction of value added co-products are the key focuses of the researchers (Klofutar et al., 2010; Likozar and Levec, 2014; Sostaric et al., 2012). Harvesting is a crucial step which separates the microalgal biomass from water, for effective downstream processing. Harvesting of microalgal biomass has posed many challenges due to the small size and low density of the microalgae (Vandamme et al., 2013). These can be harvested by different methods such as, centrifugation, filtration, sedimentation, flocculation, and flotation but the efficient harvesting of microalgae is considered to be one of the most problematic area in algal biofuels production and a key factor limiting the commercial use of microalgae (Greenwell et al., 2010). Harvesting by conventional methods of centrifugation and filtration are expensive for production of low value product like biodiesel due to relatively dilute cultures that require processing large volumes of water (Amaro et al., 2011). Development of inexpensive harvesting techniques is crucial for economic feasibility of microalgal biodiesel. An efficient microalgal harvesting process should not have deteriorating effect on extraction and quality of lipids and require minimum investment, energy, and maintenance (Poelman et al., 1997).

Microalgal cell walls have a net negative charge because of the presence of acidic polysaccharides (pectin) (Safi et al., 2014). This negative charge, if neutralized can form microalgal cell aggregates, which can be easily separated from medium. This coagulation concept can be used for harvesting of microalgal biomass (Safi et al., 2014). Many natural and chemical coagulants and flocculants can be applied for harvesting of microalgae. Addition of chemical flocculants and coagulants may require high dosages as well as could affect the biomass quality. Recently some researchers have investigated the potential of electrochemical harvesting (ECH) for microalgal biomass recovery from culture medium for commercial products like biodiesel (Kim et al., 2012a; Lee et al., 2013; Vandamme et al., 2011). Electrochemical harvesting is based on the principle of the movement of electrically charged particles in an electric field. The negative surface charge of microalgae (Amaro et al., 2011) will cause them to move towards and accumulate at the anode (positive charge) during the electrochemical harvesting. When in close proximity to the anode (+) the charges will be neutralized and algal 'aggregates' are formed (Aragón et al., 1992). The generation of hydrogen (H_2) and oxygen (O_2) gas in the electrolysis of water at the anode creates bubbles that will float the microalgal aggregates or flocks to the surface where they easily can be skimmed off. The electrochemical harvesting (ECH) process thus leads to the flocculation and flotation of the algae at the same time, without the usual addition of chemical flocculants. Vandamme et al. (2011) applied a electro-coagulation–flocculation process using aluminum anode for harvesting of two microalgal species viz. the fresh water *Chlorella vulgaris* and the marine *Phaeodactylum tricornutum*. Uduman et al. (2011) studied an electrocoagulation method for harvesting of the marine microalgae *Chlorococcum* sp. and *Tetraselmis* sp. Electrolytic methods are a potential approach to recover microalgal biomass which operates at low energy input and does not require the addition of any chemical flocculants (Alfafara et al., 2002; Gao et al., 2010a,b).

Electrochemical approaches have been used in product recovery, waste destruction (Neti and Misra, 2012) and chemical synthesis with several benefits in terms of costs and safety. The electrochemical approach has been employed by several researches for removal of microalgae from surface water and wastewater (Alfafara et al., 2002; Gao et al., 2010b). ECH processes are environmentally acceptable, non-species specific, safe and cost effective for implementation at commercial scale (Lee et al., 2013; Uduman et al., 2011).

The main limitation of the harvesting of algae by electrochemical methods has so far been the depletion of the metallic electrodes (Kim et al., 2012a,b). Depletion of electrodes could cause metallic contamination of microalgal biomass and wastewater generation. Electrode depletion not only increases the cost of harvesting but affects the quality of the harvested algae in the form of metallic contaminations (Kim et al., 2012a,b; Uduman et al., 2011). To overcome this limitation the application of non-sacrificial electrodes are required for the electrochemical harvesting of microalgae. Non-sacrificial electrodes totally avoid formation of metal hydroxide unlike metallic electrodes, and thus can be used as replacements in ECH processes overcoming challenges and improving economics. The electrochemical harvesting approach itself is novel for microalgae and application of non-sacrificial electrodes has not been thoroughly investigated. Non-sacrificial electrodes may face drawback of attrition and non-steady state operation if used in particulate form, possible solution to avoid this problem is use of thick plate electrodes which are steady during the operation.

This work aims to demonstrate the electrochemical harvesting (ECH) of microalgae (*Scenedesmus obliquus*) by using non-sacrificial carbon electrodes which is a combination of electroflocculation and electroflotation processes. The influence of several important variables on the ECH process was investigated, such as applied current, electrolyte concentration (by adding additional electrolyte) and the initial pH. Earlier investigations of electrochemical processes for harvesting of microalgal biomass have not thoroughly evaluated the effect of the ECH process on the further downstream processes for biodiesel production. In this study both the effects of varying applied current and pH as well as the addition of electrolyte for lipid extraction was evaluated.

2. Methods

2.1. Cultivation of microalgae

The oleaginous green microalgae *S. obliquus* FR751179.1 isolated from Durban region, Kwa-Zulu Natal, South Africa was used in this study. The *S. obliquus* culture was grown in open circular ponds (8000 L) using BG11 as nutrient medium (Guldhe et al., 2014b). The cultivation conditions are depicted in Table 1. Microalgal growth was assessed by optical density measurements at 680 nm (Ramanna et al., 2014) using a UV/vis-spectrophotometer (Spectroquant R Pharo 300, Merck Germany). Microalgal culture was grown with fed batch mode. All the experiments were performed by using *S. obliquus* culture from the same batch to maintain uniformity. The biomass (g L^{-1}) was measured gravimetrically (Mutanda et al., 2011).

2.2. Electrochemical harvesting of microalgae

ECH experiments were carried out in a batch reactor (Fig. 1) with configuration of 14 cm (length) × 10 cm (width) × 14 cm (height). All the experiments were carried out at room temperature

Table 1
Microalgae cultivation conditions.

Cultivation condition parameters	
Microalgal strain	*Scenedesmus obliquus*
Cultivation pond	Open circular pond
Culture capacity	8000 L
Culture medium	BG 11
Light intensity (natural sunlight)	400–1200 μmol m^{-2} s^{-1}
Culture temperature	18–27 °C
Mixing and aeration flow rate (submersible pump)	110 L min^{-1}
pH of culture	9
Microalgal biomass	2.4 ± 0.01 g L^{-1}

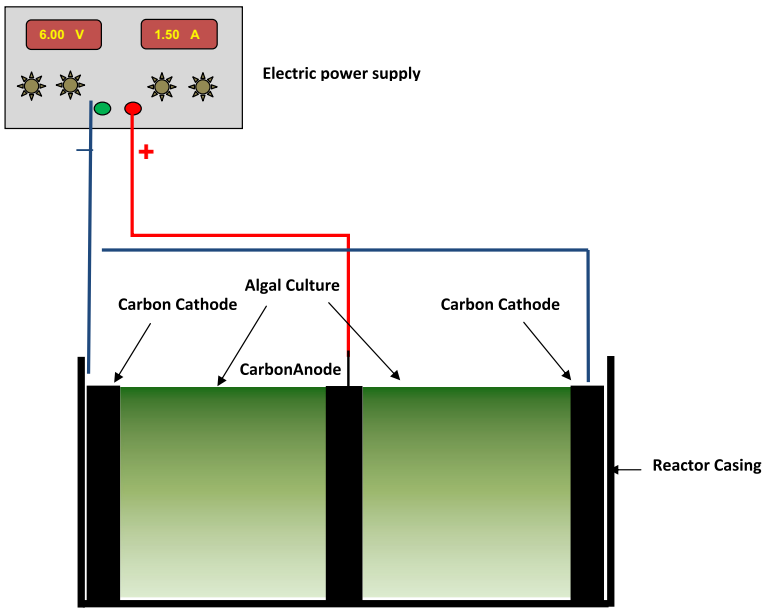

Fig. 1. Schematic diagram of electrochemical harvesting reactor.

by using 0.9 L of microalgal culture. Two carbon cathodes plates (12 cm × 10 cm × 2 cm) were kept 6 cm apart fixed to the reactor casing, and a carbon anode plate (12 cm × 10 cm × 2 cm) was kept in the middle of the reactor. Both carbon cathodes and the carbon anode were connected to negative and positive poles respectively of the Manson (HCS-3302) DC power supply. The applied current was regulated from the DC power supply, which was operated in the constant current mode. Electrolyte sodium chloride was added in the range of 2–6 g L^{-1} in the microalgae culture before the ECH process to check its effect on recovery efficiency. To determine the effect of initial pH, this was adjusted to 5 and 7 in the microalgal culture by using HCl prior to the ECH experiment.

2.3. Microalgal recovery efficiency and power consumption of ECH process

The microalgal recovery efficiency was calculated based upon the decrease in optical density of the microalgal culture (measured at 680 nm with a UV–vis spectrometer, Spectroquant Pharo 300, Merck). Samples were collected 5 cm below the water surface in the ECH reactor at regular time points (t) during the ECH process. The percentage recovery efficiency was determined using the following equation (Vandamme et al., 2011):

$$\text{Microalgal recovery efficiency } \mu_a = \left[(OD_i - OD_{f)}/OD_i\right] \times 100 \quad (1)$$

where OD_i is the optical density of the sample prior to the start of the ECH process, and OD_f is the optical density of the sample at time t.

The power consumption E (in kWh kg^{-1} of recovered microalgae) was calculated as (Vandamme et al., 2011):

$$E = (P \times t)/(1000 \times V \times \mu_a \times C_i) \quad (2)$$

where P is the power (W), t the time of the ECH treatment (h), V the volume of the microalgal solution treated (m^3), μ_a the microalgae recovery efficiency, and C_i the initial microalgae biomass concentration (kg m^{-3}).

2.4. Lipid extraction and analysis

After completion of all the ECH processes microalgal biomass was collected by skimming and freeze dried (Mini Lyotrap, LTE scientific Ltd., United Kingdom). Lipid extraction was carried out by sonication assisted solvent extraction. Dried microalgal biomass was mixed in solvent mixture of chloroform and methanol (2:1 v/v) and given sonication treatment at 20 kHz for 2 min (Misonix XL-2000-010, output power 100 W, output frequency 22.5 kHz). Solvent and biomass was separated by centrifugation (Heraeus Multifuge 4KR, USA) at 2000g for 15 min and sonication treatment was repeated with residual biomass for effective lipid extraction (Ramanna et al., 2014). Organic solvent mixture with lipids dissolved in it were pooled together and dried in an oven at 70 °C. Lipids were measured gravimetrically and compared to evaluate the effect of different ECH process on lipid recovery.

2.5. Statistical analysis

All the ECH experiments were performed in triplicates ($n = 3$) and lipid extractions in duplicates ($n = 2$). Data is represented as mean value ± SE (standard error). Significance of the results was tested at 0.05 levels by comparing mean values using one-way analysis of variance (ANOVA).

3. Results and discussion

3.1. Effect of applied current on microalgal recovery efficiency

The studied effects of the applied current on the microalgal recovery efficiency are summarized in Fig. 2. This figure shows microalgal recovery efficiency measurements before (time zero) and during the ECH process. The microalgal recovery efficiency at 60 min for 0.5 A, 1.0 A and 1.5 A current were found to be 54.2 ± 0.75%, 61.7 ± 0.45%, and 65.7 ± 0.08% respectively. It was clearly evident that with an increase in applied current, microalgal recovery efficiency also increased significantly ($p < 0.05$). The proportional decrease in optical density of microalgal broth was observed with the progress of ECH processes with various experimental designs. The initial microalgal recovery efficiency is faster

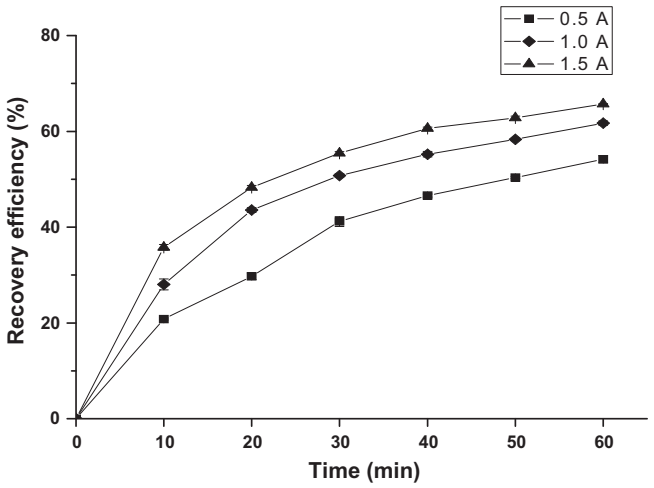

Fig. 2. Effect of applied current on microalgal recovery efficiency. Data expressed as mean ± SE (n = 3).

as 41.2 ± 1.04%, 50.8 ± 0.36% and 55.4 ± 0.33% recovery efficiency were achieved within 30 min of ECH process at 0.5 A, 1.0 A and 1.5 A respectively. After 60 min a stationary phase was reached, where no further significant increase in microalgal recovery efficiency was observed.

The supply of current to the electrochemical system determines the amount of charge released from the respective electrodes. The microalgal recovery efficiency depends on the applied current as well as the conductivity of the microalgal broth (Gao et al., 2010b). With the increase of electric field strength, the electrical charges on the electrodes as well as generation of bubbles increased accordingly. This increase in charged particles would result in the effective recovery of microalgae. The difference in the degree of increased recovery with applied current was also dependent on treatment time. When too high current is applied, there is a risk of heating the microalgal broth or ECH system with a subsequent wastage of electrical energy (Vandamme et al., 2011). Too large current density would result in high energy input and thus increasing the overall process cost.

3.2. Effect of electrolyte addition on ECH process

Utilization of an electrolyte is beneficial in reducing the power consumption of the electrochemical process (Gao et al., 2010a). NaCl was chosen as the supporting electrolyte in this study due to its ability to enhance the electrochemical process and minimize the energy input. Electrolyte concentration was varied by addition of 2 g L^{-1}, 4 g L^{-1} and 6 g L^{-1} NaCl to the microalgal culture. Together with carrying electric charge, chloride ions were found to significantly reduce the adverse effect of other anions present in the microalgal broth (Gao et al., 2010a). Fig. 3 shows the effect of different amounts of NaCl on microalgal recovery efficiency. The addition of NaCl in to the microalgal culture as additional electrolyte effectively enhances the microalgal recovery efficiency and reduces power consumption (Uduman et al., 2011). This could be attributed to the formation of active chlorine species and increased conductivity due to presence of chloride ions. Microalgal recovery efficiency peaked up at 83 ± 0.1% with the addition of 6 g L^{-1} of electrolyte (NaCl), while it was 72.8 ± 0.04% and 79.8 ± 0.05% with 2 g L^{-1} and 4 g L^{-1} additional concentration of NaCl respectively ($p < 0.05$). The addition of NaCl would also lead to the decrease in power consumption because of the increase in conductivity (Gao et al., 2010a). At large scale addition of electrolyte to the ECH process, is a feasible strategy to enhance the performance.

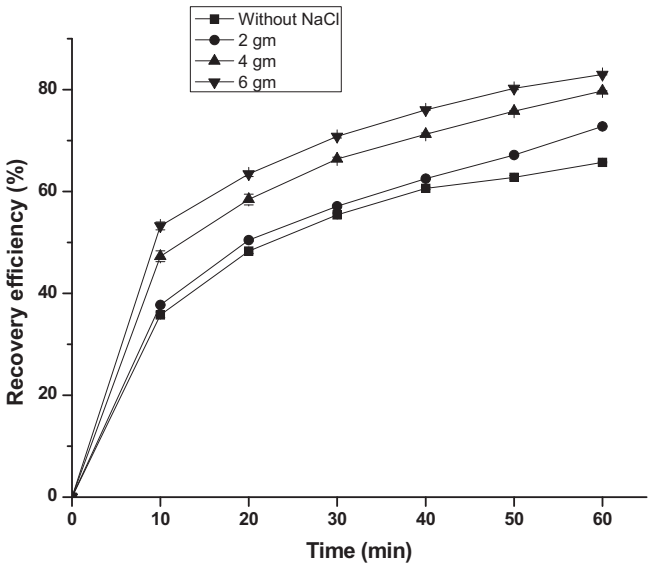

Fig. 3. Effect of electrolyte (NaCl) concentration on microalgal recovery efficiency. Data expressed as mean ± SE (n = 3).

3.3. Effect of initial pH on ECH process

The initial pH of microalgal culture is an important operating factor influencing the performance characteristics of the electrochemical process (Kim et al., 2012a). The initial pH of microalgal culture used in this study was 9. This was compared with pH 7 and 5 by adjustment with HCl for the microalgal recovery efficiency (Fig. 4). The highest removal percentage (73 ± 0.08%) was obtained at pH 5.0; with increasing pH, a decrease in the microalgal recovery efficiency (65–66%) was observed (Table 2). It is well known that pH is an important variable in ECH, as it determines speciation of charged particles in the solution and under acidic conditions, the formation of positively charged ions (Vandamme et al., 2011). When the initial pH was 5, the microalgal culture pH increased gradually during the ECH process with the time of electrolysis. The increase of pH was mainly due to the continuous formation of OH^- ions at the cathode as a consequence of the H_2

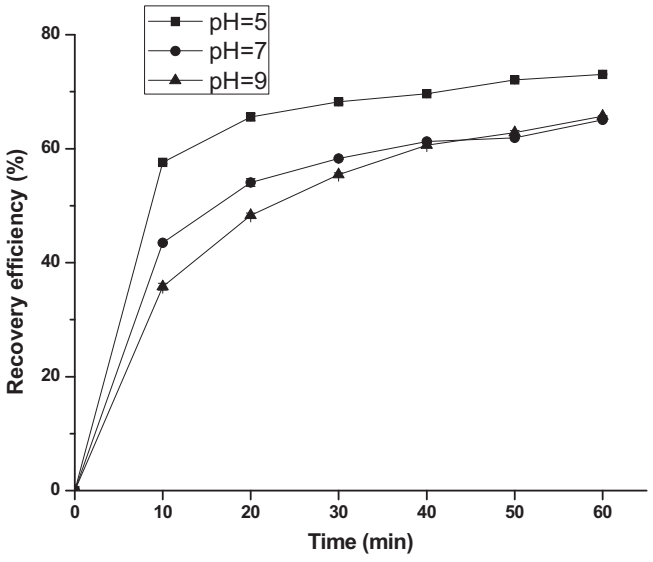

Fig. 4. Effect of initial ph on microalgal recovery efficiency. Data expressed as mean ± SE (n = 3).

Table 2
Microalgal recovery efficiency (μ_a) and power consumption (E) obtained in different ECH processes. Microalgal recovery efficiency data expressed as mean ± SE (n = 3).

Current (A)	Voltage (V)	Duration (h)	pH	Electrolyte (g L^{-1})	Microalgal recovery efficiency (%)	Power consumption (kWh kg^{-1})	Recovery efficiency per unit power consumption (%)
0.5	5.0	1	9	0	54.2 ± 0.75	2.13	25.45
1.0	7.2	1	9	0	61.7 ± 0.45	5.40	11.43
1.5	8.4	1	9	0	65.7 ± 0.08	8.87	7.41
1.5	5.7	1	9	2	72.8 ± 0.04	5.43	13.41
1.5	5.1	1	9	4	79.8 ± 0.05	4.44	17.97
1.5	4.6	1	9	6	83 ± 0.1	3.84	21.61
1.5	8.6	1	5	0	73 ± 0.08	7.32	9.97
1.5	8.9	1	7	0	65 ± 0.22	9.50	6.84

evolution process which tends to reach neutral pH. On the other hand, when the initial pH was 9, a slight decrease of the pH was detected at the beginning, which might be described by the consumption of OH^- and then almost a constant pH level was maintained during the overall ECH process. Microalgal culture normally has pH in the alkaline range; lowering the pH adds an extra step in the harvesting process which could hamper the overall economics of biodiesel production (Gao et al., 2010b). In this study the increase in recovery efficiency was observed in both cases viz. addition of electrolyte and lowering the initial pH. However, at large scale addition of electrolyte could be more feasible strategy compared to pH adjustment.

3.4. Power consumption of ECH process

Electric energy per mass is the kilowatt-hours required to obtain the recovery of a kilogram of microalgal biomass from the culture. Power consumption of ECH experiments conducted in this study is depicted in Table 2. It is clear that this novel configuration of electrochemical harvesting reactor is characterized by a lower electric energy per mass for the recovery of microalgae. In addition, the electric energy per mass increases with an increase in applied current, with respect to microalgal recovery efficiency (Table 2). It is inferred that some electrical energy was wasted in undesired side reactions at higher applied current (Vandamme et al., 2011). Highest microalgal recovery efficiency (83%) was observed in ECH process with 1.5 A applied current and 6 g L^{-1} NaCl (electrolyte) addition, with energy consumption of 3.384 kWh kg^{-1} microalgal biomass. Addition of electrolyte decreases the energy input because of increased conductivity. The energy consumption of ECH process without electrolyte addition at 1.5 A current was 8.87 kWh kg^{-1}. With the addition of electrolyte in the ECH process, energy consumption gradually decreased with increasing electrolyte concentration.

The availability of literature on electrochemical harvesting of microalgae is scanty. This novel approach has not been investigated thoroughly, particularly in terms of energy consumptions and effect on further downstream steps. The energy consumption of the ECH process in this study is comparable to previous studies on electrochemical harvesting (Kim et al., 2012b; Vandamme et al., 2011). Energy consumption of the ECH process in this study (3.384 kWh kg^{-1}) was found to be lower than other conventional harvesting processes like centrifugation (16 kWh kg^{-1}), chemical flocculation (36.81 kWh kg^{-1}), and filtration (3.58 kWh kg^{-1}) (Danquah et al., 2009; Vandamme et al., 2011). The ECH process has several advantages over currently applied harvesting and dewatering techniques. Periodic replacement of filters in filtration and metal electrodes in electrochemical processes can be avoided by ECH process with non-sacrificial electrodes. Natural evaporation is a slow process and requires substantial land area. Moreover due to radiations biochemical quality of biomass and lipid can be hampered (Guldhe et al., 2014b). Chemical flocculants could be toxic, contaminate biomass as well as leads to chemical wastewater generation. Conventional centrifugation is an effective but high energy consuming process. ECH processes have been applied at large scale in several other applications such as waste management and chemical synthesis. Sustainability and easy scalability of the ECH process and advantages of non-sacrificial electrodes makes it an excellent choice for microalgal biomass harvesting. Vandamme et al. (2011) have reported a power consumption of the electrolytic process around 2 kWh kg^{-1} of microalgal biomass harvested for *Chlorella vulgaris* under optimal conditions. The highest microalgal recovery efficiency they found was 92% with *C. vulgaris* using metal electrodes. Uduman et al. (2011) studied a electrocoagulation process using metallic electrodes for harvesting of marine microalgae. In their study the highest microalgal recovery efficiency was 99% and 98% for *Tetraselmis* sp. and *Chlorococcum* sp., respectively. In these previous studies the applied metallic electrodes, which require periodic replacement, is adding to the overall cost of the process. The present study applies non-sacrificial carbon electrodes, which avoids the periodic electrode replacement and does not give a metallic contamination of the output water and microalgal biomass. Thus ECH process could not only be applied for biodiesel production but also for other microalgal applications. The size of microalgae does influence the flocculation process (Uduman et al., 2011), thus the ECH process with non-sacrificial electrode needs further investigation with different microalgal species.

3.5. Effect of applied current and electrolyte on lipid extraction

After harvesting and drying of microalgal biomass, lipid extraction is the next most important step in biodiesel production. Microalgal lipid content depends upon number of factors like cultivation parameters, nutrient stress, light intensity, etc. Lipids are normally extracted using organic solvent extraction coupled with cell disruption. Extracted lipids are then subjected to conversion process to make biodiesel (Guldhe et al., 2014b). Kim et al. (2012b) speculated that due to formation of oxidative chemicals there is a visual change in the color of microalgal flocs and thus emphasized on the need of further studies of the effect of the ECH process on lipid extraction and conversion. In this study, lipid extraction was carried out with harvested biomass after each ECH experiments to assess its effect on lipid recovery. Biomass harvested with conventional centrifugation process was used as a control. Lipid yields were in the range of 13–16% by dry cell weight (DCW) which is shown in Figs. 5 and 6. Lipid yield of control biomass was 13.6 ± 0.16% DCW. Fig. 5 shows that there is no significant difference ($p > 0.05$) in lipid yields of biomass recovered from ECH processes done with variation in applied current and pH (13.2 ± 0.6–13.7 ± 0.05% DCW). However biomass harvested after the ECH processes with addition of electrolyte in concentrations 2 g L^{-1}, 4 g L^{-1} and 6 g L^{-1} showed slightly higher lipid yields of 14.8 ± 0.35, 15.1 ± 0.18 and 16.2 ± 0.27% DCW respectively ($p < 0.05$) (Fig. 6). This higher yield of lipids could be due to the osmotic shock produced by electrolyte (NaCl) which weakens or disrupts the cell wall of microalgae (Lee et al., 2010;

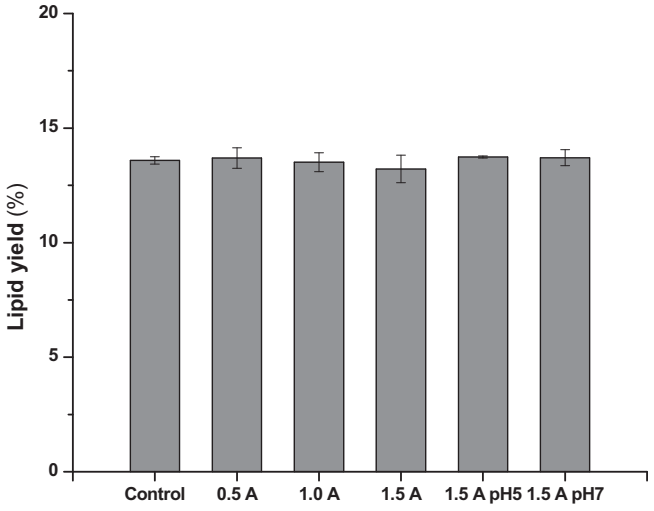

Fig. 5. Effect of applied current and initial pH on lipid yields of biomass harvested by ECH process. Data expressed as mean ± SE (n = 2).

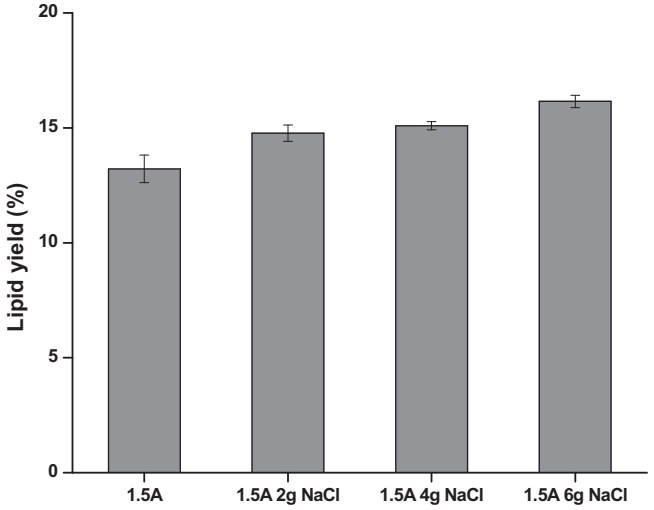

Fig. 6. Effect of electrolyte addition on lipid yields of biomass harvested by ECH process. Data expressed as mean ± SE (n = 2).

Prabakaran and Ravindran, 2011). Thus biomass subjected to lipid extraction gave higher yield because of effective cell disruption by both osmotic shock as well as sonication. Yoo et al. (2012) demonstrated that osmotic shock enhances the lipid extraction efficiency with wet biomass of microalgae *Chlamydomonas reinhardtii* (water content > 99%). Active oxidizing agents produced by addition of electrolyte and external electrical field in the ECH process weaken the microalgal cell wall. This implied that the NaCl could not only improve the algae removal in the ECH process, but also caused the breakage of cell integrity. Thus it can be concluded that the ECH process has no adverse effect on lipid extraction. Electrolyte addition strategy applied to enhance ECH performance aids in lipid extraction from microalgae.

4. Conclusion

In the present study, a novel configuration of electrochemical harvesting reactor comprising non sacrificial carbon electrode was developed for the harvesting of microalgae. With this configuration anode depletion and accompanying metallic contamination can be completely avoided. This work demonstrates that the addition of electrolyte is beneficial for the ECH process and it is also aiding in lipid extraction with higher yields. Low power consumption and high efficiency makes ECH process an environmentally acceptable and sustainable harvesting step for commercial scale microalgal biodiesel production.

Acknowledgements

Authors hereby acknowledge the National Research Foundation (South Africa) (NRF), South African Research Chair initiative (SARChI) and Durban University of Technology, South Africa for financial contribution.

References

Alfafara, C.G., Nakano, K., Nomura, N., Igarashi, T., Matsumura, M., 2002. Operating and scale-up factors for the electrolytic removal of algae from eutrophied lake water. J. Chem. Technol. Biotechnol. 77 (8), 871–876.
Amaro, H.M., Guedes, A.C., Malcata, F.X., 2011. Advances and perspectives in using microalgae to produce biodiesel. Appl. Energy 88 (10), 3402–3410.
Aragón, A.B., Padilla, R.B., Ros de Ursinos, J.A.F., 1992. Experimental study of the recovery of algae cultured in effluents from the anaerobic biological treatment of urban wastewaters. Resour. Conserv. Recycl. 6 (4), 293–302.
Danquah, M.K., Ang, L., Uduman, N., Moheimani, N., Forde, G.M., 2009. Dewatering of microalgal culture for biodiesel production: exploring polymer flocculation and tangential flow filtration. J. Chem. Technol. Biotechnol. 84 (7), 1078–1083.
Gao, S., Du, M., Tian, J., Yang, J., Yang, J., Ma, F., Nan, J., 2010a. Effects of chloride ions on electro-coagulation–flotation process with aluminum electrodes for algae removal. J. Hazard. Mater. 182 (1–3), 827–834.
Gao, S., Yang, J., Tian, J., Ma, F., Tu, G., Du, M., 2010b. Electro-coagulation–flotation process for algae removal. J. Hazard. Mater. 177 (1–3), 336–343.
Greenwell, H.C., Laurens, L.M.L., Shields, R.J., Lovitt, R.W., Flynn, K.J., 2010. Placing microalgae on the biofuels priority list: a review of the technological challenges. J. R. Soc. Interface 7 (46), 703–726.
Guldhe, A., Singh, B., Rawat, I., Bux, F., 2014a. Synthesis of biodiesel from Scenedesmus sp. by microwave and ultrasound assisted in situ transesterification using tungstated zirconia as a solid acid catalyst. Chem. Eng. Res. Des. 92 (8), 1503–1511.
Guldhe, A., Singh, B., Rawat, I., Ramluckan, K., Bux, F., 2014b. Efficacy of drying and cell disruption techniques on lipid recovery from microalgae for biodiesel production. Fuel 128, 46–52.
Kim, J., Ryu, B.-G., Kim, B.-K., Han, J.-I., Yang, J.-W., 2012a. Continuous microalgae recovery using electrolysis with polarity exchange. Bioresour. Technol. 111, 268–275.
Kim, J., Ryu, B.G., Kim, K., Kim, B.K., Han, J.I., Yang, J.W., 2012b. Continuous microalgae recovery using electrolysis: effect of different electrode pairs and timing of polarity exchange. Bioresour. Technol. 123, 164–170.
Klofutar, B., Golob, J., Likozar, B., Klofutar, C., Zagar, E., Poljansek, I., 2010. The transesterification of rapeseed and waste sunflower oils: mass-transfer and kinetics in a laboratory batch reactor and in an industrial-scale reactor/separator setup. Bioresour. Technol. 101 (10), 3333–3344.
Lee, A.K., Lewis, D.M., Ashman, P.J., 2013. Harvesting of marine microalgae by electroflocculation: the energetics, plant design, and economics. Appl. Energy 108, 45–53.
Lee, J.Y., Yoo, C., Jun, S.Y., Ahn, C.Y., Oh, H.M., 2010. Comparison of several methods for effective lipid extraction from microalgae. Bioresour. Technol. 101, 575–577.
Likozar, B., Levec, J., 2014. Effect of process conditions on equilibrium, reaction kinetics and mass transfer for triglyceride transesterification to biodiesel: experimental and modeling based on fatty acid composition. Fuel Process. Technol. 122, 30–41.
Mutanda, T., Karthikeyan, S., Bux, F., 2011. The utilization of post-chlorinated municipal domestic wastewater for biomass and lipid production by *Chlorella* spp. under batch conditions. Appl. Biochem. Biotechnol. 164 (7), 1126–1138.
Neti, N.R., Misra, R., 2012. Efficient degradation of reactive blue 4 in carbon bed electrochemical reactor. Chem. Eng. J. 184, 23–32.
Pienkos, P.T., Darzins, A., 2009. The promise and challenges of microalgal-derived biofuels. Biofuels, Bioprod. Biorefin. 3 (4), 431–440.
Poelman, E., De Pauw, N., Jeurissen, B., 1997. Potential of electrolytic flocculation for recovery of micro-algae. Resour. Conserv. Recycl. 19 (1), 1–10.
Prabakaran, P., Ravindran, A.D., 2011. A comparative study on effective cell disruption methods for lipid extraction from microalgae. Lett. Appl. Microbiol. 53 (2), 150–154.
Ramanna, L., Guldhe, A., Rawat, I., Bux, F., 2014. The optimization of biomass and lipid yields of *Chlorella sorokiniana* when using wastewater supplemented with different nitrogen sources. Bioresour. Technol. 168, 127–135.
Safi, C., Zebib, B., Merah, O., Pontalier, P.-Y., Vaca-Garcia, C., 2014. Morphology, composition, production, processing and applications of *Chlorella vulgaris*. Renew. Sust. Energy Rev. 35, 265–278.

Singh, B., Guldhe, A., Rawat, I., Bux, F., 2014. Towards a sustainable approach for development of biodiesel from plant and microalgae. Renew. Sust. Energy Rev. 29, 216–245.

Sostaric, M., Klinar, D., Bricelj, M., Golob, J., Berovic, M., Likozar, B., 2012. Growth, lipid extraction and thermal degradation of the microalga *Chlorella vulgaris*. N. Biotechnol. 29 (3), 325–331.

Uduman, N., Bourniquel, V., Danquah, M.K., Hoadley, A.F.A., 2011. A parametric study of electrocoagulation as a recovery process of marine microalgae for biodiesel production. Chem. Eng. J. 174 (1), 249–257.

Vandamme, D., Foubert, I., Muylaert, K., 2013. Flocculation as a low-cost method for harvesting microalgae for bulk biomass production. Trends Biotechnol. 31 (4), 233–239.

Vandamme, D., Pontes, S.C., Goiris, K., Foubert, I., Pinoy, L.J., Muylaert, K., 2011. Evaluation of electro-coagulation–flocculation for harvesting marine and freshwater microalgae. Biotechnol. Bioeng. 108 (10), 2320–2329.

Yoo, G., Park, W.K., Kim, C.W., Choi, Y.E., Yang, J.W., 2012. Direct lipid extraction from wet *Chlamydomonas reinhardtii* biomass using osmotic shock. Bioresour. Technol. 123, 717–722.