COMPARISON OF DIFFERENT HARVESTING METHODS OF MICROALGAE: ASSESSMENT OF EFFICIENCY AND ENERGY

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Master's Thesis

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Programme in Environmental Sciences

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ABSTRACT

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Microalgae is a promising feedstock for various applications and it has gained appreciation in recent years. The high costs related with microalgal biomass isolation hinder its large-scale applications. The application of an efficient and less costly harvesting method is fundamental for achieving a viable process. For this reason, a comparison of different microalgae harvesting technique's efficiencies was performed and energy costs were investigated in this thesis. In this work, harvesting efficiencies of *Synechocystis sp.*, *Chlorella Vulgaris sp.* and *Scenedesmus Quadricauda sp.* were compared by electrocoagulation and chemical coagulation with initial pH and adjusted values of 4, 7 and 12. The chemicals $FeSO_4$ and $Al_2(SO_4)_3.18H_2O$ were analyzed as coagulant whilst Fe and Al electrodes were tested in the electrocoagulation. The energy consumption and operating cost were calculated for EC and CC experiments. $Al_2(SO_4)_3.18H_2O$ achieved higher efficiency than $FeSO_4$ and the Al electrodes in EC experiment also was more efficient in harvesting than Fe ones. The harvesting efficiency was increased by increasing the applied electric current in electrocoagulation and with an increased dosage of the coagulant in chemical coagulation experiment. The obtained results showed that the adjusted pH values with Al electrodes presented high efficiency. The power and energy consumption rose with an increase of the applied current thereby increasing the operating costs. The electrocoagulation and chemical coagulation techniques achieved high efficiency values and appear to be suitable harvesting methods.

Keywords: Microalgae, Harvesting, Electrocoagulation, Chemical Coagulation.

ÖZET

MİKROALGLERİN FARKLI HASAT YÖNTEMLERİNİN KARŞILAŞTIRILMASI: VERİMLİLİK VE ENERJİNİN DEĞERLENDİRİLMESİ

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Mikroalg, çeşitli uygulamalar için gelecek vaat eden bir hammaddedir ve son yıllarda önemli hale gelmiştir. Mikroalgal biyokütle izolasyonu ile ilgili yüksek maliyetler, büyük ölçekli uygulamalarını engellemektedir. Verimli ve daha az maliyetli bir hasat yönteminin uygulanması, uygulanabilir bir proses elde etmek için esastır. Bu nedenle, bu tez kapsamında farklı mikroalg hasat tekniklerinin verimlilikleri karşılaştırılmıştır ve enerji maliyetleri incelenmiştir. Bu çalışmada, Synechocystis sp., Chlorella Vulgaris sp. ve Scenedesmus Quadricauda sp. mikroalg türleri için hasat verimlilikleri elektrokoagülasyon (EC) ve kimyasal koagülasyon (CC) yöntemleri kullanılarak karşılaştırılmıştır. Bu yöntemlerde ilk pH ve ayrıca 4, 7 ve 12 ayarlanmış pH değerleri kullanılmıştır. Kimyasal koagülasyon işleminde $FeSO_4$ ve $Al_2(SO_4)_3.18H_2O$ kimyasalları koagülant olarak kullanılmıştır ve elektrokoagülasyon yönteminde de Fe ve Al elektrotları test edilmiştir. EC ve CC deneyleri için enerji tüketimi ve işletme maliyetleri hesaplanmıştır. $Al_2(SO_4)_3.18H_2O$ ile, $FeSO_4$ kimyasalından, Al elektrotları ile de Fe elektrotlarından daha yüksek verimler elde edilmiştir. Hasat verimi, elektrokoagülasyonda uygulanan elektrik akımının ve kimyasal koagülasyon deneyinde koagülant dozajının artırılması ile yükselmiştir. Elde edilen sonuçlar, Al elektrotlar ve ayarlanmış pH değerlerinin yüksek verimlilik sağladığını göstermiştir. Uygulanan akımın artmasıyla güç ve enerji tüketimi artmış, dolayısıyla işletme maliyetleri artmıştır. Elektrokoagülasyon ve kimyasal koagülasyon yöntemleri ile yüksek verimlilik değerlerine ulaşıldığından dolayı, söz konusu yöntemler uygun hasat yöntemleri olarak görünmektedir.

Anahtar Sözcükler: Mikroalgler, Hasat, Elektrokoagülasyon, Kimyasal Koagülasyon.

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Halima AL-THAWR

17/08/2020

STATEMENT OF COMPLIANCE WITH ETHICAL PRINCIPLES AND RULES

I hereby truthfully declare that this thesis is an original work prepared by me; that I have behaved in accordance with the scientific ethical principles and rules throughout the stages of preparation, data collection, analysis and presentation of my work; that I have cited the sources of all the data and information that could be obtained within the scope of this study, and included these sources in the references section; and that this study has been scanned for plagiarism with "scientific plagiarism detection program" used by Eskişehir Technical University, and that "it does not have any plagiarism" whatsoever. I also declare that, if a case contrary to my declaration is detected in my work at any time, I hereby express my consent to all the ethical and legal consequences that are involved.

Halima AL-THAWR

CONTENTS

Page

LIST OF TABLES

Page

LIST OF FIGURES

Page

LIST OF IMAGES

Page

GLOSSARY OF SYMBOLS AND ABBREVIATIONS

A	Ampere
Al	Aluminum
BG11	Blue green- 11 medium
CC	Chemical Coagulation
CO_2	Carbon Dioxide
Con(mS/cm)	Conductivity
DC	Direct Current
DNA	Deoxyribonucleic acid
EC	Electrocoagulation
Eq.	Equation
Fe	Iron
g	Gram
h	Hour
I	Electric Current
kg	Kilogram
L	Liter
M	Molarity
min	Minute
mL	Milliliter
nm	Nanometer
OD	Optical Density
pH	Power of Hydrogen
RE	Recovery Efficiency
rpm	Rotation Per Minute

1. INTRODUCTION

Global warming is the effect of increased average Earth's surface and atmosphere temperature because of increased rate of GHS (Green House Gas) emissions that include carbon dioxide, methane, nitrous oxide and some other gases released in long term due to human activities (Cuellar-bermudez et al., 2020). In the recent years, the world has started to look for renewable energy sources to mitigate the risk of increased GHS and researches tend to investigate various renewable energy sources. Many renewable energy sources, such as thermal, water, wind, solar, and bioenergy, have been developed and introduced. Bioenergy that is obtained from biological sources such as biomass, has many advantages that caught the world's attention. Nowadays, bioenergy is seen as a clean and sustainable alternative to gradually replace fossil dependent energy. Carbon fixation using microalgae has become an option and is being studied extensively as part of a plan to reduce and mitigate greenhouse gases in the atmosphere (B. Wang et al., 2008). Carbon dioxide fixation can be combined with different processes such as wastewater treatment and this will be economically feasible and environmentally sustainable (Razzak et al., 2013). According to many studies, microalgae can potentially fix carbon dioxide from 10 to 50 times more than terrestrial plants (Pih & Britain, 1997). With this regard, the use of microalgae is beneficial due to their high productivity, high growth rate and unique properties (Mathimani & Mallick, 2020).

Microalgae represent a diverse range of living organisms and are considered a promising raw material for many applications, like production of food, cosmetics, medicines, carbon dioxide fixation, and biofuel production (Dominguez, 2013). Before taking any advantage of microalgae biomass, they must be harvested in an efficient and economical way. Microalgae harvesting means to separate microalgae cells from its aqueous growth medium (Kim, 2015). The methods for harvesting microalgae varies between chemical, biological, electrical and mechanical or combination of different harvesting techniques. (Mathimaniand Mallick,2018). However, microalgae harvesting processes face many challenges, depending on the process used (Barros et al., 2015; Wan et al.,2015). These difficulties include the high costs which require high input energy, expensive equipment, and microalgae contamination in the case of the use of chemical coagulants (Barros et al., 2015). Furthermore, the small cell size and relatively low concentration in the big volume of the growth medium are the main challenges (Mathimani & Mallick, 2020).

This thesis aims to explain, summarize and analyze some of the different techniques used in the harvesting of microalgae for three microalgae species *Synechocystis sp., Chlorella vulgaris sp.,* and *Scenedesmus quadricauda sp.* Furthermore, the methods of experiments will be discussed with clarification of the differences, pros, and cons. In this thesis, firstly, general characteristics of microalgae and the importance of microalgae in the various applications will be explained. Secondly, the microalgae cultivation and purification of cultures will be explained. Furthermore, experiments with different harvesting techniques, statistical analyzes for the comparison between them and the efficiencies and the results of the all experiments and analyses will be given. Finally, some solutions and future expectations and challenges of environmental pollution control will be proposed.

2. MICROALGAE

The word algae contain microscopic organisms (microalgae) and large organisms (macroalgae) and the latter generally includes seaweed (Anderson, 1996). Microalgae are microorganisms that are found in many ecosystems especially in aqueous ecosystems (Lewis and Flechtner, 2002; Flechtner, 2007). Microalgae are among the oldest living organisms on Earth (Song et al., 2008). According to their unique characteristics and their possession of a variety of groups and species, they are considered as a promising raw material for several applications and environmental treatments (Harun et al., 2010a; Brennan and Owende, 2010). Microalgae are promising feedstock of biofuels that are considered as one of the good sources of lipids and their oil content by dry weight is in the range of 20–50%, and can reach up to 70% in some microalga strains (Martins et al., 2010). Carbon dioxide bio-fixation are verified by microalgae as a strategy of reducing of CO_2 (10-50 times) more efficiently than terrestrial plants (Kumar et al., 2010; Sydney et al., 2010).

2.1. What Are Microalgae?

Microalgae or phytoplankton are prokaryotes like cyanobacteria, or eukaryotic organisms like green algae that grow with photosynthesis which is autotrophs as they absorb solar radiation by chlorophyll and converting it into organic compounds from inorganic nutrients such as nitrogen, phosphorous and carbon (Anderson, 1996). Furthermore, they are unicellular types found individually, in series or groups. Unlike higher plants, microalgae do not have roots, stems, or leaves. The cells generally range between 3-25 μm and have a negative charge cell membrane (Pugazhendhi et al.,2019).

Microalgae have great importance and a promising future, there are many applications and studies that have been conducted or are still under study. Among these features are that they produce more protein compared to terrestrial plants and are an important source of some organic molecules such as a series of long-chain unsaturated fatty acids that are important in human feeding (Spolaore et al., 2006). Microalgae are the main source of the food chain for nearly 70 % of the world's biomass (Wiessner et al.,1995). Microalgae grow up rapidly 100 times quicker than terrestrial plants and can increase their biomass within a day (Randrianarison & Ashraf, 2017). To produce one ton of biomass, it needs approximately 2 tons of CO_2, 100 kg of nitrogen and 10 kg of phosphorus (Sepulveda et al., 2019). There are some disagreements in the taxonomy of

microalgae and several branches and principles for classification. It is estimated that there are species between 200,000 and 800,000 in general, and only about 50,000 species of them are described (Manoj Kumar et al., 2018).

Microalgae have a massive biodiversity which can be exist freely or associated with other organisms, as is the case in lichens (Adl et al., 2005; Keeling et al., 2005). The classification of commercial microalgae species includes chlorophyta (green algae), phaeophyta (brown algae), pyrrophyta (Dinoflagellates), chrysophyta (Diatoms), rhodophyta (Red algae) and euglenophyta (Euglenoids) (Leliaert et al., 2012). Microalgae are distributed almost everywhere in the biosphere. Green microalgae generally grow in freshwater, seawater, and other species in highly saline environments. They may be found in the surface layer of water column or in the subsurface and few of them in the photic zone (200-300 m) below the surface layer (Kim, 2015). Furthermore, microalgae grow in swamps, deserts, rocks, snow regions and virtually microalgae exist in all terrestrial environments (Rindiet al., 2007).

The microalgae cells contain organelles, including the nucleus, which contain DNA that regulates cell functions and contain genetic codes. Additionally, the cells have a porous cell membrane that allows the materials to pass (Kim, 2015). The cell consists of polysaccharide and glycoprotein that provide the cell enormous defense in its environment, unlike animal cells. Additionally, it contains green chloroplasts that perform photosynthesis and the mitochondria that is the energy house of the cell (Kim, 2015). Microalgae have three types of photosynthesis pigments, namely chlorophyll, carotenoids and phycobiliproteins existent in the chloroplasts and green algae contain chlorophyll type A (Gouveia et al., 2008). Brownish color of microalgae forms because of the domination of carotenoids and xanthophylls, and red color is sometimes made from rising content of phycobilins. Whereas some of them possess flagella for the movement and in the others its non-existent. Reproduction in microalgae is asexually or occurs by cell division. They have the ability to convert energy, light and carbon dioxide into biomass with no need of reproductive organs and in some green algae sexual production takes place (Sekimoto, 2017).

2.2. Applications of Microalgae

Microalgae are used in various important areas including medicines, human and animal nutrients, cosmetology, fixation of carbon dioxide, wastewater treatment and

biofuel production (Dominguez, 2013). Most of the microalgae are characterized by their high productivity of biofuels because of their high lipid content, with some species accumulating more than 80% of dry weight (Chisti, 2007). Microalgae are characterized by their biological properties in different applications and their bio-products act as antioxidants, anti-cancer agents, lipids, vitamins, pigments and anti-virus agents (Spolaore et al., 2006). Additionally, microalgae are a sustainable, ecofriendly and nontoxic resource.

2.2.1. Human food and animal nutrition

Many species are used in food applications for humans and animals, the most popularly used species are applied in human and animal nutrition e.g. (Chlorella, Nannochloropsis, Phaeodactylum, Tetraselmis, pavlova and etc.) (Borowitzka, 2013). Some species are used in aquaculture as food additives for clams, mussels, shrimps and crabs and others in pets and farm animal nutrition (Kim, 2015). In addition to the fact that microalgae are the essential food for zooplankton, they also help to improve the conditions and quality of the aquaculture medium, such as improving water quality by producing oxygen and stabilizing the pH (Cellana & Hague, 2012). They are used as a feed supplement for many animals, such as cats, dogs, ornamental birds, cows, and horses because they are a good source for a large quantity of minerals and vitamins. Additionally, they can be used at the level of 5-10% as a partial substitute for traditional proteins in poultry farms (Navarro et al., 2016).

Microalgae contain the following nutritional compounds which give them value for applying in human and animal food: lipids, antioxidants, carbohydrates, vitamins A, B1, B2, B6, B12, Biotin, Folic acid, C, E and Pantothenic acid, proteins and some trace elements (Bishop & Zubeck, 2012). Due to a high percentage of proteins and other materials in microalgae content, it is considered as a possible source of human and animal nutrition in addition to being the source of vitamins and amino acids in the food systems of humans and animals (Bishop & Zubeck, 2012). In addition to their content of polyunsaturated fatty acids such as DHA (docosahexaenoic acid), EPA (eicosapentaenoic acid) etc., some microalgae such as Haematococcus algae are used as a natural food colorant.

2.2.2. Pharmaceuticals

Marine microalgae are rich in biologically active substances and toxins that are used in pharmaceutical purposes and foodstuffs and they have mighty genetic possibilities for several bioactive factors (Kim, 2015). According to Simmonset al. (2005), cytotoxic activity found that cyanobacteria as well as diatoms and conjugaphyte spirogyra plays a significant role in anticancer medicines and antiviral efficiencies (Microorganisms et al., 2017). In a research study about new antibiotics an antimicrobial activity was observed in microalgae and antifungal effectiveness were also found in cyanobacteria (LeRoy Creswell, 2010). Additionally, antifungal, antioxidants and anti-inflammatory activities have also been found and are being used in many drugs and vaccines (Miranda et al., 1998). They have been found some anti-obesity and anti-diabetic characteristics (Mayer & Hamann, 2005). Furthermore, several types of enzymes like lipolytic enzymes have been successfully produced in microalgae.

2.2.3. Nutraceuticals

Microalgae characterized by the essential biomolecules that are important for human and animal nutrition, such as lutein, chlorophyll, PUFAs, astaxanthin, and beta-carotene are effective commercial sources of these nutrients (Borowitzka, 2013). Microalgae are characterized by the production of high-value pigments which are carotenoids, phycobiliproteins, and chlorophyll that have health enhancing properties and strengthening immunity. Some commercially used microalgae in nutritional supplements are Chlamydomonas, Synechococcus, Scenedesmus and Botryococcus since they contain some basic elements such as magnesium, sodium, calcium, potassium, selenium, copper, phosphorus, iron, and sulfur (Kim, 2015). Also, microalgae are prospective sources for polyunsaturated fatty acids (PUFAs), Ω6 (arachidonic acid) and Ω3 (Epicosapentaenoic (EPA)), Docosahexaenoic (DHA) (Bishop & Zubeck, 2012). DHA is an essential fatty acid in the structural substance of the brain and eye, and also an essential component of heart tissue that supports brain, heart and cardiovascular functions

2.2.4. Cosmetics

Cosmetics products for hair and skin include some bioactive extracts or ingredients extracted from microalgae. They have been developed as refreshing or rejuvenating creams or anti-aging products enhancing collagen and thus preventing wrinkles, moisturizers and the sun protection creams (Kim, 2015). Some commercially available

species such as Spirulina, Arthrospira, Anacystis, Halymenia, and Dunaliella are used for erasing skin imperfections, promoting collagen synthesis and preventing wrinkles in many countries (Stolz & Obermayer, 2005). The pigment Phycobiliproteins is also a result of photosynthesis and used as cosmetic dyes for products such as face powder and eye shadows. Mycosporin-like amino acids (MAAs) from some microalgae and marine organisms are used in sunscreen creams to lower ultraviolet radiation damage and characterized as secondary metabolites produced by organisms that live in a high sunlight area and absorb between 310- 320 nm of ultraviolet rays that destroy biological molecules such as DNA and protein (Priyadarshani & Rath, 2012).

Squalene is utilized as a natural antioxidant, nutritional supplement and moisturizer for the skin as well as for other cosmetic objectives. All living things produce it as a biochemical medium (Patel et al., 2019). Microalgae accumulate high levels of squalene in their cells and this causes microalgae to be a potential commercial source because of its low toxicity. Also, it does not cause skin irritation and allergies, it has been included in many skin protection products, fight wrinkles and skin hyperpigmentation due to its high ability to hydrate (Ariede et al., 2017).

2.2.5. Wastewater treatment

Global water security and providing clean water to the population has become a serious global challenge. Organic and inorganic materials are released to the environment as a waste products of domestic, agricultural and industrial activities leading to pollution. Contaminated water contains phosphorous, nitrogen and heavy metals that are drained and the use of microalgae in bio-treatment is an effective step in treating wastewater (Kim, 2015). Microalgae can treat human wastewater, livestock wastes, industrial and agricultural wastes thus they have been suggested as a possible secondary treating system (Razzak et al., 2013). The tertiary treatment procedures clean off all organic ions and they are performed either biologically or chemically. The biological process performs better treatment than the chemical one which is considered costly and causes secondary pollution (Abdel-raouf, 2012).

Wastewater treatment systems including microalgae provide an optimal solution to tertiary treatment due to the ability to use both inorganic nitrogen and phosphorous in photosynthesis as well their ability to remove heavy metals from wastewater together with some poisonous organic compounds such as mercury, lead, scandium and cadmium

without causing secondary pollution (Razzak et al., 2013). On the other hand, microalgae provide oxygen to the aerobic bacteria to perform bio-decomposing of organic pollutants and to consume carbon dioxide emitted from bacterial activity (Abdel-raouf, 2012). Microalgae can also reduce unpleasant odors in wastewater (Abdel-raouf, 2012).

2.2.6. Biofuel production

In view of the fact that microalgae have important advantages such as rapid growth, high content of carbohydrates, proteins and lipids and along with their present applications in nutrition and pharmaceuticals, they are also a producer of biofuels (Martins et al., 2010). Microalgae can be used as a raw material source for many types of biofuels. For example, it is possible to produce methane by anaerobic degradation of microalgae biomass, biodiesel from microalgae oils and bio-hydrogen by photobiological reactions. Nowadays, sustainable and renewable energy is an essential need and biofuels are the best option for this task. While many factors are important in improving biofuel production from microalgae, among them the microalgae cultivation process plays a key role. Microalgae are considered the third generation in the production of biofuels because they can accumulate large amount of lipids and cultivate them in open ponds or photobioreactors on non-arable soils by utilizing sea water or even wastewater (Siqueira et al., 2016).

The techniques that convert microalgae biomass to biofuel are classified in two ways: biochemistry and thermochemistry (Tsukahara & Sawayama, 2005). It is possible to produce carbon dioxide, hydrogen and methane gas by the process of gasification (Kim, 2015). In the case of microalgae containing a high percentage of water (80-90%), it is preferable to produce fuel with liquefaction technology or bio-conversion instead of gasification (Patil et al., 2008). Biomass can be converted into biofuel, charcoal and gas by the pyrolysis process (Demirbaş & Arin, 2002). Organic components such as proteins, carbohydrates and lipids in microorganisms are broken down into smaller molecules by digestion, which may be via aerobic or anaerobic processes (Mckendry, 2002).

2.2.7. Bioremediation of carbon dioxide

The increase of CO_2 in the atmosphere has caused global warming and climate change, and among the challenges currently facing the world, different strategies are used and experimented to stabilize carbon dioxide whilst trying to find sustainable and ecofriendly energy alternatives (B. Wang et al., 2008). One of these promising potential

solutions to fix CO_2 are microalgae (B. Wang et al., 2008). They are distinguished by their ability to absorb CO_2 and produce biomass through photosynthesis (A. Kumar et al., 2010).

Microalgae can stabilize CO_2 from 10 to 50 times more than terrestrial plants (A. Kumar et al., 2010). In addition, some species of microalgae can directly capture CO_2 from the flue gas without the necessity for prior treatment and can use CO_2 from various sources such as CO_2 from the atmosphere or in the form of soluble carbonate or bicarbonate (A. Kumar et al., 2010). The carbon dioxide concentration can be measured at the inlet and outlet of the reactors of the culture to find out the amount of gas used and captured by microalgae unlike terrestrial plants that have supportive constructions such as stems and roots that are difficult to harvest. In fact, the fixation process of CO_2 by microalgae may be integrated with wastewater treatment and biogas production, or with the production of pharmaceutical materials and nutrients from microalgae (A. Kumar et al., 2010).

2.3. Cultivation systems of microalgae

The growth properties and composition of microlagae are recognized to significantly depend on the cultivation conditions (Chen et al., 2011). Microalgae production is currently carried out on a laboratory and pilot scale and from the literature view point, not many studies have mentioned the cultivation of microalgae on a large scale (Yin et al., 2020). The microalgae cultivation system has been developed from conventional open systems to well-developed photobioreactors (PBRs). There are two main different styles to cultivate microalgae for various applications, namely the open systems and closed systems (PBRs) (Brennan & Owende, 2010).

2.3.1. Open systems vs. photobioreactors (PBRs)

Microalgae are produced in the laboratory and pilot scale and there are not many studies that have cultivated microalgae in open systems (Mathimani & Mallick, 2020). Microalgae grow naturally in rivers, seas, etc., but for commercial purposes, microalgae must be cultivated in open culture systems like ponds and lakes or enclosed photobioreactors (PBRs) to increase biomass production in line with the purpose they will be used for (Kim, 2015). A bioreactor is defined as a system in which a biological process is carried out (Martins et al., 2010). There are different types of closed culture systems (PBRs) such as air lift, bubble column, fixed and floating, helical tubes and plates (Tasić

et al., 2016). However, the construction and maintenance of PBRs are costly (Okoro et al., 2019)

Open systems include scrubbers, open ponds, raceway ponds and tanks that work efficiently in wastewater treatment and the energy cost is low (Yin et al., 2020). In general, there are several factors that affect the growth of microalgae, including; 1: the external environment factors such as the light, nutrients, pH, temperature, carbon dioxide concentration, and salinity, 2: growth biological factors such as bacteria, viruses, fungi and other microalgae species, 3: artificial factors such as mixing rate, width and depth of culture system (Enamala et al., 2018). Table 2.1 shows the comparison between PBRs and open systems (Brennan & Owende, 2010; Yin et al., 2020).

Table 2.1. *The main advantages and disadvantages of the open system and PBRs.*

Cultivation sys.	Advantages	Disadvantages
Open system(tanks, open ponds,..)	The construction is more simple than closed systems. Require low energy and relatively cheap.	Poor lighting, mixing and rapid evaporation. Carbon dioxide scattering in the atmosphere. The need for a wide area of land. Contaminated by predators. Difficult to harvest due to low cell densities. Dependence on climate. Not suitable for all microalgae strains
PBRs	High biomass production the possibility of control the culture conditions (light, pH,..) prevent the culture contamination reduce the evaporation and loss of CO_2.	High operating, contraction and maintenance costs. The accumulation of O_2 can led to the low biomass production. The presence of microalgae biofilm limiting the PBRs surfaces from the penetration of light into the culture. Hard to scale-up and overheating.

Microalgae are grown by transferring them to cultures that contain nutrients and the nutrients could be obtained from chemicals, wastewater or sea water that is less expensive (Razzak et al., 2013). The ideal temperature of the microalgae growth ranges between 20-30 °C and there are species like Anacystis nidulans and Chaetoceros that live in hot springs that resist temperatures up to 80 °C (Venkata Subhash et al., 2014). The pH has a main role in the growth of microalgae. For example; microalgae capture CO_2 easily from the atmosphere in the alkaline conditions due to high biomass production (Enamala et al., 2018).

2.3.2. Nutritional mode

Microalgae cultivation is associated with several patterns of nutrition: Photoautotrophs are the most common organisms that carry out photosynthesis for the most microalgae species. The energy from sunlight and carbon dioxide and water are converted into organic materials (Enamala et al., 2018). Heterotrophs use organic carbon (glucose) as a carbon source and energy for producing biomass, while Mixotrophic is a combination of heterotrophic and phototrophic organisms and the cell number is increased due to high lipid accumulation (Manoj Kumar et al., 2018). Amphitrophy, subtype of mixotrophy means that organisms can grow autotrophically or heterotrophically, depending on the concentration of organic compounds and light intensity (Martins et al., 2010).

2.4. Harvesting of Microalgae Biomass

After cultivation of microalgae for the desired purpose, harvesting the biomass must be performed (Okoro et al., 2019). Harvesting is the separation of microalgae from the medium in which they grow with the assistance of different separation techniques (Enamala et al., 2018). The choice of the appropriate harvesting method depends on the type of microalgae used, the density and size of the cell, the purpose for which they will be cultivated for, and the possibility of reusing the culture medium (Amaro et al., 2011). Microalgae harvesting is performed by several methods that may be physical, chemical, biological and electrical (Al et al., 2015). There are various techniques that must be developed and some of them are still under the study to obtain the best harvesting technique with the highest efficiency at lowest cost. The cost of harvesting can range between 20-30% of the total biomass production cost (Suparmaniam et al., 2019). Various techniques have been used in the harvesting of microalgae such as sedimentation, filtration, flotation, flocculation and electrocoagulation or two techniques can be used together to improve harvesting efficiency (Martins et al., 2010). In this section, the current techniques that are used in the harvesting of microalgae will be reviewed to evaluate their economic and technical capabilities.

The efficiency of the harvesting procedures can be described as recovery efficiency (RE) and concentration factor (CF) and they are explained as follows (Singh & Patidar, 2018):

- RE: mass of the cell isolated / mass of the cells in initial culture

- CF: concentration of algae in last product/ initial concentration of algae in culture.

The biomass concentration of microalgae can be calculated by measuring chlorophyll pigment, counting cell number, optical density and dry weight (Singh & Patidar, 2018). Six criteria play a major role in determining the appropriate harvesting method for the main applications of microalgae (fuel, human and animal food, high value materials, wastewater treatment and CO_2 emission fixation). These criteria; 1: Biomass quantity (BQn): a massive amount of biomass must be produced, 2: The quality of the biomass (BQI): the harvested cells must be in good condition,3: Costs (C) must be supplied as low as possible, 4: The processing time (PT) should be fast, 5: Specific species (SS): method must include species following with high dewatering capability and 6: Toxicity (T): the method used should not cause toxicity to the biomass as much as possible (Al et al., 2015).

2.4.1. Sedimentation

A technique that relies on the forces of gravity to separate liquid and solid materials from one another (Al et al., 2015). The big difference between density will lead to faster sedimentation value and small particles with small density need longer time to settle by gravity (Milledge & Heaven, 2013). Over time, the biomass settles to the bottom of the tanks or tubes and can be collected afterwards. Typically, sedimentation tanks are cylindrical with a ground funnel so that microalgae settle near the outlet that is at the bottom and are easily assembled from it (Al et al., 2015). Due to the convergence of the densities between marine microalgae and seawater, likewise with freshwater microalgae, this method makes it less efficient and causes some challenge (Al et al., 2015).

The temperature, light, time and cell age are the other major factors that affect the efficiency of sedimentation (Al et al., 2015; Lean & Pick, 1996). The temperature has a significant effect on the sedimentation method and it has been observed that the colder water has a slower rate of sedimentation due to increased viscosity. On the other hand, the sedimentation in the high temperature leads to cell damage (Greenwell et al., 2010; Harith et al., 2009). As for the cell age, the harvesting by sedimentation in the stationary phase is higher than it is in the exponential growth phase (Choi et al., 2006). Certainly, to increase the sedimentation rate, microalgae have to take a longer time of more than 24 h to settle (Z. Wang et al., 2013). Although it is a low-cost method, it is not a practical

for use in factories and requiring a long time for settling and the biomass produced from this method is very low. The efficiency of the method may increase if it is integrated with the flocculation method (Al et al., 2015; Enamala et al., 2018).

2.4.2. Filtration

This is porous technique used to harvest microalgae, and preserve the biomass while permitting the liquid to pass through (Yin et al., 2020). It requires a pressure difference across the filter that can be operated by vacuum, pressure or gravity (Yin et al., 2020). The pressure required to push the liquid through the membrane reduces with increasing pore size of the membrane. There are several types of filter such as a vacuum, a suction, a belt, and a pre-coated drum one. Each of these different types of filters have different uses, advantages and disadvantages (Al et al., 2015; Grima et al., 2003). Filtration is an effective method for large microalgae cells such as Coelastrum proboscideum, Spirulina and S. Platensis and not suitable for smaller species like Chlorella, Dunaliella and Scenedesmus (Grima et al., 2003; Yin et al., 2020). There are three kinds of membrane filtration methods namely macro filtration (bigger than 10 μm), microfiltration pore diameter (100 nm-10000 nm), ultrafiltration (1nm-100nm) and reverse osmosis (less than 0.001 μm) (Brennan & Owende, 2010). Filtration is characterized to be more efficient than sedimentation in the harvesting of microalgae, preserve cells after the recovery process and is non-toxic to the cell, which can be used in the food production and high-va lued materials (Mathimani & Mallick, 2020).

In general, the disadvantages of this technique are; constant need for membrane replacement and frequent washing is needed to avoid clogging the pores of the membrane and it requires high energy (Mathimani & Mallick, 2020; Yin et al., 2020). Flocculation and filtration can be combined to reduce the energy requirements and additional costs, thus this method is not economically viable for major scale production (Singh & Patidar, 2018).

2.4.3. Centrifugation

Centrifugation is a method that uses centrifugal forces instead of gravity forces to isolate the microalgae from their growth medium (Yin et al., 2020). This technique depends on the settling characteristics of cells, cell slurry retention time in the centrifuge (Singh & Patidar, 2018). The feasibility of harvesting by centrifugation depends on the species of microalgae and the type of centrifuges. High efficiencies of up to 90% can be

achieved at low flow rates and high energy use (Singh & Patidar, 2018). There are two common types of centrifuges used in harvesting of microalgae (Milledge & Heaven, 2013):

1) Disc stack centrifuges are the most common industrial centrifuge and are widely used in commercial high value algal product plants and algal biofuel pilot plants. A disc stack centrifuge consists of a cylindrical container containing a number of closely spaced metal cones (discs) that rotate. The mixture to be separated is fed to the center of the stack of discs. After that, the dense phase travels outwards under the influence of centrifugal force and the lighter phase is displaced to the center. It is commonly used in biofuel production and high value products (Al et al., 2015). The separation of the materials depends on the densities and it is suitable for microalgae cell size with 3-30 μm and 0.02–0.05% content of microalgae cells (Milledge & Heaven, 2013). This technique is mechanically complex and costly and also the space is small between discs that makes it difficult to clean (Sharma et al., 2013).

2) Decanter centrifuge adopts the use of a sedimentation tank where the suspended solids are deposited due to the forces of gravity (Milledge & Heaven, 2013). The device continuously pumps the microalgae to the centrifuge bowl and the suspended particles in the culture are forced to the bottom of the bowl (Al et al., 2015). This device achieves a higher harvest rate than disc centrifuge, however it is suitable for suspensions with higher solid particles more than 15 μm and not suitable for microalgae suspensions (Milledge & Heaven, 2013). One of the disadvantages of this device is that it consumes more energy than the disc centrifuge and high costs of equipment is required to process large volumes (Milledge & Heaven, 2013).

2.4.4. Coagulation and flocculation

Microalgae cells have a negative charge (Zeta potential) and their densities close to the growth medium and found in a scattered condition. So, this system results in slow physical sedimentation (Amer et al., 2011; Edzwald, 1993; Singh & Patidar, 2018). Flocculation - coagulation is the most efficient method for harvesting of microalgae by surface charge neutralization, whereby suspended microalgae cells can be grouped into large, adjacent agglomerations (LeRoy Creswell, 2010; Okoro et al., 2019). There are many organic and inorganic coagulants such as metal ions and polymers that coagulate

microalgae cells by assembling the cells (coagulation) followed by sedimentation to the bottom of the cultivation device (C. Y. Chen et al., 2011; Uduman et al., 2011).

The efficacy of multivalent coagulants such as iron and aluminum salts is based on their electronegativity and solubility. Furthermore, electronegative ions have quicker coagulation and also low solubility salts are more efficacious (Papazi et al., 2010; Rawat et al., 2011). It has been noted that aluminum salts are more effective than ferric salts and cationic polymers bind to cells while anionic and non-anionic fail to form microalgae masses because of the electrical repulsion. Coagulation efficacy decreases by increasing the molecular weight of the coagulant (Papazi et al., 2010).

The pH plays an important role as its value changes in the solution and helps the coagulation process as a result of the negative charge connected with the surface of the cells (Branyikova et al., 2018). For the success of this technique, the chemical coagulants must be sustainable, not contaminate the biomass, allow to reuse the broth of culture, inexpensive, nonpoisonous, and efficient in small doses (Branyikova et al., 2018; Milledge & Heaven, 2013). This method is expensive for wide use, it has high pH sensitivity, and limited uses for food purposes. Likewise, the subsistence of microalgae culture nutrients, different microalgae strains and culture temperature can affect the optimum coagulation dose (Show et al., 2015). It is difficult to control the appropriate amount of the coagulants due to the changes occurring during the microalgae growth cycle. Low doses reduce the association and coagulation between the cells and high doses also reduce the bridging due to the electrostatic hindering. During the stationary growth phase, low zeta potential and low metabolic activity with high intercellular interactions can be a positive factor for harvesting of microalgae in this way (Mathimani & Mallick, 2020). In general, it is preferable to use coagulation - flocculation integration with other harvesting methods for more economic harvesting (Singh & Patidar, 2018).

2.4.4.1. Bio-flocculation

This technique works with microorganisms added to the culture medium and attached to the microalgae cells, which cause a heavy weight so cells can settle to the bottom (Singh & Patidar, 2018). Usually bacteria are used and it is found that the bacterium Paenibacillus sp. and Bacillus licheniformis has achieved an effective harvesting (Al et al., 2015; Oh et al., 2001). Factors that affect bio-flocculation involve the concentrations of the bio-flocculent, pH and the quality of the microorganisms

selected (Al et al., 2015). The efficiency of the harvesting depends on the ratio of the concentrations of the bio-flocculent to the concentrations of microalgae in the growth medium. Harvesting efficiency is increased when the concentration of bio-flocculent is higher than the concentration of microalgae (Salim et al., 2011). The pH has an effect on the harvesting efficiency as it converts the surface charge of the molecules in the medium, which affects attraction and repulsion. When the pH increases, the flocculation efficiency increases especially when it rises from 3 to 11. The pros of this method are biodegradability, nonpoisonous, the products are not toxic pollutants and it consumes very little energy (Salehizadeh & Shojaosadati, 2001). Its cons are that the organisms used for flocculation are specific to species, the recycling of these organisms is hard and microbiological pollution of the microalgae biomass can occur in case microalgae are used in the food and feed industries (Vandamme et al., 2013).

2.4.5. Electrocoagulation

Electrocoagulation (EC) is a simple technique that relies on metal electrodes commonly made of iron or aluminum and few amount of electricity in specific time is applied (Yin et al., 2020). Electrocoagulation includes three phases, namely the consistence of coagulants, destabilization of the suspended matters, and the gathering of destabilized suspended matters to form the flocs that settle and deposit by gravity (Mollah et al., 2004). This technique is used to remove a variety of pollutants and elements such as Pb, Cr, Cd, Ni, As, Hg, F^-, and colloidal matters from wastewater (Mollah et al., 2004). The harvesting of microalgae by EC has been studied in small scale in general and it is a promising technology because it has several advantages such as being ecofriendly, safe, and no direct addition of chemicals and low energy use (Barros et al., 2015; Yin et al., 2020). Additionally, it has effective recovery values that can range from 80% to 100% in optimum conditions (C. Y. Chen et al., 2011). This technique depends on some parameters such as current density, pH, initial cell density of microalgae, distance between cathode and anode, time, and conductivity (Singh & Patidar, 2018).

Since microalgae have a negative surface charge, they can be neutralized by coagulant ions that are generated in the situ where the anode electrode dissolves to stabilize the biomass later and can be harvested (Yin et al., 2020). The oxygen and hydrogen are emitted into the anode and cathode, for example if the electrodes used are made of aluminum, then all the forms in the first part of the reaction are in the form of

$Al(OH)_3(S)$ and at the end of the reaction $Aln(OH)_3n(S)$ is formed and the formation of all these forms rely on the pH (Brahmi et al., 2019). It is possible to calculate the microalgae recovery efficiency (MRE) in the EC by the following law (Fayad et al., 2017):

$$MRE = (OD_0 - OD_{st})/OD_0 \times 100$$

where OD_0 is the optical density of the initial culture before the EC treatment and ODst is the optical density at the selected settling time after EC. The microalgae harvested suffer from metallic contamination due to the metal ions released during the reaction and so it is needed to change the anode regularly (Yin et al., 2020).

2.4.6. Flotation

Flotation is a simple method that works very similarly to a sink and float process, where the density characteristics of the materials, with respect to that of the medium where they are placed are at the base of the separation. Gas bubbles pass through a liquid - solid suspension that cause microalgae to float on the surface by sticking to the gas bubbles (Barros et al., 2015). Microalgae with cell diameter range less than 10 μm can be efficiently harvested by flotation and this method is usually used with flocculation to harvest microalgae in wastewater (J. Kim et al., 2013). It is characterized by a physiochemical gravity separation process that is quicker than sedimentation, has a recovery efficiency of 50-90% and somewhat has a low cost (Al et al., 2015). Flotation is categorized according to bubble size into different types such as dissolved air flotation, dispersed air flotation (DAF), electrolytic flotation, and ozonation dispersed flotation (ODF) (Yin et al., 2020)

The size of the bubbles impacts the harvesting efficiency. The most important parameter determining the efficiency of this method is the instability of suspended matters, as well as the size of cells or particles. Sizes less than 500 μm are preferred to increase the chance of lifting it up by the bubbles (Suparmaniam et al., 2019; Yin et al., 2020). pH is one of the most significant factors that influence flotation operations and it was found that the pH range between 5-8 enhance the harvesting efficiency by rates up to 95% and above 8 the efficiency decreases due to the change of the surface charge (Y. M. Chen et al., 1998). Alkalinity, the air flow rate, the culture medium, recycle rate, time, and hydraulic loading are other important factors affecting the efficiency of the flotation

method (Al et al., 2015). Although harvesting of microalgae by flotation is better and faster than sedimentation, it is an energy consuming method with high operational costs for producing small bubbles (Hanotu et al., 2012).

Table 2.2. *The main advantages and disadvantages of different harvesting methods*

Harvesting process	**Harvesting methods**	**Advantages**	**Disadvantages**
Physical	Sedimentation	Inexpensive and simple. Not damage to the cells No biomass contamination so Suitable for food and pharmaceutical production	Loss of time Low recovery efficiency Not effective
	Filtration	High recovery efficiency. Less energy consuming and relatively low costs. Without chemical addition Suitable for food and feed uses.	The frequent membrane replacement High maintenance costs. formation of the bacterial films on the microfabric surface.
	Centrifugation	Fast and high recovery efficiency. Suitable for most microalgae strains.	High energy requirement. increase of gravitational force and shear stresses could damage the cells.
	flotation	less energy and cost. less operation time. well flexibility.	Not suitable for marine water microalgae. Requires chemical addition. High energy requirement.
Chemical	Chemical coagulation	Suitable for most microalgae species. Short time, simple and high efficiency. Less energy .	highly cost on large scale Not suitable for food and feed uses. The recycling of the medium is limited. Dependent on pH. Contamination the biomass by chemicals.
Electrical	Electro-coagulation	Suitable for most microalgae strains. Fast and high efficiently.	High cost on large scale. cathode fouling. Very high energy input. High electric current could damage the cells.
Biological	Bio-flocculation	efficient. Low cost.	Unclear method need more study. Microbiological contamination. Need long time.

3. LITERATURE REVIEW

Microalgae harvesting methods have different advantages and drawbacks. The harvesting process for microalgae should be applicable to all strains of microalgae and require moderate costs for operation and maintenance (Shelef & Sukenik, 1984). In general, some studies in the literature have tested various methods for harvesting of microalgae, some of which covered costs, energy consumption, advantages and disadvantages. This section gives a summary of the most important findings in some relevant studies, which have focused on electrocoagulation, coagulation and sedimentation that are related to the subject of this study

Uduman et al., (2011) investigated the electrocoagulation harvesting method and found that the recovery efficiency increased to 99% for *Tetraselmis sp.* and 98% for *Chlorococcum sp.* with voltage and run time. This can be clarified by the increasing of coagulants with the increase of current and time. Experiments were performed in photobioreactor with different applied voltage. The ideal minimum energy required to achieve the maximum level of microalgae recovery was obtained as 9.16 kWh/kg for *Tetraselmis sp.* and 4.44 kWh/kg for *Chlorococcum sp.* in 15 min. They also found that temperature increases reduce high energy requirements and lower salinity reduces the recovery efficiency of microalgae. They noticed that there was no significant effect of variations of pH in experiments. According to the results of our study and different studies in the literature it was found that pH had an effect in recovery efficiency of microalgae.

Golzary et al., (2015) performed electrical (EC) and chemical coagulation (CC) processes and showed that optimum high recovery efficiency (96.8%) was obtained with low cost by applying electric current (I) = 1.6 mA/cm^2 for 17.65 min. During chemical coagulation by using alum it was found that 450 mg/L concentration and pH= 6 were optimum conditions to reach 85% efficiency. Additionally, they observed that EC was less sensitive to pH variations than CC and EC which had good implementation in natural pH of microalgae medium and the highest recovery efficiency was observed at pH = 6. They considered the cost of industrial alum about 3 USD/kg, the cost of coagulant was 1.5 USD/m^3 and the cost of alkalinity should be added to this value. On the other hand, the cost of EC in optimum conditions was evaluated as 0.024 US $\$/m^3$ and they did not need extra cost to adjust alkalinity that means EC was more cost-effective than CC. The authors investigated three parameters such as time, current density and PH for EC and

used different dosage of alum in CC during experiments. It was noticed that during the experiments initial pH level was reduced with addition of the coagulant and the highest flocs were formed when the pH was 5. It was preferable that the initial pH was a little alkaline to obtain the highest ratio of floc formation and separation efficiency

Shi et al., (2017) found that the use of a higher current density produced a faster harvesting. The energy consumption at current density of 66.7 A m^{-2} was approximately 1.32 and 3.38 times higher than those at current density of 44.4 and 22.2 A m^{-2}, respectively, to harvest 98% of C. vulgaris. But, the use of a higher current may not be economically due to high energy consumption. There were no considerable changes in pH and temperature in the medium and the operation mechanisms were based on the application of low electric energy, the charge neutralization, bridging and bubble flotation. The cost of harvesting of microalgae by Al electrolysis was evaluated as 1.47 x 10^{-3} US$ g^{-1} biomass.

Matos et al., (2013) found that maximum recovery (97%) was obtained with a current density of 8.3 mA cm^{-2} in 10 min of operating time and after 30 min of sedimentation/flotation which were sufficient and optimal for harvesting. Applied current was increased to 16.7 mA cm^{-2}, but no significant biomass recovery was noted. Electrocoagulation was accomplished using aluminum electrodes and the concentrations of the total lipid, fatty acids, pigments and aluminum ion were identified in the microalgae medium. It was found that the accumulation of aluminum after EC in the microalgae was 0.56% and 1.39% when current densities are 3.3 and 8.3 mA cm^{-2}, respectively, and the result was consistent with (Vandamme et al., 2011) that indicated the release of metal from the electrodes is directly proportional to the current density and operating time. They investigated the area of electrodes with the volume of microalgae culture medium and found the highest recovery efficiency (98.9%) for an electrode area / volume of 180 cm^2 L^{-1}. pH, salinity and temperature were not identified as major parameters in this study.

Das et al., (2016) found that higher harvesting efficiencies were noticed at lower initial pH for the same dose, for 72 mg/L $FeCl_3$. RE in the original culture was 72.9%; however, for the same dose, RE in the pH 6.5 adjusted culture was 97.2%. It is possible to use an acidic extract solution to extract a large part of the iron, which is acidic in nature that will increase the efficiency of the harvest and reduce the coagulant requirements in the subsequent biomass harvesting. In this study, they reused iron as a coagulant and

determined it in the solution and then extracted it. When the coagulant was reused in the culture media of microalgae, growth of *Scenedesmus sp.* was not affected up to two times of recycling.

Fayad et al., (2017) investigated electrocoagulation-flocculation (ECF) harvesting method and found that aluminum electrodes were more effective than iron electrodes for Chlorella vulgaris recovery and the RE increased with the increasing current density. Additionally, ECF did not affect the lipid and pigment content of microalgae. In addition to that, the cost of ECF can be reduced by operating a low current density and a lower inter-electrode distance. The study also tested the influence of mixing speed and found the highest RE of 99 ± 1% after 60 min of electrolysis at the stirring speed of 250 rpm, pH=8, current density of 6.7 mA/cm^2, and inter-electrode distance of 1 cm, with an Al electrode.

Vandamme et al., (2011) have tested electrocoagulation-flocculation (ECF) by using Al and Fe electrodes and found that Al anodes were more efficient and recommended a sedimentation period after removal of the microalgae from ECF. ECF had lower Al consumption compared to chemical coagulation-flocculation (CCF) and lower energy consumption than centrifugation. Inter-electrode distance as an important parameter that influences energy consumption was not tested in this study.

Eldridge et al., (2012) tested different inorganic and organic coagulants in different microalgae species and found that Al^{3+}or Fe^{3+} were more effective than cationic polyacrylamides. Ferric sulphate is a likeable coagulant for its low toxicity and allows reuse growth medium. They investigated the features that affect coagulation like cell surface chemistry, motility, cell size and wall structure.

Bleeke et al., (2015) tested electro-flocculation (EF) by using various electrode materials and found that the highest RE was obtained by Mg electrodes followed by Al, Zn, Cu, Fe and brass. All electrodes presented the highest flocculation at electrical current of 40 V and the lowest at 10 V.

(Luis & Maceiras, 2017) investigated that pH induced flocculation was an efficient method for harvesting of microalgae such as *Skeletonema Costatum* and *Chaetoceros Gracilis* by reducing the costs of the separation. They observed high recovery efficiency by flocculation with basic pH values and another study carried out by (Mixson et al.,

2014) was noted to give same result. Furthermore, oil extraction for microalgae was performed by ultrasound and Soxhlet extraction and it was found that their content on saturated fatty acids were very high. This means that they are promising species for the production of biofuels.

4. MATERIALS AND METHODS

4.1. Microalgae Species and Cultivation

Twenty different microalgae species that were preserved in the laboratory have been purified (Image 4.1) and three species have been selected, which are observed to be the purest and fastest growing cultures. Experiments were performed with three microalgae species *Synechocystis sp*, *Chlorella vulgaris sp.*, and *Scenedesmus quadricauda sp.* were looked under the light microscope to verify them. *Synechocystis species* is a small (2 μm) spherical photosynthetic unicellular cyanobacteria that lives in fresh water (J. S. Choi et al., 1999) (Image 4.2). *Chlorella vulgaris species* is green freshwater photosynthetic microalgae and spherical unicellular (2-10 μm diameter) eukaryotic having a thick cell wall (Ru et al., 2020) (Image 4.3). *Scenedesmus quadricauda* species is a green fresh water microalgae and cells attach side by side in small colonies of 2 or 4 or 8 cells (Image 4.4) (Shawky et al., 2015).

Microalga samples have been looked under the light microscope to choose pure cultures and purify them. The three species have been purified by mixing 17 g agar in 1 L BG11 and the mixture autoclaved then it was poured in petri dishes in small amounts while the mixture was hot and left to cool down. The purification process was done in the microbiology lab aseptically to decrease the contamination risk. Then one drop from the three species was taken from the sample's tube and placed in the agar petri dishes and named group A. From the microalgae sample's tube 1 mL sample was taken and put in the flasks filled with 80 mL of BG11 for diluting them and the flasks neck were closed with sterilized cotton and left to grow for one week with a sufficient light source. At the same time, one drop from the diluted cultures was taken and put in group B of petri dishes for more purification purposes. The petri dishes were covered with Teflon bands to decrease the potential of contamination. Image 4.1 shows the petri dishes with purified cultures.

The cultures in petri dishes were left until the microalgae had grown and the small spots of the cultures cleared. To prepare 1 L of BG11 (the medium which microalgae were grown in) the following materials were added to 1 L flask filled with distilled water (Rippka et al., 1979) (Table 4.1). The trace element solution was added to 1 L of distilled water with the following materials in Table 4.1.

Table 4.1. *The BG11 medium components for 1L and the trace elements component*

BG 11	g/L	Trace elements	g/L
$NaNO_3$	1.5	H_3BO_3	2.860
K_2HPO_4	0.04	$C_{l2}H_8MnO_4$	1.810
$MgSO_4.7H_2O$	0.075	$ZnSO_4.\ 7H_2O$	0.222
$CaCl_2.2H_2O$	0.036	$H_4MoNa_2O_6$	0.390
Citric acid	0.006	$CuSO_4.5H_2O$	0.079
$C_6H_8FeNO_7$	0.006	$Co(NO_3)_2.6H2O$	0.0494
EDTA	0.001		
Na_2CO_3	0.02		
Trace elements	1 ml		

From the petri dishes of the group A and B one small spot was taken by loop and put in 50 mL flasks filled with BG11 and had cotton and foil to cover them. Likewise, one spot was taken and transferred to the glass tubes with a plastic cover. All the tubes and flasks were put in a cool place with a light source at room temperature and were gently shaken every day. The cultures were left until the microalgae apparently grew then 15 mL from that stock transferred to 1 L Erlenmeyer flasks filled with BG11 for each microalgae species.

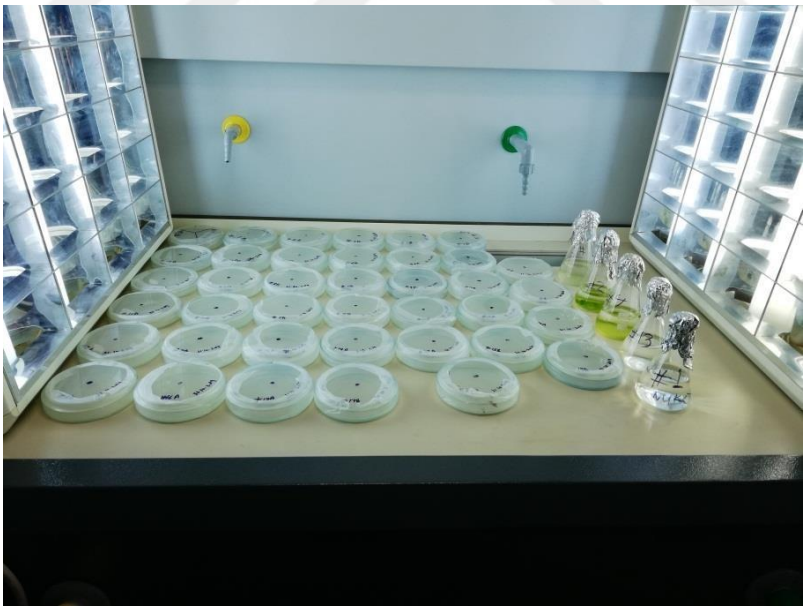

Image 4.1. *The purified microalgae cultures in petri dishes*

The cultivation of each purified microalgae were followed using one liter flasks as photobioreactor. They were continuously aerated with tubes connected from an air pumper where a two stage air compressor with approximate equal flow rates of 1 volume of gas per volume of liquid per minute (vvm) to mix the cultures and provide a CO_2 source

at the same time. The air flow was passed through a 0.2 μm PTFE membrane filters (Fluoropore, Merck Millipore) to provide more sterilization. The flasks neck was sealed with cotton and foil to reduce contamination risk. All the equipment and materials were washed with distilled water using a water purification system (Thermo scientific, Germany) and autoclaved before cultivation processes.

Monitoring cell growth of each microalgae species was determined by measuring absorbance capacity of the microalgae sample using a spectrophotometer every day at 680 nm via absorbance (UV 1800 UV-VIS spectrophotometer Shimadzu, Japan). A calibration curve was obtained to convert between biomass density and optical density. 1 L microalgae cultures were grown for 20 days until they reached a high growth rate and when the OD_{680nm} after 1.000 nm reading the UV spectrophotometer was not able to read values correctly, so the concentration of microalgae was diluted each time when the OD was bigger than 1.000 nm and OD_{680nm} was measured after dilution. The dilution of the samples was followed by mixing 2 ml of the microalgae culture with 2 ml distilled water. To even increase the cultivation volume, 1 L Erlenmeyer flasks with microalgae species were transferred to the 10 L sterilized flasks filled with autoclaved BG11 which separated into two volumes so, each species had two active 5 L cultivation flasks with air supply and daily growth was monitored.

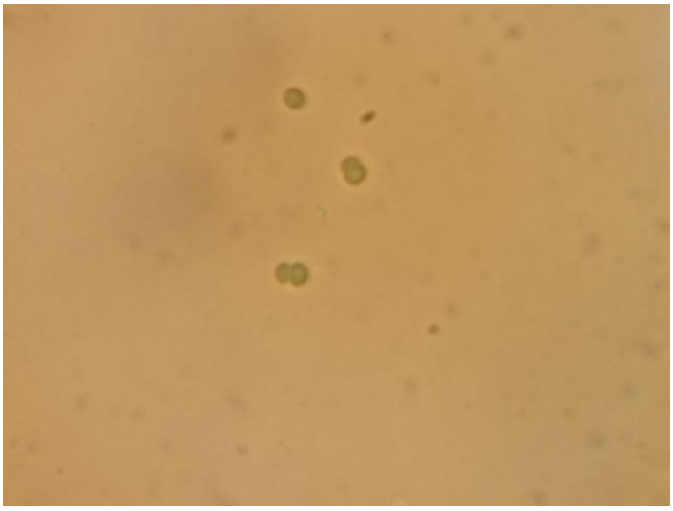

Image 4.2. *Microscopic view of Synechocystis sp. Scale bar 100 (oil immersion).*

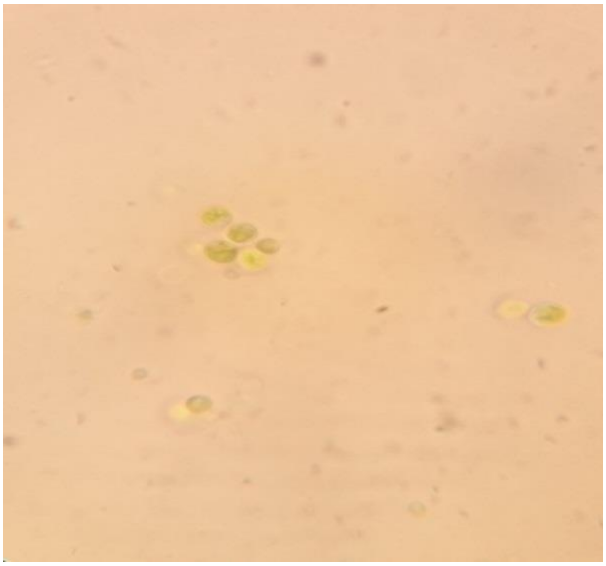

Image 4.3. *Microscopic view of Chlorella vulgaris sp. scale bar 100 (oil immersion).*

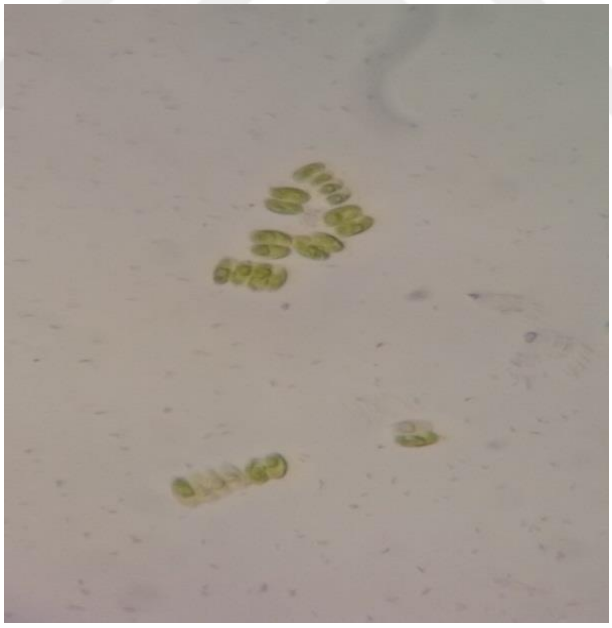

Image 4.4. *Microscopic view of Scenedesmus quadricauda sp. scale bar 100 (oil immersion).*

4.2. Calibration Curve for Optical Density- Biomass Dry Weight

In this study a calibration curve was needed to modify the optical density (OD_{680nm}) to concentration (mg/L) in dry weight basis. The dry weight of the three species was obtained by taking a 100 mL sample from each cultivated species well grown culture and centrifugation via (Centurion Scientific - C2 Series Centrifuges, UK). The sedimented biomass was washed resuspended by ultrapure distilled water and centrifuged again and the supernatant was discarded to remove minerals. The sedimented biomass was like a paste and dried in aluminum dishes at 110°C for 24 h. The process was done two times for each microalgae species. Then the dishes were cooled down in the desiccator and weighed. Image 4.5 shows the dried biomass for the three microalgae species in the aluminum dishes. The dry weight biomass was calculated as the difference of:

$$\text{weight of Al dishes(g)}_0 - \text{weight of Al dishes (g)}_1 \qquad \text{Eq.1}$$

the weight of empty aluminum dishes $(g)_0$ and the weight of the aluminum dishes with microalgae after drying $(g)_1$. Two millimeters of microalgae culture were diluted and the OD_{680nm} was read. The graph between microalgae concentration and OD was drawn and was linear as shown on Figure 5.1. The optical density was the difference between the initial optical density of the microalgae species and OD of the blank. The concentration of microalgae (g/L) was also calculated by:

$$\text{Concentration(g/L)} = \text{dry weight(g)} / \text{sample volume(L)} \qquad \text{Eq.2}$$

The calibration curve of the OD_{680nm} with dry weight biomass (g/L) was calculated and written by MS Excel program. The slope between concentration of microalgae (g/L) and the $OD_{initial}$ of the microalgae was calculated and also drawn by MS Excel program. The linear region of the curve was taken as reference for further conversions. The R-square root was 0.995, 0.993 and 0.998 for *Synechocystis sp.*, *Chlorella Vulgaris sp.* and *Scenedesmus Quadricauda sp*, respectively which shows the high precision required for this study.

Image 4.5. *The dried biomass.*

4.3. Harvesting Experiments

4.3.1. Microalgae natural settling

The natural settling behavior of the microalgae was investigated to test the natural settling efficiency in the three microalgae. The natural settling experiments were performed at room temperature and evaluated after a short and long time interval of 30 min and of 24 h, respectively. The samples were taken when $OD_{initial}$ was approximately 1.000 nm. Two sets of samples were collected from the three cultures and left to settle down naturally in 10 mL tubes for 30 min and for 24 h settling time. The optical density was read to determine settling efficiency. The samples were taken from the clear top zone tubes. The recovery efficiency was calculated by (Oh et al., 2001):

$$\text{Recovery efficiency } \% = (OD_{initial} - OD_{t}/OD_{initial}) \times 100 \qquad \text{Eq.3}$$

Where $OD_{initial}$ is the initial optical density of sample before the harvesting experiment, OD_t is the optical density of microalgae concentration after 30 min or 24 settling time.

4.3.2. Electrocoagulation (EC) experiments

Electrocoagulation experiments were performed under room temperature conditions for the three microalgae species and three different electric currents were applied 0.2 A, 0.5 A and 0.8 A. Aluminum and iron electrodes were used in EC experiments and each electrode consisted of 6 parallel plates. Before starting the experiment, the iron electrodes were soaked in diluted H_2SO_4 for 24 h to remove rust likewise, the Aluminum electrodes were soaked in diluted HCl for 24 h. The EC cell consisted of a 500 mL beaker equipped with Fe electrodes connected to a DC power

supply with magnetic stirrer, Figure 4.1 shows the EC reactor design. The sample volume was 300 mL and the EC experiment operating time was 30 min. The effect of the time was investigated for both electrocoagulation and coagulation experiments. For the experiment, each sample was left to settle for 30 min before reading optical density (OD), also was later left 24 h for more settling time before testing OD.

The absorbance of each microalgae were measured before the experiment via UV Spectrophotometer at 680 nm and the values were close to each other 1.018 nm for *Synechocystis sp.*, 0.966 nm for *Chlorella Vulgaris sp.* and 0.962 nm for *Scenedesmus Quadricauda sp*. The OD_{680nm} values were the difference between initial OD_{680nm} and OD of the blank (BG11).

The first run of the EC experiment for the first microalgae species was performed with Fe electrodes and the electric current was 0.2 A. The mixture was rotated at 200 rpm for 30 min by a magnetic stirrer (Biosan MSH-300, Magnetic Stirrer with hot plate, Latvia). After that the samples were taken after 5, 10, 20, and 30 min time intervals. The voltages were recorded from the screen of the DC power supply over the experiment for each specific taken time. The three first runs of the experiment were performed with the initial values of pH medium. The samples were taken to 10 mL tubes and left for 30 min before testing the OD. And the recovery efficiency was calculated for each sample by Eq.3.

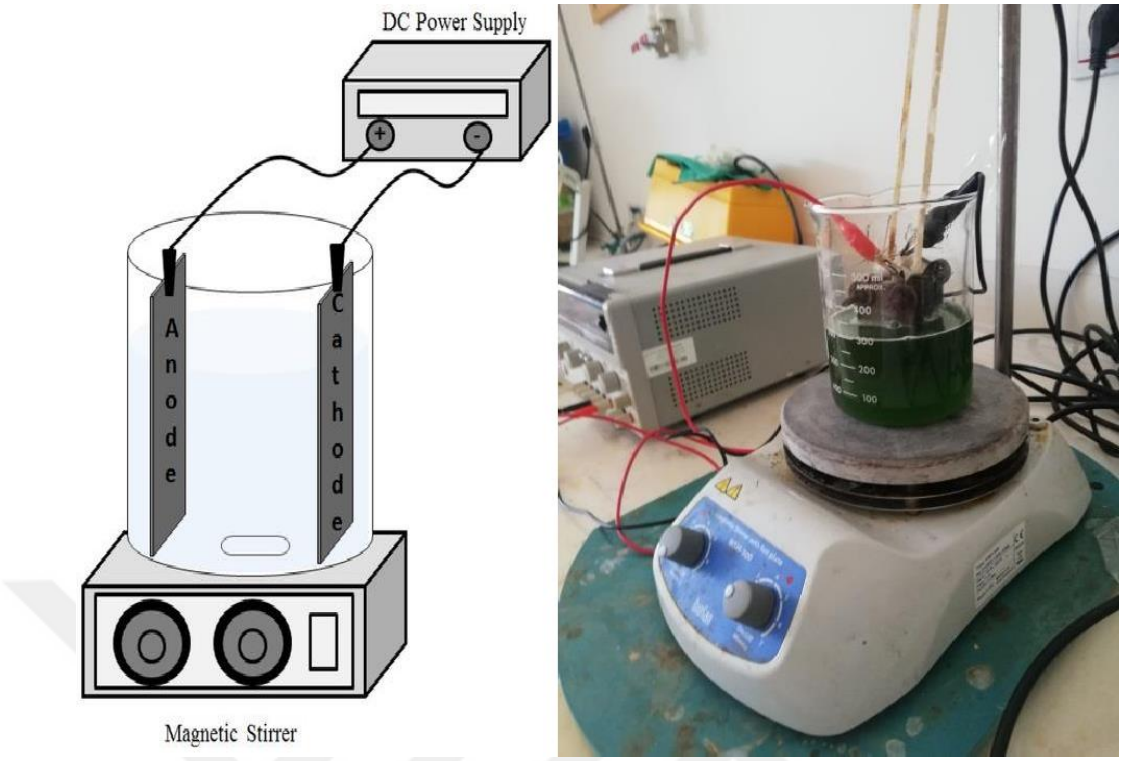

Figure 4.1. *EC experiment reactor design.*

The pH was measured by the pH meter (edge® Multiparameter pH Meter - HI2020, USA) for the initial microalgae culture and for each sample, likewise the conductivity was measured by conductivity meter (WTW Inolab conductivity meter Level 1, Germany). The second and third runs of the experiment were carried out at electric current of 0.5 A and 0.8 A, respectively with original pH of the culture. The fourth, fifth, and sixth runs were done with the same processes, but with adjusted pH whose values were 4, 7 and 12 at I: 0.5 A as an average point. The adjustment of pH was done by adding appropriate amounts of 1M NaOH and 1M H_2SO_4 was added to the microalgae solution. To prepare 1M NaOH and 1M H_2SO_4 the following equation was applied:

Molarity= (moles of solute/ liters of solution) Eq.4

For preparing 1M NaOH, 40 g NaOH was taken and dissolved in 1L balloon with distilled water and for the 1M H_2SO_4, 54.3 mL H_2SO_4 was taken and made up 1 L in balloon.

The same experimental processes were repeated for the second and third microalgae species. For the Al electrodes the same processes were done with the original pH of the microalgae cultures and at electric current: 0.2 A, 0.5 A and 0.8 A. Also, the experiments were performed with the three adjusted pH values at current of 0.5 A.

The electrode consumption for Al and Fe electrodes was calculated by Faraday 's law (Gao et al., 2010):

$$W = I \times t \times M / n \times F \qquad \text{Eq.5}$$

Where: W is dissolved metal (gram of Al or Fe), I is electric current A, t is the time of electrolysis (seconds), M is molecular weight of Al electrodes or Fe electrodes which (26.98 g mol^{-1}) and (55.8450 g mol^{-1}), respectively. F is Faraday's constant (96.487 C mol^{-1}), n is the number of electrons transferred for Al (3) and Fe is (2).

After that, microalgae mass (g) for each sample after reaction time 30 min and settling time 24 h was calculated by:

$$(\text{Sample volume mL} \times OD_{initial} \times \text{Slope value} \times \text{Recovery efficiency}) /100 \qquad \text{Eq.6}$$

Where: the sample volume was 300 mL, slope values were calculated for each microalgae species by MS Excel program and the results shown in Table 5.1. Recovery efficiency either after 30 min of reaction time or 24 h of settling time.

The power (watt) was calculated for each time run and electrode current applied by:

$$\text{Power (watt)} = \text{Voltage} \times \text{Electric current applied} \qquad \text{Eq.7}$$

Also, the energy was calculated for each run time by:

$$\text{Power (watt)} \times \text{Time (s)} \qquad \text{Eq.8}$$

Additionally, energy per microalgae biomass j/kg after the reaction time 30 min and 24 h of settling rate was calculated by:

$$\text{Microalgae biomass g/ Energy j} \qquad \text{Eq.9}$$

To evaluate the efficiency of the EC experiment from the cost view point, the cost was calculated in USA $ by (Geraldino et al., 2015):

$$a \times C_{energy} + b \times C_{el} \qquad \text{Eq.10}$$

where a: is the energy cost that was considered as the value of the electric current in Turkey from Global Petrol Price website and C_{energy} is the energy consumed j. b is the cost of the electrodes (Al and Fe) and found that the price of the Al metal was1.79 $/kg and Fe was 0.424 $/kg. The values were taken from Alibaba website in Turkey. C_{el} is the electrode consumption in g (Al and Fe) that was calculated by Eq.5. Furthermore, the costs per microalgae mass $/kg after 30 min of reaction time and 24 h settling time were calculated by:

$$\text{Microalgae Mass g/ The cost \$} \times 1000 \qquad \text{Eq.11}$$

4.3.3. Chemical coagulation experiments

The chemical coagulation experiments were performed with chemicals that include iron sulphate ($FeSO_4$) and aluminum sulphate ($Al_2(SO_4)_3.18H_2O$). The concentrations of each chemical was calculated according to the result of electrode consumption by Eq.5 from the EC experiment and the results shown in Table 5.2. For the Fe electrodes the electrode consumptions of the three electric currents 0.2 A, 0.5 A and 0.8 A were 0.1041 g, 0.2604 g and 0.4167 g, respectively. Then the unit (g) was changed to molarity for the Fe electrodes consumed g by the equation of:

$$\text{(Metal consumed g)} \times \text{(mole/molecular weight of Fe or Al)} \qquad \text{Eq.12}$$

The molecular weight of Fe is 55.845 g mol^{-1} and for Al is 26.98 g mol^{-1}.

The values of consumed metal g in molarity were as:

1.9×10^{-3} M, 4.7×10^{-3} M and 7.5×10^{-3}M, respectively.

The molarity was changed to milliliter by equation of:

$$C_1 \times V_1 = C_2 \times V_2, \qquad \text{Eq.13}$$

where C_1 and V_1 are the concentration *M* and the volume (mL) of the starting solution, C_2 and*d* V_2 are the concentration (M) and the volume (mL) of the desired solution. The results were: 1.14 mL $FeSO_4$, 2.82 mL $FeSO_4$ and 4.5 mL $FeSO_4$.

The Al electrodes consumptions were calculated by Eq.5 and found that:

0.03355 g, 0.08388 g and 0.13422g,

then Eq.12 was applied and it was found that:

1.23×10^{-3} M, 3×10^{-3} M and 5×10^{-3}, respectively.

After that by Eq.13, it was found that:

0.74 mL, 1.92 mL and 3 mL.

The concentrations M of each chemical were determined, but to prepare the stock solutions of each chemical, the following calculation must be done: for the $FeSO_4$ coagulant:

Molecular mass of the chemical × the volume of water needed to dissolve the chemical × the desired concentration M Eq.14

The molecular mass of $FeSO_4$ is 151.91 g/mol, the volume of needed water was 0.1 L and the desired concentration was 0.5 M . The result was 7.6 g.

For $Al_2(SO_4)_3.18H_2O$, coagulant molecular mass = 666.4844 g/mol , then by Eq.14:

(0.5 M) × (0.1 L) × (666.4844 g/mole) = 33.32 g .

The experiments of chemical coagulation were carried out using 500 mL beaker and the sample volume was 300 mL. The initial OD_{680nm} were 1.002 nm for *Synechocystis sp.*, 0.926 nm for *Chlorella Vulgaris sp.* and 1.004 nm for *Scenedesmus Quadricauda sp.* The OD_{680nm} values were the difference between initial OD_{680nm} and OD of the blank (BG11).

The experiments were performed at room temperature and all the equipment was washed with distilled water. The first coagulant value was (7.6 g $FeSO_4$) dissolved in distilled water in 100 mL. The experiments were carried out in 500 mL beaker filled with 300 mL microalgae culture (sample volume). For the first run, $FeSO_4$ solution was added to the mixture of microalgae from the stock (100 mL of $FeSO_4$) with 1.14 mL. Then the mixture was agitated at 250 rpm for 30 s during a high mix condition followed by a slow mix for 20 min at 150 rpm rotated by a magnetic stirrer.

The pH was measured at the 0, 2, 5, 10, and 20 min time interval which were the experiment operating time. The samples were taken in 10 mL tubes. After that the samples were left to settle down for 30 min before testing the OD_{680nm} and again for 24 h. The recovery efficiency was calculated for each sample after 30 min and 24 h by Eq.3.

The first, second, and third runs of $FeSO_4$ coagulant were performed with the original pH of the medium with the following dosages: 1.14 mL, 2.82 mL, and 4.5 mL, respectively. The effect of pH was investigated by adjusting the pH at 4, 7 and 12 values with appropriate amounts of 1M NaOH and 1M H_2SO_4 . The fourth, fifth, and sixth runs were performed with pH adjustment then the coagulant (2.82 mL of $FeSO_4$) was added with an average dose for each pH adjusted value run . The same steps were tested with other microalgae species.

For $Al_2(SO_4)_3.18H_2O$ coagulant, the f dosages of 0.744 mL, 1.92 mL and 3 mL were tested, respectively with the three microalgae species. After the adjustment of pH the coagulant was added with dosage of 1.92 mL as an average dose. The optical density after 30 min and 24 h settling time were measured, then the RE was calculated by Eq.3.

5. RESULTS AND DISCUSSION

5.1. Calibration Curve for Optical Density-Biomass Dry Weight

Microalgae dry weight (g) and the concentration (g L^{-1}) are shown in Table 5.1, sp.1 for *Synechocystis sp.,* sp.2 for *Chlorella Vulgaris sp*. and sp.3 for *Scenedesmus Quadricauda sp.* According to the data in Table 5.1 the calibration curve between the optical density and concentration of microalgae g L^{-1} was drawn and shown in Figure 5.1 (a, b and c). The average biomass dry weight were 0.244 g, 0.192 g and 0.512 g for *Synechocystis sp., Chlorella Vulgaris sp*. and *Scenedesmus Quadricauda sp.,* respectively. *Scenedesmus Quadricauda sp.* dry weight was the highest one compared to the other two species. The concentrations g/L of the three microalgae species were done twice according to the dry weight that was done twice. They were 1.203 and 1.240 g/L for *Synechocystis sp.,* 1.058 and 0.866 g/L for *Chlorella Vulgaris sp.,* and 2.602 and 2.514 g/L for *Scenedesmus Quadricauda sp.* The slopes were 0.20531, 0.30513 and 0.43193 for *Synechocystis sp., Chlorella Vulgaris sp*. and *Scenedesmus Quadricauda sp.,* respectively.

Table 5.1. *The concentration of microalgae and calibration curve data.*

$OD_{initial}$ Sp.1	$OD_{initial}$ Sp.2	$OD_{initial}$ Sp.3	Con. g/L Sp.1	Con. g/L Sp.2	Con. g/L Sp.3	Dry w. g Sp.1	Dry w. g Sp.2	Dry w. g Sp.3
3.045	2.243	3.106	1.222	0.962	2.558	0.120	0.1058	0.2602
2.260	1.390	2.608	0.611	0.481	1.279	0.124	0.0866	0.2514
1.434	0.755	1.439	0.305	0.241	0.639			
0.789	0.343	0.702	0.153	0.120	0.319			
0.360	0.137	0.317	0.076	0.060	0.159			
0.146	0.034	0.124	0.038	0.030	0.079			
0.038	0	0.027	0.019	0	0.039			
0		0	0		0			

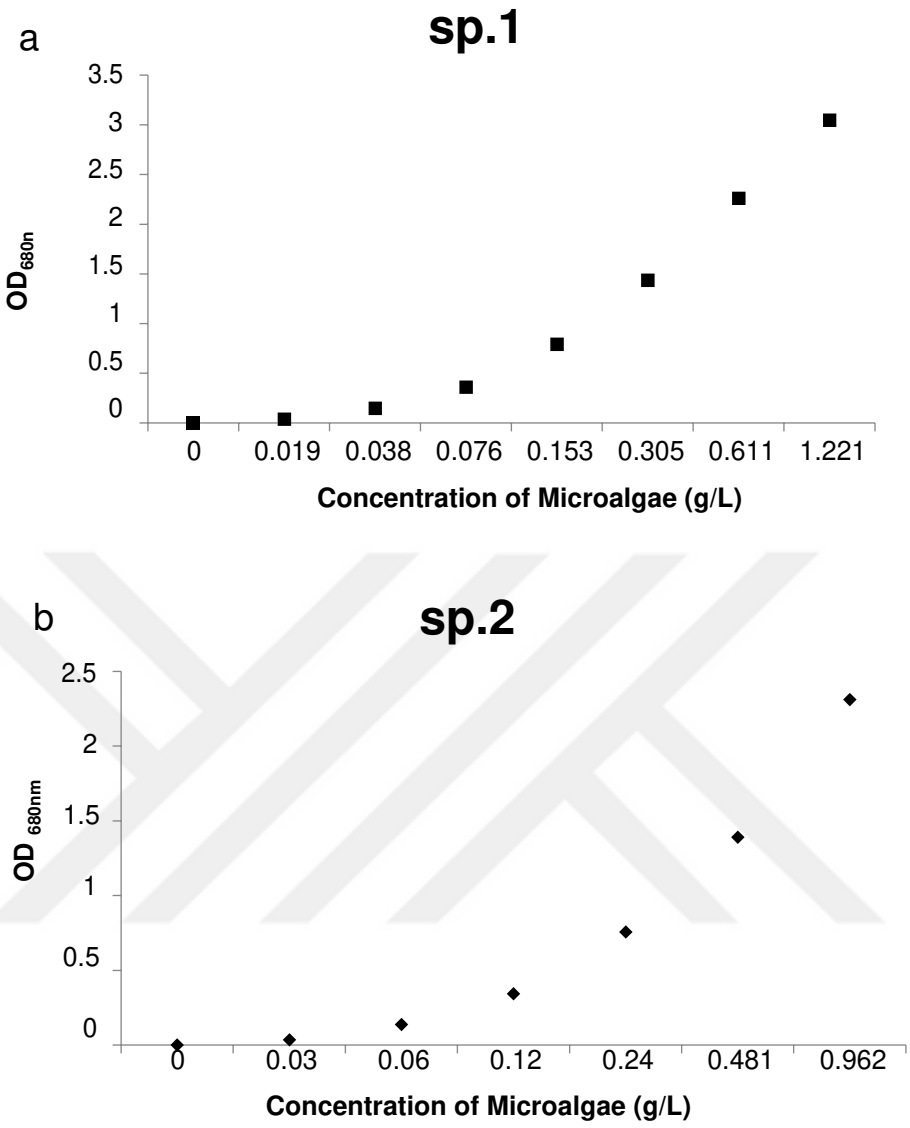

Figure 5.1. *(a, b and c) The calibration curve of the microalgae species, a: Synechocystis sp., b: Chlorella Vulgaris sp., c: Scenedesmus Quadricauda sp.*

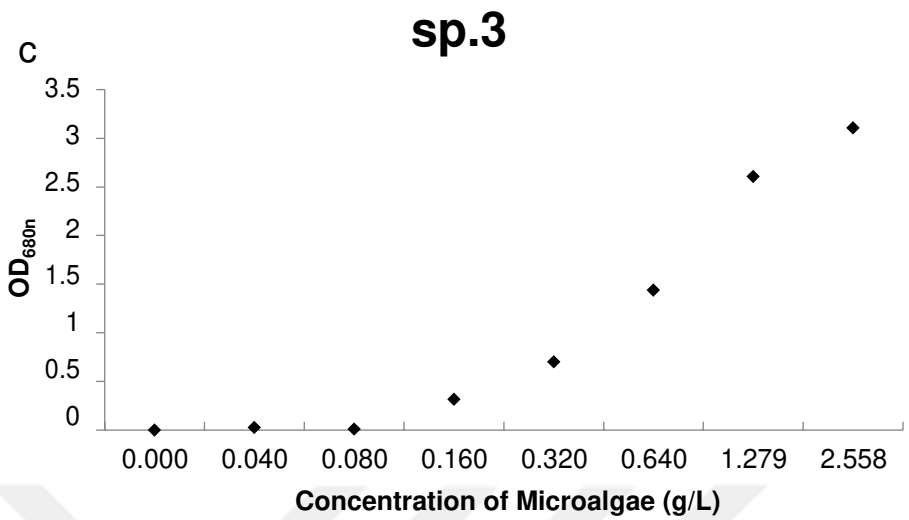

Figure 5.1. (cont.) *(a, b and c) The calibration curve of the microalgae species, a: Synechocystis sp., b: Chlorella Vulgaris sp., c: Scenedesmus Quadricauda sp.*

5.2. Microalgae Species and Cultivation

The cultivation of microalgae in 1 L for *Synechocystis sp.*, *Chlorella Vulgaris sp.* and *Scenedesmus Quadricauda sp.* is shown in Figure 5.2. 5 L microalgae cultivation is shown in Figure 5.3 with one species having two volumes. The growth rate for *Synechocystis sp.* was very high and in a few days the growth curve increased rapidly and showed the highest growth rate from the other species. The growth rate for the three species was significantly high due to the optimal cultivation conditions. It was provided with enough light source, nutrients, CO_2 and suitable temperature. Additionally, the contamination of the cultures was also very low which was contributed to successful rapid growth for the three species.

As in Figures 5.2 and 5.3, the growth rates were different according to the days and *Chlorella Vulgaris sp.* growth rate was higher than *Scenedesmus Quadricauda sp.* The three microalgae species reached the required growth rate which was close to 1.000 nm in different days under optimal cultivation conditions.

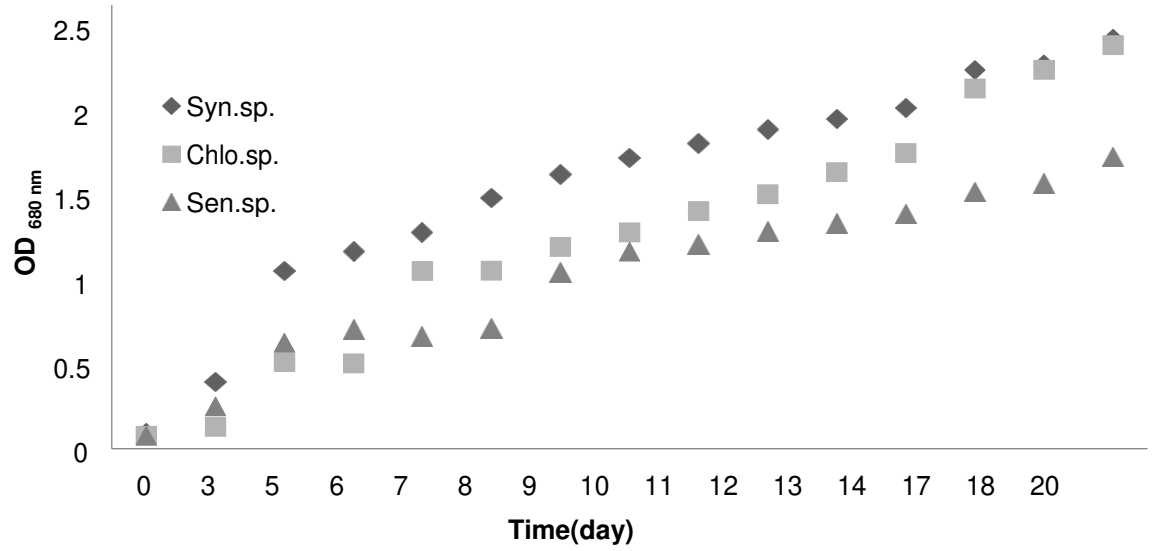

Figure 5.2. *Microalgae species growth rate in 20 days for 1L sp.1 Synechocystis sp., Chlorella Vulgaris sp. and Scenedesmus Quadricauda sp.*

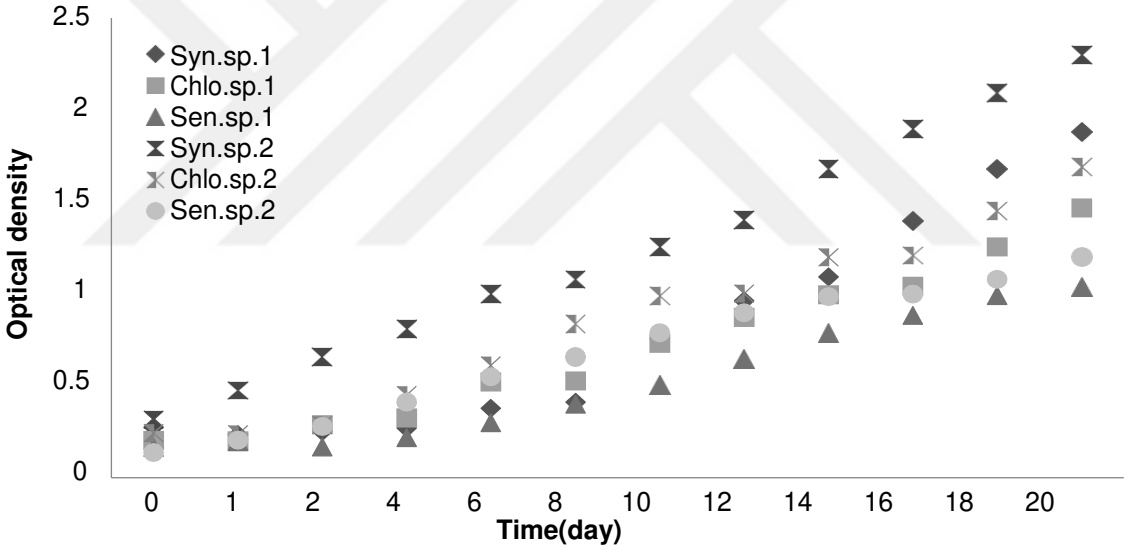

Figure 5.3. *Microalgae species growth rate in 20 days for 5L.*

5.3. Microalgae Natural Settling

The determination of the microalgae natural settling was very important to test the settling efficiency of each microalgae species and the species with higher settling efficiency have higher harvesting efficiency (Parthasarathy & Narayanan, 2014). In this study the microalgae natural settling behavior was investigated as a harvesting technique and to determine the settling efficiency that related to the other harvesting method's efficiency. Natural settling test of the three microalgae species was the simplest method

and it was less costly compared to the other used harvesting methods. The initial OD_{680} was tested after 30 min and 24 h of the settling time and it was found that the natural settling of the three species was significantly different. *Synechocystis sp.* had a very poor natural settling rate compared to the other two microalgae species. Image 5.1 shows the settling samples after 30 min in the right tube and the left one settling after 24 h.

For *Synechocystis sp.* the OD_{680} after 30 min was 1.012 nm and OD_{680} after 24 h was 0.948 nm with the recovery efficiency calculated as RE (30 min) = 7.5 % and RE(24h) = 13.3%. *Chlorella Vulgaris sp.* had a good natural sedimentation rate compared to *Synechocystis sp.* which the OD_{680} (30 min) was 0.766 nm and the OD_{680}(24h) was 0.221 nm corresponding to the recovery efficiency of RE (30 min) = 26.5%, RE(24 h)= 78.8%. Image 5.2 shows the settling after 30 min and after 24 h of *Chlorella Vulgaris sp.* For *Scenedesmus Quadricauda sp*., OD_{680} (30 min) was 0.172 nm and the OD_{680} (24h) was 0.093 where the RE (30min) was 83.4% and RE (24h) was 91% respectively. Image 5.3 shows the settling after 30 min and after 24 h for *Scenedesmus Quadricauda sp*.

Scenedesmus Quadricauda sp. had the highest settling rate compared to the other species and the same results were found by Derakhshandeh et al., (2020). Figure 5.4 shows the RE of the three microalgae species with natural settling after 30 min and 24 h. The settling efficiency of the microalgae depend on some factors such as, the cell size, the growth medium density ratio to the microalgae species' density and the growth phase of the microalgae, where dead cells tend to settle faster (S. K. Choi et al., 2006). *Synechocystis sp. (*blue green microalgae) showed poor settling efficiency compared to *Chlorella Vulgaris sp.* and *Scenedesmus Quadricauda sp.* (green microalgae). It was concluded that the green microalgae species can be harvested more efficiently and probably less costly as the same results were also reported by Derakhshandeh et al., (2020).

Image 5.1. *Natural settling of Synechocystis sp. in the solution.*

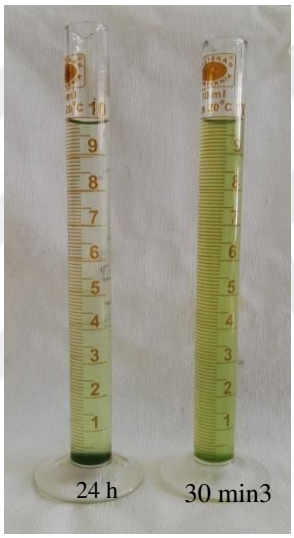

Image 5.2. *Natural settling of Chlorella Vulgaris sp. in the solution.*

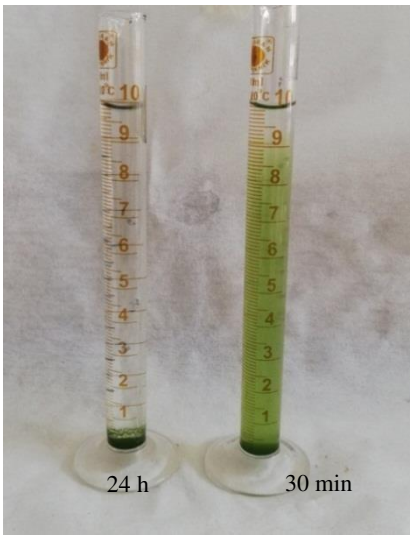

Image 5.3. *Natural settling of Scenedesmus Quadricauda sp. in the solution*

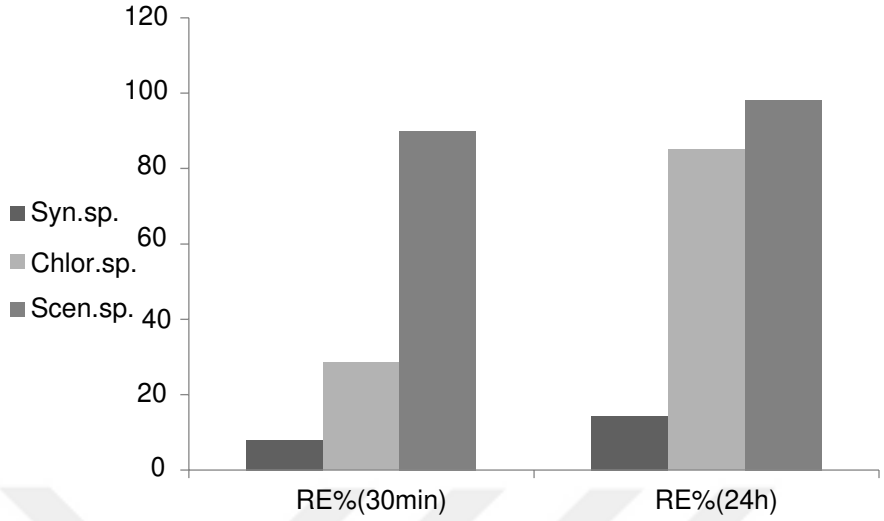

Figure 5.4. *RE of the three microalgae species with natural settling.*

5.4. Electrocoagulation Experiments

The EC experiments were applied with different voltages, pH, electrodes and run times. Table 5.2 provides the different RE conducted on both of *Synechocystis sp.*, *Chlorella Vulgaris sp*. and *Scenedesmus Quadricauda sp.* The mechanism of the EC experiment is must be shown to understand the process.

The EC process is formed as:

At the anode, $M \rightarrow M^{n+} + ne$, (1)

at the cathode, $n\ H_2O + ne \rightarrow {}_{n/2}H_2 + nOH^-$ (2)

where, M= is anode material and n= is the number of electrons contributed in the oxidation/reduction reaction. Metal (Al, Fe) ions are produced at the anode and interact with OH^- ions that are produced at the cathode. Insoluble metal hydroxides interact with suspended material then settle down as (Merzouk et al., 2009);

$M^{n+} + nOH^- \rightarrow M(OH)_n$ (3)

In the case of using Al electrodes these reactions occur at the anode

$Al \rightarrow Al^{3+} + 3e^-$ (1)

$$xAl^{3+} + y\ OH^{-} \rightarrow Alx(OH)_y^{2+} \quad (2)$$

and with Fe electrodes

$$Fe \rightarrow Fe^{2+} + 2e^{-}, \quad (1)$$

$$Fe^{2+} + 2OH^{-} \rightarrow Fe(OH)_2, \quad (2)$$

or

$$Fe \rightarrow Fe^{3+} + 3e^{-}, \quad (3)$$

$$Fe^{3+} + 3OH^{-} \rightarrow Fe(OH)_3 \quad (4)$$

The mechanisms regarding the formation of the Fe^{2+} or Fe^{3+} ions during the EC are not clear and Fe^{2+} can oxidize to Fe^{3+} in the presence of O_2. However, Fe^{2+} in the EC solution produces brown- green hydroxide precipitates, while Fe^{3+} produces yellow precipitates (Vandamme et al., 2011).

In the present study, visual observation of the mixture through the EC experiment showed a greenish-brownish color of insoluble hydroxides precipitates with Fe electrodes and milky precipitates with Al electrodes. The green- brown precipitates were formed and it was proposed that Fe^{2+} ions rather than Fe^{3+} were released during EC experiment as shown in Image 5.4. Vandamme et al., (2011) also noticed the brown-green precipitates with Fe electrodes in EC experiment.

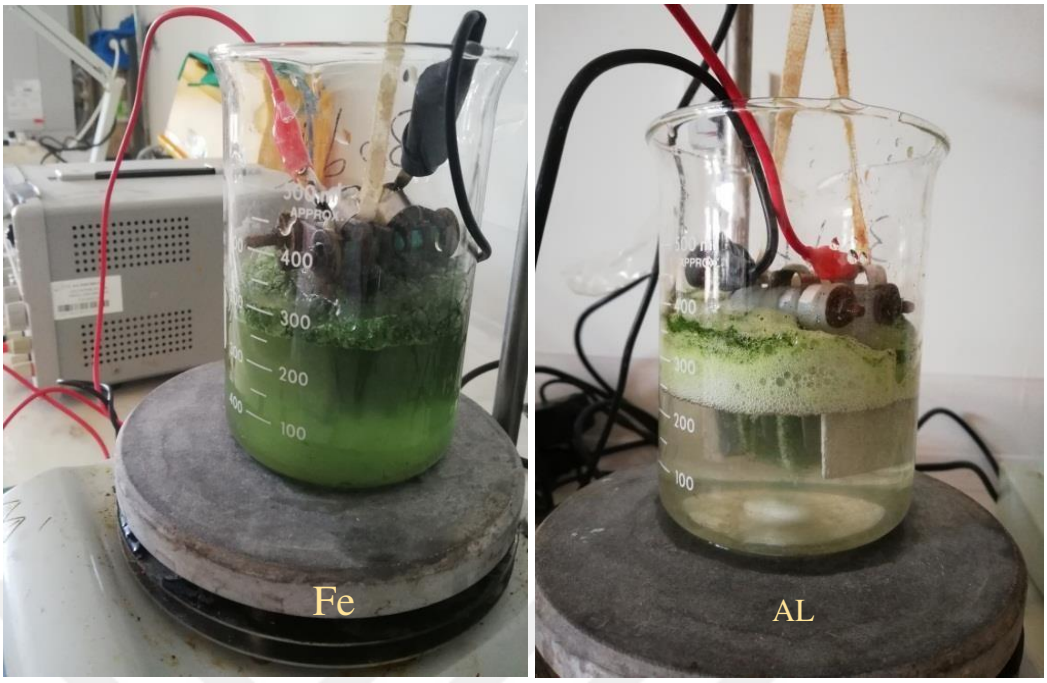

Image 5.4. *Fe and Al electrodes during the EC process.*

This study aimed to achieve the highest recovery efficiency of the microalgae with minimal processing cost. Furthermore, three parameters were investigated including original and adjusted pH, electric current and time on the microalgae harvesting efficiency. The voltage, time, harvesting efficiency, energy and electrode consumption were calculated and shown in Tables 5.2, 5.3 and 5.4 for both Fe and Al electrodes in the order for the three species.

5.4.1. The effect of the electric current and time

Figure 5.5. and 5.6 shows the effect of different electric currents for 24 h of settling time. Also, as in Figures 5.9, 5.10 and 5.11, changes in the electric current had an effect on the efficiency of harvesting microalgae. Figures 5.9a, 5.10a and 5.11a show the linear relationship between the applied electric current and the harvesting efficiency with operating time 30 min and 24 h of settling time with Fe electrodes for *Synechocystis sp., Chlorella Vulgaris sp.* and *Scenedesmus Quadricauda sp*., respectively. As an example, the current 0.2 A with the Fe electrodes was showed lower RE after 30 min and after 24 h compared to the higher applied electric current in the experiment and Golzary et al., (2015) found the same result. The current 0.5 A and 0.8 A with *Synechocystis sp.* resulted in a higher recovery efficiency with original pH values unchanged as that of the microalgae cultures after 30 min reached 62% and 67% sp., respectively Figure 5.9 (a).

The recovery efficiency at electric applied 0.8 A with Fe electrodes on *Chlorella Vulgaris sp.* and *Scenedesmus Quadricauda sp.* were higher than *Synechocystis sp.* and reached approximately 63 % and 97% after 30 min, respectively as shown in Figure 5.10 (a) and 5.11 (a). The increase of the settling time after the EC experiment led to a rise of the recovery efficiency. Figure 5.6 (a, b and c) shows the samples after 24 h of settling time at electric current 0.8 A and the RE was higher with Chlorella Vulgaris sp. and Scenedesmus Quadricauda sp. than Synechocystis sp.

According to Faraday's Law Eq.5, the electric current has a direct effect on the rate of dissolution of the anode and has a significant impact on the performance of the EC experiment (Golzary et al., 2015). Therefore, with an increase in the electric current, the amount of coagulant ions increases, and therefore the microalgae isolation rate increases (Mayank Kumar et al., 2009). Moreover, the amount and the speed of hydrogen gas depends strongly on the electric current strongly so that the increase of the electric current results in the increase of the hydrogen produced (Nanseu-Njiki et al., 2009). Then, a higher amount of the isolated microalgae float on the liquid surface by the gas flow (Golzary et al., 2015). In order to increase the efficiency of the microalgae harvesting, the electric current is increased. According to Faraday's law, the increase of the electric current results in an increase in energy and electrodes consumption, thus an increase in operating costs is not avoidable (Martínez-Villafañe et al., 2009).

The time of the applied current was an effective parameter in the EC experiment. Based on Faraday's Law Eq.5, the amount of the dissolved electrodes is proportional to the time of the reaction (Martínez-Villafañe et al., 2009). It has been noticed that the increase of the electric current and reaction time led to an increase of the efficiency and energy consumption as found by (Golzary et al., 2015; Uduman et al., 2011). Figure 5.5 a, b and C shows the EC experiment with the reaction times of 5, 10, 20 and 30 min and Fe electrodes when applied on the three microalgae species after a settling time of 24 h at I=0.2 A and *Scenedesmus Quadricauda sp.* had the highest recovery efficiency. In some cases, a slight decrease in the separation efficiency may occur with an increase of electric current and time due to the electrostatic repulsion forces between the flocs, which reduces their sedimentation (Golzary et al., 2015). RE of *Scenedesmus Quadricauda sp.* at I= 0.8 A was slightly lower comparing to I: 0.2 A and 0.5 A.

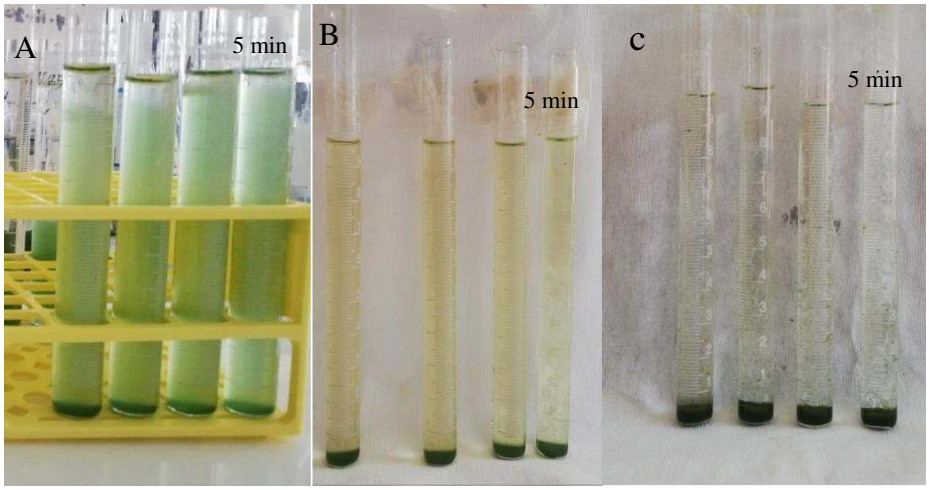

Figure 5.5. *EC with Fe electrode at I=0.2 A after 24 h of settling time, A: Synechocystis sp., B: Chlorella Vulgaris sp. and C: Scenedesmus Quadricauda sp.*

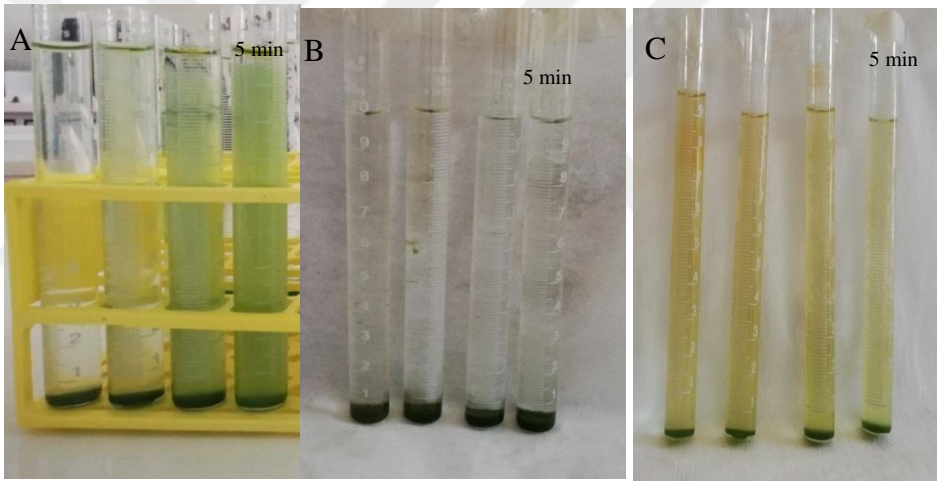

Figure 5.6. *EC with Fe electrode at I=0.8 A after 24 h of settling time, A: Synechocystis sp., B: Chlorella Vulgaris sp. and C: Scenedesmus Quadricauda sp.*

5.4.2. The effect of the pH

The pH of the medium plays a significant role in the performance of the EC experiments (Nanseu-Njiki et al., 2009). In this study it was noticed that the effect of the original pH compared to the current and time had a significant effect on the efficiency of harvesting. Additionally, if the original pH of the medium overtakes the original value from 8 to 9, the RE would reduce, and if it decreases from 6 to 5, the harvest efficiency also may decrease (Golzary et al., 2015). In the present study the original pH of the

culture medium in the three microalgae species was higher than pH 8 and the highest RE reached 91% after 30 min.

As with the Al electrodes, various Al hydroxides are formed at different pH values, at pH<4, $Al(H_2O)_6^{3+}$ is formed and with a high pH increase, these species are formed $Al(OH)_4$, $Al_2(OH)_2^{4+}$, $Al(OH)_5^{2}$ and polymerized to hydroxide forms (Golzary et al., 2015). The ability of these polymeric forms to neutralize and absorb the colloidal particles in the mixture are effective (Rebhun & Lurie, 1993). $Al(OH)_4$ is formed when the pH is above 9 and is not efficient with harvesting microalgae by EC (Gomes et al., 2007). When the culture medium is slightly acidic ($6 < pH < 7$) the dominant mechanism of isolation is absorption and neutralization whereas when the pH is alkaline, the sweeping mechanism is dominant, it therefor seems that the neutralization has a better separation efficiency than sweeping (Gao et al., 2010). It can be concluded that the acidic medium is more efficient than the alkaline one.

In this study the pH was adjusted into values of 4, 7 and 12 the RE of the sample with a pH of 4 reached maximum values of 100% in most of the runs. For the pH 7 the RE was not high compared to the pH values 4 and 12. The pH 12 value with Fe electrodes had a significant effect in the EC experiment and *Scenedesmus Quadricauda sp.* had the maximum RE values either after 30 min or 24 h. The original pH values and the conductivity in the three species were increased with increasing applied current and the time as shown in Table 5.2. The maximum recovery efficiency was observed after 10 min in the three species. It can be concluded that 10 min was optimum for high recovery efficiency and less energy consumption.

EC experiments were performed with Al electrodes under the same conditions of Fe electrodes and it was found that Al electrodes were more efficient as higher RE values were achieved in a shorter time than Fe electrodes as was also found by (Bleeke et al., 2015). The Al electrodes were more efficient than the Fe mostly, because the latter have less current efficiency and they separate less than the Al electrodes (Barros et al., 2015).

The RE values were high with the electric current of 0.2 A for all the three species with the medium's original pH values after 10 min of reactions. The RE values were close to each other for the three species either after 30 min or 24 h settling time. It can be

concluded that the reaction time of 10 min with original pH values with the electric current of 0.2 A was enough with Al electrodes in the three species. The RE was high with a pH 4 value in the three species and reached 100% after 10 min with 30 min of settling time. For the pH 7, the results were near to that of pH 4 values in the three species. Since *Synechocystis sp.* had poor natural settling rate and its RE was lower in most of the run times even with pH 12 and Al as electrode, the RE after 30 min was lower than the other species with the same run settings. The pH 12 value had higher RE values with *Scenedesmus Quadricauda sp.* and the maximum value was 100% achieved after 30 min with 24 h of settling time. Figure 5.7 (a, b and c) shows the EC experiment with Al electrodes at currents of 0.2 A, 0.5 A and 0.8 A with original pH with *Scenedesmus Quadricauda sp.* after 24 h settling time and the highest RE was achieved in the three runs. Because *Scenedesmus Quadricauda sp.* had high settling rate that can be harvested easily. Figure 5.8 (a, b and c) shows the EC experiment with Al electrodes at adjusted PH 4, 7 and 12 with *Scenedesmus Quadricauda sp.* after 24 h of settling time. The RE was significantly high with pH of 12 and had achieved this after 5 min of reaction time.

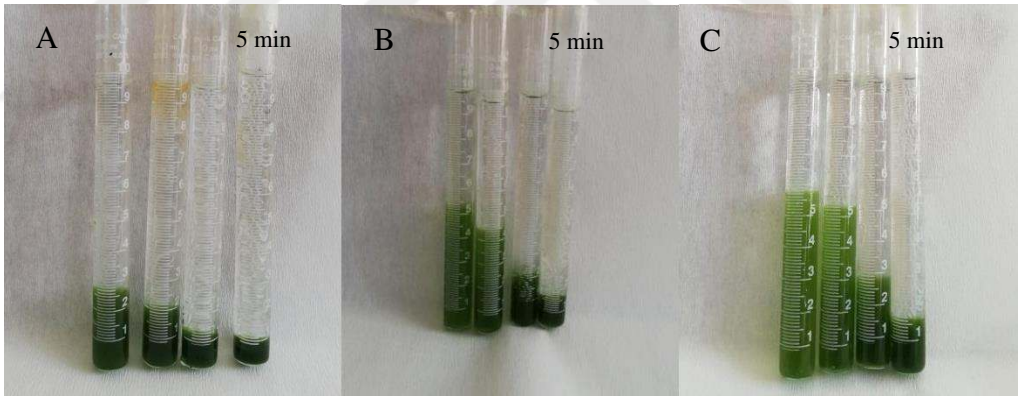

Figure 5.7. *EC experiment with Al electrodes A:0.2 A, B:0.5 A, C:0.8 A with Scenedesmus Quadricauda sp.*

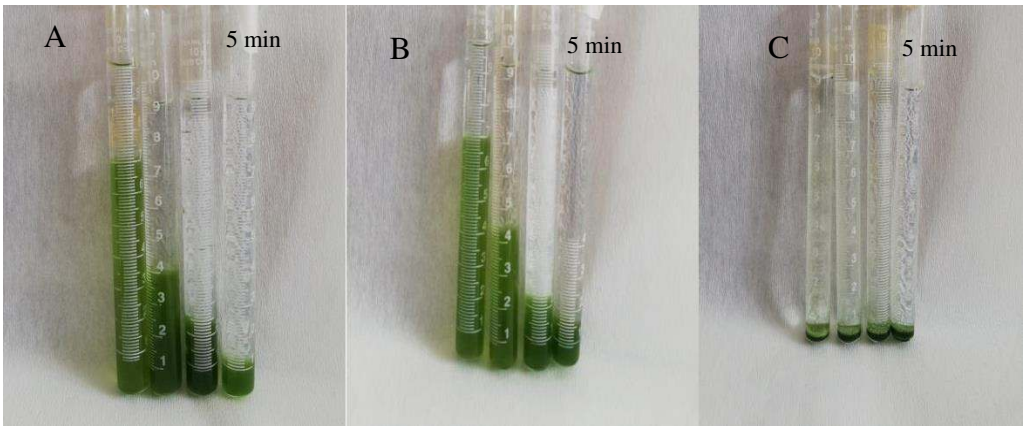

Figure 5.8. *EC with Al electrodes with adjusted pH A:PH 4, B: pH 7, C: pH 12 with Scenedesmus Quadricauda sp.*

The conductivity and pH values were increased with the increasing applied current and run times in original pH values of the cultures with Fe electrodes. However, the conductivity was decreased with Al electrodes in the original pH runs. It could be explained as the decrease in conductivity might be assigned to the decrease in chloride ions by the formation of chlorinated solid compounds as $Al_{45}O_{45}(OH)_{45}Cl$ with Al electrodes (Bejjany et al., 2017). All the results of RE (24 h) were higher than RE (30 min) which can be explained that the electrocoagulation experiment is more efficient with the increase of settling time.

5.4.3. Energy consumption and the costs

Experimental results showed that the harvesting efficiencies could be obtained by applying a high electric current (0.8 A) and Fayad et al., (2017) noticed the same result. For example, it was found that the harvesting efficiency reached the highest value of 100% for *Chlorella Vulgaris sp.* and *Scenedesmus Quadricauda sp*. at the electric currents of 0.5 A and 0.8 A after 30 min. The power and the energy consumed were calculated and shown in Tables 5.2, 5.3 and 5.4 in the order for the three species.

Energy per microalgae mass j/g was calculated and it was found that increasing the applied current increased the consumed energy and microalgae mass and the highest values were obtained after 30 min of reaction time. It was found that the consumed energy was lower when low electric current was applied and the same result was found by (Vandamme et al., 2011). Although a higher applied current leads to faster coagulation

of the microalgae, applying a lower current is more efficient, from an energy consumption's point of view (Hakizimana et al., 2017). Figures 5.12 and 5.13 show the increasing rate of energy with rising of electric current for *Synechocystis sp.* Figures 5.16 and 5.17 for *Chlorella Vulgaris sp.* and *Scenedesmus Quadricauda sp.,* respectively show an increase of the consumed energy with increasing applied current which I= 0.8 A having the highest energy consumption in the three microalgae species. The highest value of the energy consumed per microalgae mass was 170 j/g and noticed with Al electrodes for *Chlorella Vulgaris sp.* with I= 0.8 A. Furthermore, the energy consumption was higher with Al electrodes than Fe electrodes. The energy consumption with pH 12 was also higher in all the species and at the reaction time 30 min. At the original pH values the energy consumption was not high compared to the pH value of 12 in all the species. However, to increase the efficiency of the recovered microalgae using a low current for lower energy consumption, it demands more reaction time (Golzary et al., 2015). Nevertheless, adding more time to the experiment should be taken into account, because it could influence the quality of the harvested microalgae cells. It was found that increasing the applied current increases the electrode consumption, and consequently increases the total cost. The EC experiment with Al electrodes was costlier than Fe electrodes in all the runs.

At the experiment runs of the adjusted pH values it was found that the RE had maximum values with Al electrodes at pH 12 and at pH 4 with Fe electrodes and the applied current in these runs was the same and relatively low (0.5 A). It could be concluded that the pH adjustment method was successful for higher recovery efficiency and lower energy consumption. The difference in energy consumption in harvesting microalgae between the different species is due to the use of marine and fresh water microalgae cultures, where freshwater microalgae have higher energy requirements due to the lower conductivity of the mixture (Uduman et al., 2011).

EC experiment operating cost includes the applied electric current and the consumed metal. The operating cost of the EC using Al electrodes was higher than that from Fe electrodes is due to the price of the metal, Al (1.79 $/kg) and Fe (0.424 $/kg). Additionally, the cost of the EC experiment was increased with an increase of the applied current and can be explained by Faraday's law due to the rising current and electrode consumption (Fuente et al., 2019). The cost (electrode+ electricity) per kilogram of

obtained microalgae ($/kj) was calculated for the three microalgae species as shown in Tables 5.2, 5.3 and 5.4. Figure 5.14 (a and b) shows the cost (Electrode + Electricity) per Kilogram of Obtained Microalgae ($/kg) with *Synechocystis sp.*, a: iron electrodes, original pH, I= 0.5 A, after 30 min of reaction time and 24 of settling time. B: Al electrodes, I= 0.5 A, after 30 min of reaction time and 24 h of settling time. Additionally, Figure 5.15 (a and b) shows the cost (Electrode + Electricity) per Kilogram of Obtained microalgae ($/kg) with *Synechocystis sp*. that a: Fe and Al electrodeswith original pH, I= 0.2 A, 0.5 A and 0.8A after 24 h of settling time. c: with original and adjusted pH values, Al and Fe electrodes, I= 0.5 A and after 24 h of settling time. Figures 5.18 and 5.22 (a and b) show the cost per Kilogram of Obtained Microalgae ($/kg) with *Chlorella Vulgaris sp.* and *Scenedesmus Quadricauda sp*., respectively that a: iron electrodes, original pH, I= 0.5 A, after 30 min of reaction time and 24 of settling time. b: Al electrodes, I= 0.5 A, original pH and after 30 min of reaction time and 24 h of settling. Figures 5.19 and 5.23 (a and b) show the cost (Electrode + Electricity) per Kilogram of Obtained microalgae ($/kg) with *Chlorella Vulgaris sp.* and *Scenedesmus Quadricauda sp*. that a; Fe and Al electrodes with original pH, I= 0.2 A, 0.5 A and 0.8 A after 24 h of settling time. b: with original and adjusted pH values, Al and Fe electrodes, I= 0.5 A and after 24 h of settling time. time It was found that the cost rose with applied current increase and highest values were obtained during the reaction time. With the electric current of 0.8 A, the highest cost values were attained with the three species. With the adjusted pH values the cost values were close to each other because a single electric current was applied 0.5 A.

The EC experiments should be done in pilot- scale to determine whether the power consumption can be extrapolated to industrial scale and thereby estimate the full costs of the operation. Another parameter that may affect the energy consumption is the distance between the electrodes which was not investigated in our work (Holt et al., 2005). The operation cost is a fundamental factor in defining the applicability of any method in large scale. For the lab scale applications, the operating cost comes mainly from the energy and electrode material (Hashim et al., 2017).

Table 5.2. *Fe and Al electrodes runs with Synechocystis sp.*

Time min	Voltage	pH	Con. mS/cm	OD 30min	OD 24 h	RE 30min	RE 24 h	power watt	Energy j	Mic. mass g 30 min	Mic. mass g 24 h	Electrode consu. g	Energy g/30min	Energy g/24 h	Cost $	Cost $/kg algae 30 min	Cost $/kg algae 24 h
Run 1 I: 0.2 A Fe electrodes																	
0	3.3	9.93	2.02	1.018	-	-	-	0.66	0	-	-	-	-	-	-	-	-
5	3.3	10.15	2.09	1.006	0.784	1.179	22.986	0.66	198	0.001	0.014	0.017	267.888	13.738	1.30E-05	17.625	0.904
10	3.4	10.3	2.14	0.914	0.454	10.216	55.403	0.68	402	0.006	0.035	0.035	62.757	11.572	2.62E-05	4.094	0.755
20	3.4	10.65	2.16	0.709	0.358	30.354	64.833	0.68	810	0.019	0.041	0.070	42.559	19.926	5.26E-05	2.765	1.2955
30	3.5	10.92	2.18	0.543	0.334	46.660	67.191	0.7	1230	0.029	0.042	0.104	42.042	29.196	7.94E-05	2.713	1.884
Run 2 I:0.5 A																	
0	4.5	9.93	-	-	-	-	-	2.25	0	-	-	-	-	-	-	-	-
5	4.6	10.71	2.11	0.976	0.777	5.926	23.674	2.3	690	0.004	0.015	0.043	185.707	46.484	3.81E-05	10.019	2.622
10	4.7	10.89	2.13	0.912	0.543	12.213	46.660	2.35	1395	0.008	0.029	0.087	182.174	47.681	7.67E-05	7.470	3.859
20	4.8	11.33	2.17	0.7	0.367	33.038	63.949	2.4	2835	0.021	0.040	0.174	136.856	70.703	0.000155	5.920	4.625
30	4.8	11.47	2.22	0.398	0.201	62.704	80.255	2.4	4275	0.039	0.050	0.261	108.734	84.954	0.000233	10.267	2.570
Run 3 I: 0.8 A																	
0	5.5	9.93	2.02	-	-	-	-	4.4	0	-	-	-	-	-	-	-	-
5	5.7	10.85	2.07	0.972	0.776	6.319	23.77	4.56	1368	0.004	0.015	0.069	345.288	91.778	6.86E-05	17.312	4.602
10	5.8	11.23	2.15	0.927	0.448	10.739	55.992	4.64	2760	0.007	0.035	0.139	409.885	78.615	0.000138	20.474	3.927
20	5.8	11.48	2.21	0.543	0.269	48.460	73.576	4.64	5544	0.030	0.046	0.278	182.457	120.174	0.000276	9.097	5.992
30	5.8	11.64	2.36	0.35	0.125	67.419	87.721	4.64	8328	0.042	0.055	0.417	197.006	151.411	0.000415	9.816	7.545
Run 4 I: 0.2 A Al electrodes																	
0	4.3	9.93	2.02	-	-	-	-	0.86	0	-	-	-	-	-	-	-	-
5	4.3	10.25	2.07	0.95	0.449	6.680	55.894	0.86	258	0.004	0.035	0.006	61.510	7.362	1.7E-05	4.153	0.496
10	4.3	10	2.05	0.141	0.007	86.149	99.312	0.86	516	0.054	0.062	0.011	9.5525	8.286	3.5E-05	0.644	0.559
20	4.4	9.87	2	0.052	0.003	94.892	99.705	0.88	1044	0.059	0.063	0.022	17.547	16.610	6.1E-05	1.175	1.118
30	4.4	9.88	1962µS/cm	0.003	0	99.705	100	0.88	1572	0.0625	0.063	0.034	25.145	25.071	0.00010	1.681	1.675
Run 5 I: 0.5 A																	
0	5.8	9.93	2.02	-	-	2.9	-	-	0	-	-	-	-	-	-	-	-
5	5.8	9.66	2	0.039	0.007	96.169	99.312	2.9	870	0.060	0.062	0.014	14.428	13.971	4.10E-05	0.828	0.802
10	6	9.76	1950µS	0.006	0.002	99.410	99.804	3	1770	0.062	0.063	0.028	28.396	28.284	0.000101	1.615	1.609
20	6.4	9.81	1970 µS	0.003	0.006	99.705	99.411	3.2	3690	0.062	0.062	0.056	59.024	59.199	0.000206	3.290	3.210
30	6.4	10.08	1959 µS	0	0.001	100	99.902	3.2	5610	0.063	0.063	0.083	89.471	89.559	0.000311	4.955	4.960
Run 6 I: 0.8 A																	
0	7	9.93	2.02	-	-	-	-	5.6	0	-	-	-	-	-	-	-	-
5	7	9.89	2.03	0.087	0.086	92.048	92.139	5.6	28	0.057	0.062	0.022	29.297	27.059	8.81E-05	1.537	1.419
10	7.1	9.8	1960 µS	0.09	0.076	91.773	93.053	5.68	84.8	0.057	0.063	0.044	59.204	53.970	0.000177	3.095	2.821
20	7.3	10	1966 µS	0.083	0.077	92.413	92.961	5.84	201.6	0.058	0.063	0.089	119.605	109.962	0.000357	6.203	5.703
30	7.8	10.2	1970 µS	0.081	0.075	92.596	93.144	6.24	388.8	0.058	0.063	0.134	184.223	169.565	0.000544	9.434	8.683
Run 7 I:0.5 A PH=4 Fe electrodes																	
0	4.2	4	2.3	-	-	-	-	2.1	0	-	-	-	-	-	-	-	-
5	4.5	5.83	2.09	0.801	0.345	21.316	0.801	2.25	675	0.013	0.041	0.043	50.502	16.283	3.77E-05	2.822	0.910
10	4.6	6.29	2.12	0.765	0.322	24.853	0.765	2.3	1365	0.016	0.043	0.087	87.595	31.841	7.59E-05	4.868	1.770
20	4.7	9	2.08	0.592	0.022	41.847	0.592	2.35	2775	0.026	0.061	0.174	105.76	45.235	0.000153	5.832	2.494
30	4.8	9.88	2.08	0.367	0.008	63.949	0.367	2.4	4215	0.040	0.062	0.260	105.12	67.756	0.000231	5.762	3.714
Run 8 PH= 7 I: 0.5 A																	
0	4.2	7	2.08	-	-	-	-	2.1	0	-	-	-	-	-	-	-	-
5	4.4	8.05	2.06	0.989	0.25	2.849	75.442	2.2	660	0.002	0.047	0.043	369.500	13.952	3.73E-05	20.876	0.788
10	4.5	9.41	2.06	0.961	0.225	5.510	77.898	2.25	1335	0.004	0.049	0.087	380.255	27.332	7.50E-05	21.365	1.536
20	4.6	10.29	2.12	0.813	0.212	20.138	79.175	2.3	2715	0.013	0.050	0.173	215.023	54.690	0.000151	11.983	3.048
30	4.8	10.89	2.13	0.686	0.199	32.613	80.452	2.4	4155	0.020	0.050	0.260	203.190	82.367	0.000229	11.214	4.546

Table 5.2 (cont.) Fe and Al electrodes runs with *Synechocystis sp.*

							Run 9 I: 0.5 A PH= 12										
0	3.5	12	3.21	-	-	-	-	1.75	0	-	-	-	-	-	-	-	-
5	3.8	12.05	3.19	0.493	0.259	51.572	74.558	1.9	570	0.032	0.047	0.043	17.627	12.193	3.47E-05	1.074	0.743
10	4	12.03	3.03	0.345	0.184	66.110	81.925	2	1170	0.041	0.0514	0.087	28.225	22.777	7.03E-05	1.696	1.368
20	4	12.15	3	0.204	0.073	79.961	92.829	2	2370	0.050	0.0582	0.174	47.271	40.718	0.000141	2.821	2.430
30	4	12.2	2.73	0.194	0.063	80.943	93.811	2	3570	0.051	0.0588	0.260	70.341	60.692	0.000213	4.188	3.614
							Run 10 I:0.5 A PH=4 Al electrodes										
0	7.4	4	2.3	-	-	-	-	3.7	0	-	-	-	-	-	-	-	-
5	7.4	5.87	2.08	0.006	0	99.411	100	3.7	1110	0.062	0.063	0.014	17.808	17.703	5.68E-05	0.911	0.906
10	8	6.54	2.02	0.005	0	99.509	100	4	2310	0.062	0.063	0.028	37.023	36.841	0.000116	1.861	1.852
20	8.01	9.06	2.01	0.002	0	99.804	100	4.005	4713	0.063	0.063	0.056	75.313	75.165	0.000235	3.755	3.747
30	8.06	9.47	1996µs/cm	0.014	0.003	98.625	99.705	4.03	7131	0.062	0.063	0.084	115.315	114.065	0.000354	5.728	5.665
							Run 11 I:0.5 A PH=7										
0	6.5	7	2.18	-	-	-	-	3.25	0	-	-	-	-	-	-	-	-
5	6.5	8.15	2.15	0.063	0.001	93.811	99.902	3.25	975	0.059	0.063	0.014	16.576	15.565	5.29E-05	0.8100	0.845
10	6.7	8.58	2.11	0.001	0.001	99.902	99.902	3.35	1980	0.063	0.063	0.028	31.609	31.609	0.000107	1.703	1.703
20	6.7	9.39	2.1	0	0	100	100	3.35	3990	0.063	0.063	0.056	63.635	63.635	0.000214	3.417	3.417
30	7.4	9.81	2.09	0	0	100	100	3.7	6210	0.063	0.063	0.084	99.040	99.040	0.000328	5.228	5.229
							Run 12 I: 0.5 A PH=12										
0	5	.2	3.23	-	-	-	-	2.5	0	-	-	-	-	-	-	-	-
5	5.7	.1.64	2.43	0.62	0.411	39.096	59.627	2.85	855	0.025	0.037	0.014	34.878	22.869	4.95E-05	2.019	1.324
10	6.5	.1.07	2.4	0.313	0.087	69.253	91.454	3.25	1830	0.043	0.057	0.028	42.144	31.913	0.000102	2.358	1.786
20	9.9	.0.56	2.38	0.016	0.016	98.428	98.428	4.95	4800	0.062	0.062	0.056	77.775	77.775	0.000237	3.847	3.847
30	14.2	.0.57	2.32	0.01	0.014	99.018	98.625	7.1	9060	0.062	0.062	0.084	145.927	146.509	0.000409	6.594	6.620

Table 5.3. *Fe and Al electrodes runs with Chlorella Vulgaris sp.*

Time min	Voltage	Con mS/cm	pH	OD 30 min	OD 24 h	RE 30 min	RE 24 h	Power watt	Energy j	Mic. mass g 30 min	Mic. mass g 24 h	Electrode cons. g	Energy g/30min	Energy g/ 24 h	Cost $	Cost $/kg algae 30 min	Cost $/kg algae 24 h
Run 1 I:0.2 A Fe electrodes																	
0	2	2.14	9.80	-	-	-	-	0.64	0	-	-	-	-	-	-	-	-
5	3.5	2.07	9.91	0.691	0.213	28.468	77.950	0.66	210	0.025	0.069	0.017	8.342	3.047	1.34E-05	0.531	0.194
10	3.6	2.08	9.89	0.648	0.151	32.919	84.369	0.66	426	0.029	0.075	0.035	14.634	5.710	2.69E-05	0.925	0.361
20	3.7	2.1	10.2	0.57	0.119	40.994	87.681	0.7	870	0.036	0.078	0.069	24.000	11.221	5.43E-05	1.410	0.701
30	3.8	2.11	10.42	0.46	0.077	52.381	92.029	0.72	1326	0.046	0.081	0.104	28.628	16.2943	8.21E-05	1.773	1.009
Run 2 I: 0.5 A																	
0	4.6	-	9.80	-	-	-	-	2.3	0	-	-	-	-	-	-	-	-
5	4.8	2.06	9.9	0.569	0.111	41.097	88.509	2.4	720	0.036	0.078	0.043	19.812	9.199	3.90E-05	1.073	0.498
10	5.1	2.05	9.93	0.546	0.109	43.478	88.716	2.55	1485	0.038	0.078	0.087	38.625	18.930	7.93E-05	2.063	1.011
20	5.3	2.08	10.3	0.489	0.106	49.379	89.027	2.65	3075	0.044	0.079	0.174	70.424	39.061	0.000161	3.701	2.053
30	5.4	2.08	10.57	0.441	0.095	54.348	90.166	2.7	4695	0.048	0.080	0.260	97.695	58.886	0.000245	5.093	3.070
Run 3 I: 0.8 A																	
0	5.9	-	9.80	-	-	-	-	4.72	0	-	-	-	-	-	-	-	-
5	6	2.05	9.94	0.53	0.147	45.135	84.783	4.8	1440	0.040	0.075	0.069	36.080	19.208	7.06E-05	1.770	0.942
10	6.2	2.05	10.01	0.431	0.146	55.383	84.886	4.96	2928	0.049	0.075	0.139	59.788	39.008	0.000143	2.913	1.901
20	6.4	2.11	10.39	0.386	0.137	60.041	85.818	5.12	6000	0.053	0.076	0.278	113.010	79.066	0.000289	5.452	3.814
30	6.6	2.18	10.76	0.356	0.11	63.147	88.6128	5.28	9168	0.056	0.078	0.417	164.187	117.002	0.000439	7.862	5.603
Run 4 I: 0.5 A pH: 4																	
0	4.1	2.92	4	-	-	-	-	2.05	0	-	-	-	-	-	-	-	-
5	4.3	2.73	5.5	0.418	0.149	56.729	84.576	2.15	645	0.050	0.075	0.043	12.858	8.624	3.69E-05	0.735	0.493
10	4.6	2.72	6.14	0.424	0.096	56.108	90.062	2.3	1335	0.050	0.080	0.087	26.908	16.763	7.50E-05	1.512	0.942
20	4.6	2.84	7.4	0.417	0.096	56.832	90.062	2.3	2715	0.050	0.080	0.174	54.025	34.091	0.000151	3.011	1.810
30	4.7	2.78	8.87	0.404	0.091	58.178	90.580	2.35	4125	0.051	0.080	0.260	80.183	51.500	0.000228	4.441	2.852
Run 5 pH: 7 I: 0.5 A																	
0	4.3	2.33	7	-	-	-	-	2.15	0	-	-	-	-	-	-	-	-
5	4.4	2.26	7.81	0.53	0.154	45.135	84.058	2.2	660	0.040	0.074	0.043	16.537	8.879	3.73E-05	0.934	0.502
10	4.6	2.29	8.92	0.457	0.142	52.692	85.300	2.3	1350	0.047	0.075	0.087	28.974	17.898	7.54E-05	1.619	1.000
20	4.8	2.35	9.79	0.426	0.114	55.901	88.199	2.4	2790	0.049	0.078	0.174	56.442	35.773	0.000153	3.104	1.967
30	5	2.31	10.16	0.367	0.065	62.008	93.271	2.5	4290	0.055	0.082	0.260	78.239	52.015	0.000233	4.253	2.827
Run 6 pH: 12																	
0	3.6	3.75	12	-	-	-	-	1.8	0	-	-	-	-	-	-	-	-
5	3.8	3.7	12.07	0.253	0.164	73.810	83.023	1.9	570	0.065	0.073	0.043	8.733	7.764	3.47E-05	0.532	0.473
10	3.9	3.73	12.08	0.189	0.136	80.435	85.921	1.95	1155	0.071	0.076	0.087	16.239	15.202	6.99E-05	0.982	0.919
20	3.9	3.83	12.13	0.144	0.108	85.093	88.820	1.95	2325	0.075	0.079	0.174	30.899	29.603	0.000140	1.862	1.784
30	4	3.9	12.16	0.112	0.098	88.406	89.855	2	3525	0.078	0.079	0.260	45.092	44.364	0.000211	2.703	2.659
Run 7 I: 0.2 A Al electrodes																	
0	4.3	-	9.80	-	-	-	-	0.86	0	-	-	-	-	-	-	-	-
5	4.4	2.06	9.82	0.532	0.057	44.928	94.099	0.88	264	0.040	0.083	0.006	6.645	3.173	1.76E-05	0.442	0.211
10	4.4	2.03	9.85	0.135	0.007	86.025	99.275	0.88	528	0.076	0.088	0.011	6.941	6.015	3.51E-05	0.462	0.400
20	4.5	2.08	9.89	0.012	0	98.758	100	0.9	1068	0.087	0.088	0.022	12.230	12.078	7.06E-05	0.808	0.798
30	4.6	2.1	10.03	0	0	100	100	0.92	1620	0.088	0.088	0.034	18.320	18.320	0.000106	1.203	1.203
Run 8 I: 0.5 A																	
0	6.5	-	9.80	-	-	-	-	3.25	0	-	-	-	-	-	-	-	-
5	6.5	2.04	9.84	0.055	0.004	94.306	99.586	3.25	975	0.083	0.088	0.014	11.692	11.072	5.29E-05	0.635	0.601
10	6.7	2.01	9.89	0.02	0.001	97.930	99.896	3.35	1980	0.087	0.088	0.028	22.865	22.415	0.000107	1.232	1.208
20	6.7	1979µS	10	0.006	0	99.379	100	3.35	3990	0.088	0.088	0.056	45.404	45.122	0.000214	2.438	2.423
30	7	2.01	10.06	0	0	100	100	3.5	6090	0.088	0.088	0.084	68.871	68.871	0.000324	3.669	3.669

Table 5.3 (cont.) *Fe and Al electrodes runs with Chlorella Vulgaris sp.*

								Run 9 I: 0.8 A									
0	7.6	-	9.80	-	-	-	-	6.08	0	-	-	-	-	-	-	-	-
5	7.7	2.06	9.84	0.044	0.006	95.445	99.379	6.16	1848	0.084	0.088	0.022	0.088	21.896	9.29E-05	1.101	1.057
10	7.7	2.01	9.9	0.053	0.01	94.513	98.965	6.16	3696	0.084	0.088	0.045	0.088	44.224	0.000186	2.224	2.124
20	8.1	2.01	10.04	0.002	0.002	99.793	99.793	6.48	7584	0.088	0.088	0.089	0.088	85.944	0.000377	4.274	4.274
30	9.2	2.01	10.08	0	0	100	100	7.36	12000	0.088	0.088	0.134	0.088	135.706	0.000582	6.510	6.510
								Run10 I: 0.5 A pH 4									
0	6	2.49	4	-	-	-	-	3	0	-	-	-	-	-	-	-	-
5	6	2.2	5.74	0.027	0	97.205	99.172	3	900	0.086	0.088	0.014	10.471	10.263	5.08E-05	0.591	0.579
10	6.2	2.2	6.36	0.019	0	98.033	100	3.1	1830	0.087	0.088	0.028	21.110	20.695	0.000102	1.181	1.158
20	6.3	2.19	7.83	0.012	0	98.758	100	3.15	3720	0.087	0.088	0.056	42.598	42.069	0.000207	2.365	2.336
30	6.5	2.2	9.01	0	0	100	100	3.25	5670	0.088	0.088	0.084	64.121	64.121	0.000312	3.533	3.533
								Run11 I: 0.5 A pH 7									
0	6.1	2.4	7	-	-	-	-	3.05	0	-	-	-	-	-	-	-	-
5	6.2	2.35	8	0.086	0.004	91.097	99.586	3.1	930	0.080	0.088	0.014	11.545	10.561	5.16E-05	0.641	0.586
10	6.2	2.34	8.43	0.012	0.001	98.758	99.896	3.1	1860	0.087	0.088	0.028	21.299	21.0562	0.000103	1.183	1.169
20	6.3	2.32	9.33	0.004	0	99.586	100	3.15	3750	0.088	0.088	0.056	42.585	42.408	0.000207	2.355	2.345
30	6.5	2.31	9.73	0	0	100	100	3.25	5700	0.088	0.088	0.084	64.460	64.4602	0.000313	3.542	3.542
								Run12 I: 0.5 A pH 12									
0	7	3.76	12	-	-	-	-	3.5	0	-	-	-	-	-	-	-	-
5	7.1	3.48	11.95	0.267	0.257	72.360	73.395	3.55	1065	0.064	0.065	0.014	16.644	16.410	5.55E-05	0.867	0.855
10	12.3	3.38	11.8	0.191	0.133	80.228	86.232	6.15	2910	0.071	0.076	0.028	41.019	38.163	0.000133	1.879	1.748
20	16.6	3.2	11.71	0.128	0.108	86.749	88.820	8.3	7890	0.077	0.079	0.056	102.855	100.458	0.000326	4.248	4.149
30	22.2	2.79	11.61	0.036	0.021	96.273	97.826	11.1	14550	0.085	0.087	0.084	170.912	168.200	0.000566	6.654	6.548

Table 5.4. *Fe and Al electrodes runs with Scenedesmus Quadricauda sp.*

Time min	Voltage	Con. mS/cm	pH	OD 30min	OD 24h	RE% 30min	RE% 24h	power watt	Energy j	Mic. mass g 30min	Mic. mass g 24 h	Electrode consu. g	Energy g/30min	Energy g/24 h	Cost $	Cost $/kg algae 30 min	Cost $/kg algae 24 h
Run 1 I:0.2 A Fe electrodes																	
0	3.2	2.17	10.4	-	-	-	-	0.64	0	-	-	-	-	-	-	-	-
5	3.3	2.11	10.07	0.043	0.011	95.530	98.857	0.66	198	0.119	0.123	0.017	1.663	1.607	1.30E-05	0.109	0.106
10	3.3	2.1	10.23	0.042	0.007	95.634	99.272	0.66	396	0.119	0.124	0.035	3.322	3.200	2.60E-05	0.219	0.211
20	3.5	2.13	10.43	0.038	0	96.050	100	0.7	816	0.120	0.125	0.069	6.815	6.546	5.28E-05	0.441	0.424
30	3.6	2.13	10.62	0.027	0	97.193	100	0.72	1248	0.121	0.125	0.104	10.300	10.012	7.99E-05	0.659	0.641
Run 2 I: 0.5 A																	
0	4.2	-	10.4	-	-	-	-	2.1	0	-	-	-	-	-	-	-	-
5	4.3	2.13	10.36	0.08	0.022	91.684	97.713	2.15	645	0.114	0.122	0.043	5.644	5.295	3.69E-05	0.323	0.303
10	4.4	2.13	10.59	0.054	0.002	94.387	99.792	2.2	1305	0.118	0.124	0.087	11.091	10.490	7.41E-05	0.630	0.596
20	4.4	2.19	11.09	0.051	0	94.699	100	2.2	2625	0.118	0.125	0.174	22.237	21.058	0.00014	1.260	1.193
30	4.5	2.25	11.47	0.043	0	95.530	100	2.25	3975	0.119	0.125	0.260	33.380	31.888	0.00022	1.882	1.798
Run 3 I:0.8 A																	
0	5.3	-	10.4	-	-	-	-	4.24	0	-	-	-	-	-	-	-	-
5	5.3	2.16	10.5	0.233	0.007	75.780	99.272	4.24	1272	0.094	0.124	0.069	13.466	10.279	6.58E-05	0.697	0.532
10	5.4	2.2	10.8	0.231	0.001	75.988	99.896	4.32	2568	0.095	0.125	0.139	27.111	20.622	0.00013	1.397	1.063
20	5.4	2.24	11.29	0.15	0	84.407	100	4.32	5160	0.105	0.125	0.278	49.041	41.394	0.00027	2.523	2.129
30	5.4	2.52	11.6	0.119	0	87.630	100	4.32	7752	0.109	0.125	0.417	70.966	62.188	0.00040	3.648	3.1967
Run 4 I: 0.5A pH 4																	
0	2.2	2.73	4	-	-	-	-	1.1	0	-	-	-	-	-	-	-	-
5	4	2.53	5.94	0.057	0.012	94.075	98.753	2	600	0.117	0.123	0.043	5.116	4.874	3.56E-05	0.303	0.289
10	4.1	2.51	6.76	0.045	0.001	95.322	99.896	2.05	1215	0.119	0.125	0.087	10.225	9.757	7.16E-05	0.602	0.575
20	4.2	2.51	9.5	0.029	0.001	96.985	99.896	2.1	2475	0.121	0.125	0.174	20.472	19.875	0.000144	1.195	1.160
30	4.3	2.51	10.3	0.04	0.007	95.842	99.272	2.15	3765	0.119	0.124	0.260	31.514	30.425	0.000218	1.826	1.763
Run 5 I: 0.5 A																	
0	4.4	2.35	7	-	-	-	-	2.2	0	-	-	-	-	-	-	-	-
5	4.4	2.33	7.79	0.047	0.003	95.114	99.688	2.2	660	0.119	0.124	0.043	5.567	5.311	3.73E-05	0.315	0.300
10	4.5	2.32	9.5	0.044	0.002	95.426	99.792	2.25	1335	0.119	0.124	0.087	11.223	10.732	7.50E-05	0.631	0.603
20	4.6	2.2	10.27	0.03	0.001	96.882	99.896	2.3	2715	0.121	0.125	0.174	22.481	21.803	0.000151	1.253	1.215
30	4.7	2.34	10.81	0.034	0.004	96.466	99.584	2.35	4125	0.120	0.124	0.260	34.304	33.230	0.000228	1.810	1.840
Run 6 I: 0.5 A																	
0	4	3.54	12	-	-	-	-	2	0	-	-	-	-	-	-	-	-
5	4	3.48	12	0.084	0.009	91.268	99.064	2	600	0.114	0.123	0.043	5.274	4.859	3.56E-05	0.313	0.288
10	4	3.48	12.5	0.093	0.012	90.333	98.753	2	1200	0.113	0.123	0.087	10.657	9.748	7.11E-05	0.632	0.578
20	4.1	3.52	12.1	0.084	0.001	91.268	99.896	2.05	2430	0.114	0.125	0.174	21.359	19.514	0.000143	1.258	1.150
30	4.2	3.73	12.15	0.086	0.003	91.060	99.688	2.1	3690	0.114	0.124	0.260	32.508	29.694	0.000216	1.903	1.738
Run 7 I: 0.2 A Al electrodes																	
0	4.5	-	10.4	-	-	-	-	0.9	0	-	-	-	-	-	-	-	-
5	4.6	2.11	9.9	0.117	0.003	87.838	99.688	0.92	276	0.109	0.124	0.006	2.521	2.221	1.79E-05	0.164	0.144
10	4.6	2.12	10	0.066	0	93.139	100	0.92	552	0.116	0.125	0.011	4.754	4.428	3.58E-05	0.308	0.287
20	4.7	2.11	10.5	0.021	0	97.817	100	0.94	1116	0.122	0.125	0.022	9.153	8.953	7.20E-05	0.590	0.577
30	4.8	2.12	10.59	0.002	0.003	99.792	99.688	0.96	1692	0.124	0.124	0.034	13.602	13.616	0.000108	0.872	0.873
Run 8 I: 0.5 A																	
0	6.6	-	10.4	-	-	-	-	3.3	0	-	-	-	-	-	-	-	-
5	6.8	2.12	9.99	0.11	0.002	88.565	99.792	3.4	1020	0.110	0.124	0.014	9.239	8.200	5.42E-05	0.491	0.436
10	6.8	2.1	10.06	0.034	0.002	96.466	99.792	3.4	2040	0.120	0.124	0.028	16.965	16.400	0.000108	0.902	0.872
20	7.1	2.1	10.3	0.025	0	97.401	100	3.55	4170	0.121	0.125	0.056	34.345	33.452	0.000219	1.807	1.760
30	7.7	2.15	10.44	0.038	0.005	96.050	99.480	3.85	6480	0.120	0.124	0.084	54.121	52.255	0.000336	2.803	2.706
Run 9 I: 0.8 A																	
0	9	-	-	-	-	-	-	7.2	0	-	-	-	-	-	-	-	-
5	9	2.11	10	0.026	0.002	97.297	99.792	7.2	2160	0.121	0.124	0.022	17.809	17.364	0.000101	0.840	0.819
10	9.3	2.1	10.8	0	0.001	100	99.896	7.44	4392	0.125	0.125	0.045	35.233	35.270	0.000206	1.651	1.652
20	10.7	2.12	10.45	0.014	0.005	98.545	99.480	8.56	9528	0.123	0.124	0.089	77.564	76.834	0.000433	3.523	3.490
30	15.6	2.14	10.6	0.004	0	99.584	100	12.48	17016	0.124	0.125	0.134	137.075	136.504	0.000727	5.857	5.833

Table 5.4 (cont.) *Fe and Al electrodes runs with Scenedesmus Quadricauda sp*

									Run 10 I: 0.5 A pH4								
0	6.6	2.61	4	-	-	-	-	3.3	0	-	-	-	-	-	-	-	-
5	6.8	2.41	6.18	0.019	0	98.025	100	3.4	1020	0.122	0.125	0.013	0.125	8.347	5.42E-05	0.444	0.444
10	6.9	2.4	6.73	0.018	0	98.129	100	3.45	2055	0.122	0.125	0.028	0.125	16.700	0.000109	0.890	0.890
20	7.4	2.39	8.22	0.003	0	99.688	100	3.7	4275	0.124	0.125	0.056	0.125	34.402	0.000222	1.790	1.790
30	7.8	2.37	9.44	0.005	0.002	99.480	99.792	3.9	6615	0.124	0.124	0.083	0.124	53.344	0.000339	2.737	2.737
									Run 11 I:0.5 A pH 7								
0	6.9	2.26	7	-	-	-	-	3.45	0	-	-	-	-	-	-	-	-
5	7.3	2.25	7.81	0.044	0.002	95.426	99.792	3.65	1095	0.119	0.124	0.014	9.205	8.803	5.64E-05	0.474	0.453
10	7.3	2.23	8.8	0.001	0	99.896	100	3.65	2190	0.125	0.125	0.028	17.587	17.568	0.000113	0.905	0.904
20	7.6	2.19	9.56	0.004	0.001	99.584	99.896	3.8	4470	0.124	0.125	0.056	36.008	35.896	0.000228	1.837	1.831
30	8.1	2.17	9.7	0.003	0	99.688	100	4.05	6900	0.124	0.125	0.084	55.526	55.353	0.000348	2.797	2.788
									Run12 I: 0.5 A pH 12								
0	5.7	3.87	12	-	-	-	-	2.85	0	-	-	-	-	-	-	-	-
5	6.7	3.43	11.8	0.073	0.009	92.412	99.064	3.35	1005	0.115	0.123	0.014	8.724	8.138	5.38E-05	0.467	0.436
10	8.7	3.17	11.7	0.066	0.004	93.139	99.584	4.35	2310	0.116	0.124	0.028	19.896	18.609	0.000116	1.000	0.936
20	13.3	3.1	11.37	0.056	0.002	94.179	99.792	6.65	6300	0.117	0.124	0.056	53.663	50.645	0.000280	2.388	2.254
30	16.6	2.94	11.2	0.049	0	94.906	100	8.3	11280	0.118	0.125	0.084	95.346	90.490	0.000473	3.997	3.794

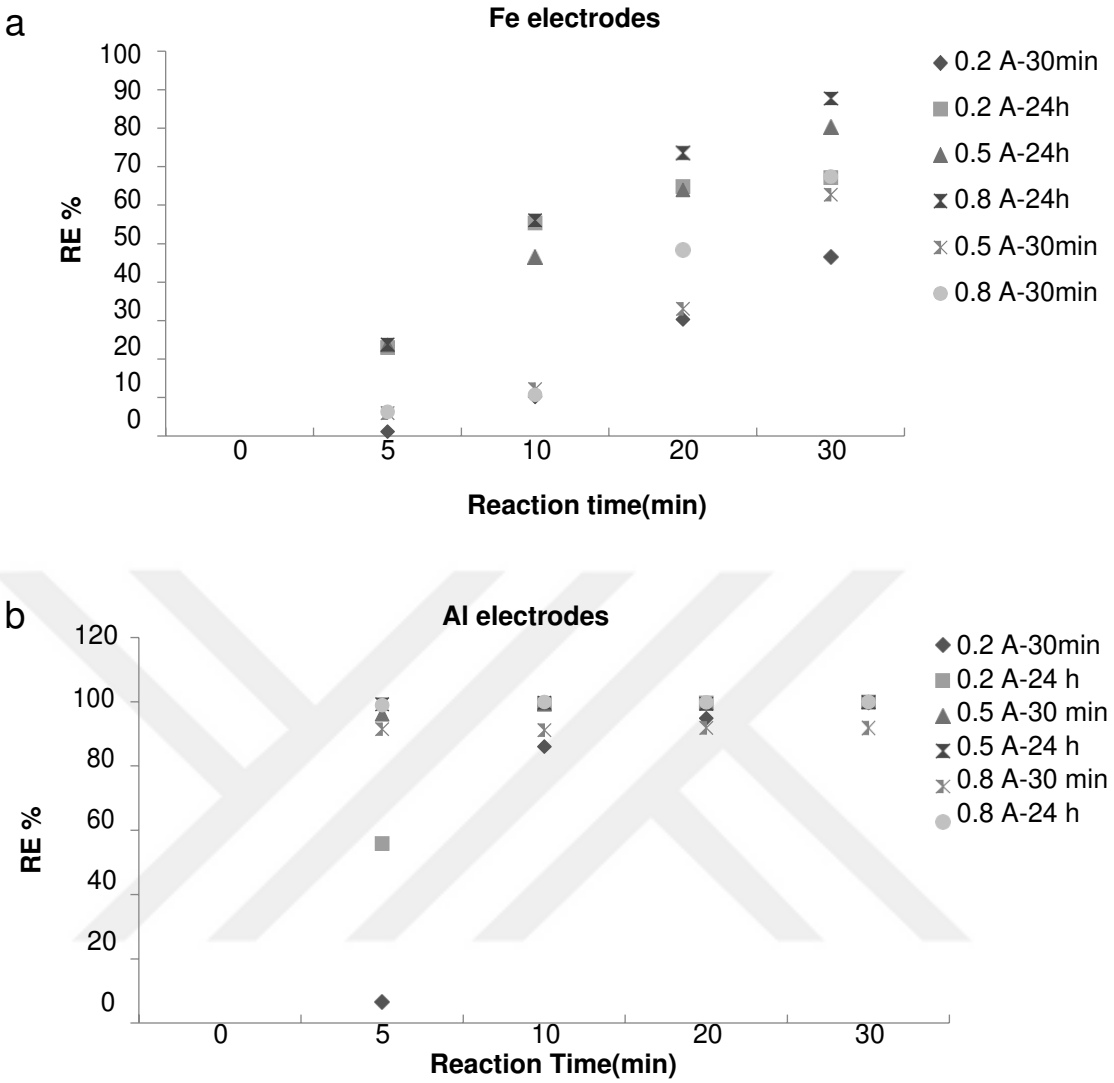

Figure 5.9. *(a, b, c and d) RE of Synechocystis sp. with EC experiment Fe and Al electrodes, 30 min reaction time and 24 h of settling time (a and b): original pH.*

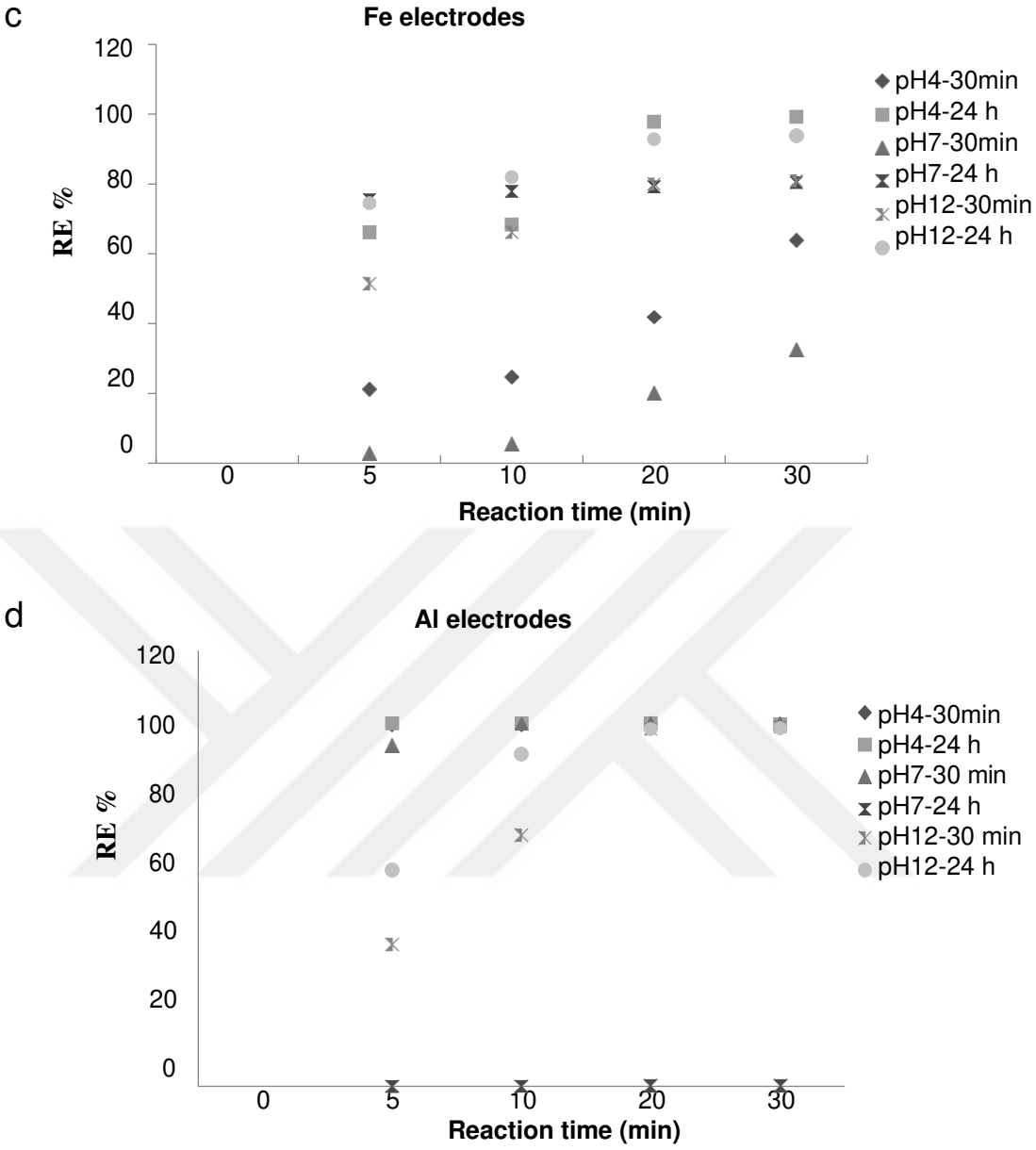

Figure 5.9. (cont.) *(a, b, c and d) RE of Synechocystis sp. with EC experiment Fe and Al electrodes, 30 min reaction time and 24 h of settling tim. (a and b): original pH and (c and d) adjusted pH 4, 7 and 12.*

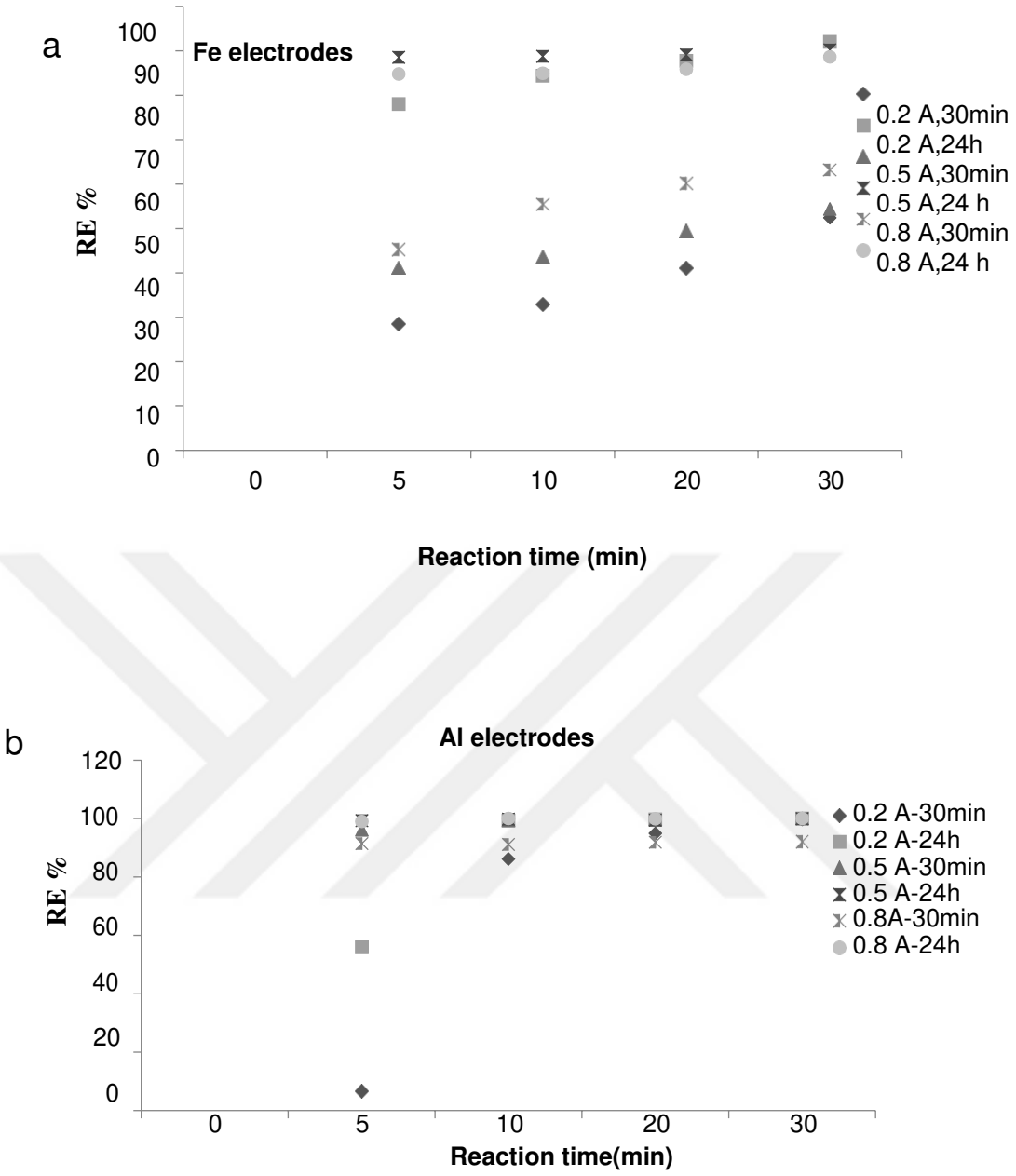

Figure 5.10. *(a, b, c and d) RE of Chlorella Vulgaris sp with EC experiment Fe and Al electrodes, reaction time 30 min and 24 h of settling time. (a and b): original pH.*

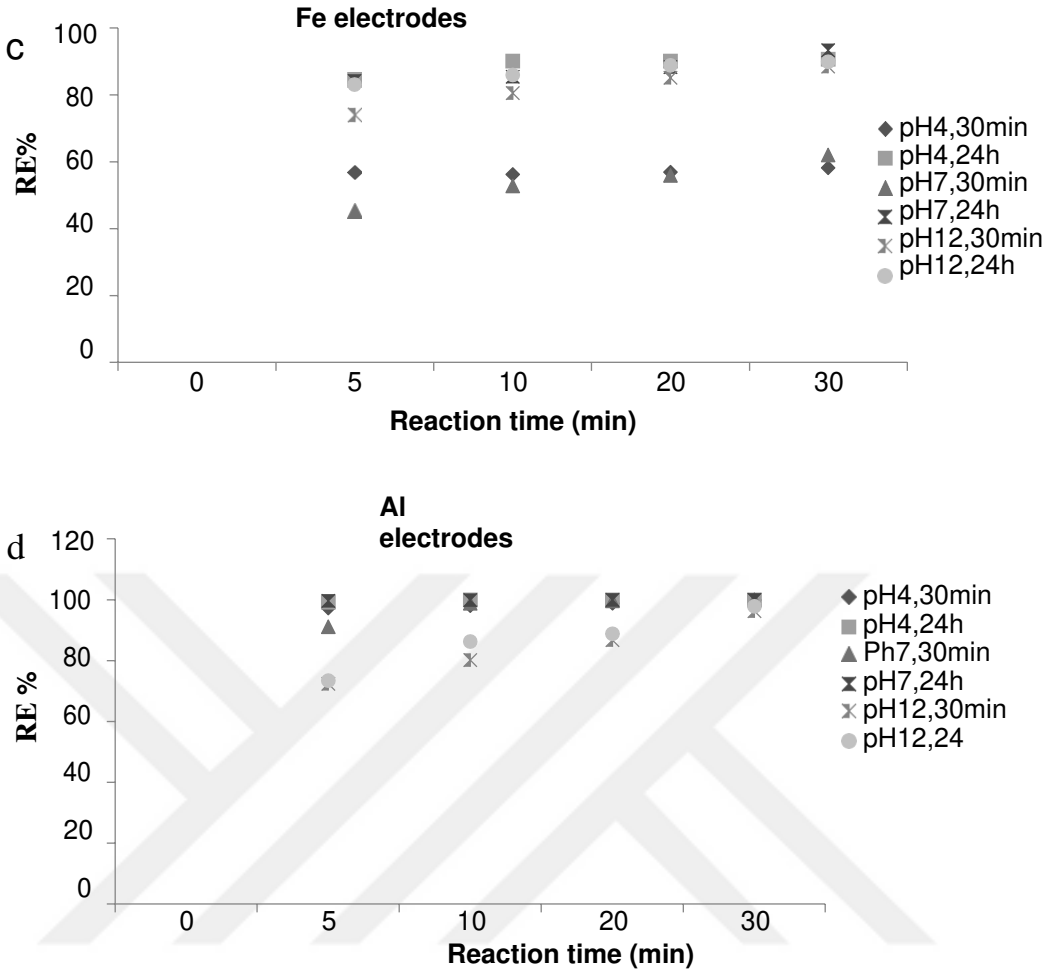

Figure 5.10. (cont.) *(a, b, c and d) RE of Chlorella Vulgaris sp with EC experiment Fe and Al electrodes, reaction time 30 min and 24 h of settling time. (a and b): original pH and (c and d): adjusted values 4, 7 and 12.*

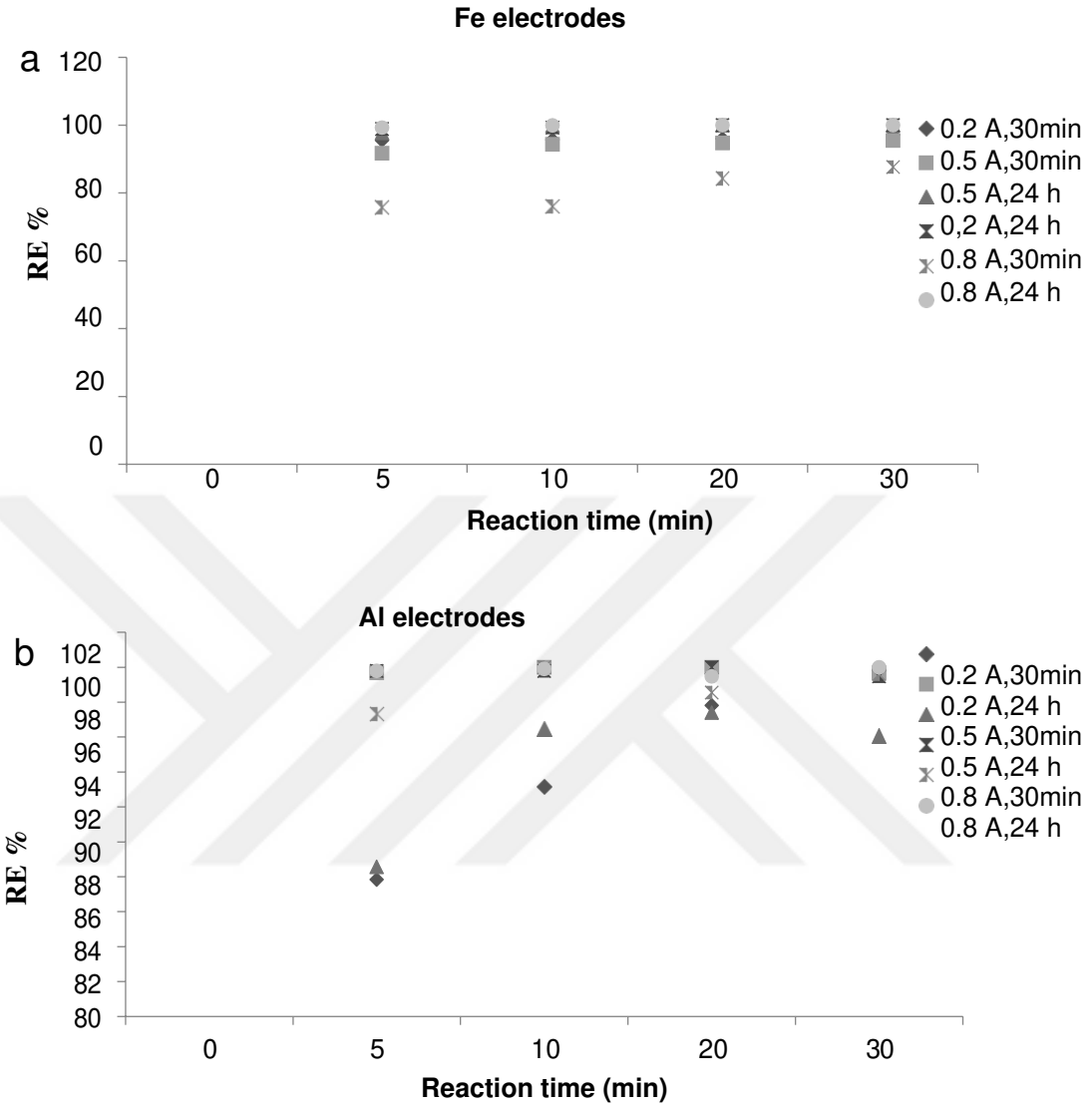

Figure 5.11. *(a, b, c and d) RE of Scenedesmus Quadricauda sp. with EC experiment Fe and Al electrodes, after 30 min of reaction time and 24 h of settling time. (a and b): original pH.*

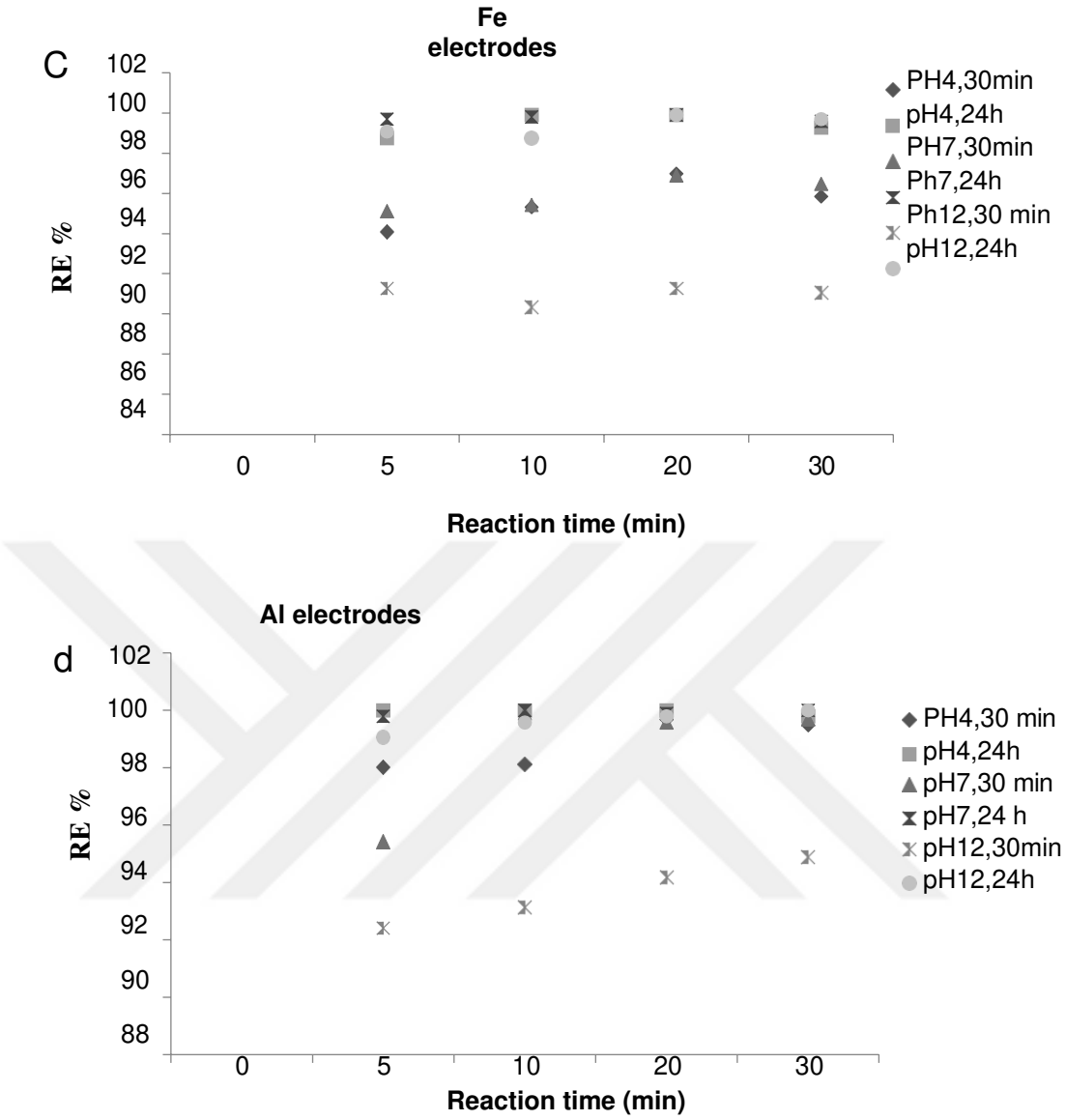

Figure 5.11. (cont.) *(a, b, c and d) RE of Scenedesmus Quadricauda sp. with EC experiment Fe and Al electrodes, after 30 min of reaction time and 24 h of settling time. (a and b): original pH and (c and d) adjusted value 4, 7 and 12.*

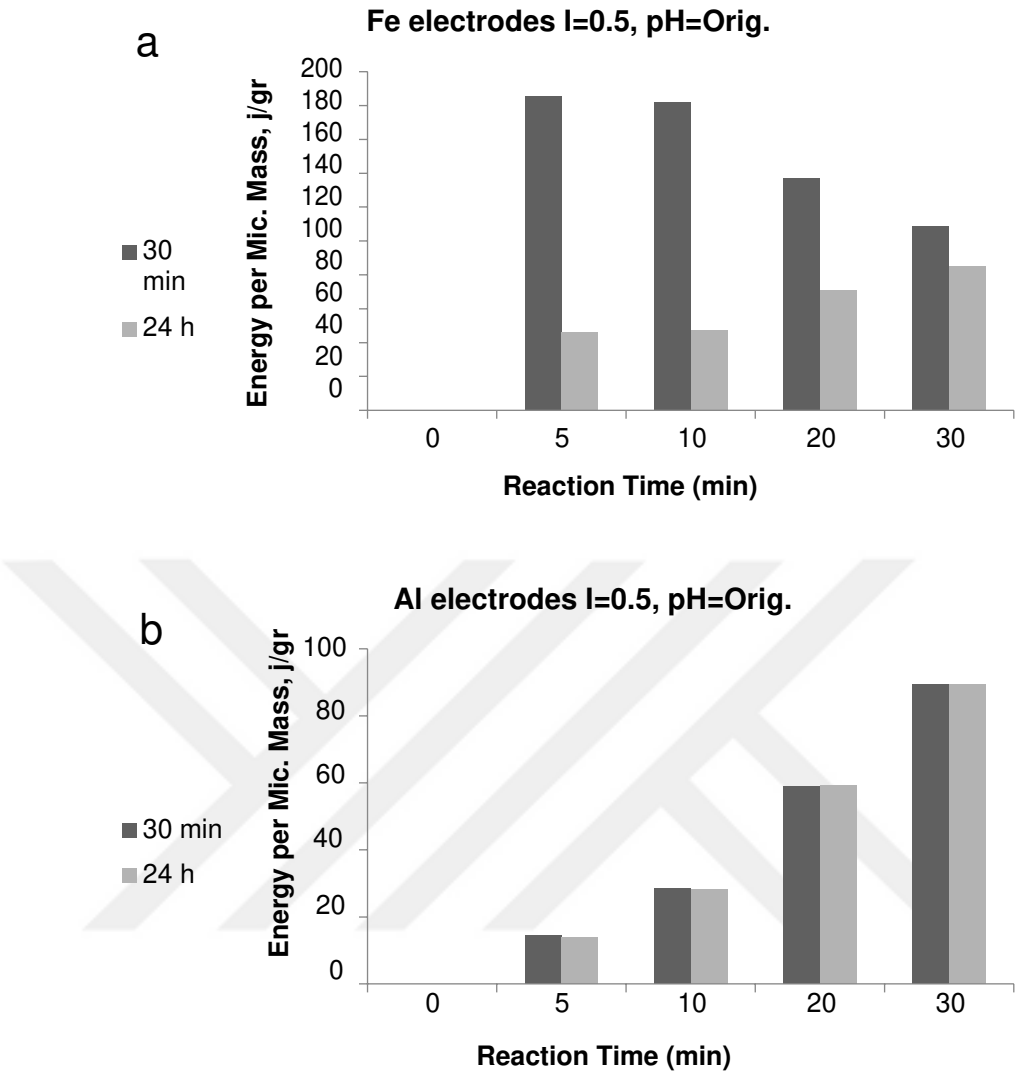

Figure 5.12. *Consumed energy per obtained grams of microalgae (j/g) with Synechocystis sp., a: Fe electrodes, original pH, I= 0.5 A, after 30 min of reaction time and 24 h settling time. B: Al electrodes, original pH, I=0.5 A, after 30 min of reaction time and 24 h settling time.*

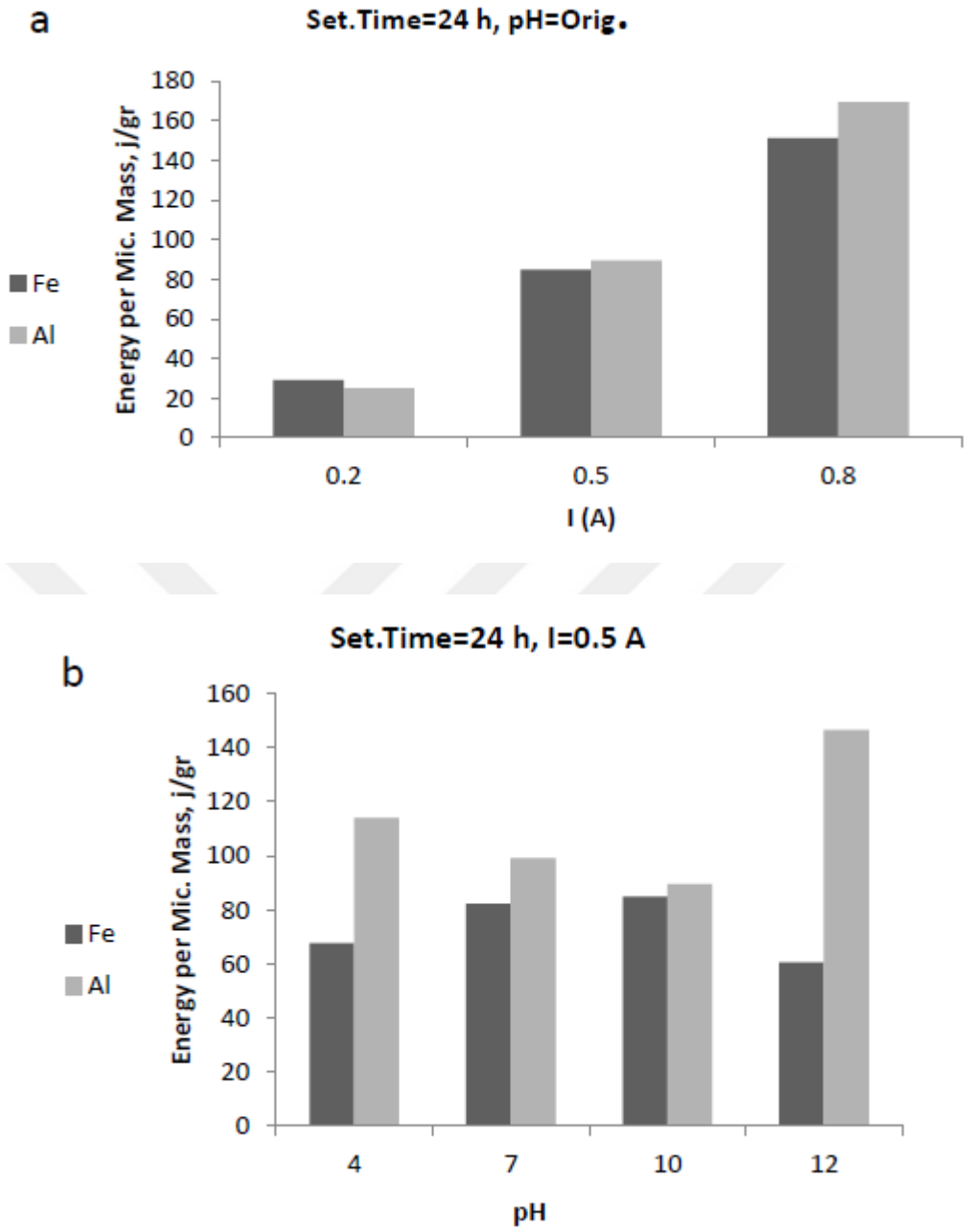

Figure 5.13. *Consumed energy per obtained grams of microalgae (j/g) with Synechocystis sp.,a: original pH, Fe and Al electrodes , I= 0.2A, 0.A5 and 0.8A. and after 24 h of settling time. B: adjusted and original pH value, Fe and Al electrodes, I= 0.5 A and after 24 h of settling time.*

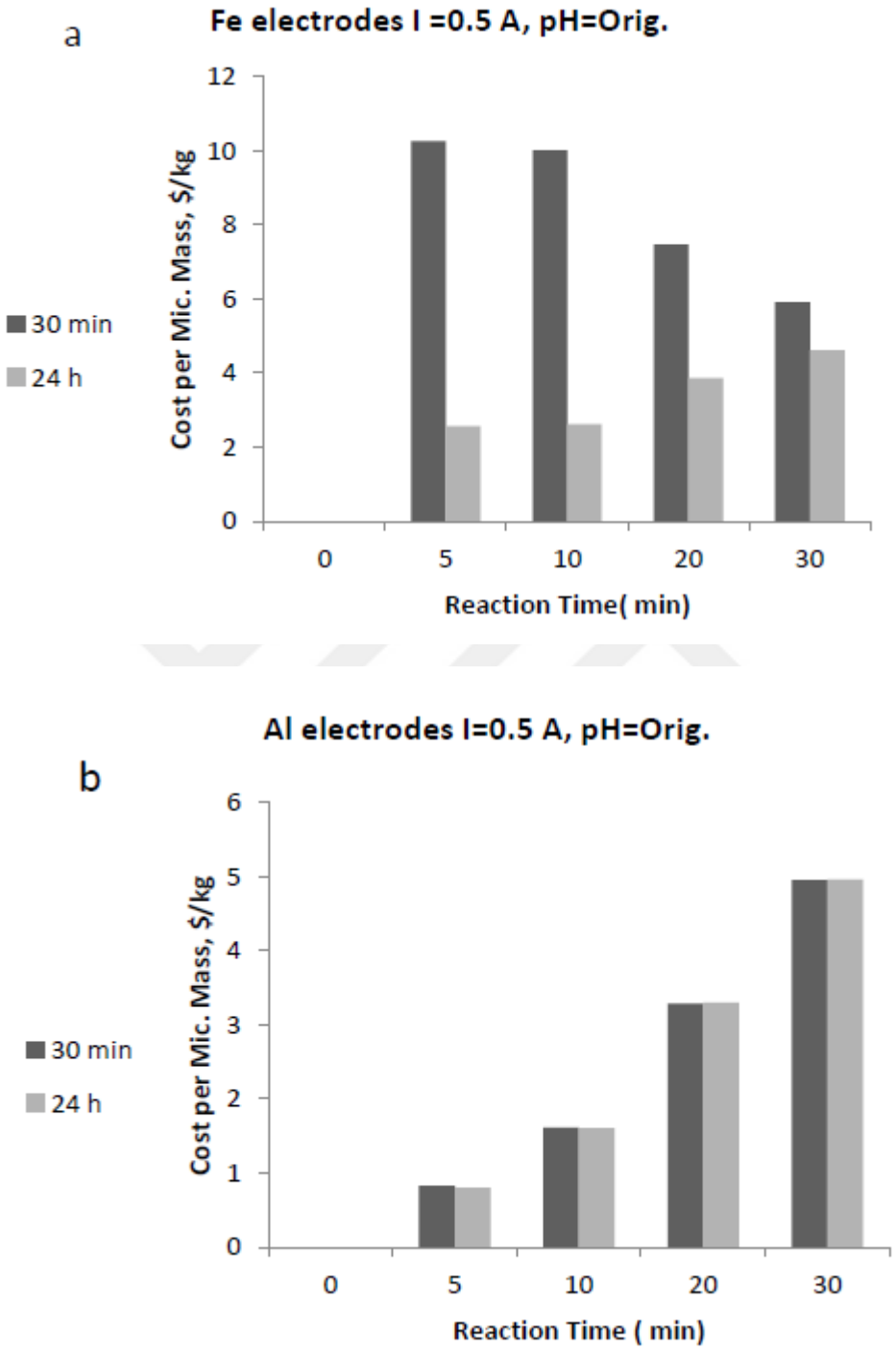

Figure 5.14. *Cost (Electrode + Electricity) per Kilogram of Obtained Microalgae ($/kg) with Synechocystis sp., a: iron electrodes, original PH, I= 0.5 A, after 30 min of reaction time and 24 of settling time. b: Al electrodes, I= 0.5 A, after 30 min of reaction time and 24 h of settling time.*

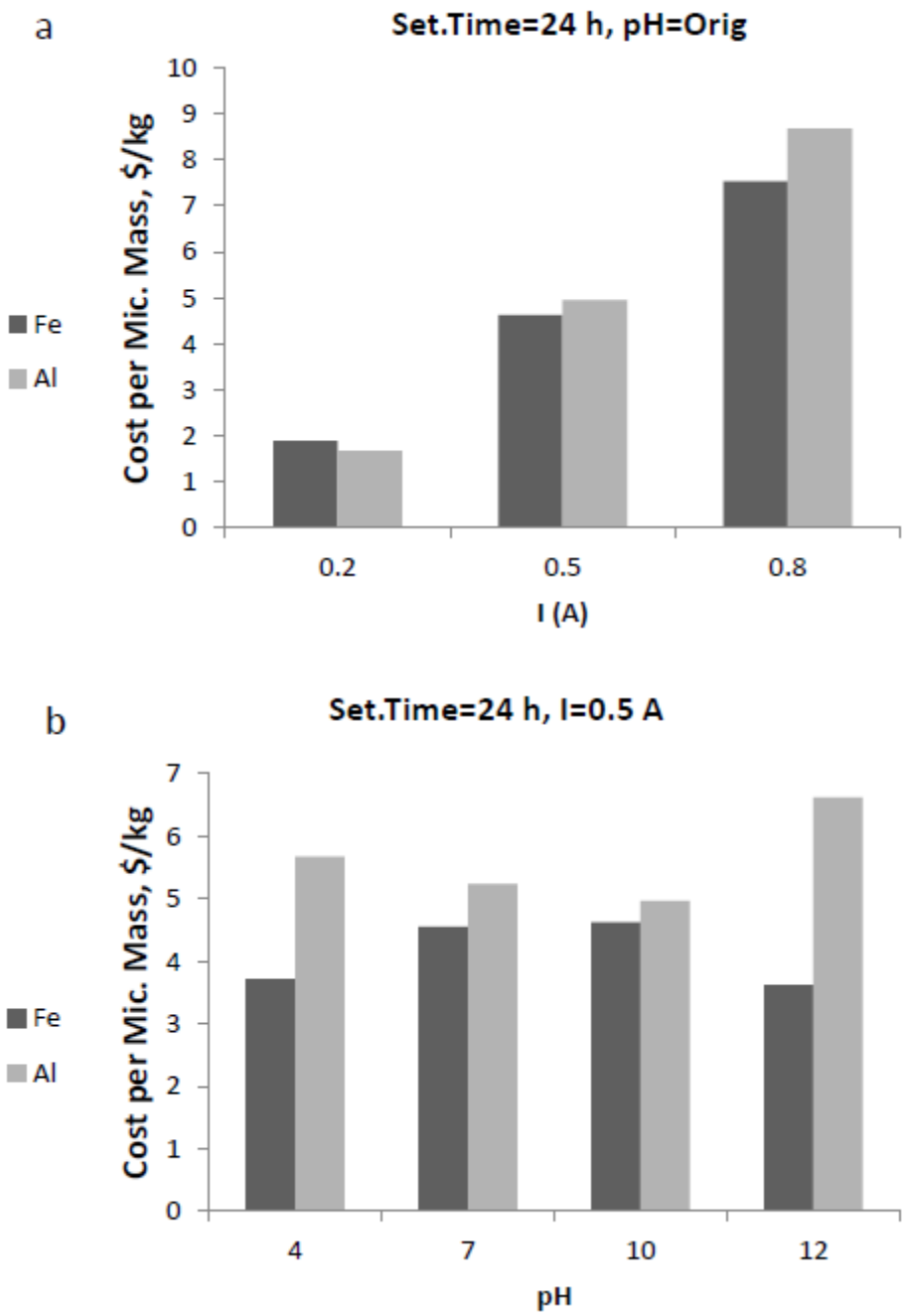

Figure 5.15. *Cost (Electrode + Electricity) per Kilogram of Obtained Microalgae ($/kg) with Synechocystis sp., a; Fe and Al electrodes with original pH, I= 0.2 A, 0.5 A and 0.8A after24 h of settling time. b: with original and adjusted pH values, Al and Fe electrodes, I= 0.5 A and after 24 h of settling time.*

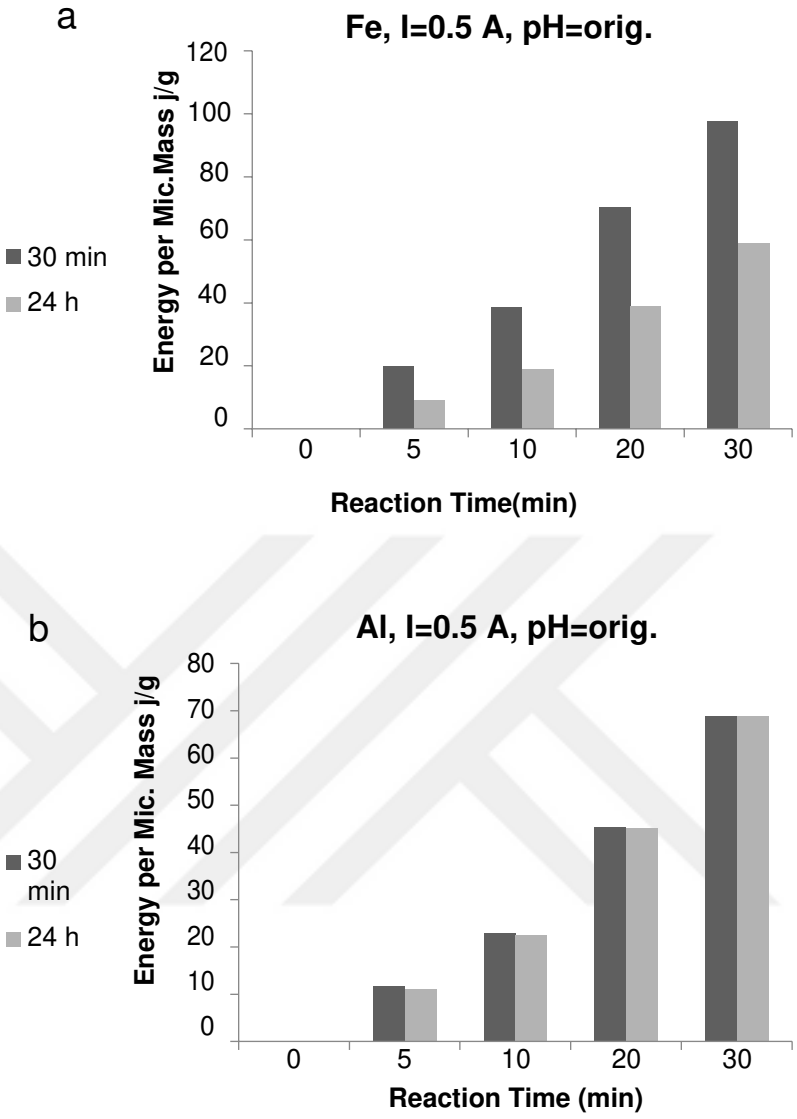

Figure 5.16. *Consumed energy per obtained grams of microalgae (j/g) with Chlorella Vulgaris sp., a: with Fe electrodes, I = 0.5 A, original pH, after 30 min of reaction time and 24 h settling time. b: with Al electrodes, original pH, I=0.5 A, after 30 min of reaction time and 24 h settling time.*

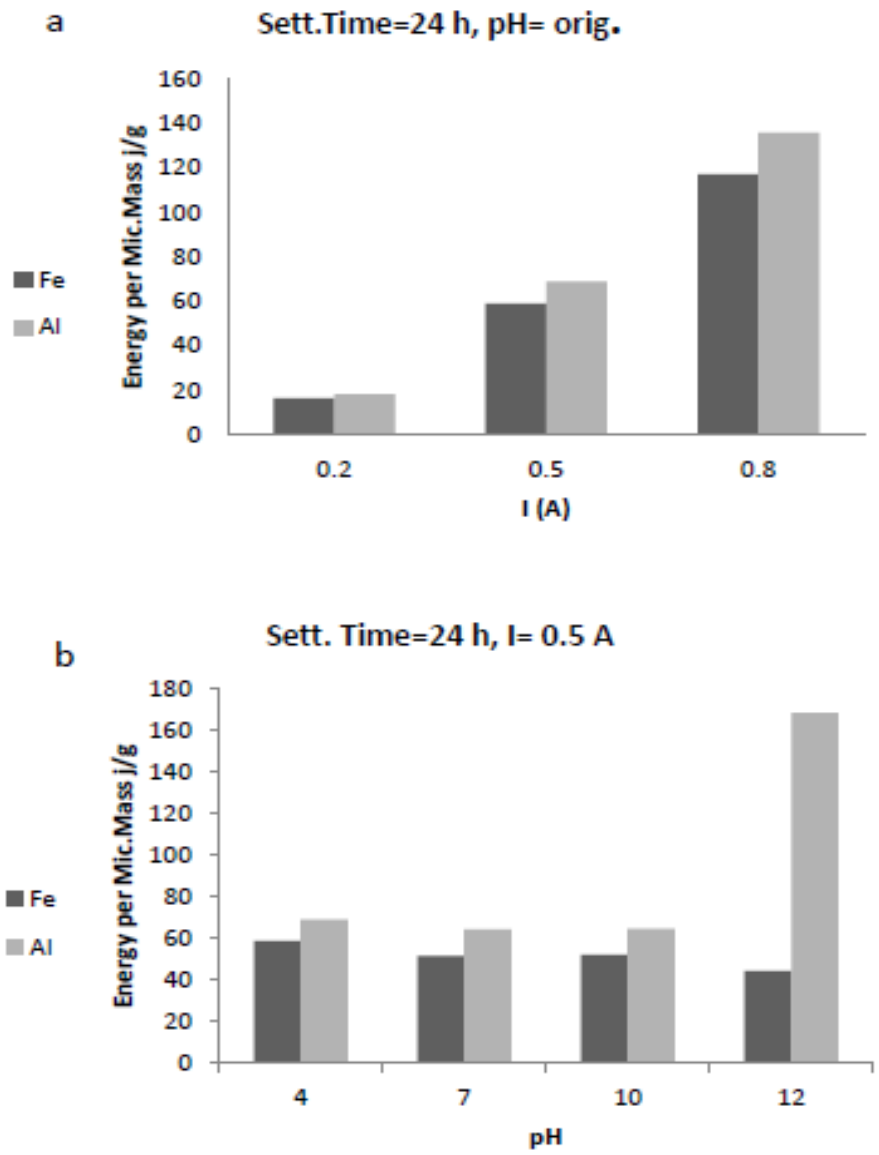

Figure 5.17. *Consumed energy per obtained grams of microalgae (j/g) with Chlorella Vulgaris sp.,a: original pH, Fe and Al electrodes, I= 0.2 A, 0. 5 A and 0.8 A, adjusted pH values after 24 h of settling time. b:with original and adjusted pH values, I= 0.5 A, Fe and Al electrodes and after 24 of settling time.*

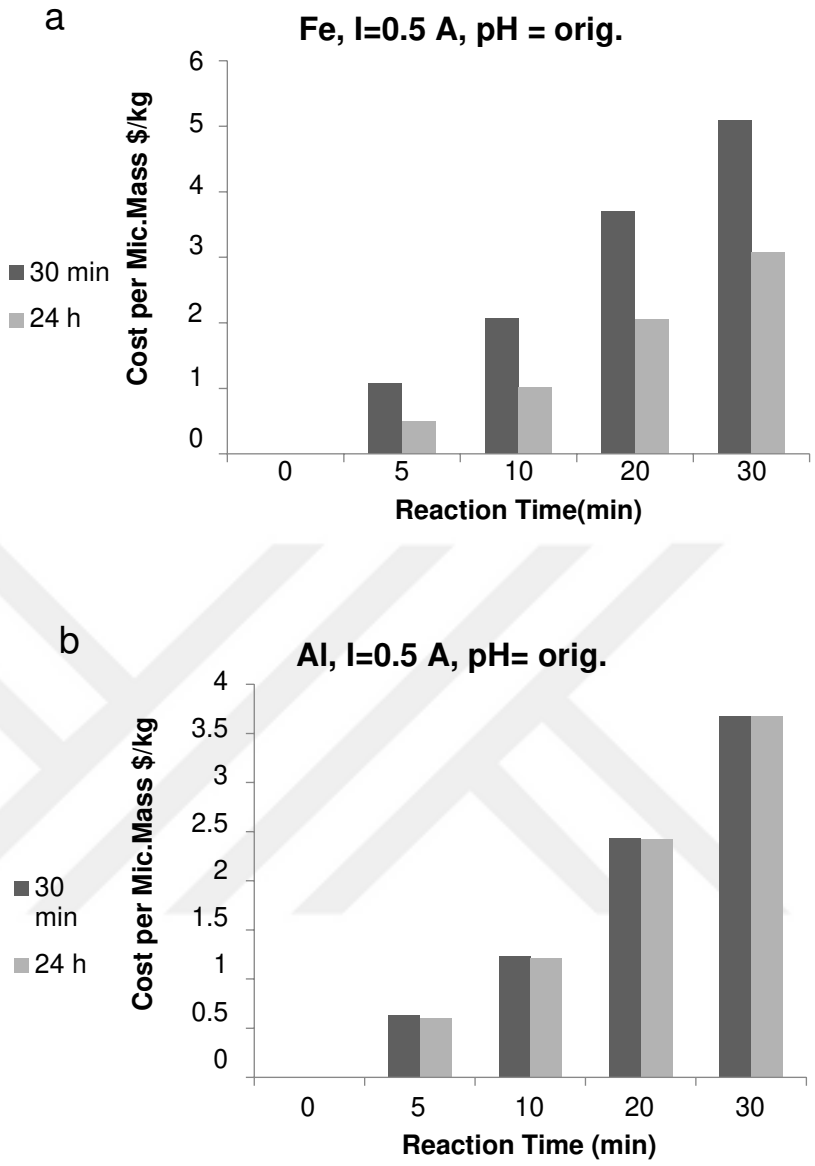

Figure 5.18. *Cost (Electrode + Electricity) per Kilogram of Obtained Microalgae ($/kg) with Chlorella Vulgaris sp., a:iron electrodes, original pH, I= 0.5 A, after 30 min of reaction time and 24 of settling time. b: Al electrodes, I= 0.5 A, original pH and after 30 min of reaction time and 24 h of settling time.*

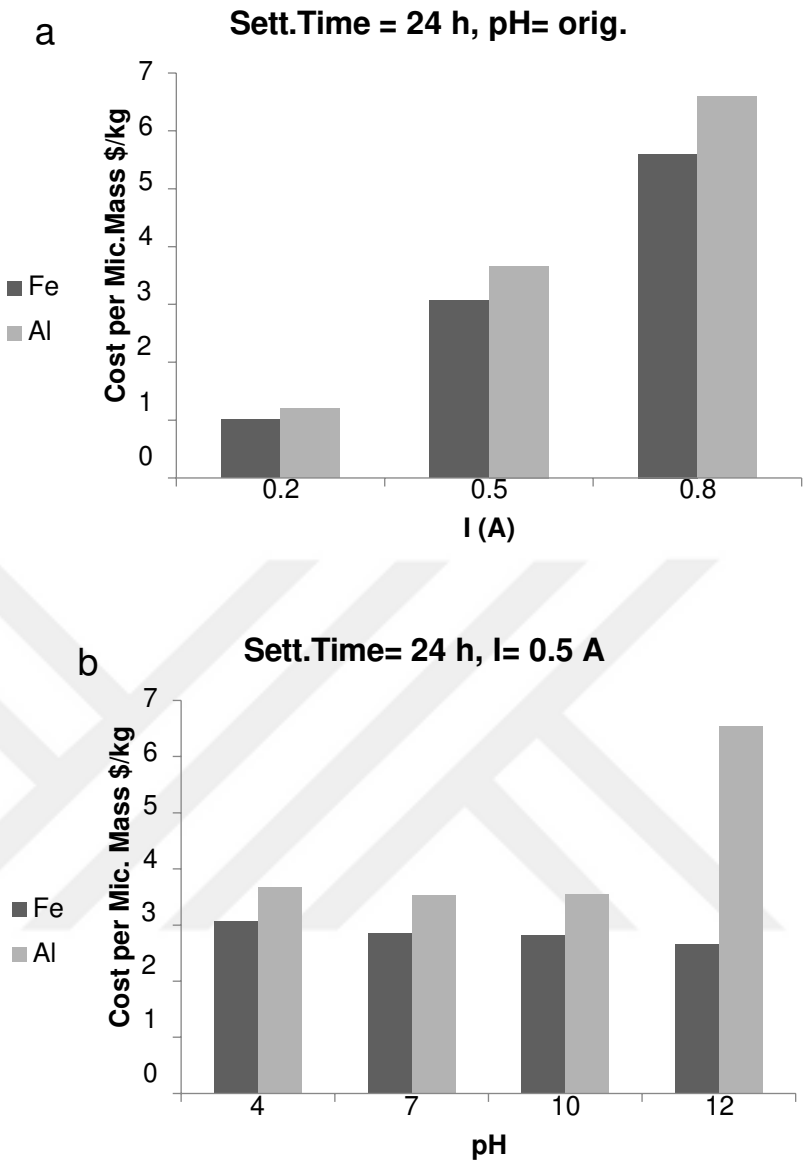

Figure 5.19. *Cost (Electrode + Electricity) per Kilogram of Obtained Microalgae ($/kg) with Chlorella Vulgaris sp, a; Fe and Al electrodes with original pH, I= 0.2 A, 0.5 A and 0.8A after 24 h of settling time. b: with original and adjusted pH values, Al and Fe electrodes, I= 0.5 A and after 24 h of settling time.*

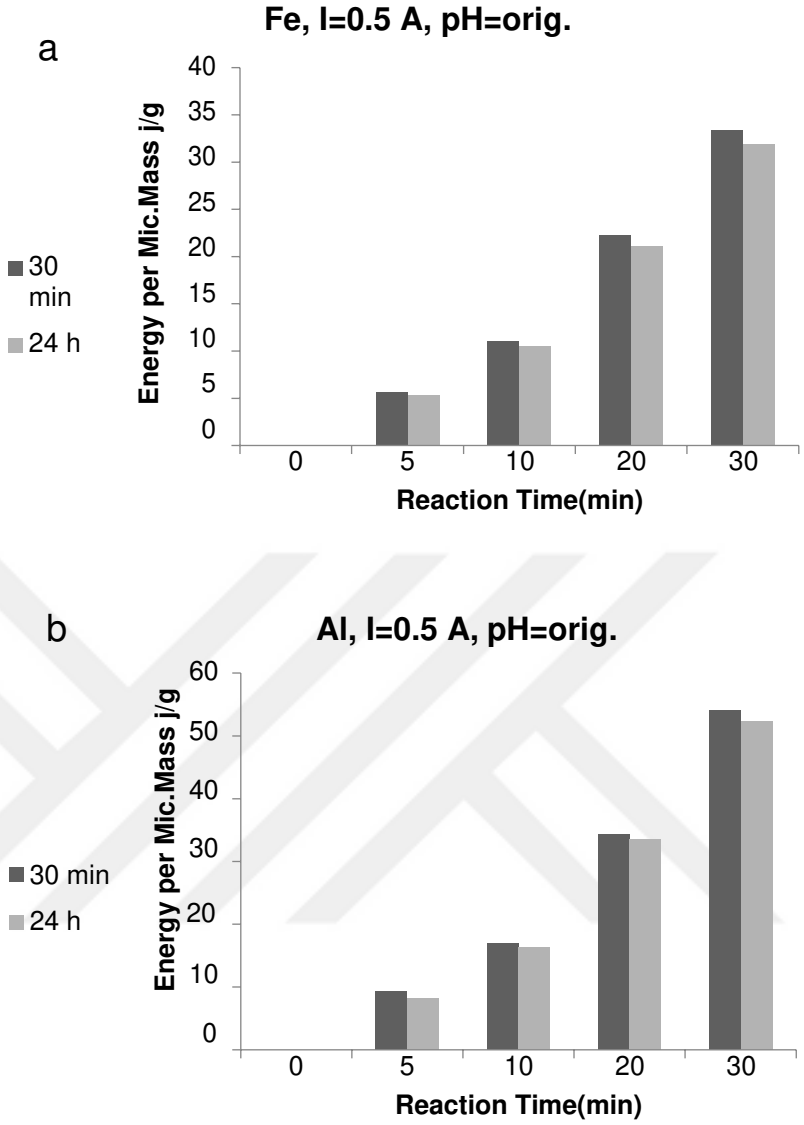

Figure 5.20. *Consumed energy per obtained grams of microalgae (j/g) with Scenedesmus Quadricauda sp.,a;with Fe electrodes, I = 0.5 A, original pH, after 30 min of reaction time and 24 h settling time. b: with Al electrodes, original pH, I=0.5 A, after 30 min of reaction time and 24 h of settling time.*

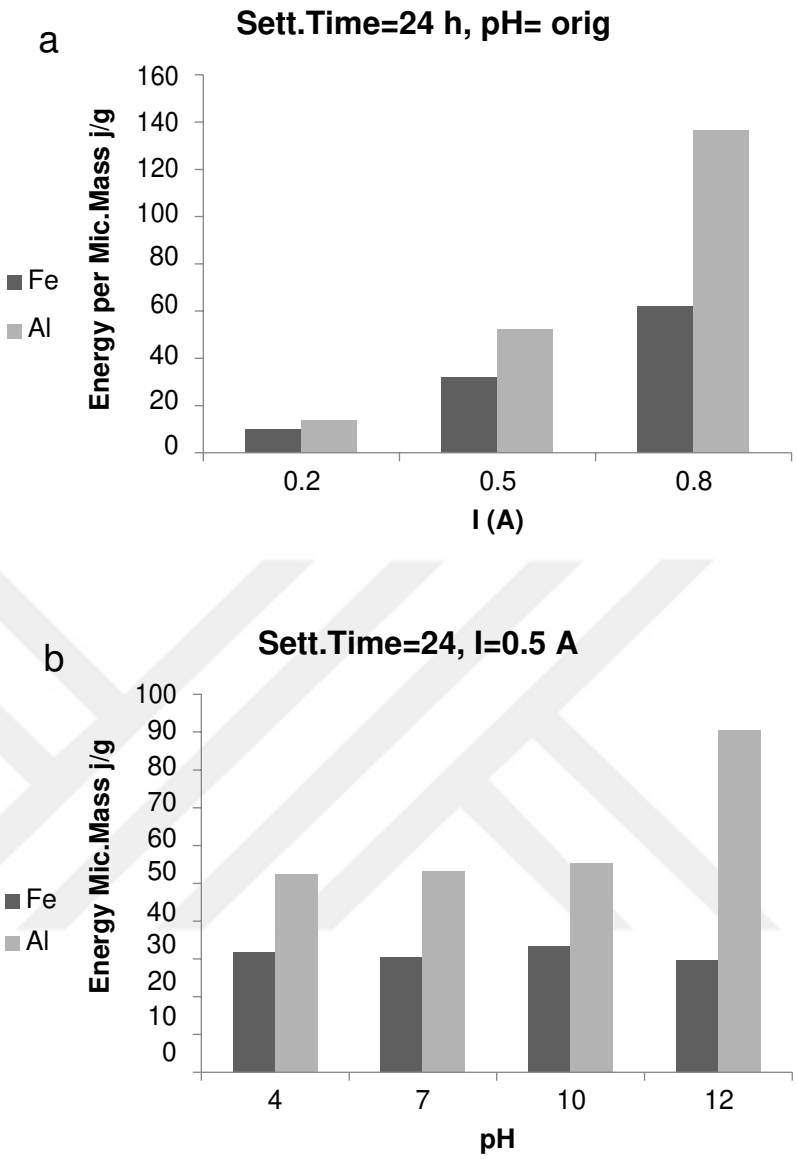

Figure 5.21. *Consumed energy per obtained grams of microalgae (j/g) with Scenedesmus Quadricauda sp.,a; original pH and I= 0.2 A, 0.5 A and 0.8A, Fe and Al electrodes, after 24 of settling time. b: with adjusted and original pH values, I= 0.5 A, Fe and Al electrodes and after 24 h of settling time.*

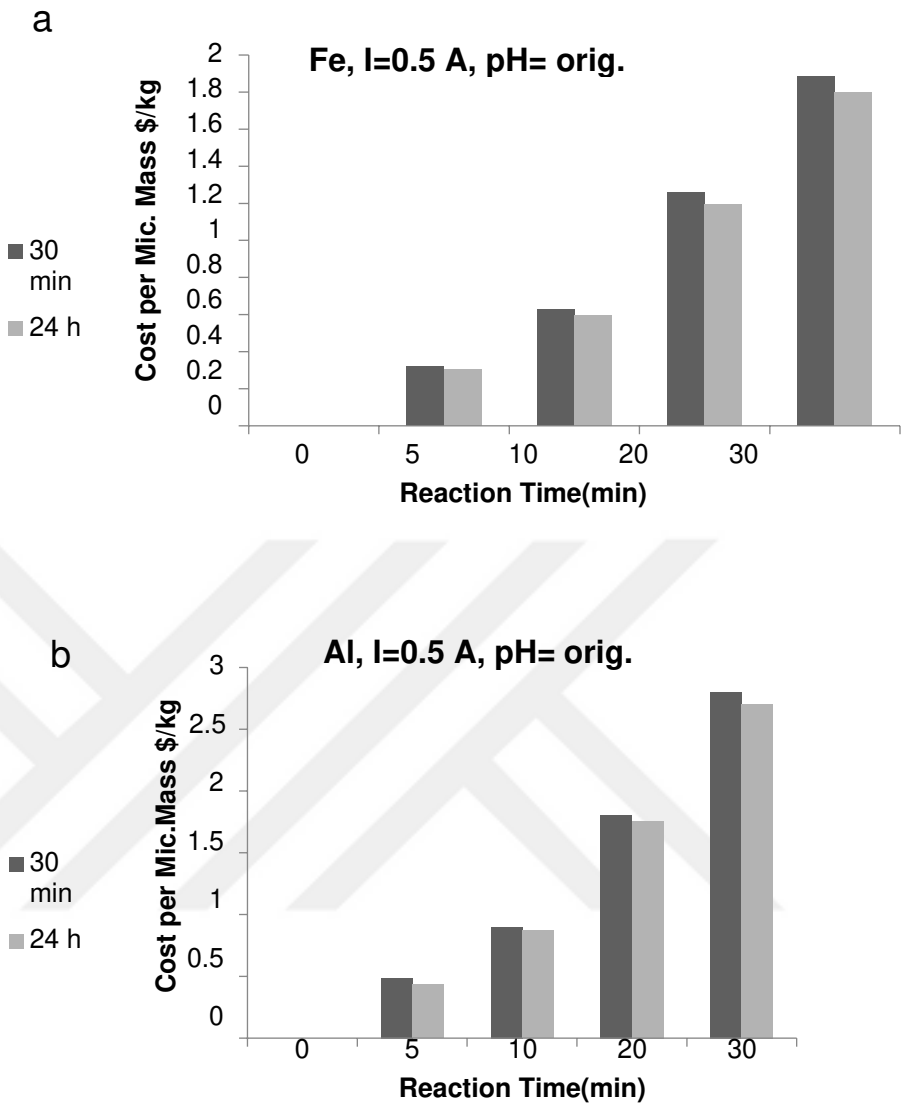

Figure 5.22. *Cost (Electrode + Electricity) per Kilogram of Obtained Microalgae ($/kg) with Scenedesmus Quadricauda sp, a:iron electrodes, original pH, I= 0.5 A, after 30 min of reaction time and 24 of settling time. b: Al electrodes, original pH, I = 0.5 A, after 30 min of reaction time and 24 h of settling time.*

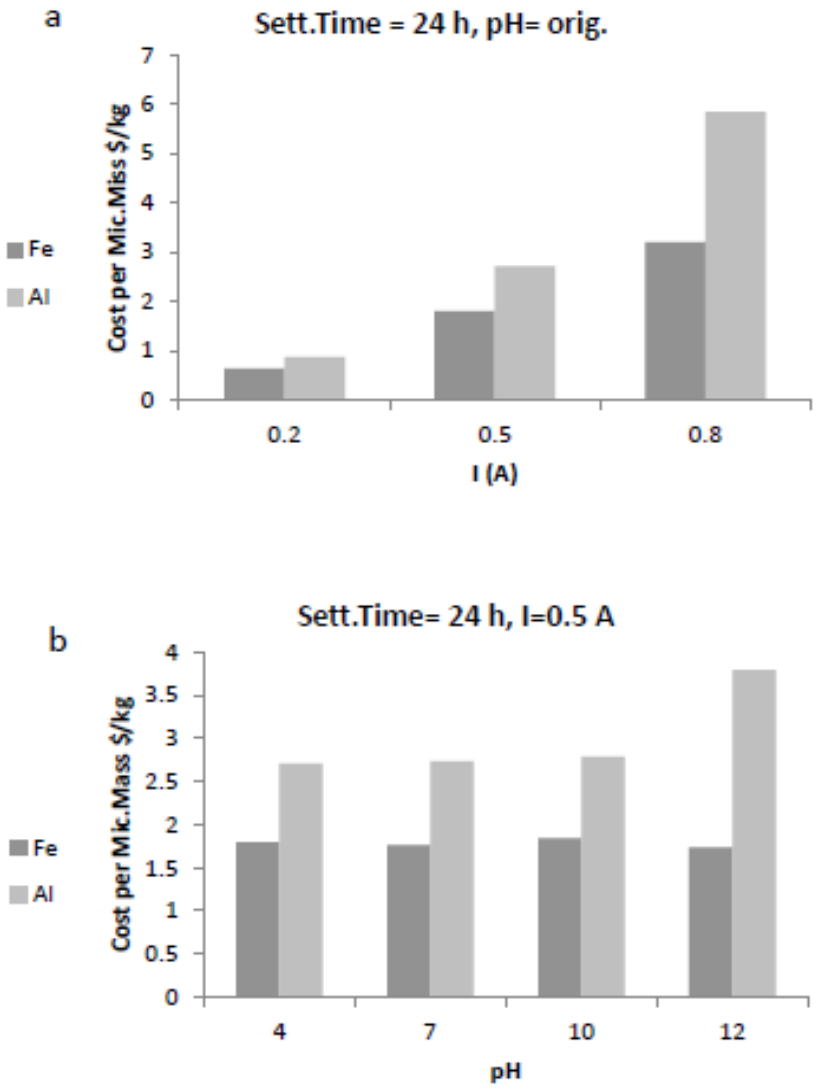

Figure 5.23. *Cost (Electrode + Electricity) per Kilogram of Obtained Microalgae ($/kg) with Scenedesmus Quadricauda sp, a; Fe and Al electrodes with original pH, I= 0.2 A, 0.5 A and 0.8A after 24 h of settling time. b: with original and adjusted pH values, Al and Fe electrodes, I= 0.5 A and after 24 h of settling time.*

5.5. The Chemical Coagulation Experiment

Microalgae have a negative cell surface charge and addition of the chemical coagulants will reduce or neutralize the charge. Dissolution of the salt in the solution release its cations ($Al^{3+,}$ Fe^{3+},...) which lower electrostatic repulsive force between the cells and can form big floc (Mathimani & Mallick, 2020). The higher efficiency of the CC experiment is related to the higher activity of the released cations and the efficacy of the cations determined by their electronegativity (Papazi et al., 2010). Al and Fe have higher electronegativity than the Ca, Mg and NH_4 so Al and Fe are considered as efficient chemical coagulants (Papazi et al., 2010). In this study, time, pH and coagulant dosage were investigated as the important parameters affecting the efficiency of the chemical coagulation. The present study of the CC experiments was applied for *Synechocystis sp., Chlorella Vulgaris sp.* and *Scenedesmus Quadricauda sp.* with the coagulants $FeSO_4$ and $Al_2(SO_4)_3.18H_2O$ including all the experiment time runs. The obtained results are shown in Tables 5.5, 5.6 and 5.7.

5.5.1. The effect of the pH

The pH plays a prominent role in coagulation efficiency as it promotes cell gathering (Al et al., 2015). The recovery efficiency with $FeSO_4$ at the original pH of the culture media was surprisingly high and achieved 92% after 24 h with *Synechocystis sp.* and *Scenedesmus Quadricauda sp.* The adjusted pH 4 value had a good effect in RE% for all species, while the RE with *Chlorella Vulgaris sp.* was lower than the other species. This is because of the small size of *Chlorella Vulgaris sp.* (5-10 μm) which could be flocculated less efficiently by the pH decreased induced flocculation process. The microalgae cells with small size have a good stability in the growth medium and hard to flocculate (Liu et al., 2014). Figure 5.24 (a, b and c) shows the chemical coagulant $FeSO_4$ with Chlorella Vulgaris sp. at initial pH after 30 min A:1.14 mL, B:2.82, C:4.5 that achieved high RE values.

In the pH 7 RE was high after 30 min and 24 h, but has a low effect with *Chlorella Vulgaris sp.* As well as pH 12 which had a significant effect with the coagulant in increasing the RE after 30 min and it can be explained that basic pH values were more efficient with coagulation as was also proposed by (Luis & Maceiras, 2017). Additionally, since microalgae cells are negatively charged, their association is difficult due to the repulsive forces. When adding sodium hydroxide to the medium to raise the pH to the

value 12, a huge amount of the positive charges is obtainable and thus a neutralization of the charges occurs (Luis & Maceiras, 2017). Therefore, microalgae cells are close together and aggregate into big flocks due to its weight which tend to settle down at the bottom of the container (Liu et al., 2014).

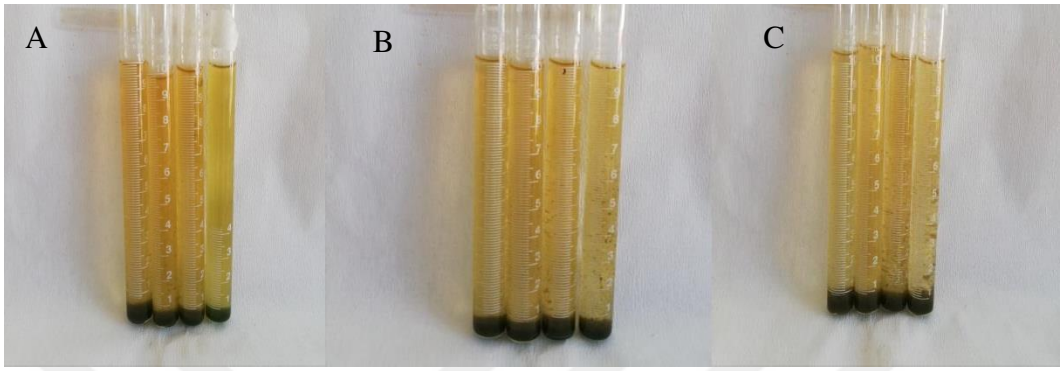

Figure 5.24. *Chemical coagulant $FeSO_4$ with Chlorella Vulgaris sp. at initial pH after 30 min A:1.14 mL, B:2.82, C:4.5*

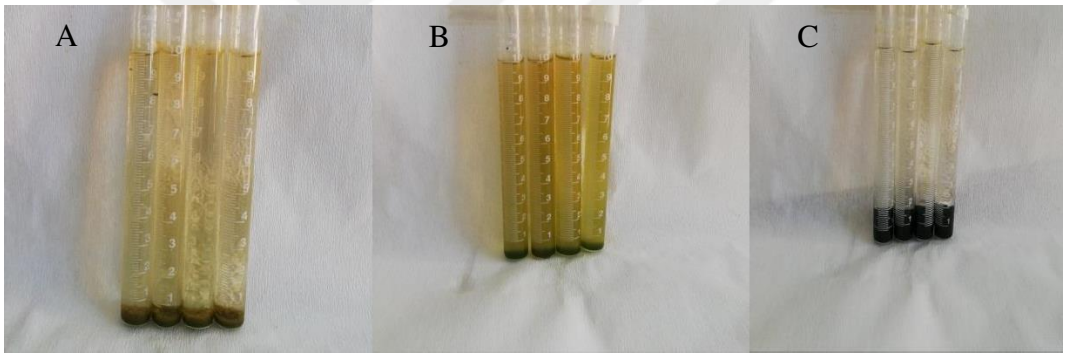

Figure 5.25. *Chemical coagulant $FeSO_4$ with Chlorella Vulgaris sp. after 30 min with adjusted pH A:pH 4 , B:pH 7, C:pH 12*

Adjusted pH value 7 had lower effect compared to the adjusted pH 4 and 12 values in the three species. Liu et al., (2013) found that when the pH value was 4 the microalgae cells coagulated more and settled in few minutes and the maximum efficiency rate reached at pH 4 and then reached a plateau. Figure 5.25 (a, b and c) shows the chemical coagulant $FeSO_4$ with *Chlorella Vulgaris sp*. after 30 min with adjusted pH A: pH 4, B: pH 7, C: pH 12 that pH 12 showed the maximum RE in 30 min of reaction time. This can be explained in the pH (5-1.5) where the zeta potential of the culture increased and was near to 0 mv (isoelectric point) that corresponding to the maximum efficiencies of flocculation (Liu et al., 2014). When the pH of the medium was set around 7 both $Al^{3+}$and Fe^{3+} have limited solubility due to deposit of an amorphous hydroxide, which could have a role in coagulation and flocculation (Duan & Gregory, 2003). When adding

ferric salts, the color of the medium turned from green to yellow- brown and influenced the microalgae pigments.

5.5.2. The effect of the coagulant dosage and the time

The relationship between microalgae concentration and coagulant dosage is not clear but it seems linear (Papazi et al., 2010). The RE was increased with increasing coagulant dosage and run time (Figures 5.7, 5.8 and 5.9). Higher dosage of coagulant led to charge neutralization and destabilization of the microlagae cells (Duan & Gregory, 2003). *Scenedesmus Quadricauda sp.* had a higher RE either after 30 min and 24 h settling times compared to the other species. The natural sedimentation rate of each microalgae species had a significant effect to the RE and settling time after the experiments. For more maximum efficient rate, the CC experiment could need more settling time after sampling. Sanyano et al., (2013) tested the effect of settling time on microalgae RE by aluminum sulphate and ferric chloride and the greatest RE (100) was within 60 min and 40 min by aluminum sulphate and ferric chloride, respectively.

$Al_2(SO_4).18H_2O$ has a higher recovery efficiency in a shorter time compared to $FeSO_4$. This could be explained by the formed cation's molecular weight and the charge where, Al ion's charge density is higher than Fe ions, which led to expanded molecular formation and gathering of the cells, hence improving charge neutralization of microalgae cells (Papazi et al., 2010). Aluminum salts have lower molecular weight than ferric salts and were better as coagulant, despite the charge in the ferric and aluminum salts being equal, the higher molecular weight of the ferric salts reduced their solubility, hence caused a reduction of their efficiency as a coagulant (Papazi et al., 2010). Figure 5.26 (a, b, c, d, e and f) shows the chemical coagulant $Al_2(SO_4)_3.18H_2O$ with *Synechocystis sp.*at adjusted pH a: pH 4 after 30 min, b: pH:7 after 30 min, c: pH 12 after 30 min, d: pH 4 after 24 h, e: pH 7 after 24 h, f: pH 12 after 24 h. It can be concluded that at pH 12 maximum RE values in 30 min of reaction time was achieved and at pH 4 and 7 maximum values were achieved after 24 h of settling time. Furthermore, the coagulant $Al_2(SO_4)_3.18H_2O$ showed the need of the settling time after reaction time 30 min to reach maximum RE values.

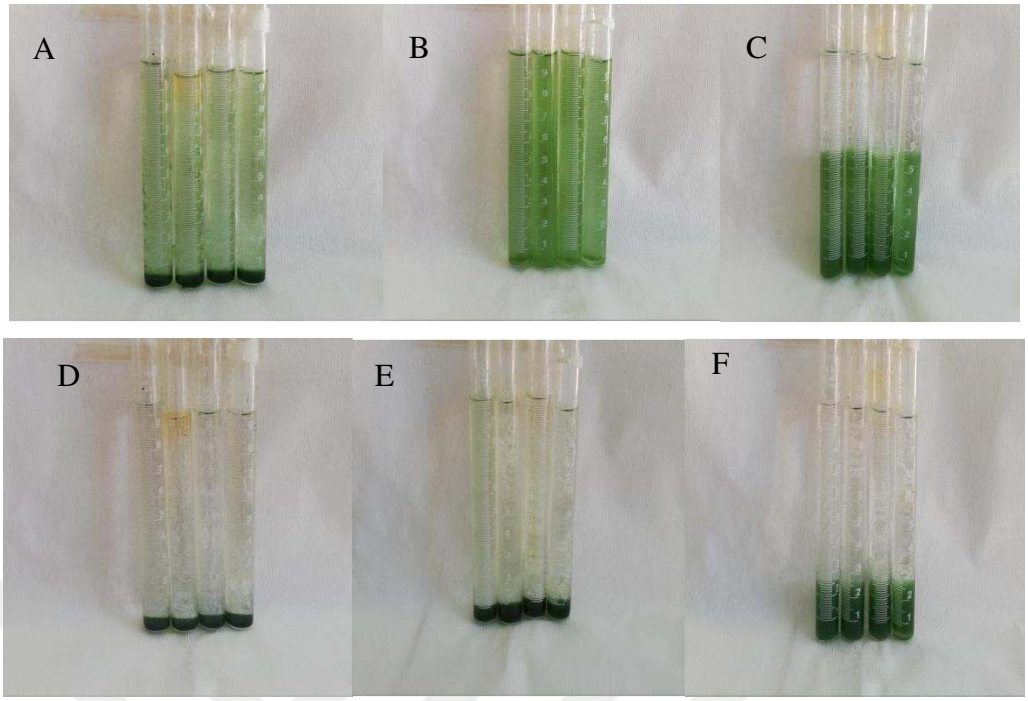

Figure 5.26. *Chemical coagulant $Al_2(SO_4)_3.18H_2O$ with Synechocystis sp.at adjusted pH A:PH 4 after 30 min , B: pH:7 after 30 min ,C: pH 12 after 30 min, D: pH 4 after 24h, E: pH 7 after 24 h, F: pH 12 after 24 h.*

5.5.3. The operating cost of the chemical coagulation experiment

The operating cost of the chemical coagulation depend mainly on the cost of the chemicals used and a higher dosage of coagulant can result in higher cost (Al et al., 2015). Generally, the CC experiment is less expensive than other harvesting methods such as centrifuge and electrocoagulation which require intensive energy (Milledge & Heaven, 2013). In this research, the cost of the CC experiment was calculated for $FeSO_4$ and $Al_2(SO_4).18H_2O$. The estimation of the total operating cost of the CC experiment was 0.0205 US$/ kg for dried microalgae of $FeSO_4$ coagulant and 0.1013 US$/ kg for $Al_2(SO_4).18H_2O$. It was found that the coagulant $Al_2(SO_4).18H_2O$ is costlier than $FeSO_4$, but more efficient in harvesting than $FeSO_4$.

Table 5.5. *The CC experiment time runs of Synechocystis sp.*

Time min	pH	OD 30min	OD 24h	RE 30min	RE 24h	Mic. Mass g 30 min	Mic. Mass g 24 h
Run 1 1.9×10^{-3}M $FeSO_4$							
0	10.23	1.002	-	-	-	-	-
2	7.35	0.12	0.006	88.024	99.401	0.054	0.061
5	6.97	0.117	0.006	88.323	99.401	0.055	0.061
10	6.72	0.091	0.002	90.918	99.800	0.056	0.062
20	6.65	0.084	0	91.617	100	0.057	0.062
Run 2 4.7×10^{-3} M $FeSO_4$							
0	5.54	1.002	-	-	-	-	-
2	5.65	0.114	0.019	88.623	98.104	0.055	0.061
5	5.76	0.092	0.008	90.818	99.202	0.056	0.061
10	5.77	0.099	0.007	90.120	99.301	0.056	0.061
20	5.73	0.117	0.006	88.323	99.401	0.055	0.061
Run 3 7.5×10^{-3} $FeSO_4$							
0	5.54	1.002	-	-	-	-	-
2	5.68	0.141	0.015	85.928	98.503	0.053	0.061
5	5.62	0.205	0.01	79.541	99.002	0.049	0.061
10	5.59	0.186	0.011	81.437	98.902	0.050	0.061
20	5.53	0.21	0.007	79.042	99.301	0.049	0.061
Run 4 4.7×10^{-3} M $FeSO_4$ pH 4							
0	3.76	1.002	-	-	-	-	-
2	3.84	0.547	0.005	45.409	99.501	0.028	0.061
5	3.72	0.491	0	50.998	100	0.031	0.061
10	3.75	0.542	0.001	45.908	99.900	0.028	0.061
20	3.8	0.422	0	57.884	100	0.036	0.062
Run 5 4.7×10^{-3} M $FeSO_4$ pH 7							
0	4.33	1.002	-	-	-	-	-
2	4.37	0.392	0.002	60.878	99.800	0.038	0.062
5	4.28	0.356	0.001	64.471	99.900	0.040	0.062
10	4.25	0.356	0	64.471	100	0.040	0.062
20	4.23	0.392	0.001	60.878	99.900	0.038	0.062
Run 6 4.7×10^{-3} M $FeSO_4$ pH 12							
0	8.74	1.002	-	-	-	-	-
2	7.96	0.062	0.022	93.812	97.804	0.058	0.060
5	7.46	0.07	0.045	93.014	95.509	0.057	0.059
10	6.9	0.116	0.035	88.423	96.507	0.055	0.060
20	6.83	0.06	0.023	94.012	97.705	0.058	0.060

Table 5.5 (cont.) *CC experiment time runs of Synechocystis sp*

Run 7 **1.23×10^{-3}M $Al_2(SO_4)_3.18H_2O$**							
0	4.57	1.002	-	-	-	-	-
2	4.61	0.961	0.497	4.092	50.399	0.003	0.031
5	4.61	0.927	0.488	7.485	51.297	0.005	0.032
10	4.61	0.921	0.443	8.084	55.788	0.005	0.034
20	4.59	0.914	0.424	8.782	57.685	0.005	0.036
Run 8 **3×10^{-3}M $Al_2(SO_4)_3.18H_2O$**							
0	4.89	1.002	-	-	-	-	-
2	4.74	0.89	0.212	11.178	78.842	0.007	0.049
5	4.72	0.925	0.226	7.685	77.445	0.005	0.048
10	4.64	0.924	0.268	7.784	73.253	0.005	0.045
20	4.64	0.914	0.282	8.782	71.856	0.005	0.044
Run 9 **5×10^{-3}M $Al_2(SO_4)_3.18H_2O$**							
0	4.55	1.002	-	-	-	-	-
2	4.43	0.964	0.073	3.792	92.715	0.002	0.057
5	4.41	0.892	0.072	10.978	92.814	0.007	0.057
10	4.4	0.917	0.079	8.483	92.116	0.005	0.057
20	4.4	0.901	0.073	10.080	92.715	0.006	0.057
Run 10 **3×10^{-3}M $Al_2(SO_4)_3.18H_2O$** **PH 4**							
0	3.86	1.002	-	-	-	-	-
2	3.9	0.781	0.036	22.056	96.407	0.014	0.060
5	3.9	0.777	0.034	22.455	96.607	0.014	0.060
10	3.89	0.768	0.032	23.353	96.806	0.014	0.060
20	3.88	0.772	0.043	22.954	95.709	0.014	0.0591
Run 11 **3×10^{-3}M $Al_2(SO_4)_3.18H_2O$** **pH 7**							
0	4.31	1.002	-	-	-	-	-
2	4.28	0.907	0.065	9.481	93.513	0.006	0.058
5	4.27	0.917	0.057	8.483	94.311	0.005	0.058
10	4.27	0.914	0.054	8.782	94.611	0.005	0.058
20	4.26	0.874	0.053	12.774	94.711	0.008	0.058
Run 12 **3×10^{-3}M $Al_2(SO_4)_3.18H_2O$** **pH 12**							
0	5.21	1.002	-	-	-	-	-
2	5.01	0.021	0.005	97.904	99.501	0.060	0.061
5	5	0.009	0	99.102	100	0.061	0.062
10	4.9	0.007	0	99.301	100	0.061	0.062
20	4.9	0.002	0	99.800	100	0.062	0.062

Table 5.6. *The CC experiment time runs of Chlorella Vulgaris sp.*

Time min	pH	OD 30min	OD 24h	RE 30min	RE 24h	Mic. Mass g 30 min	Mic. Mass g 24h
				Run1 1.9×10^{-3}M FeSO4			
0	7.16	0.926	-	-	-	-	-
2	6.9	0.621	0.121	32.937	86.933	0.028	0.074
5	6.76	0.601	0.104	35.097	88.769	0.030	0.075
10	6.68	0.527	0.094	43.089	89.849	0.037	0.076
20	6.6	0.395	0.086	57.343	90.713	0.049	0.077
				Run 2 4.7×10^{-3} M FeSO4			
0	6.72	0.926	-	-	-	-	-
2	6.46	0.503	0.097	45.680	89.525	0.039	0.076
5	6.44	0.383	0.086	58.639	90.713	0.050	0.077
10	6.26	0.372	0.084	59.827	90.929	0.051	0.077
20	6.16	0.26	0.081	71.922	91.253	0.061	0.077
				Run 3 7.5×10^{-3} M FeSO4			
0	6.31	0.926	-	-	-	-	-
2	6.25	0.167	0.09	81.965	90.281	0.069	0.077
5	6.09	0.156	0.083	83.153	91.037	0.070	0.077
10	5.98	0.154	0.07	83.369	92.441	0.071	0.078
20	6	0.152	0.068	83.585	92.657	0.071	0.079
				Run 4 4.7×10^{-3} M FeSO4 pH 4			
0	3.69	0.926	-	-	-	-	-
2	3.68	0.686	0.015	25.918	98.380	0.022	0.083
5	3.65	0.697	0.049	24.730	94.708	0.021	0.080
10	3.65	0.694	0.029	25.054	96.868	0.021	0.082
20	3.67	0.699	0.033	24.514	96.436	0.021	0.082
				Run 5 4.7×10^{-3} M FeSO4 pH 7			
0	6.6	0.926	-	-	-	-	-
2	6.52	0.796	0.104	14.039	88.769	0.012	0.075
5	6.5	0.727	0.095	21.490	89.741	0.018	0.076
10	6.3	0.653	0.088	29.482	90.497	0.025	0.077
20	6.24	0.589	0.082	36.393	91.145	0.03	0.077
				Run 6 4.7×10^{-3} M FeSO4 pH 12			
0	10.69	0.926	-	-	-	-	-
2	10.44	0.019	0.001	97.948	99.892	0.083	0.085
5	10.49	0.007	0	99.244	100	0.084	0.085
10	10.36	0.003	0	99.676	100	0.084	0.085
20	10.45	0.002	0	99.784	100	0.085	0.085

Table 5.6 (cont.) *CC experiment time runs of Chlorella Vulgaris sp.*

				Run 7			
				1.23×10^{-3}M $Al_2(SO_4)_3.18H_2O$			
0	5.31	0.926	-	-	-	-	-
2	5.22	0.168	0.002	81.857	99.784	0.069	0.085
5	5.12	0.164	0.001	82.289	99.892	0.070	0.085
10	5.05	0.186	0.006	79.914	99.352	0.068	0.084
20	4.96	0.161	0.004	82.613	99.568	0.070	0.084
				Run 8			
				3×10^{-3}M $Al_2(SO_4)_3.18H_2O$			
0	4.75	0.926	-	-	-	-	-
2	4.73	0.268	0.007	71.058	99.244	0.060	0.084
5	4.68	0.263	0.003	71.598	99.676	0.061	0.084
10	4.63	0.15	0.001	83.801	99.892	0.071	0.085
20	4.55	0.175	0.009	81.102	99.028	0.069	0.084
				Run 9			
				5×10^{-3}M $Al_2(SO_4)_3.18H_2O$			
0	4.58	0.926	-	-	-	-	-
2	4.48	0.353	0.068	61.879	92.657	0.054	0.079
5	4.53	0.294	0.059	68.251	93.629	0.058	0.079
10	4.47	0.219	0.047	76.350	94.924	0.065	0.080
20	4.5	0.2	0.044	78.402	95.248	0.066	0.081
				Run 10			
				3×10^{-3}M $Al_2(SO_4)_3.18H_2O$			
				pH 4			
0	3.79	0.926	-	-	-	-	-
2	3.88	0.669	0.081	27.754	91.253	0.024	0.077
5	3.88	0.661	0.068	28.618	92.657	0.024	0.079
10	3.83	0.623	0.031	32.721	96.652	0.028	0.082
20	4	0.505	0.03	45.464	96.760	0.039	0.082
				Run 11			
				3×10^{-3}M $Al_2(SO_4)_3.18H_2O$			
				pH 7			
0	4.62	0.926	-	-	-	-	-
2	4.56	0.485	0.026	47.62	97.192	0.040	0.082
5	4.6	0.433	0.021	49.1	97.732	0.042	0.083
10	4.55	0.313	0.017	61.1	98.164	0.052	0.083
20	4.7	0.273	0.027	65.1	97.084	0.055	0.082
				Run 12			
				3×10^{-3}M $Al_2(SO_4)_3.18H_2O$			
				pH 12			
0	6	0.926	-	-	-	-	-
2	6.1	0.019	0	100	100	0.083	0.085
5	6.19	0.015	0	100	100	0.083	0.085
10	5.82	0	0	100	100	0.085	0.085
20	5.76	0	0	100	100	0.085	0.085

Table 5.7. *The CC experiment time runs for Scenedesmus Quadricauda sp.*

Time min	pH	OD 30m	OD 24h	RE 30min	RE 24h	Mic. Mass. g 30 min	Mic. Mass. g 24 h
Run 1 1.9×10^{-3}M $FeSO_4$ pH $_{initial}$ 9.90							
0	7.13	1.004	-	-	-	-	-
2	7	0.168	0.002	83.267	99.801	0.108	0.130
5	6.93	0.11	0.001	89.044	99.900	0.116	0.130
10	6.83	0.097	0	90.34	100	0.118	0.130
20	6.55	0.098	0	90.239	100	0.117	0.130
Run 2 4.7×10^{-3} M $FeSO_4$							
0	6.44	1.004	-	-	-	-	-
2	6.41	0.076	0.044	92.430	95.618	0.120	0.124
5	6.38	0.054	0.035	94.622	96.514	0.123	0.126
10	6.11	0.047	0.014	95.319	98.606	0.124	0.128
20	5.86	0.04	0.007	96.016	99.303	0.125	0.129
Run 3 7.5×10^{-3} $FeSO_4$							
0	6.24	1.004	-	-	-	-	-
2	6.18	0.04	0.029	96.016	97.112	0.125	0.126
5	6.15	0.03	0.026	97.012	97.410	0.126	0.127
10	5.89	0.034	0.006	96.614	99.402	0.126	0.129
20	5.8	0.053	0.016	94.721	98.406	0.123	0.128
Run 4 4.7×10^{-3} M $FeSO_4$ pH 4							
0	3.86	1.004	-	-	-	-	-
2	3.92	0.089	0.005	91.135	99.502	0.119	0.129
5	3.86	0.081	0.008	91.932	99.203	0.120	0.191
10	3.9	0.075	0.004	92.530	99.602	0.120	0.130
20	3.84	0.052	0	94.820	100	0.123	0.130
Run 5 4.7×10^{-3} M $FeSO_4$ pH 7							
0	6.18	1.004	-	-	-	-	-
2	6.12	0.055	0.014	94.522	98.606	0.123	0.128
5	6.03	0.031	0.012	96.912	98.805	0.126	0.129
10	5.89	0.024	0.01	97.610	99.004	0.127	0.129
20	5.65	0.044	0.018	95.617	98.207	0.124	0.128
Run 6 4.7×10^{-3} M $FeSO_4$ pH 12							
0	11.63	1.004	-	-	-	-	-
2	11.71	0.003	0.002	99.701	99.801	0.130	0.130
5	11.94	0.001	0	99.900	100	0.130	0.130
10	11.69	0	0	100	100	0.130	0.130
20	11.66	0	0	100	100	0.130	0.130
Run 7 0.74 ml $Al_2(SO_4)_3.18H_2O$							
0	5.16	1.004	-	-	-	-	-
2	5.09	0.06	0.003	94.023	99.701	0.122	0.130
5	5.04	0.056	0.001	94.422	99.900	0.123	0.130
10	4.87	0.054	0	94.622	100	0.123	0.130
20	4.83	0.043	0	95.717	100	0.125	0.130

Table 5.7 (cont.) *CC experiment time runs for Scenedesmus Quadricauda sp*
.

Run 8 **1.23×10^{-3}M $Al_2(SO_4)_3.18H_2O$**							
0	4.66	1.004	-	-	-	-	-
2	4.65	0.051	0.001	94.920	99.900	0.123	0.130
5	4.59	0.049	0	95.120	100	0.124	0.130
10	4.58	0.038	0	96.215	100	0.125	0.130
20	4.55	0.044	0	95.618	100	0.124	0.130
Run 9 **5×10^{-3}M $Al_2(SO_4)_3.18H_2O$**							
0	4.54	1.004	-	-	-	-	-
2	4.43	0.015	0	98.506	100	0.128	0.130
5	4.4	0.003	0	99.701	100	0.130	0.130
10	4.36	0.001	0	99.9004	100	0.130	0.130
20	4.28	0	0	100	100	0.130	0.130
Run 10 **3×10^{-3}M $Al_2(SO_4)_3.18H_2O$** **pH 4**							
0	3.85	1.004	-	-	-	-	-
2	3.91	0.04	0.004	96.01594	99.602	0.125	0.130
5	3.86	0.039	0.001	96.11554	99.900	0.125	0.130
10	3.91	0.036	0	96.41434	100	0.125	0.130
20	3.93	0.047	0.003	95.31873	99.701	0.1240	0.130
Run 11 **3×10^{-3}M $Al_2(SO_4)_3.18H_2O$** **pH 7**							
0	4.52	1.004	-	-	-	-	-
2	4.5	0.096	0.006	90.438	99.402	0.118	0.130
5	4.45	0.094	0.003	90.637	99.701	0.118	0.130
10	4.41	0.09	0.001	91.036	99.900	0.118	0.130
20	4.42	0.104	0.004	89.641	99.602	0.117	0.130
Run 12 **3×10^{-3}M $Al_2(SO_4)_3.18H_2O$** **pH 12**							
0	8.94	1.004	-	-	-	-	-
2	8.77	0.003	0	99.701	100	0.130	0.130
5	8.6	0.001	0	99.900	100	0.130	0.130
10	8.55	0	0	100	100	0.130	0.130
20	8.43	0	0	100	100	0.130	0.130

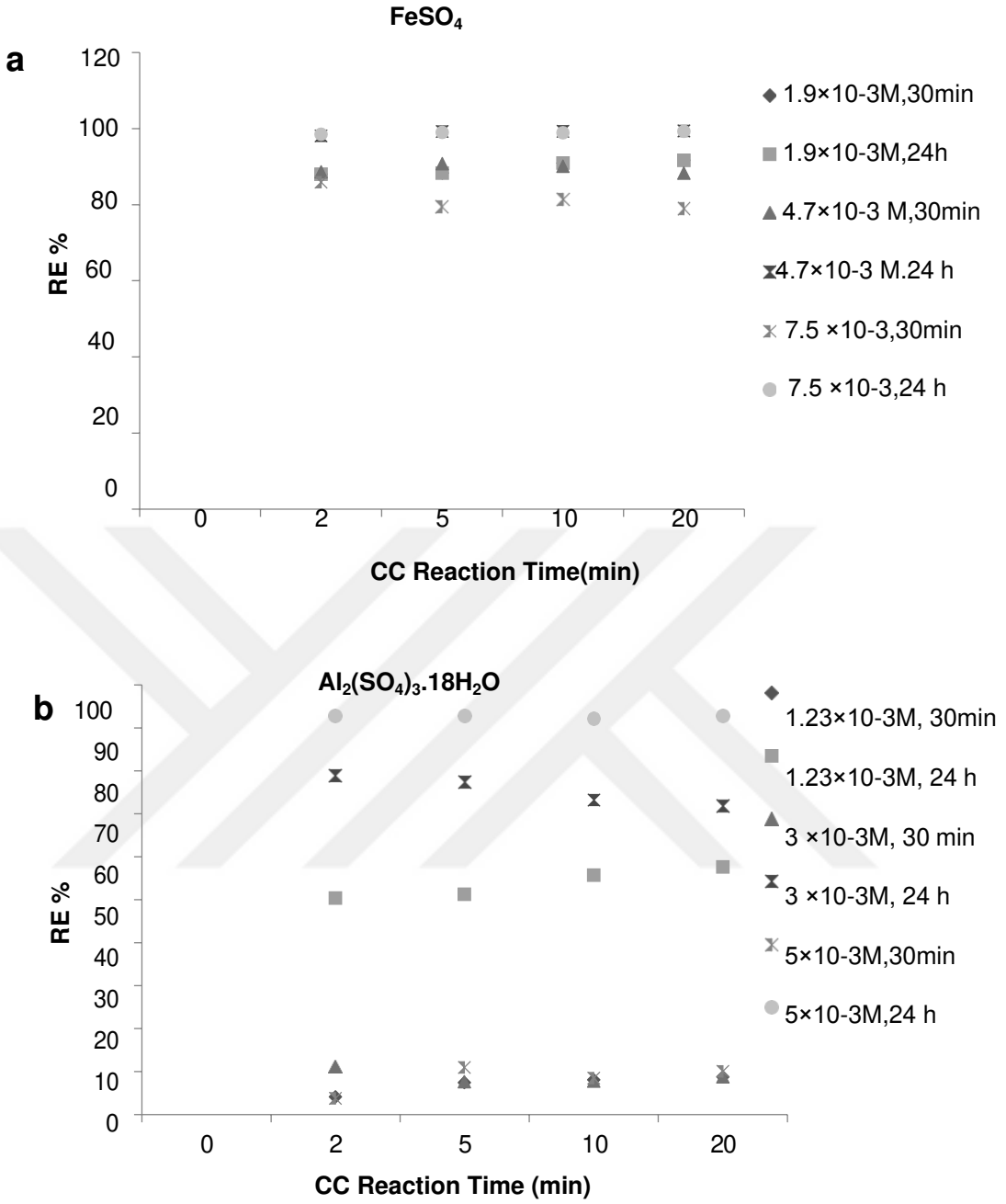

Figure 5.27. *RE of Synechocystis sp. with CC experiment operating times 30 min and settling time 24 h, (a and b) with initial pH.*

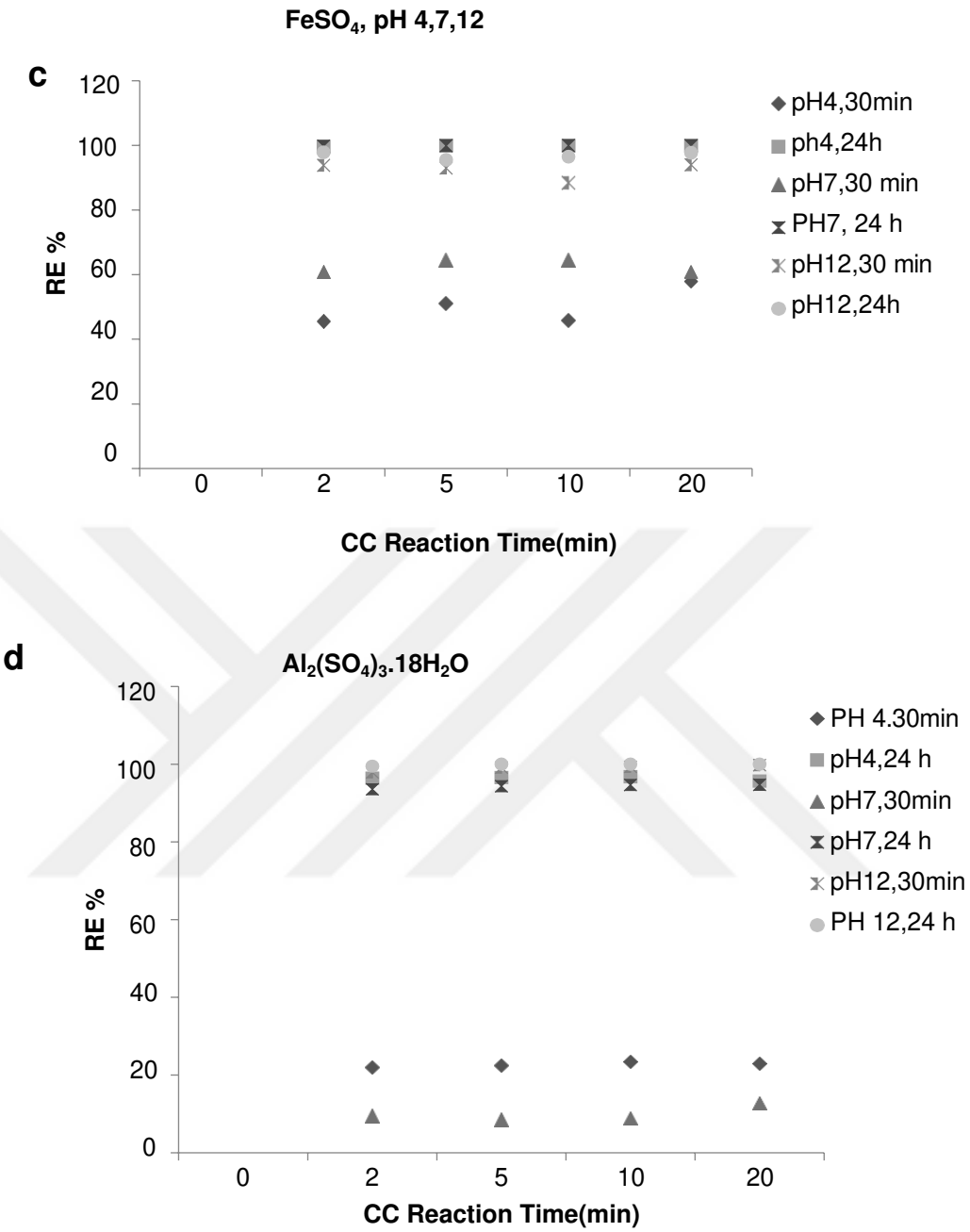

Figure 5.27 (cont.) *RE of Synechocystis sp. With CC experiment operating times 30 min and settling time 24 h, (a and b) with initial pH and (c and d) with adjusted pH (4, 7 and 12).*

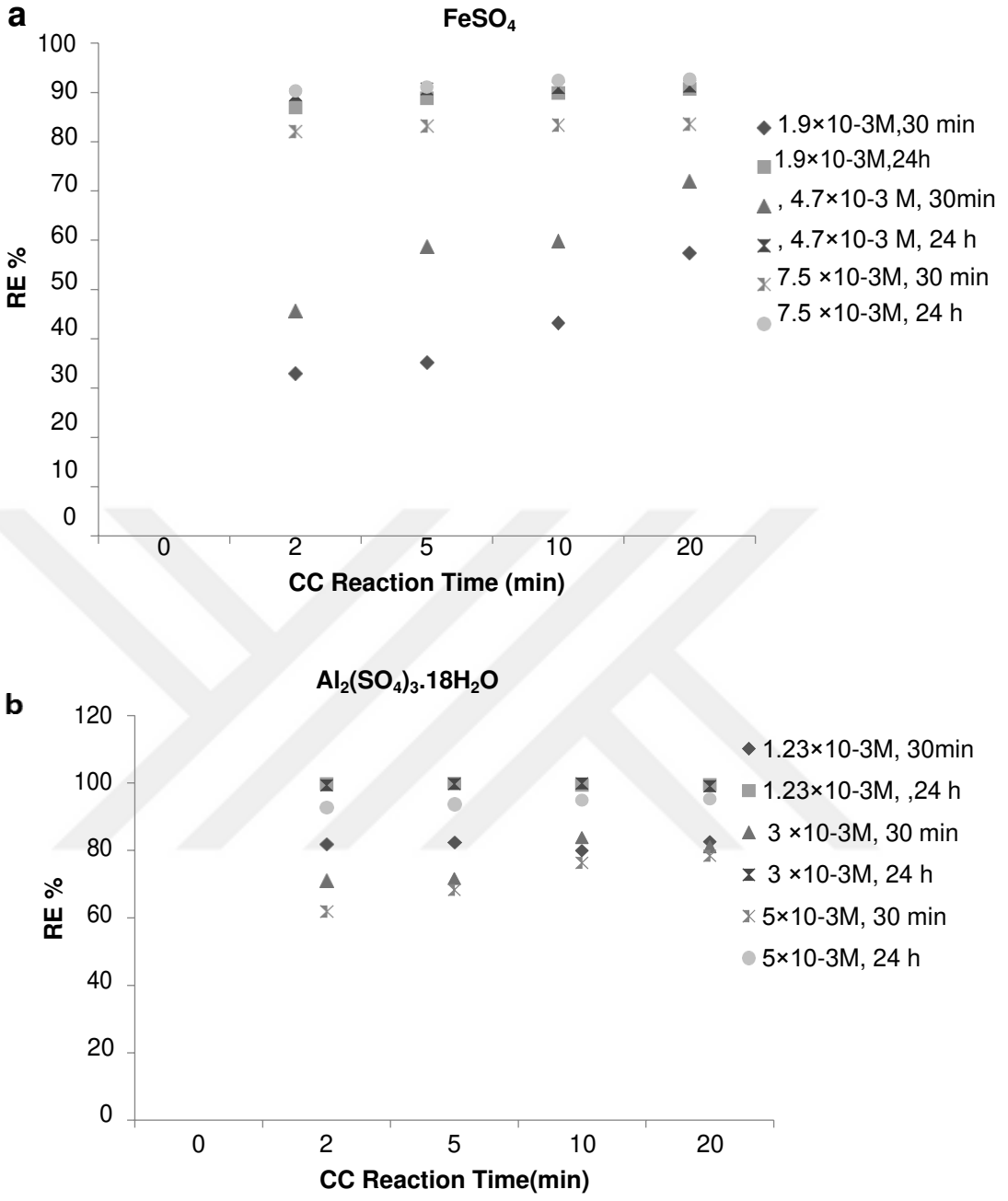

Figure 5.28. *RE of Chlorella Vulgaris sp. with CC experiment operating times30 min and settling time 24h, (a and b) with initial pH.*

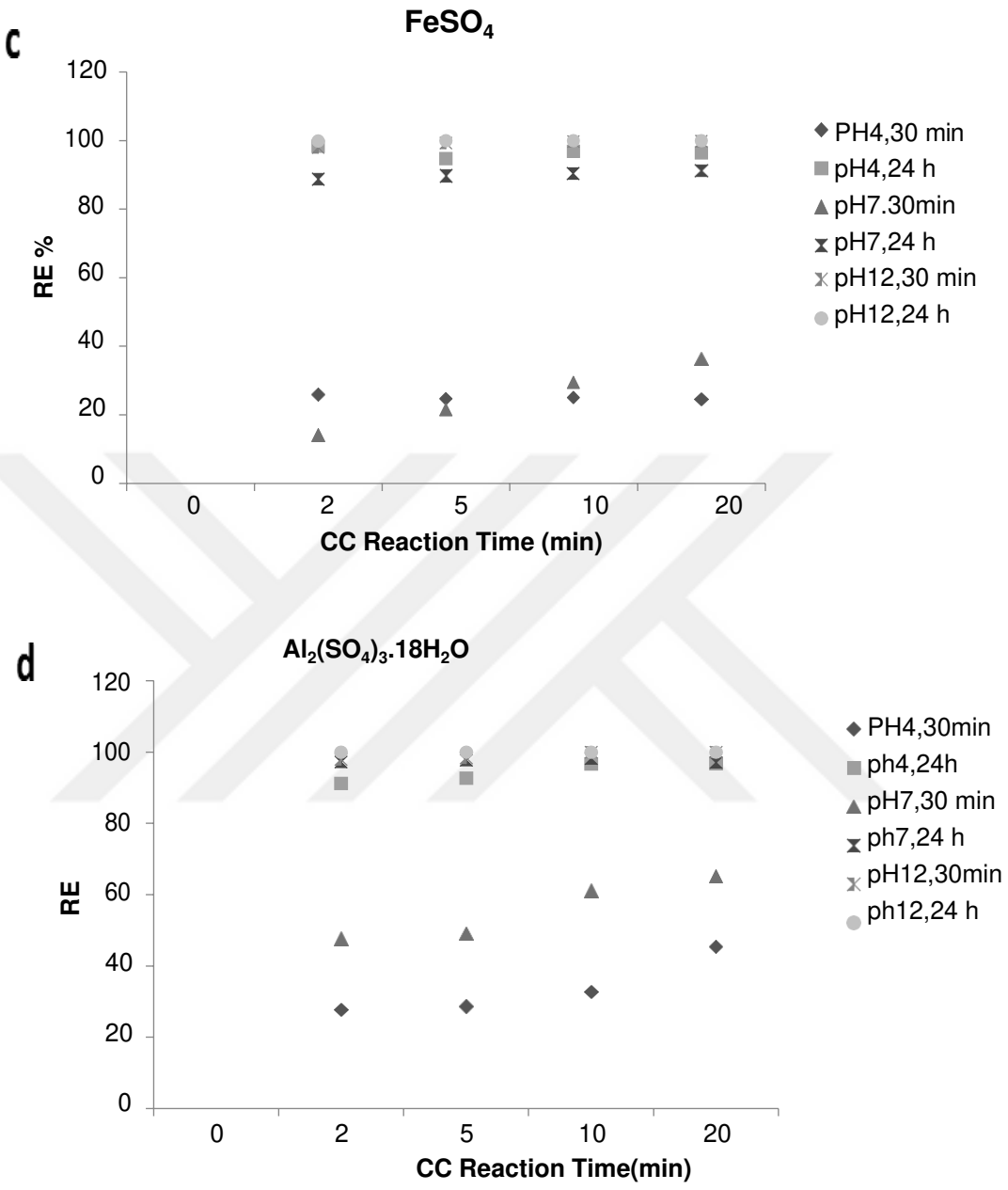

Figure 5.28. (cont.) *RE of Chlorella Vulgaris sp. with CC experiment operating times30 min and settling time 24h, (a and b) with initial pH and. (c and d) with adjusted pH (4, 7 and 12).*

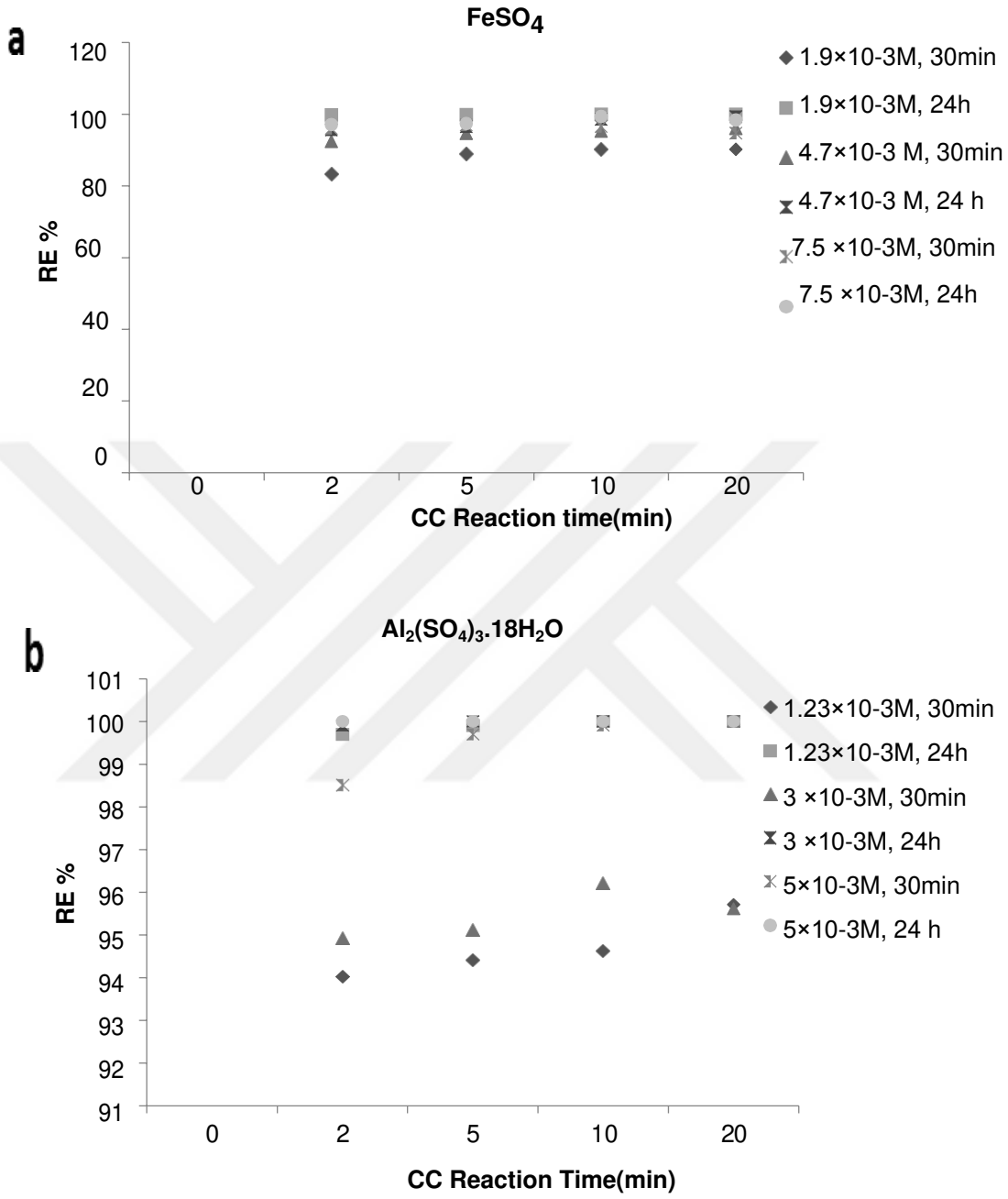

Figure 5.29. *RE of Scenedesmus Quadricauda sp. with CC experiment 30 min and 24 h settling time, (a and b) with initial pH.*

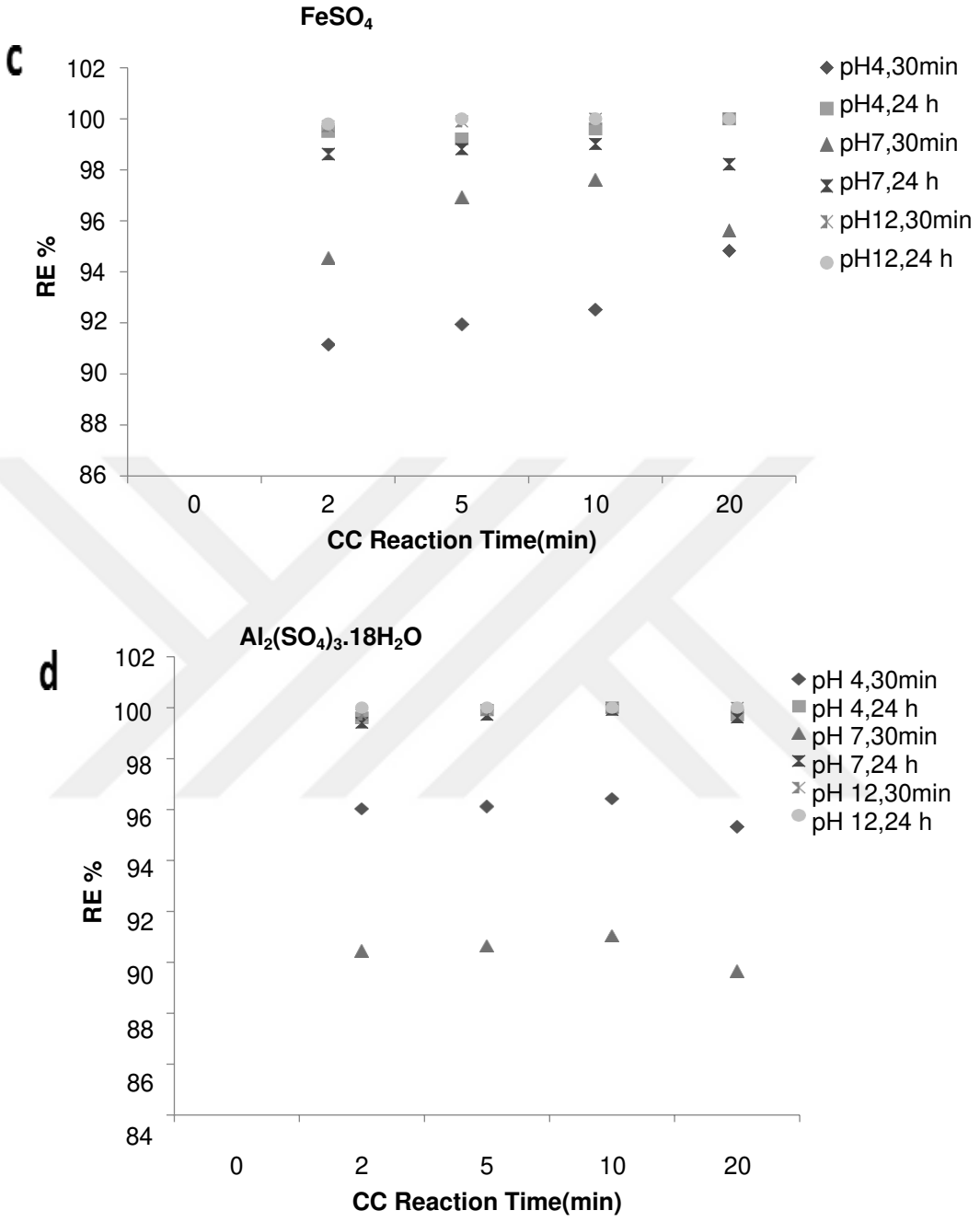

Figure 5.29. (cont.) *RE of Scenedesmus Quadricauda sp. with CC experiment 30 min and 24 h settling time, (a and b) with initial pH and (c and d) with adjusted PH (4, 7 and 12).*

6. CONCLUSION

Three different microalgae species *Synechocystis sp., Scenedesmus Quadricauda sp.* and *Chlorella Vulgaris sp.* were successfully harvested by electrocoagulation and coagulation and reached the maximum values after 30 min and 24 h. The achieved recovery efficiency of the three species was attained with significantly high initial pH values and adjusted values. It can be concluded that the adjusted pH on electrocoagulation was a successful method on harvesting microalgae and the recovery efficiency reached 100% with less energy consumption and costs. However, the cost of this method was slightly high because of the operating costs such as the energy consumption and the need of replacing the electrodes frequently. The chemical coagulation experiment was less energy consuming compared to the EC and achieved high recovery efficiency with low dosage of the coagulant in short time. However, the addition of the chemicals to the microalgae medium was contaminating the harvested biomass and this biomass may not be applicable to every commercial sector as for example in food and pharmaceutical products.

The researches carried out in the harvesting microalgae over last years have made a lot of progress. Researchers have optimized different harvesting methods which had become more energy efficient. There is a necessity to optimize the current methods to be less costly and environmentally friendly and develop a combination of more than one harvesting method that can reach the best harvesting efficiency with less energy consumption and cost. However, to choose the best harvesting method it depends on many factors including the cost, the energy efficiency, less contamination on the harvested biomass and on the environment. Intensive research works has to be conducted to fill the knowledge gap of the issues in each harvesting method to find out their advantages and disadvantages. Finally, most of the harvesting experiments were tested at laboratory level and studies must be carried out at pilot scale to check their compatibility and estimate the full costs.

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