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Department of Bioresource Engineering

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ENGINEERING DESIGN 3



Design 3 Final Report

Design of a Small Scale Algae Cultivation System to Produce Biodiesel

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Executive Summary

Biodiesel can be produced from crop feedstock, waste oil, and animal fats. Algae are a highly productive feedstock with great potential in the biodiesel industry. The purpose of this project is to design a small-scale algae cultivation system for the production of biodiesel. This design incorporates the techniques and findings of advanced research with materials and an operating procedure that are simple, accessible to the common man, and less costly than laboratory units. The final design is based on an extensive literature review, computer modeling, and a physical model.

Introduction

Demand for fossil fuels is sky-rocketing and fossil fuels are a limited, non-renewable energy source. As this resource is depleted, prices will likely rise exponentially, until stocks are completely exhausted. Furthermore, harmful emissions from the combustion of fossil fuels contribute to climate change (EIA). As that resource is depleted, an alternative source of energy will need to replace it. Over the last decades, billions of dollars have been poured into research in a massive effort to find such an alternative fuel. Biodiesel is a renewable, potentially carbon neutral transport fuel (OEE). The process occurs according to the following chemical reaction.

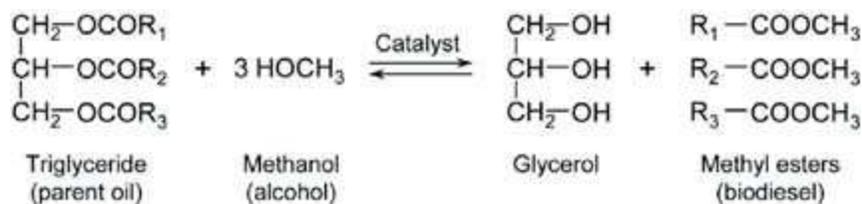


Figure 1: Transesterification of oil to biodiesel (Christi 2007)

An oil, or triglyceride, is combined with an alcohol, generally methanol, and goes through transesterification to become methyl esters (biodiesel). The by-product, glycerol, is also of commercial value in the pharmaceutical industry.

Biodiesel is a cleaner burning fuel than conventional diesel, and produces fewer greenhouse gas emissions. Biodiesel blends of any concentration can be used in any diesel engine. Technology producing and using biodiesel has been available for over 50 years (Christi 2007). Therefore, biodiesel is potentially an ideal alternative to fossil fuel use.

The obstacle then becomes producing sufficient quantities of the oils needed to make biodiesel. In the United States, biodiesel is produced primarily from soybean oil. It is also produced, to a lesser extent, from canola oil, palm oil, corn oil, waste cooking oil, animal fat, and jatropha oil. Unfortunately, biodiesel production from any of the conventional feedstocks mentioned here cannot realistically meet the tremendous demand for transport fuels. It would require 0.53 billion m³ of biodiesel to replace all the transport fuels consumed in the United States. Producing even half of this would require an unsustainably large land base (Chisti 2007). Therefore, a new feedstock is required. Algae, like all plants, uses sunlight to produce oils, but it does so more efficiently than crop plants. The table below summarizes the land area required for various feedstocks to meet 50% of the transport fuel needs of the United States.

Table 1 - Land area required to meet 50% of U.S. transport fuel needs

Comparison of some sources of biodiesel			
Crop	Oil yield (L/ha)	Land area needed (M ha) ^a	Percent of existing US cropping area ^a
Corn	172	1540	846
Soybean	446	594	326
Canola	1190	223	122
Jatropha	1892	140	77
Coconut	2689	99	54
Oil palm	5950	45	24
Microalgae ^b	136,900	2	1.1
Microalgae ^c	58,700	4.5	2.5

^a For meeting 50% of all transport fuel needs of the United States.

^b 70% oil (by wt) in biomass.

^c 30% oil (by wt) in biomass.

(Chisti 2007)

The land base required for microalgae production is just a tiny fraction of the land required to harvest the same quantity of oil using any other known feedstock. Many species of microalgae are extremely rich in oil (Rodolfi et al.). An oil content of 20-50% by weight on a dry mass basis is easily attainable. Algae also reproduce very quickly, and commonly have doubling times of 24 hours or less. They are largely a non-food resource. Algae production can also take place on non-productive land, such as desert land, that is of too poor quality for conventional crops (NREL). They can utilize brackish or saline water as well as sequester waste CO₂ from other processes (NREL). Algae, for the preceding reasons, are an ideal feedstock for biodiesel production.

Problem Identification

Algae is an ideal feedstock for the production of biodiesel. A great deal of research has been conducted to optimize algae cultivation. Models for algae growth have been developed to isolate ideal temperature, light, and nutrient input ranges. Techniques have been formulated to maximize algae productivity. Advanced algae cultivation systems have been designed using the most expensive equipment, with operating procedures that require a state-of-the-art laboratory setting.

One remaining challenge is that none of the products of this research are currently available to the general public. Biodiesel produced from cultivated algae is not yet marketed commercially. However, there is adequate scientific research available for a person to be able to construct and operate their own optimized algae cultivation system. The challenge is that although the information is available through various sources, nowhere is the research integrated into a clear, easily comprehensible format. What's more, the technology remains inaccessible to the general public because the systems proposed in literature are not explicit in their descriptions and require complex structures, hard to acquire materials, and fully equipped laboratories to operate.

Objective and Scope

This project, therefore, aims to bring algae cultivation to those members of the public who are interested in biodiesel production. The design incorporates the findings of research that is largely inaccessible to the public in the development of an algae cultivation system. This algae cultivation system, however, while making the most of the extensive research that has been conducted, is simple to build, and utilizes materials that are readily available. The project furthermore attempts to keep implementation of the design relatively affordable. In brief, the design makes algae cultivation accessible to the general public without compromising performance.

The goal of the project is to design an algae cultivation system encompassing, the selection of the algae, the operating procedure and the system in which it is grown. The subsequent harvesting of the algae and oil extraction are beyond the scope of this design, although a literature review of the techniques and processes currently available is provided in the Appendix.

Early design decisions

1) Choice of algal species

a) *The Algae Selection Process*

The first step in the design process was the selection of an algal strain for the purpose of biodiesel production. Certain key design characteristics were identified as desirable for the purpose of the project:

Table 2: Desired algae characteristics

<u>Lipids</u>	
High lipid productivity:	40 mg/l/day under optimal conditions
Lipid content enhancement due to nitrogen deprivation	
Easily convertible lipids to biodiesel	
<u>Growth conditions</u>	
Temperature	20-25°C
pH: Wide tolerance	
Light inhibition	>1400 $\mu\text{mol photons /m}^2/\text{s}$
Resilience to variation in growth media composition	
Resistance to contamination: competitive strain	
Fresh water strains preferred	
<u>Other</u>	
Inexpensive	
Easy to procure	
Information widely available	

These characteristics were based on the likely conditions within the system. For instance, the system would likely not be air tight, allowing for a small degree of contamination, this meant that the algae strain needed to be resilient to contamination.

Another crucial parameter was the availability of information. Many strains lack the proper documentation and background research to give sufficient information as to its optimal growth conditions or limitations, making the selection of such strains risky. Moreover, the lack of sufficient background literature would make it difficult to validate the result of the project since no literature information would be available for comparison.

The selection process began with choosing the algal strains from the following list identified by Rodolfi et al (2008) as being the most promising in terms of lipid productivity:

Table 3: Biomass productivity, lipid content and lipid productivity of 30 microalgal strains (Rodolfi et Al 2008)

Microalgae	Biomass productivity (g/L/day)	Lipid content (% biomass)	Lipid productivity (mg/L/day)
Marine strains			
<i>Porphyridium cruentum</i>	0.37	9.5	34.8
<i>Tetraselmis suecica</i> F&M-M33	0.32	8.5	27.0
<i>Tetraselmis</i> sp. F&M-M34	0.30	14.7	43.4
<i>Tetraselmis suecica</i> F&M-M35	0.28	12.9	36.4
<i>Phaeodactylum tricoratum</i> F&M-M40	0.24	18.7	44.8
<i>Nannochloropsis</i> sp. F&M-M26	0.21	29.6	61.0
<i>Nannochloropsis</i> sp. F&M-M27	0.20	24.4	48.2
<i>Nannochloropsis</i> sp. F&M-M24	0.18	30.9	54.8
<i>Nannochloropsis</i> sp. F&M-M29	0.17	21.6	37.6
<i>Ellipsoidium</i> sp. F&M-M31	0.17	27.4	47.3
<i>Nannochloropsis</i> sp. F&M-M28	0.17	35.7	60.9
<i>Nannochloropsis</i> CS 246	0.17	29.2	49.7
<i>Isochrysis</i> sp. (T-ISO) CS 177	0.17	22.4	37.7
<i>Pavlova salina</i> CS 49	0.16	30.9	49.4
<i>Pavlova lutheri</i> CS 182	0.14	35.5	50.2
<i>Isochrysis</i> sp. F&M-M37	0.14	27.4	37.8
<i>Skeletonema</i> sp. CS 252	0.09	31.8	27.3
<i>Thalassiosira pseudonana</i> CS 173	0.08	20.6	17.4
<i>Skeletonema costatum</i> CS 181	0.08	21.1	17.4
<i>Chaetoceros muelleri</i> F&M-M43	0.07	33.6	21.8
<i>Chaetoceros calcitrans</i> CS 178	0.04	39.8	17.6
Freshwater strains			
<i>Chlorococcum</i> sp. UMACC 112	0.28	19.3	53.7
<i>Scenedesmus</i> sp. DM	0.26	21.1	53.9
<i>Chlorella sorokiniana</i> IAM-212	0.23	19.3	44.7
<i>Chlorella</i> sp. F&M-M48	0.23	18.7	42.1
<i>Scenedesmus</i> sp. F&M-M19	0.21	19.6	40.8
<i>Chlorella vulgaris</i> F&M-M49	0.20	18.4	36.9
<i>Scenedesmus quadricauda</i>	0.19	18.4	35.1
<i>Monodus subterraneus</i> UTEX 151	0.19	16.1	30.4
<i>Chlorella vulgaris</i> CCAP 211/11b	0.17	19.2	32.6

The flasks were incubated at 25°C under continuous illumination in an orbital shaker flushed with CO₂ enriched air.

Strains which lacked documentation on key parameters such as growth conditions were discarded in favor of better known strains. Both marine and fresh water strains were investigated but the fresh water strains were given priority in order to eliminate, if possible, the added time and cost associated with making salt water.

b) Algal species investigated

Four algal species were selected on the basis of their lipid productivity and then investigated:

Three fresh water strains: - *Chlorella sp*: 42.1 mg/L/day

- *Chlorococcum sp*: 53.7 mg/L/day

- *Scenedesmus sp*: 53.9 mg/L/day

One marine strain: - *Nannochloropsis sp*: 61.0 mg/L/day

Nannochloropsis sp displayed high lipid productivity and grew well under the specified conditions. It was also fairly well documented and reacted well to nitrogen deprivation. However this species was not easy to find and required salt water with an optimal salinity range, meaning the growth medium would be more complex to make. These two factors were difficult to overcome in our design since widely available algae species was deemed essential for the project. Procurement difficulties and salt water issues therefore led to the abandon of this species as a possible choice. It is important to note that the algae presented above are a type of algal species comprised of a number of strains. However most algae strains of the same species display similar behavior. For instance, all *Nannochloropsis sp*. strains have high lipid productivity on average as compared to other marine species. For the purpose of this project, it is therefore

assumed that the chosen strain displays the productivity and properties of the species that it is from according to literature values.

c) The Chosen Algae: *Chlorella* sp.

Out of the three remaining productive fresh water strains, the only strain for which sufficient information was available was the *Chlorella* sp which has the advantage of being easy to procure and cheap. It is also known for its resistance to contamination and tolerance to varying growth medium conditions (Amos 2005). This algae strain is widely used in research and a lot of literature review is available making it easier to interpret results. Indeed *Chlorella* strains have been investigated both as a food supplement as well as for biofuel production purposes. Algae optimized for growth at 26°C for the project were obtained from *Fussman Lab* with the help of Anil Patel.

Chlorella is a genus of single-celled green algae, belonging to the phylum Chlorophyta. It has a spherical shape, about 2 to 10 µm in diameter, and is without flagella. *Chlorella* contains the green photosynthetic pigments chlorophyll-a and -b in its chloroplast.



Figure 2: Photo of *Chlorella* sp.
(Chisti, 2007)

It is important to note that *Chlorella sp* has relatively low lipid content but high biomass productivity. This is what explains the fact that lipid productivity is reasonable. The high biomass productivity results in the high chlorophyll content of this strain giving it a higher photosynthetic efficiency. It takes around 20 days for the algae to go from lag phase to exponential phase and then stationary phase. (Borowitzka, 1980)

Chlorella sp. enhances its lipid content due to nitrogen deprivation but is less responsive than marine species such as *Nannocloropsis sp.* The sensitivity depends on the particular strains but almost all strains display an increase in lipid content under nitrogen deprived conditions.

Table 4: Comparative characterization of Chlorella strains grown on a medium with or without nitrogen source

Strains	Temp-erature (°C)	Variant	% dry weight		
			Proteins	Carbo-hydrates	Fatty acids
<i>Chlorella ellipsoidea</i> SK	26	+N	43.0	15.0	2.3
		-N	22.6	21.0	26.8
<i>C. pyrenoidosa</i> 82	26	+N	59.3	24.0	16.7
		-N	15.6	37.3	47.1
<i>C. pyrenoidosa</i> 82T	26	+N	55.0	31.8	13.2
		-N	22.5	67.9	9.6
<i>C. pyrenoidosa</i> TKh-7-11-05	36	+N	57.0	10.0	10.1
		-N	27.3	44.2	12.6
<i>Chlorella sp.</i> K	36	+N	46.2	18.4	7.9
		-N	24.3	54.5	10.1
<i>C. vulgaris</i> 157	26	+N	49.9	10.3	3.1
		-N	16.5	44.0	11.5

(Michael A Borowitzka, 1980)

d) Nutrient requirements

During growth *Chlorella* consumes nutrients in a certain proportion shown in table 5 and these nutrient requirements can be met by cultivating the algae in a growth media made from the following nutrient batch solutions:

Table 5: : Growth media for *Chlorella* sp.

Nutrients (mg l ⁻¹)	A
KNO ₃	1250
Urea	
KH ₂ PO ₄	1250
MgSO ₄ ·7H ₂ O	1000
CaCl ₂	83.5
FeSO ₄ ·7H ₂ O	49.8
A ₅ solution (ml) ^a	
H ₃ BO ₃	114.2
ZnSO ₄ ·7H ₂ O	88.2
MnCl ₂ ·4H ₂ O	14.2
MoO ₃	7.1
CuSO ₄ ·5H ₂ O	15.7
Co(NO ₃) ₂ ·6H ₂ O	4.9
Citric acid	
EDTA	500

(Michael A Borowitz, 1980)

An example of growth media used for *Chlorella* is shown in the Appendix, and was in fact used during the experimentation with the *Chlorella* algae.

Overall process of photosynthetic biomass production for the mixed culture is found to be (S. John Pirt et al, 1980):



However the above equation only takes into account the production of new biomass disregarding the maintenance energy and nutrient requirements of the existing biomass. A good ratio for the amount of

carbon actually needed to grow and maintain the algae is the growth yield defined as the amount of biomass produced (dX) through the consumption of a unit quantity of substrate (Christi 2007):

$$Y_{x/s} = dX / ds = \frac{100 \text{ gBiomass}}{183 \text{ gCO}_2} = 0.546 \text{ g Biomass / gCO}_2$$

With this ratio in mind, it is possible to obtain an estimate of the carbon input required for the algae to grow without suffering from carbon deficiency. This estimation of the carbon required would later be essential in calibrating the air flow rate and CO₂ enrichment level. 183 g of CO₂ consumed for each 100g of algae produced. Under optimal conditions, grown outside in sunlight, chlorella can produce a maximum of up to 0.45 kg/m³/day (James C Ogbonna).

Assuming a volume of 20L of culture this translates into a CO₂ usage of:

$$\max CO_{2\text{ usage}} = 0.45 \text{ kgBiomass / m}^3 / \text{d} \times 1.83 \text{ kgCO}_2 / \text{kgBiomass} \times 0.02 \text{ m}^3 = 0.0165 \text{ kg CO}_2 / \text{day}$$

Assuming sub optimal growing conditions, a lower CO₂ input of as little as 5 to 10g of CO₂ per day would prevent carbon shortages in the growth media. Only experimental data from the algae growth system in operation can provide a more precise estimate of the carbon required. Feedback from the actual system would then be used to adjust the CO₂ inputs more precisely.

2) Choice of Algae Cultivation System

a) *Open Systems*

Open systems were the first method of algae cultivation developed (see Appendix). Because they require a large land base, they were deemed inappropriate for this design, as it is limited to the space available on an average individual's property. Such a large open surface area would also leave the algae vulnerable to contamination.

b) *Photobioreactors*

A photobioreactor is a closed system that contains a biologically active environment which is sustained with light, energy, heat and nutrients. They have the main advantage of being a closed system, which eliminates the risk of contamination but more importantly allows for a better control of the conditions surrounding the cultivation of microalgae. Different types of algae need different conditions in order to reproduce at their most effective rate, which is easily facilitated by the photobioreactor, which allows parameters to be controlled more tightly. Because the photobioreactor is a closed system, it requires an airlift. The airlift is the means by which the gas exchanges occur. It is a "device that serves to circulate the culture through the solar receiver (tubes)" (Fernandez et al., 2001). Carbon dioxide enters the system while oxygen is removed. It is of the greatest importance to evacuate oxygen from the system because it accumulates within the tubes and prevents photosynthesis. As the fluid circulates through the system, a gas-liquid separator found at the top of the air-lift prevents the oxygen from going back into the system (Fernandez et al., 2001). So far, the air-lift has been the mechanism the best suited to perform this function (Richmond et al., 1993). Carbon dioxide is necessary to the cultivation of micro-algae and the air-lift pump also helps to control the concentrations of CO₂ that will be sent into the system. Another advantage to the air-lift is that

is causes mixing within the system, allowing the algae to get more sunlight. This component is usually very small compared to the rest of the system as it is one of the darkest parts. If the fluid stays in it too long productivity will be diminished due to a lack of light. Researchers also prefer this system because the flow patterns and circulation times within are easy to follow and understand (Eriksen, 2008).

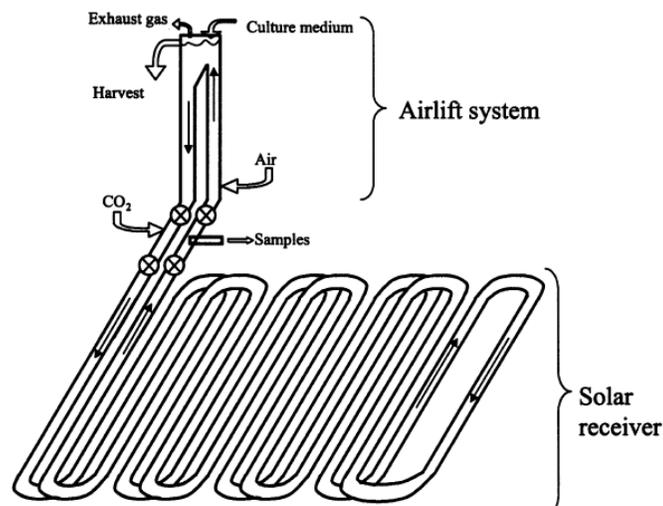


Figure 3: Airlift photobioreactor system
(Molina et al., 2001)

c) *Tubular Photobioreactor*

A tubular photobioreactor consists of an air-lift system and a solar receiver. The solar receiver is essentially long lines of tubes that intertwine over a large surface. The tubes are usually made of transparent glass or plastic. The diameters vary between 3 to 6 cm and the lengths between 10 and 100 m (Janssen et al., 2001). They can be placed in many different ways of which the most common is laid horizontally or set up vertically. Its main advantage is that it has a large surface area. Since it is spread on a long and large distance and because it has tubes of a small diameter, it allows more of the medium to be in contact with sunlight, allowing for good biomass productivities (Ugwu et al., 2007). It is also one of the cheapest systems as the pipes do

not have to be of high quality. In his research, Miyamoto used commercially available glass tubes that were at a very low cost. Also, instead of cleaning and sterilizing the tubes they could just be thrown away and replaced by new ones. On a small scale this works, but on a larger scale it may get more complicated. In terms of productivity, tubular photobioreactors have a higher biomass density, but vertical-column photobioreactors have a higher biomass yield.

As great as this system may seem it still has some major flaws. The first one is that it has poor mass transfer, problem which increases as the system is scaled up. Oxygen tends to build-up within the tubes if there is poor mixing and circulation. There are also problems with the circulation of carbon dioxide. This becomes a problem because re-carbonation is needed when the pH gradient within the tubes gets too high (Ugwu et al., 2001). The final problem is land-space. This systems demand a lot of space as the tubes can extend for long distances. Fortunately, since it is a closed system, any type of flat non-arable land can be used to hold the system.

d) Vertical-Column Photobioreactor

Vertical-column photobioreactors can also use the airlift system and they can have one or many vertical tubes that are lined up. Unlike the tubular photobioreactor, the medium does not pass through all the tubes and the columns have a bigger diameter. The tubes have diameters in the ranges of 15 to 20 cm and they can go up to 5m high. Since the tubes are much larger, a good mixing system has to be put in place. For vertical columns this system is referred to as the bubble column (Janssen et al., 2001). They are aerated from below which causes air bubbles to rise and cause mixing within the medium. This allows the algae that is not exposed to light to get more and the light/dark cycle can be profitable to the algae. Small fans can also be installed within the column to mix the fluid. Yet, the high velocities and turbulence flow caused by the fans create shear stresses which have been shown to damage the microalgae (Eriksen, 2008).

This type of mixing exerts a low shear stress on the walls of the column and demands little energy (Ugwu et al., 2008). In some cases, columns with much larger diameters have used internal light sources. Although this is efficient it is not cost effective. Another advantage is that they are quite easy to clean and to sterilize since they are rather large and can be opened from the top and the bottom allowing the waste water to flow out of the tubes.

Its disadvantages are that the materials for the column are much more expensive as the column must be made sturdier (Ugwu et al., 2008). There is also a smaller surface area of irradiance.. As was said before internal illumination systems have been tried, yet have turned out to be way too expensive for the little more biomass it would produce.



Figure 4 - Vertical column photobioreactors

Source: <http://www.sunxenergy.com/research.htm>

e) Choice of the System

After carefully examining the different types of systems, it was concluded that a vertical cylindrical photobioreactor with a bubble column and internal illumination would be the best choice. A vertical cylindrical photobioreactor is the best one to take as it would allow the maximum light penetration and the best gas holdup within the column. A vertical photobioreactor is also much simpler to build as there much fewer components and they are larger and easier to work with. It also takes up very little space and the scaling up to multiple units is simple. Because a column photobioreactor has a relatively large diameter, illumination

at the center is not optimal, and therefore it was decided that an internal illumination system be incorporated into the design to compensate for this. It would allow the algae in the middle of the photobioreactor to have access to as much light as possible. A batch system was preferred over a continuous flow system. This was because one individual will be operating the system, and it would be highly inconvenient for him to harvest algae daily. Since this system will be used to cultivate algae for biofuels, high lipid content is necessary. A technique called nitrogen deprivation is talked about in literature (see Appendix). This technique increases lipid content in algae cells and will therefore be incorporated in the design.

Design Refinement

1) Preliminary Model

The first step in the design of the model was to determine what type of photobioreactor was ideal, taking into consideration the different growth parameters and how it would operate. It was previously determined that a vertical cylindrical photobioreactor would be the best type of system. It was also determined that a mix of internal illumination and external lighting would be the most efficient. The internal light source would be powerful enough to supply light to the algae in the interior half of the outer tube, while external light source would supply light to the algae in the exterior half of the outer tube. A light/dark cycle of 18 hours of light and 6 hours of dark would be set and a timer would be used to control the lights. The light/dark cycle is good for algae growth as it enhances the photosynthesis process of the algae. (Amos et al, 2003)

Turbulence had to be generated in the tube. It was impossible to put a fan at the center of the tube because it is occupied by the light, and it was problematic to try and insert an electric wire through the side to supply electricity to the fan while keeping the main cylinder watertight. The simplest solution was to make holes in the top through which a tube would pass in a U shape at the bottom. The tube would be attached to an air pump and small holes would be pierced at intervals to create small bubbles that would constitute create air flow. Carbon dioxide would be provided to the system through the air flow which would be enriched to 0.5% carbon dioxide.

Nutrient inputs would also be put in from the top of the cylinder. In order to increase the lipid content of algae, a nitrogen deprivation cycle was established in which nitrogen would be supplied to the system for 10 days, and after it would be deprived for another 10 days before the algae was harvested and the oil was extracted. The 20 day cycle was chosen because it is the most well-cited batch cycle length (Liu et al., 2007). It was found during the literature review that, given the needs of the *Chlorella* sp., standard liquid lawn fertilizer with a high N:P ratio would supply the *Chlorella* sp. with all its required nutrients. This was much simpler than trying to acquire the individual components and mixing growth medium from scratch.

A batch cultivation system was also chosen. Once the growth cycle was complete and the algae was ready to be harvested, the whole photobioreactor would be tipped over so that the medium would pour out of the hole in the top of the photobioreactor. A small algae sample would be taken and then used to start the following batch. Cleaning of the photobioreactor could be done chemically in between batches by using a sponge soaked with a 70% by volume ethanol solution

Below is a diagram representing the proposed system design:

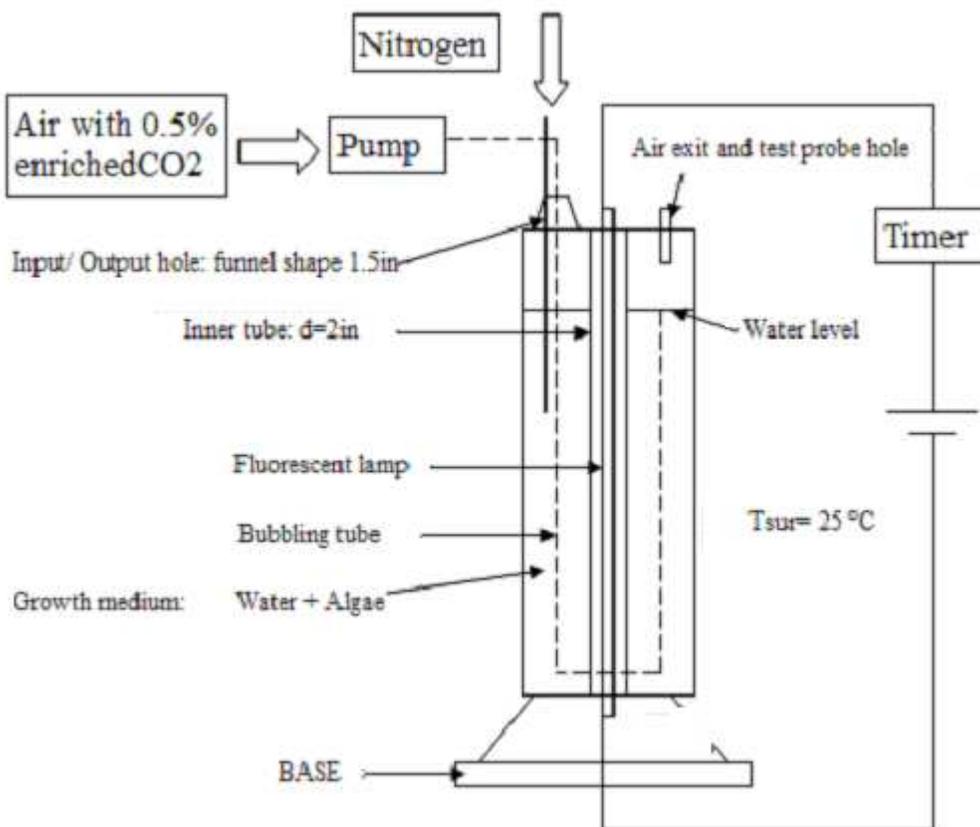


Figure 5: Preliminary design

2) Dimensions

a) *Height*

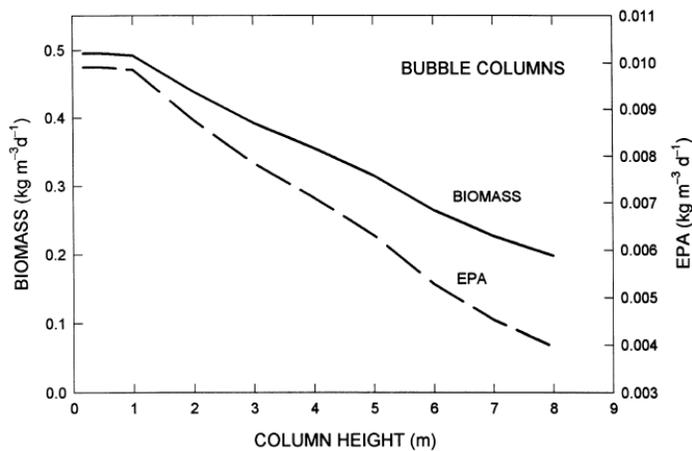


Figure 6: Biomass vs. Column height
(Sanchez Miron et al., 1999)

For a bubble column, the height should not exceed 1m. As the figure above shows, biomass productivity declines steadily as the height increases past 1m. This is because the gas holdup within the column is reduced at greater heights. Most of the carbon dioxide from the tube will be absorbed by the algae at the bottom before it makes it to the stop, diminishing the growth rate and making the photobioreactor less efficient. The turbulence at the top will also be reduced and this can affect the rapid light/dark cycle that the algae should experience.

b) Diameter

The optical path is defined as the distance between the center of the photobioreactor to the light source and the surface to volume ratio is defined as the ratio between the illuminated surface area of a reactor and its volume and it determines the amount of light which enters the system per unit volume. These parameters are paramount in importance since the higher the surface to volume ratio, the higher the cell concentration achievable and the higher the volumetric productivity of the culture. The high concentration lowers the cost of harvest. However, when cells achieve high concentration in the medium, the light is blocked by the algae closer to the light source, leaving no light for algae at the center of the photobioreactor. Penetration depth is therefore a function of algal density. (Amos, 2005)

Decreasing the pipe diameter is the best way of increasing the surface to volume ratio and therefore increases both volumetric productivity (productivity per unit of reactor volume) and illuminated surface productivity (ISP: productivity per unit of illuminated surface reactor area). It is important to note that ISP is distinct from areal productivity (productivity per unit of illuminated ground surface area occupied by the reactor). So the surface to volume ratio and therefore volumetric and illuminated surface productivities are enhanced with smaller diameter tubes. (Amos, 2005)

A simulation of the growth rate was done in order to estimate the growth rate of the algae in the system in two different light regimes: one with external illumination at latitude corresponding to that of Montreal on average over the year, and one where only internal illumination was used using a fluorescent light.

With Sunlight:

The irradiance is the density of luminous flux per surface area.

$$I = (-7.6 * D) + 1100$$

(Source: Perez et al., 2008)

$$I = (-7.6 * 7.2 \text{cm}) + 1100 = 980.68 \text{ } \mu\text{mol/ cm}^2\text{s}^{-1}$$

Where: I = Irradiance or Photon Flux ($\mu\text{mol/ cm}^2\text{s}^{-1}$)

D = Distance from outer surface (cm)

$$\mu = \frac{\mu_{\max} I}{I + K_i}$$

(Source: Perez et al., 2008)

$$\mu = (0.065 \text{ h}^{-1} * 345.28 \mu\text{mol/ cm}^2\text{s}^{-1}) / (345.28 \mu\text{mol/ cm}^2\text{s}^{-1} + 10.2 \mu\text{E /m}^2\text{s}^{-1}) = 0.0643 \text{ h}^{-1}$$

Where: μ Growth Rate (h^{-1})

$\mu_{\max} = 0.065 =$ Maximum Growth Rate (h^{-1})

I = Photon flux ($\mu\text{mol/ m}^2\text{s}^{-1}$)

$K_i = 10.2 =$ Half-Life Constant ($\mu\text{E /m}^2\text{s}^{-1}$)

The first equation is used to determine the irradiance of the sunlight into the medium according to the distance from the inner tube wall. An initial value of $1100 \mu\text{mol/ m}^2\text{s}^{-1}$ is assumed at the wall of the outer tube (Amos et al, 2003). As the rays of light from the sun penetrate into the medium the irradiance value decreases. The growth rate of the algae varies

according to the irradiance as the sunlight received by the algae will determine the rate at which photosynthesis occurs.

From the Internal Light Source: (Source: Perez et al., 2008)

$$\mu = \frac{\mu_{\max} I}{I + K_i}$$

$$\mu = (0.022 \text{ h}^{-1} * 48 \mu\text{mol}/\text{cm}^2\text{s}^1) / (48 \mu\text{mol}/\text{cm}^2\text{s}^1 + 10.2 \mu\text{E}/\text{m}^2\text{s}^1) = 0.0181 \text{h}^{-1}$$

Where: μ Growth rate (h^{-1})

$\mu_{\max} = 0.022 = \text{Maximum Growth Rate } (\text{h}^{-1})$

$I = 48 = \text{Photon flux } (\mu\text{mol}/\text{m}^2\text{s}^1)$ at 7.2 cm away from light source and it is measured

$K_i = 10.2 (\mu\text{E}/\text{m}^2\text{s}^1)$

The external illumination source provided a photon flux of $1100 \mu\text{mol}/\text{cm}^2\text{s}^1$. Once again, this value will decrease as it goes deeper into the medium and as the algae grows. The irradiance also depends on the algae growth, as the more algae there is, the less light will penetrate.

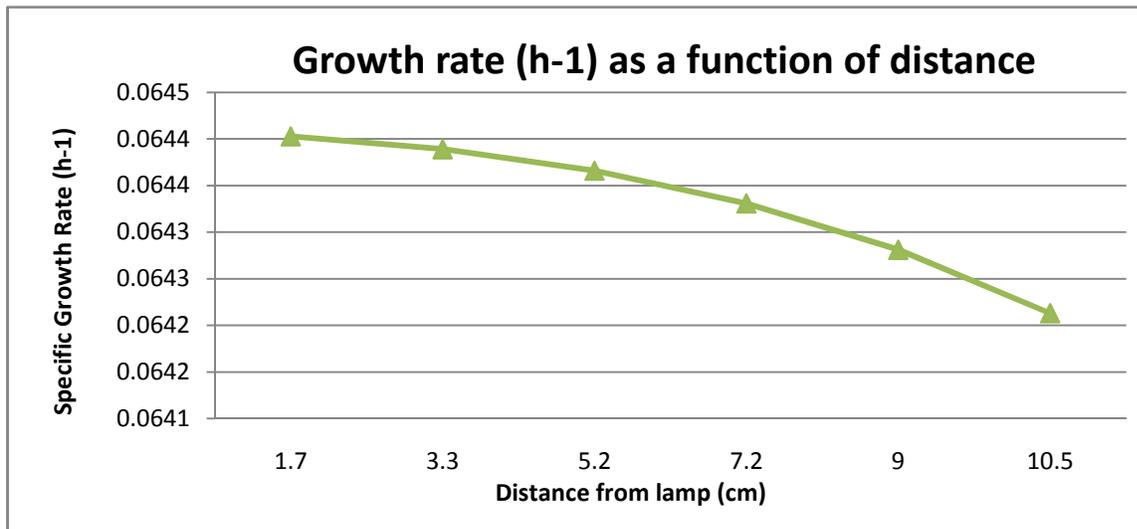


Figure 7: Growth rate vs. distance from fluorescent lamp

As the graph above demonstrates, the growth rate decreases as the algae is further from the outer wall of the photobioreactor. It is necessary to supply the most light possible to the algae without causing photoinhibition, which occurs when the capacity for photosynthesis of the algae is reduced due to a very high exposure of light. This reduces the growth rate of the algae and can damage it. If the algae do not receive enough light the photosynthesis process will not occur as rapidly. This graph helped to determine the desired tube diameter, because the growth rate decreases rapidly as the diameter increases. The chosen dimension was a 10cm radius for two reasons. The use of internal illumination reduces the light path to 5cm, which maintains a high growth rate throughout all of the photobioreactor, even though the diameter is large. Also, as the tube diameter increases, the capacity volume increases, but so does the price. The graph shows that the most cost effective choice is the 8" diameter tube, at \$7,757.75 per m³ capacity.

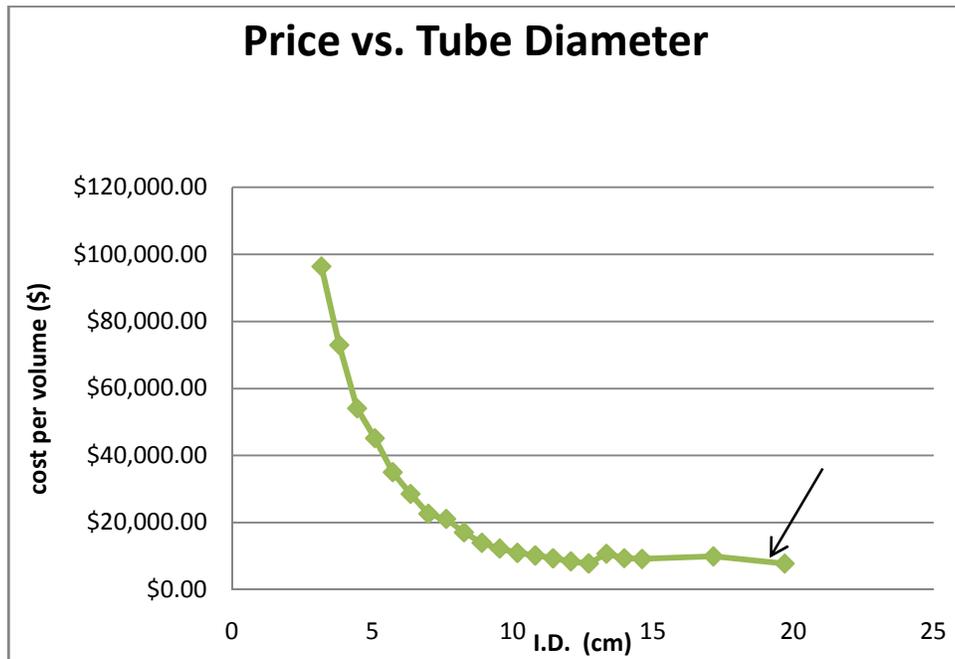


Figure 8: Price vs tube diameter

3) Gas inputs

In order to get a better idea of the mass transfer coefficient and of the desired flow rate needed for a proper functioning of the system a simulation was done to find the range at which our system would be the most efficient.

Superficial Gas Velocity: (Source: Sanchez Miron et al., 1999)

$$U_G = \frac{Q}{(A_o - A_i)}$$

$$U_g = 0.00008 \text{ m}^3/\text{s} / ((\pi * (0.18\text{m} / 2)^2) - (\pi * (0.0508\text{m} / 2)^2)) = 0.0034 \text{ m/s}$$

Where: U_g = Superficial Gas Velocity (m/s)

$Q = 0.00008$ = Air Flow From Tube (m^3/s)

A_o = Outer Tube Area (m^2)

A_i = Interior Tube Area (m^2)

Superficial gas velocity is the volume flow of gas per cross-sectional area of pipe. By changing the values of the flow rate in this equation and the values of the mass transfer coefficient a balance will be established at the point where the desired mass transfer coefficient will be attained while maintaining a reasonable flow rate that won't damage the algae.

Power Input:

$$\frac{P_G}{V_L} = \rho_L g U_g$$

(Source: Sanchez Miron et al., 1999)

$$P_G/V_L = 1000 \text{ kg/m}^3 * 9.81 \text{ m/s}^2 * 0.0034 \text{ m/s} = 33.5097 \text{ W/m}^3$$

Where: P_G/V_L = Power Input (W/m^3)

$P_w = 1000$ = Density of Tap Water (kg/m^3)

$g = 9.81$ = Gravitational Acceleration (m/s^2)

U_g = Superficial Gas Velocity (m/s)

The power input is the rate of energy that must be supplied to the pump in order to get the desired flow rate within the system.

Gas Holdup:

Gas holdup is described as the reactor volume taken up by the gas. It can be estimated as the volume of the liquid displaced by the gas. Gas holdup is very important in photobioreactor design as it determines the circulation rate, the gas residence time, as well as the overall mass transfer rate (kLa). Calculations are done using empirical correlations. These correlations

establish relationships between bubble size, gas liquid interfacial surface area as well as with the mass transfer coefficient. (C.U Ugwu et al., 2007)

$$\epsilon = (3.317 \times 10^{-4}) * (P_G / V_L)^{0.97}$$

(Source: Sanchez Miron et al., 1999)

$$\epsilon = (3.317 \times 10^{-4}) * (33.5097 \text{ W/m}^3)^{0.97} = 0.01$$

Where: ϵ = Gas Holdup

$$P_G/V_L = \text{Power Input (W/m}^3)$$

Mass Transfer Coefficient:

The mass transfer coefficient is the rate at which the gases (in this case air and carbon dioxide) will be diffused into the medium.

$$k_L a_L = 2.39 \times 10^{-4} \left(\frac{P_G}{V_L} \right)^{0.86}$$

(Source: Sanchez Miron et al., 1999)

$$k_L a_L = (2.39 \times 10^{-4}) * (33.5097 \text{ W/m}^3)^{0.86} = 0.0049 \text{ s}^{-1}$$

Where: $k_L a_L$ = Mass Transfer Coefficient (s^{-1})

$$P_G/V_L = \text{Power Input (W/m}^3)$$

The mass transfer coefficient is important because it allows the quantity of carbon dioxide that will penetrate the medium to be determined. Carbon dioxide is necessary for the proper growth of the algae. If there is too little carbon dioxide, the algae growth rate is limited by

carbon dioxide and drops significantly, but if there is too much, the growth medium will become acidic, and the growth nearly draws to a halt. The air flow that is circulated through the photobioreactor is to be enriched to 0.5% of carbon dioxide. Since the algae requires a certain quantity of carbon dioxide, the flow rate will be determined by the desired carbon dioxide input.

CO₂ Flow Rate:

$$Q_{CO_2} = CO_2 \text{ Enrichment} * Q$$

$$Q_{CO_2} = 0.005 * 0.00008 \text{ m}^3/\text{s} = 4 * 10^{-7} \text{ m}^3/\text{s}$$

Where: Q_{CO_2} = CO₂ Enrichment Flow Rate (m³/s)

$$CO_2 \text{ Enrichment} = 0.005$$

$$Q = \text{Flow Rate (m}^3/\text{s)}$$

This equation is used to determine the flow rate of carbon dioxide that will come out from air flow.

CO₂ Mass Flow Rate:

$$Q_{CO_2m} = Q_{CO_2} * \rho_{CO_2}$$

$$Q_{CO_2m} = 4 * 10^{-7} \text{ m}^3/\text{s} * 1.98 \text{ kg/m}^3 = 0.000792 \text{ g/s}$$

Where: Q_{CO_2m} = Mass Flow Rate of CO₂

$$Q_{CO_2} = CO_2 \text{ Enrichment Flow Rate (m}^3/\text{s)}$$

$$\rho_{CO_2} = 1.98 = \text{Density of CO}_2 \text{ (kg/m}^3\text{)}$$

This equation is used to determine the mass of carbon dioxide that will be transferred from the air flow to the growth medium.

Mass of CO₂ Transferred to Water Per Day:

$$Q_{CO_2ToW} = Q_{CO_2m} * k_L a_L * V_w$$

$$Q_{CO_2ToW} = 0.000792 \text{ g/s} * 0.0049 \text{ s}^{-1} * 0.0211 \text{ m}^3 * 86400 \text{ days} * 1000 = 7.06 \text{ g/day}$$

Where: Q_{CO_2ToW} = Mass of CO₂ Transferred to Water Per Day

Q_{CO_2m} = Mass Flow Rate of CO₂

$k_L a_L$ = Mass Transfer Coefficient (s⁻¹)

$V_w = 0.0211$ = Working Volume (m³)

This equation determines the mass of carbon dioxide that will be supplied to the medium per day.

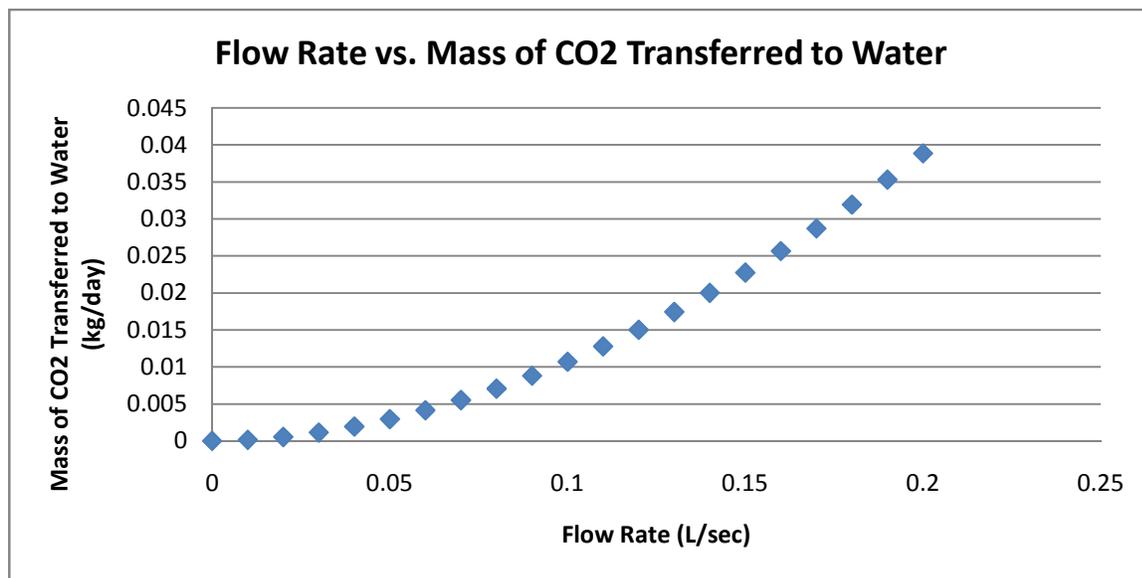


Figure 9: Mass transfer coefficient of CO₂ vs. Flow rate of air

The graph demonstrates that for the desired input rate of carbon dioxide into the medium (7 g/day), the necessary air flow rate Q is 0.08 L/s. Complete turbulence is achieved for this value of flow and increasing the turbulence more could result in damage to the algae. According to Sanchez Miron (1999) it was found that growth rates start declining after a superficial certain threshold. Indeed, when the flow rate is too high or the bubbles within the tube are too big (bigger than 1cm in diameter) it can damage the algae by exerting shear stresses that can break the algae. It was assumed that turbulence having been achieved, increasing the flow further would result in algae damage.

This bubbling method of generating gas transfer and turbulence is very energy efficient, since no mechanical stirring is necessary. In fact the power input is independent of the height of the reactor. The bubbling is also an efficient source of carbon for algae growth since CO_2 enriched air can be used. This type of reactor displays up to 90% CO_2 utilization. The bubble column reactor allows for the optimization of both CO_2 utilization and productivity. This type of reactor also offers a quick fix to the problem of O_2 build-up so common in the tubular family of reactors by allowing O_2 to bubble up the reactor and escape. Other advantages include no build-up of algae at the wall, because bubble-generated turbulence prevents it (K. Myamoto., 1988).

4) pH Simulation and Temperature

pH and temperature have a great impact on the growth kinetics of the algae and on system performance. Temperature is dependent on the environmental conditions and is not a parameter that can be adjusted if the photobioreactor is placed outside. If inside it will be maintained at room temperature, that is between 20 and 25°C. It was therefore found to be irrelevant to investigate the influence on growth rate of temperature given it would either remain constant or simply be impossible to manage. On the other hand pH was deemed adjustable and is directly a function of CO_2 inputs. That is why evaluating the impact of pH

changes was estimated to be of interest. The result from this simulation could then be compared to experimental values.

Growth Rate assuming sunlight exposure: (Source: Perez et al., 2008)

$$\mu = \frac{\mu_{\max}}{1 + ([H^+] / K_1) + (K_2 / [H^+])}$$

$$\begin{aligned} \mu &= 0.065 \text{ h}^{-1} / 1 + (1.58 \cdot 10^{-8} \text{ (mol/L)} / 2.3 \cdot 10^{-6} \%^{-1}) + (1.2 \cdot 10^{-6} \%^{-1}) / 1.58 \cdot 10^{-8} \text{ (mol/L)} \\ &= 6.41 \cdot 10^{-2} \text{ h}^{-1} \end{aligned}$$

Where: μ Growth rate (h^{-1})

$\mu_{\max \text{ external}} = 0.065 = \text{External Maximum Growth Rate (h}^{-1}\text{)}$

$H^+ = \text{Dissolved Hydrogen Ion Concentration (mol/L)}$

$K_1 = 2.3 \cdot 10^{-6} = \text{Kinetic Constant (\%}^{-1}\text{)}$

$K_2 = 1.2 \cdot 10^{-6} = \text{Kinetic Constant (\%}^{-1}\text{)}$

Growth Rate with internal illumination only: (Source: Perez et al., 2008)

$$\mu = \frac{\mu_{\max}}{1 + ([H^+] / K_1) + (K_2 / [H^+])}$$

$$\begin{aligned} \mu &= 0.022 \text{ h}^{-1} / 1 + (1.58 \cdot 10^{-8} \text{ (mol/L)} / 2.3 \cdot 10^{-6} \%^{-1}) + (1.2 \cdot 10^{-6} \%^{-1}) / 1.58 \cdot 10^{-8} \text{ (mol/L)} \\ &= 2.17 \cdot 10^{-2} \text{ h}^{-1} \end{aligned}$$

Where: μ Growth rate (h^{-1})

$\mu_{\max \text{ internal}} = 0.022 = \text{Internal Maximum Growth Rate (h}^{-1}\text{)}$

H^+ = Dissolved Hydrogen Ion Concentration (mol/L)

$K_1 = 2.3 \cdot 10^{-6}$ = Kinetic Constant ($\%^{-1}$)

$K_2 = 1.2 \cdot 10^{-6}$ = Kinetic Constant ($\%^{-1}$)

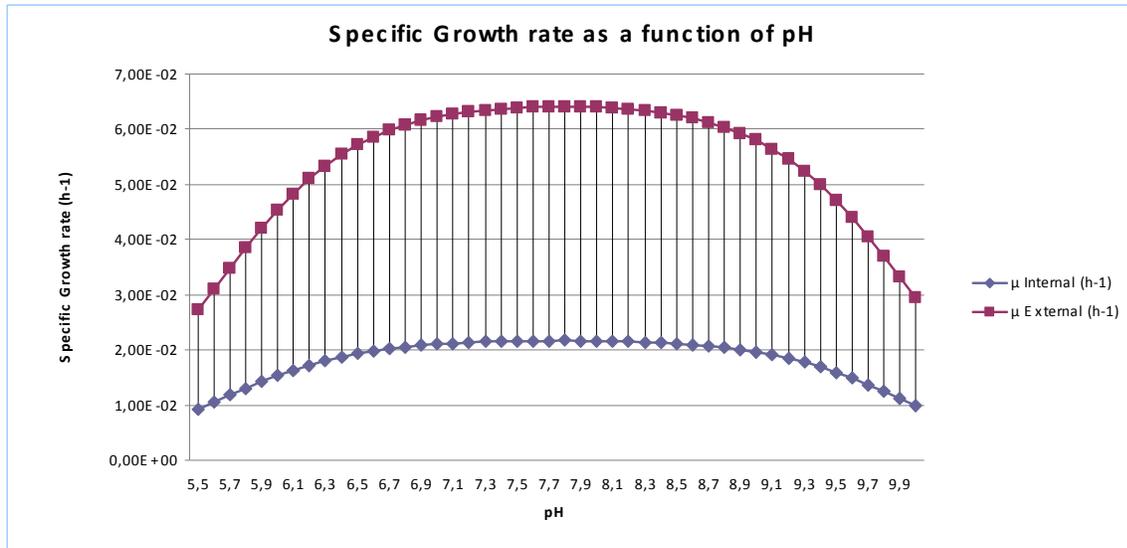


Figure 10: Growth rate vs. pH

The pH within the medium will vary according to the carbon dioxide that is found within the medium, and having a proper pH is necessary to help get the maximum lipid content from the algae. The pH will vary according to the amount of algae within the tube as the more algae there is, less carbon dioxide will be left within the medium. At the beginning of both curves, one can notice that the growth rate is much lower than in the middle. When the process starts the pH is very high because not all the carbon dioxide is being absorbed by the algae. When the medium starts to get more and more saturated with algae, all the carbon dioxide is used and the pH goes down. Since there is less available carbon dioxide, the growth rate starts to decrease and so does the lipid content. According to the graphic the best pH to maintain would be in a range of 7.3 to 8.3 as it is the point when the growth rate is at its highest. The pH can be measured with a probe or with the special papers. A way to maintain a good pH level would be

to control the carbon dioxide input and increase or decrease it according to the needs of the algae. This demands a lot of surveillance as the pH varies quite a bit from day to day.

5) Other Design Factors

a) *Batch System*

An important consideration is the fact that sustained high productivities can only be achieved by a regular partial replenishment of the growth media. Indeed, as the algae grow substrates are depleted and products accumulate until the growth ceases due to depletion of limiting nutrients. To sustain cell growth, the limiting nutrients need to be replenished by adding fresh culture medium. The original batch volume is set at 20L and fresh growing medium is to be added as needed (probably approximately 2.5L) every 2 days for the first 10 days. This volume is roughly equivalent to the volume lost over 2 days due to evaporation. The following 10 days are to be left with no replenishment of the medium and therefore a gradual depletion of nitrogen essential to synthesis. Volume lost to evaporation during these last 10 days is to be replenished using sterilized water instead. The algae, suffering nitrogen deprivation, will be subjected to stress and redirect its photosynthesis towards the production of lipids. (Rodolfi et al., 2008)

b) *Instrumentation and Control*

High productivity can only be achieved by remaining in the right temperature and pH range for the given organism. It was therefore decided to have probe holes which provide easy access to the reactor and allow measurements. This would allow readings to be made every day to

monitor the results and adjust the growing conditions accordingly. Air and CO₂ flow rates need to be monitored very carefully using flow rate controllers. A timer would be necessary to control the lighting. Temperature and pH would be measured daily, and CO₂ inputs reduced if the pH drops below 7.

c) Cleaning and Maintenance

Maintenance of the system would be greatly facilitated by having easy access to the interior of the photobioreactor. It was decided that the top of the reactor could be removable to allow inspection of the photobioreactor's interior and access to bubbling tubes.

d) Practical Considerations: Delivering Inputs and Emptying the Photobioreactor

The air flow tube was fixed to the photobioreactor using a barbed tube fitting. In the preliminary design it was determined that there would be a single large hole in the top plate and the photobioreactor would be flipped over so the medium could be dumped out. This is not feasible, because the system is relatively heavy, hard to manipulate, and fragile. Abiding to the idea of keeping things simple it was determined that a simple valve plugged at the bottom would allow for easy evacuation as gravity would do all the work. A relatively large diameter valve would have to be used to ensure that algae flocs could not clog the valve.

e) Building Materials

The fundamental criterion for designing the reactor is the material used for the photostage (main cylinder). This material should have high transparency, high mechanical strength, high durability, chemical stability, no toxicity and be low cost. Suitable materials can either be glass

or plastic. The cost of glass rules it out as a possibility for the project, so cast acrylic and polyethelene are the only two remaining cost effective solutions.

Table 1. Characteristics of photobioreactor materials and the energy content of tubular photobioreactors.

Material	Material energy content (MJ.kg ⁻¹)	Material lifespan (y)	Material density (kg.m ⁻³)	Proposed wall thickness (mm)	Energy content (MJ.m ⁻²)	Lifespan - weighted energy content (MJ.m ⁻² .y ⁻¹)
Glass	25	20	2470	1.6	310	15.5
LDPE	78	3	920	0.18	40.5	13.5
Acrylic	131	20	1180	3.0	1456	72.8

	Cast Acrylic (1/8 in)	Polyethelene (1/8 in)
Optical Refractive Index:	1.49	1.52
Light Transmission (%):	90	92
Tensile Strength (psi):	9600	6240
Modulus of Elasticity (psi):	425000	530000
Shear Strength (psi):	9000	10500
Melting Point (C):	140	136
Autoclavable :	No	No

Figure 11: Comparative properties of photobioreactor materials

As shown above the two materials, acrylic and polyethylene have similar properties. However acrylic is stronger and more durable and it was therefore selected as final choice. It should be noted that acrylic is brittle and tends to fracture easily. Cracks also expand rapidly leading to failure of the materials. This implies that the reactor should be handled carefully.

The base of the photobioreactor is to be made out of wood, since it is easy to work with and is inexpensive.

Small tubes for the air flow are to be made of tygon tubing, which is flexible, strong, non-toxic and cheap to procure.

A garden tap is decided to be the most effective, inexpensive, easy to use and widely available valve. Therefore, it is chosen as the exit valve for evacuation of the photobioreactor.

6) *Physical Model*

The design decisions made above came together as part of a proposed design which would then be built in the workshop and then tested in order to estimate its efficiency in growing *chlorella sp.* Since, living matter is at the heart of this project, it was deemed essential construct the design at the real scale and run a batch of chlorella in it in order to determine its practicality and potential. Indeed, designing a live system on a computer does not allow for an accurate representation of the system and does not bring much in terms of learning experience. Only experimental data can provide the basis of further work. Troubleshooting as well as learning how to use tools in the workshop for practical or experimental purposes or is essential in forging the skills necessary to undertake more complex projects in this field. Working in the biology lab is also a great part of the life of any scientist or engineer that wished to seriously work in this field. The construction of the photo bioreactor and its experimentation with algae therefore acts as a validation of concept and is essential in order for further work to be conducted.

a) *Deviations from Design Specifications*

In certain areas, absolute adherence to the design specifications could not be achieved. Though the design calls for plastic fittings, metal fittings were used because plastic fittings would have had to be ordered and could not be obtained within the necessary time frame. It was possible to substitute the plastic fittings with metal ones because the model was to be operated only for a short duration of time, and therefore, no adverse chemical reactions would have had time to

take place and contaminate the growing medium. Plastic pipe with $\frac{1}{4}$ " wall diameter was used instead of the specified $\frac{1}{8}$ " wall diameter, because the plastic pipe was donated, and that was what was available. The wall thickness, however, so long as it is thick enough to withstand the pressure of the growth medium within, has no effect on the functioning of the system. It influences only the cost analysis of the system.

b) Construction Challenges

Several obstacles were encountered during the construction of the physical model. The first was a persistent leakage problem. Leakage occurred at the bottom, between the two base plates. Capillary action worsened the problem and accelerated the leak. Though an o-ring had been inserted between the two plates to prevent this very thing from happening, it was found, upon dismantling the base, that the plates were not pressing the o-ring tightly enough to allow it to create a watertight seal. Several screws were protruding from the upper portion of the base, preventing the two plates from being in complete contact. The problem was remedied by sawing off the ends of these screws, adding silicone between the two plates, and re-assembling the base.

Because the tube used for the main cylinder of the photobioreactor was donated to the project, it had already been punctured with small holes along its length for previous use in another project. These were plugged with silicone, but continued to leak, and had to be re-sealed several times before the leakage was eliminated. Though this was not a problem relating to the design of the system, it consumed a great deal of time, as the silicone sealant requires a minimum of 24 hours to dry, and therefore the date at which testing the model could begin was much delayed.

c) Bubbling and turbulence

When the model was complete, the first parameter to be tested was the bubbling. Bubbling is the way uniform turbulence is achieved. It is important that the bubbles be regularly distributed, with a diameter of 5-10mm, and that there is the correct quantity of bubbling. There are no mathematical equations to predict these variables. They must be observed visually and adjusted accordingly.

As per the design specifications, a flexible tube, twice the length of the height of the photobioreactor's main cylinder, was fed through the photobioreactor in a U-shaped loop. It was punctured with 1mm diameter holes at 15mm intervals throughout its length. To observe bubbling, the photobioreactor was filled with water. It was immediately observed that when the tube filled with air, it became buoyant, and floated close to the surface, so that the lower portion of photobioreactor did not experience the effects of bubbling at all. Furthermore, it was found that, when the tube was pushed down to its proper position, the air pressure was not sufficient to cause bubbling from the lower portion of the tubing. Too much air was escaping through the punctured holes in the first quarter of the length of tubing, so that beyond that, no bubbling was produced. Finally, bubbling, when the piping was pushed down to its correct placement and the air flow increased to unrealistic levels to create bubbling throughout the piping, was unevenly distributed. Only one side of the photobioreactor was affected by the rising bubbles.

Therefore, two important modifications were made to the design. Firstly, the tubing was weighted down with a heavy ring. It was fixed in a circle, along this ring, so that the tubing rested on the bottom of the photobioreactor and well-distributed bubbling would be produced throughout the entire photobioreactor. Secondly, the tubing was puncture only along the circular portion which rested along the bottom of the photobioreactor. Therefore, no air pressure was lost due to bubbling from holes in the upper portion of the tubing.

Once the design was modified, air flow was varied to create different rates of bubbling and bubble sizes. While the bubbling rate was evaluated by visual assessment, bubbling size was measured by photographing the bubbles within the photobioreactor next to a ruler for scale. This was because the bubbles are constantly in motion. Therefore, it is much simpler to measure them from a photograph. The ideal air flow was thus determined based on the bubble size and bubbling rate that air flow generated.

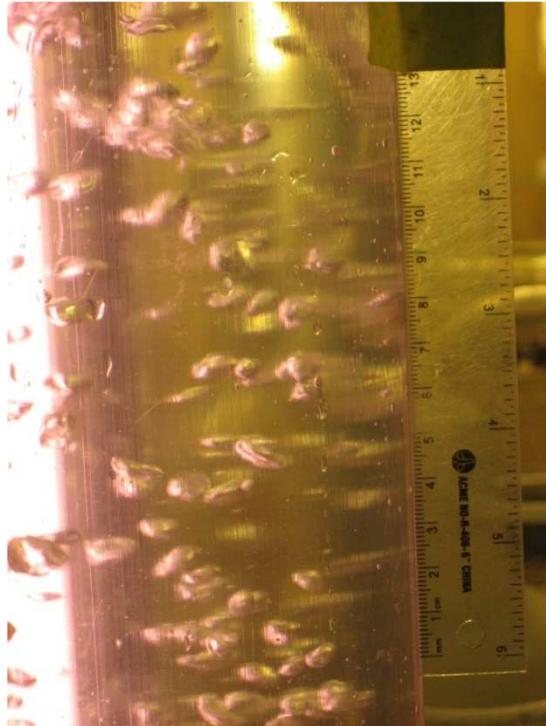


Figure 12: Example of photograph used to determine bubble size

Next, that air flow had to be converted from a gauge setting to an actual value. The gauge on the air flow valve was not calibrated, and so measurements of the air flow at various settings had to be taken to produce the gauge's calibration curve. A simple procedure was devised to do so. A clear, 1L beaker was placed upturned in a large basin. The basin was filled with enough

water to cover the beaker, and it was ensured that the beaker was filled with water and free of any air bubbles. Then, the tube attached to the air pump was fed into the basin, and into the beaker. The air flowing through the tube pushed the water out of the beaker, gradually filling it with air. The time it took for the beaker to fill to 1L with air was measured using a stopwatch.



Figure 13: Calibration of air flow rate

Left: the air valve gauge

Right: the materials used to calculate air flow

This was repeated at 4 different gauge settings, with 3 repetitions at each setting to avoid error. The resulting curve is shown below:

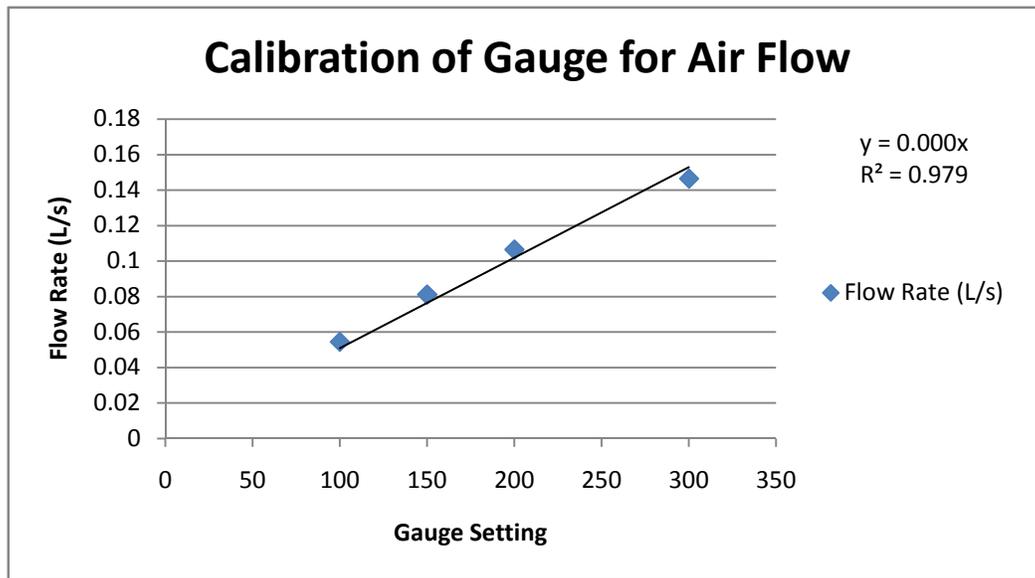


Figure 14: Calibration of gauge for airflow

This allowed the experimentally determined ideal air flow to be expressed in terms of liters per second (L/s).

d) Monitoring pH

Carbon dioxide must be input into the system to optimize the growth rate of algae. However, carbon dioxide has the negative effect of lowering the pH. Acidity negatively impacts the growth of algae. The optimal pH for algae growth lies between 7.3 and 8.3, and a pH below 6 is highly detrimental to the algae, almost halting growth. To determine approximately to what extent the introduction of carbon dioxide into the growth medium would affect the pH, the photobioreactor was filled with water and carbon dioxide was bubbled through the system at a rate of 0.0021L/s, and pH was measured every 5 minutes for 1 hour. The results are displayed in the graph below:

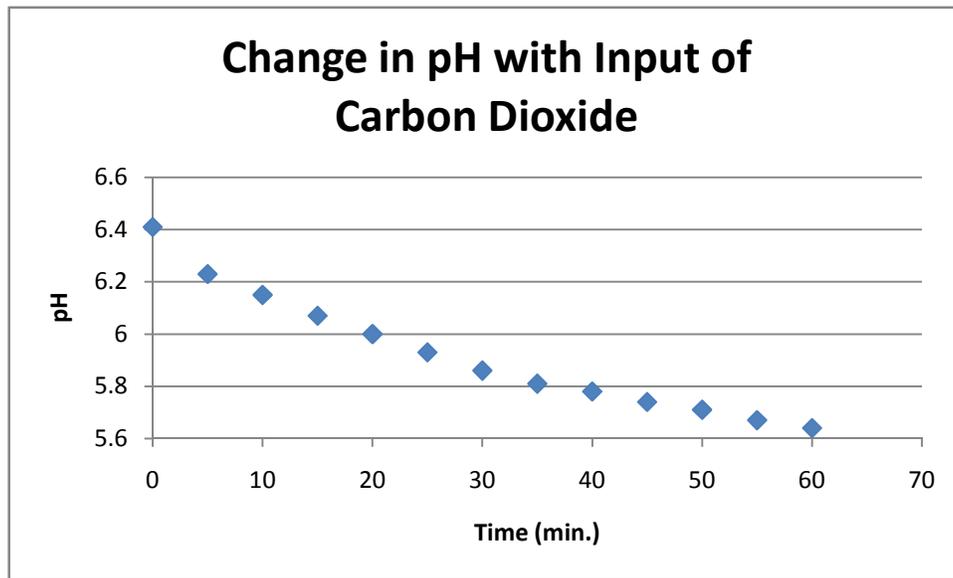


Figure 15: Change in pH as a function of carbon dioxide input

The pH dropped from 6.41 to 5.64 in 1 hour. It can be observed that the pH drops most noticeably during the first 10 minutes, and then begins to level off somewhat.

It is important to note that this experiment does not replicate exactly the actual conditions during operation of the photobioreactor. Firstly, the rate at which carbon dioxide was bubbled through the photobioreactor during the experiment was 5.5 times the actual rate at which carbon dioxide would be introduced to the system during operation of the system. This was because the simple valve on the carbon dioxide tank did not allow the flow to be reduced any further without completely shutting it off. In recognition of this, a needle valve was later added to the design. Secondly, during actual operation of the photobioreactor, much of the carbon dioxide would be uptaken by the growing algae, and would thus not affect pH. For these two reasons, it was assumed that the effect on pH would be negligible during actual operation of the photobioreactor. A recommendation was, however, included in the design of the operating procedure that pH be monitored daily. Though in the experiment, a digital pH monitor of high accuracy was used, the design recommends the use of litmus paper, as it is much less costly and is readily available at any pet store. Furthermore, the high degree of accuracy the digital pH

monitor provides is entirely unnecessary for this purpose. By monitoring pH, it would be immediately recognized if the algae were not uptaking the carbon dioxide as quickly as it were being delivered and the flow of carbon dioxide could be reduced accordingly.

3) *Experimental results : Validating the Design*

a) Challenges

A large obstacle was encountered during the operation of the physical model. According to the design procedure, the photobioreactor must first be sterilized in preparation for algae growth. This prevents contamination of the photobioreactor by eliminating both undesirable strains of algae and agents that could hinder algae growth before algae growth begins. This disinfection was conducted by flushing out the photobioreactor using a diluted alcohol solution. However, immediately upon administering this solution, cracking began to develop along the walls of photobioreactor. The alcohol reacted unexpectedly with the polyethylene and caused severe cracking in the main cylinder of the photobioreactor. Therefore, a new method of sterilizing the photobioreactor had to be devised. A literature review was conducted, and no adverse reactions of acrylic to bleach were discovered. Thus, bleach, a highly inexpensive and widely available cleaning product, replaced alcohol as the disinfecting agent in the design of the operating procedure.

The problem remained, however, that the physical model was severely compromised. Heavy duty sealant was used to patch up the cracks to the greatest extent possible, but leakage through cracks in the base could not be resolved. The photobioreactor was repaired to the extent that it could still be used to conduct a rough operating test, but continued to leak badly.

b) Biomass yields

An experimental batch of algae was grown in the photobioreactor. The algae was grown with a starting algal density of 0.021kg/m^3 and was cultivated indoors with internal lighting only (average irradiance $100\ \mu\text{mol photon m}^2/\text{s}$). Under such low lighting conditions and given our previous simulations for lighting, the specific growth rate should be approximately $0.020\ \text{h}^{-1}$. Optical density (OD) readings at 625nm wavelength were taken every day to determine the growth rate of the algae and to monitor the system performance. Other physical parameters such as temperature and pH were also monitored daily. Losses of growth medium due to leakage and evaporation amounted to $1\text{-}1.5\text{L}$ per day.

A calibration curve established the following dependence between the optical density (OD_{625}) and the dry biomass concentration (Astero Sanchez et al., 2002) (C_b , kg m^{-3}):

$$C_b = 0.38 * \text{OD}_{625}$$

The measurements of the biomass concentration were made during the daylight hours only, everyday at 4pm. This is important since the algae undergo lipid losses during dark hours therefore reducing biomass and OD readings.

$$K = \frac{\ln N_t - \ln N_o}{t}$$

The instantaneous growth rate is given by:

The dry biomass obtained after the first 10 days of culture are given below:

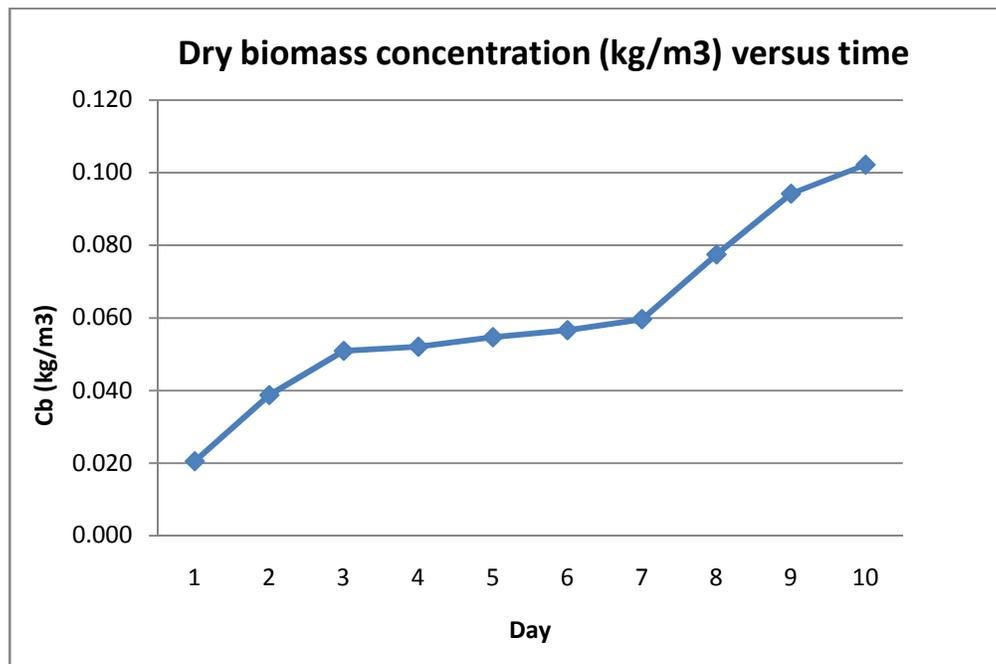
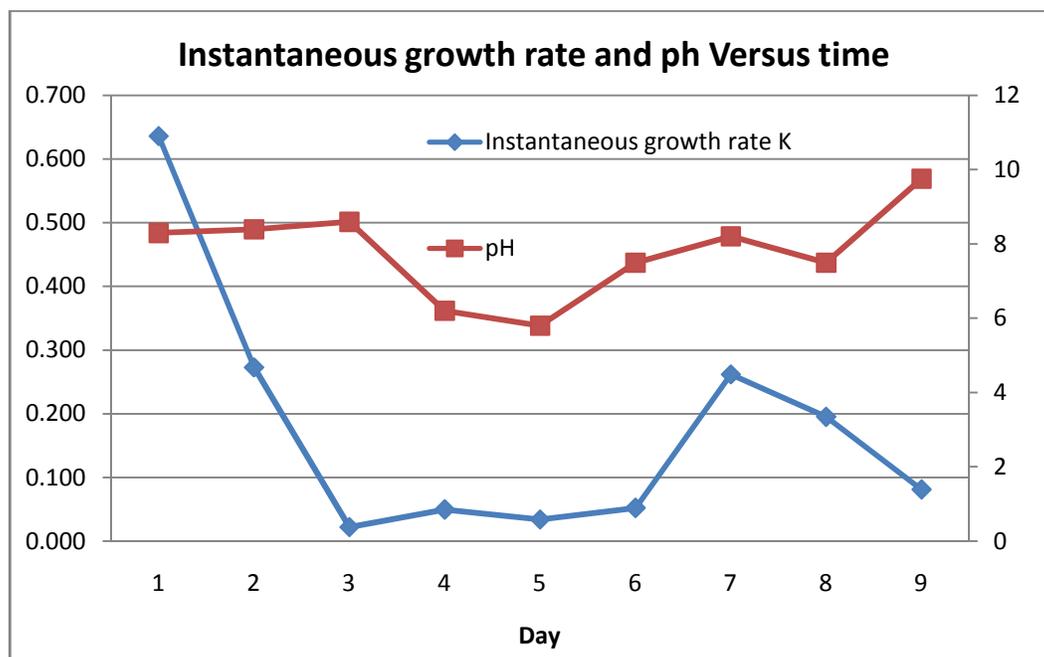


Figure 16: Dry biomass concentration vs time

The drop in the growth rate was plotted versus pH to explain the lower growth rate between day 3 and day 7:



It is clear that the experimental instantaneous growth rate is closely correlated to the pH. This is expected. Indeed, when the pH drops to less than 6, the growth rate plummets. The pH changes are closely linked to the CO₂ inputs and the CO₂ flow rate of 0.003L/s used between Day 3 until Day 5 led to excessive acidification of the medium. More careful monitoring of CO₂ inputs should be implemented. It is clear that a needle valve, which was not used, is essential to ensure proper flow rate of CO₂.

Assuming oil content of 50% on a dry weight basis, the oil yield would after the first 10 days was:

$$\text{Oil Yield} = C_b \times \text{Volume of Culture} \times 50\% = 0.104 \text{kg} \cdot \text{m}^{-3} \times 0.02 \text{m}^{-3} \times 0.5 = 1.04 \text{g}$$

c) Experimental conclusions

This clearly demonstrates that internal lighting on its own would not suffice to produce favorable results. The combination of low lighting, excessive acidification of the medium due to inputting carbon dioxide at a rate 5.5 times the design rate (due to the lack of a proper valve), and loss of growth medium due to heavy leakage, explain this low yield. Furthermore, the experiment was conducted for only 10 days, whereas a normal growth cycle is 20 days.

However, this experiment was not conducted to run the photobioreactor according to design conditions. It was conducted to ensure that an algae culture would survive and multiply within the photobioreactor, and to observe the effects of varying conditions on the productivity of the algae. In both these goals, the experimentation with the physical model was successful. It suggested that algae would flourish within the system given the right conditions, and confirmed literature stating the importance of pH control and maximizing lighting.

Final Design

1) *Dimensions*

Photobioreactor:			
<u>Dimensions:</u>			
Height:	35	inches	
Outer Tube Diameter:	8	inches	
Inner Tube Diameter:	2	inches	
Light Diameter:	1	inches	
Light Power:	32	Watts	
Top/bottom Plate Diameter:	11	inches	
<u>Tubing:</u>			
Diameter:	0.75	inches	
Length:	6	feet	
Holes:	0.0039	inches	
Hole Spacing:	0.59	inches	
<u>Exit Valve:</u>			
Valve Diameter	1	inch	Gate Valve

Base:		
<u>Dimensions:</u>		
Width:	15.75	inches
Length:	15.75	inches
Height:	6	inches

Materials
8 in plastic tube (1m)
2 in plastic tube (1.25 m)
plastic sheeting 1/4" thick (36"x24")
20 feet flexible tube
silicone sealant (10oz.)
screws (25)
particle board
2"x4" beam
plastic fittings (3)
fluorescent light and bracket
garden tap, plastic
2 outlet 4.5L/min air pump
timer
mass controller (air control valve)
CO2 detector
Aluminum CO2 cylinder
UTEX algae culture

Figure 17: Dimensions and Materials

2) Photobioreactor construction

- 1- Cut 8" diameter tube to 1m height, if necessary.
- 2- Cut the 2" diameter pipe to 1.25m.

- 3- From the acrylic sheet, cut two circular shapes with an 11" diameter.
- 4- Take these two plates and cut 1.98" holes in the center of each. Then take the 2" diameter tube and pass in the middle to make sure that it fits, but also that it is as tight as possible to aid sealing of the system.
- 5- Take the top plate and make three 0.75" threaded holes for the nutrient inputs and two 1" that will be used for the different measurement probes.
- 6- Take the bottom plate and make a 1" threaded hole. Then attach the garden tap to the bottom plate. Seal it well with silicone.
- 7- Attach the bottom plate to the outer tube with glue and seal it with silicone. The top plate only needs to fit on top of the main cylinder, and is neither glued nor sealed. Pass the inner tube through the holes in the plates and seal the bottom plate to the inner 2" diameter tube. The top is not sealed as it needs to be removed to check on the tygon tubing. Insert the input barbed tube fittings into the input holes and attach the tygon tube to one of the fittings.
- 8- The base is made out of wood. Two 2"x 4" pieces of wood and cut them into 4 lengths of 15". Put them in pairs side by side, with the 2" side resting on the particle board base and align them so that the photobioreactor sits steadily on them. Leave enough space for the exit valve. Nail these to the base. In the middle put a little block that the fluorescent light can sit on.
- 9- Break the fluorescent light out of its bracket while keeping the battery, the plug and the light connections intact.
- 10- Nail the photobioreactor to the base. Insert the light bulb into the inner tube and make sure that it sits on the block in the base.

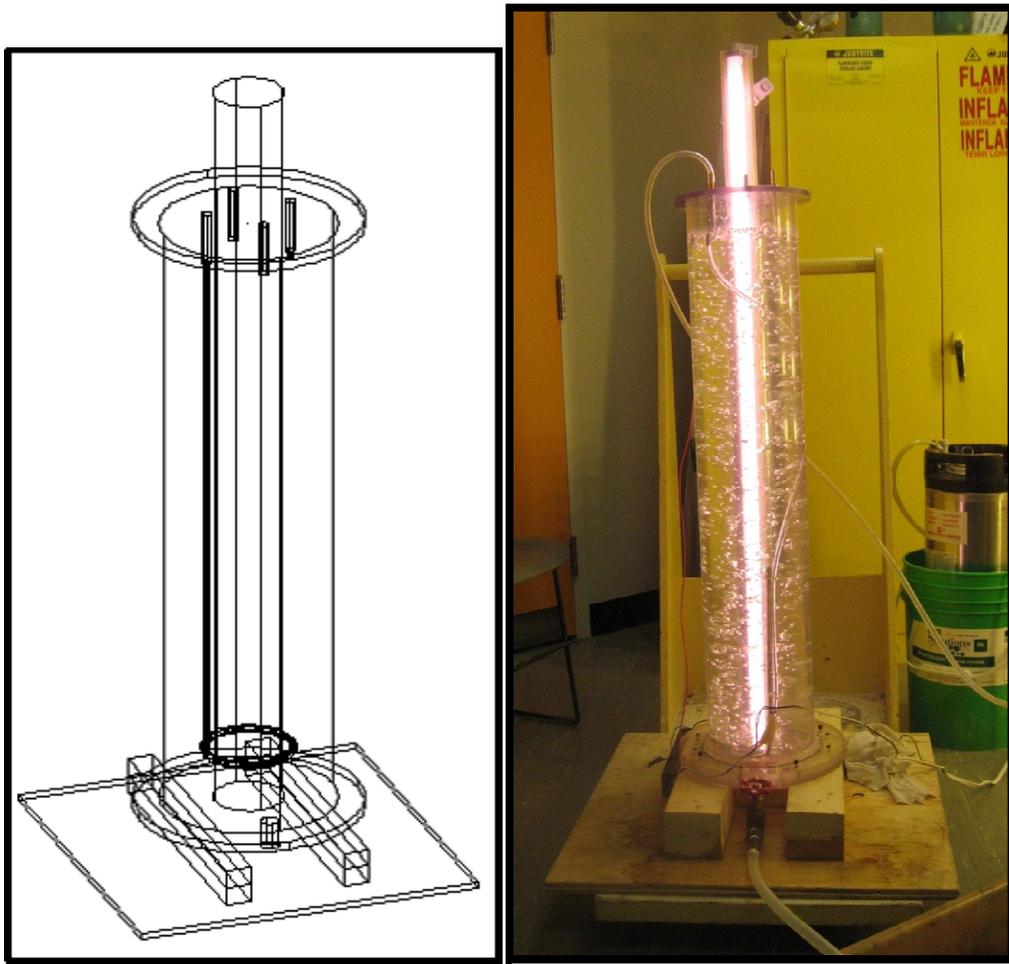


Figure 18: Autocad 3D view and photo of photobioreactor

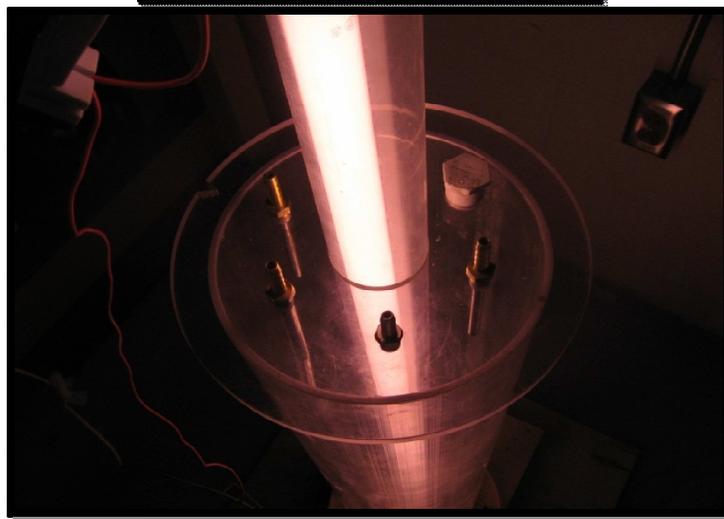
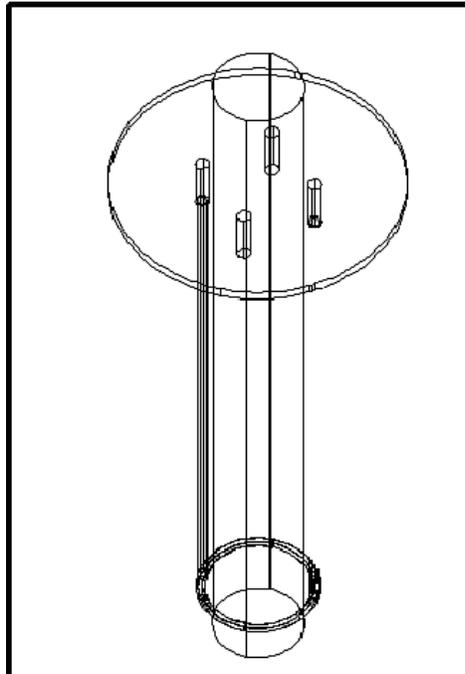


Figure 19: Autocad and photo of top of Photobioreactor

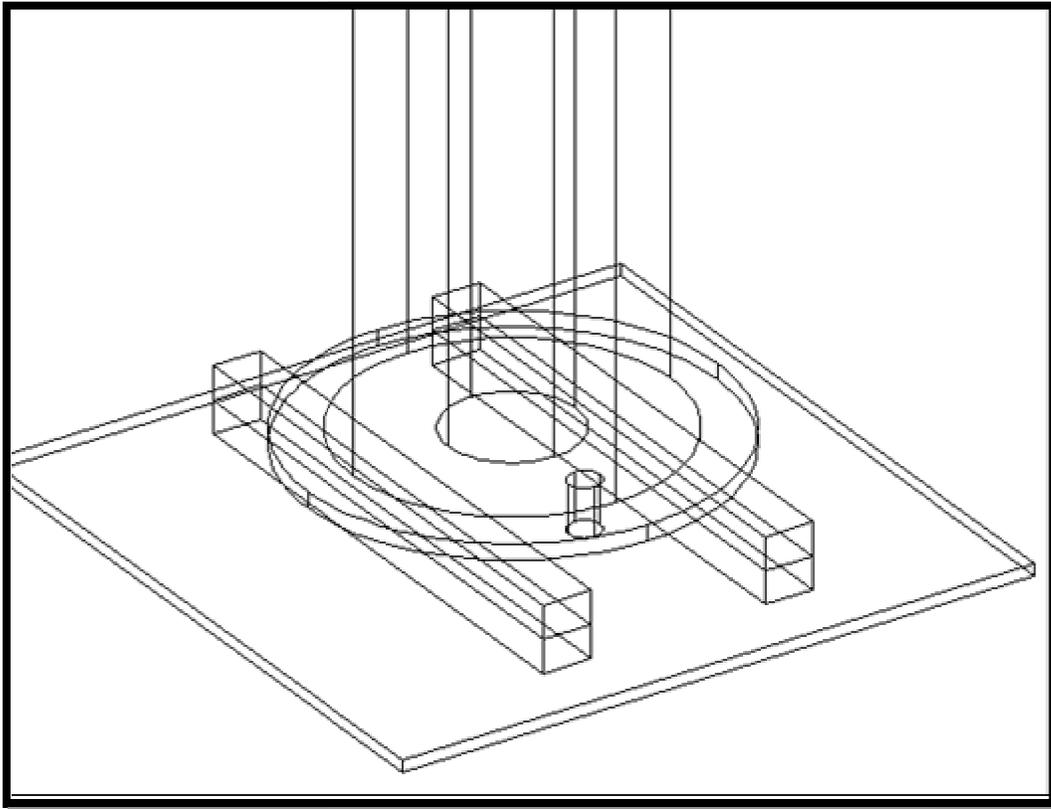


Figure 20: Autocad view of the base



Figure 21: Photo of bottom, drain valve and wooden base

3) Operating procedure

The design would be operated according to the following operating procedure:

Table 6: Operating procedure

Batch type

Total Volume	23L	
Initial volume	15 L	Add 2.5l every 2 days of fresh growing medium until day 10.
20 day cycle	10 days N rich:	
	10 days N poor:	No addition of growth medium

Light

32 W lamp optimized for plant growth (400 -700nm wavelength)

Average light intensity:	Internal lighting	100	($\mu\text{mol m}^{-2} \text{s}^{-1}$)
	Sunlight	1100	($\mu\text{mol m}^{-2} \text{s}^{-1}$)

Light cycle: 18 h of light, 6 h dark using timer

Aeration rate

Turbulent flow

Air flow rate	0.08	L/s
Gas holdup	1	%
Mass transfer coefficient	0.005	s ⁻¹
CO2 enrichment	0.5	% of volume
daily CO2 dissolved	7	g/day

Maintenance

pH

Maintain in range 7 to 8.5
Measure once a day: if too low, reduce CO2 input

Temperature

Maintain around 25 degrees +/- 3 degrees
Measure once a day

Biomass

Algae strain: *Chlorella sp.*

μ_{max}	Internal lighting	0.020 h ⁻¹
	Sunlight	0.068 h ⁻¹

Initial cell concentration: 0.020kg/m³

The main cavity of the photobioreactor must be flushed with a bleach solution prior to use, in order to prevent contamination. Add 16 mL of bleach for every liter of water. Then rinse the photobioreactor with sterilized water. An autoclave is used to sterilize water in a laboratory setting. However, a pressure cooker achieves the same result, and most households already own one. The liquid should be exposed to steam at 121 °C under 15lb pressure (103.5kPa) for 20 minutes. This will kill both bacteria and spores (Brewing Techniques, 1993). It should be

noted that boiling alone will not remove all contamination. Spores can survive in water boiled at atmospheric pressure.

Make the growth medium. According to the nutrient requirements of *Chlorella*, mix 2g fertilizer per liter of water. The growth medium must be sterilized in the pressure cooker in the same manner as the water. Note that the average-sized pressure cooker will not have the 20L capacity the photobioreactor does. Therefore, two or more batches may be required.

Add the growth medium and algae culture to the photobioreactor.

Turn on the air pump and open the carbon dioxide valve. Adjust the air pump to 0.08 L/s and carbon dioxide valve to allow 0.00038L/s flow.

Set the timer for the lighting so that the light is on for 18 hours a day, and off for 6. If the photobioreactor is housed indoors, set the overhead lighting to the same timer.

Monitor the pH and temperature daily to ensure it stays within the optimal range. If the pH falls below 7, lower the carbon dioxide flow.

Every three days, add growth medium to compensate for the loss of volume due to evaporation and the nutrients uptaken by algae.

At 10 days, stop adding fertilizer to the photobioreactor. Add only sterilized water. This will allow nitrogen deprivation to take place, and will increase the oil content of the algae.

At 20 days, harvest the algae. Flush a large basin with the same bleach solution used to sterilize the photobioreactor, and then rinse it with sterilized water. Open the valve at the bottom of the photobioreactor and allow the algae solution to flow into the basin.

Conserve 250mL of the harvested algae for the next batch.

4) Expected Results

The indoor setting was inferior to the design operating procedure and excessively high CO₂ inputs led to a severe decrease in the growth rate of the algae. Under design operating conditions, a much higher yield could be expected.

An average productivity of 0.3 g biomass/ m³/day is taken from literature (James C. Ogonna 1998). This experiment is relevant to our design since it was a bubble column photobioreactor internally illuminated using optical fibers. It was used to grow a Chlorella strain, called Chlorella Sorokiniana, outdoors in Tsukuba, Japan. This experiment showed that on a sunny day with a photon flux of 450 μmol m⁻² s⁻¹, the growth obtained was of 0.34 g dry weight of biomass /l/day. Under cloudy conditions with solar irradiance falling to 50 μmol m⁻² s⁻¹, the growth obtained was of 0.11 g dry weight of biomass /l/day. (Rouke et al., 2007)

Assuming these figures are relevant to predict the potential of our design, the biomass obtained after 20 days would be of 136g dry biomass. Assuming 50% oil content, that would translate into 68g of oil per batch in well illuminated conditions. Under poor illumination, for example if the photobioreactor were housed indoors, and external lighting was provided by overhead fluorescent lamps, a yield of 44g dry biomass after 20 days yielding 22g of oil.

Cost Analysis

There are two main costs associated with this design. The first is the initial capital cost associated with purchasing the materials to build the unit, and the second is the operating cost.

1) Initial Capital Cost

The cost of materials was calculated according to the prices any person could obtain, which is to say, not discounted due to bulk purchasing, limited time offers or professionals' rates. It is therefore a conservative estimate of the actual capital cost.

The design itself is supplied free of charge. There are no engineering costs associated with the design itself as the designers received academic credit for producing the design. It is furthermore assumed that the owner of the system will be supplying the labor free of charge. This is a realistic assumption for two reasons. Firstly, the capacity of this system is such that its yields will be large enough to meet only one person's needs. The system does not produce enough oil for this design to be operated commercially, with the intent of selling the oil. Therefore, the owner is assumed to be undertaking this project out of personal interest, and should thus be willing to provide his time free of charge. Secondly, purchasing the materials and constructing the system is a one-person job. There should be no need for additional assistance. Therefore, all labor can be assumed to be available at no cost. Below is a summary of the initial cost of building the photobioreactor.

Table 7: Total cost of fabricating the photobioreactor

Component	Unit	Price per Unit	Quantity required	Total
8 in plastic tube (1m)	5ft	\$236.10	1m	\$154.92
2 in plastic tube (1.25 m)	ft	\$5.14	1.25m	\$21.08
plastic sheeting 1/4" thick (36"x24")	36"x24"	\$56.75	1 sheet	\$56.75
20 feet flexible tube	ft	\$0.33	20ft	\$6.60
silicone sealant (10oz.)	10 oz. tube	\$6.18	1 tube	\$6.18
screws (25)	100	\$12.00	25	\$3.00
particle board	2'x4'	\$40.00	2'x2'	\$20.00
2"x4" beam	ft	\$0.45	6ft	\$2.70
plastic fittings (3)	1 fitting	\$1.35	3	\$4.05
fluorescent light and bracket	1 bracket & 3 bulbs	\$30.00	1 bracket & 1 bulb	\$30.00
garden tap, plastic	1 tap	\$6.21	1 tap	\$6.21
2 outlet 4.5L/min air pump	1 pump	\$25.00	1 pump	\$25.00
timer	1 timer	\$11.00	1 timer	\$11.00
mass controller (air control valve)	1 valve	\$3.25	2 valves	\$6.50
CO2 detector	1 detector	\$45.00	1 detector	\$45.00
Aluminum CO2 cylinder	1 cylinder	\$74.95	1 cylinder	\$74.95
UTEX algae culture	1 living culture	\$75.00	1 culture	\$75.00
Total Cost				\$548.94

The initial capital cost can furthermore be broken down into two subdivisions: the cost of building the first unit, and the cost of every unit thereafter. Because there are a number of one-time expenses, such as the carbon dioxide tank and the carbon dioxide monitor, the cost per unit decreases with the construction of multiple units.

Table 8: Onetime costs

One Time Costs	
Timer	\$11.00
Mass Controller (air & CO2 control valves)	\$6.50
CO2 detector	\$45.00
Aluminum CO2 cylinder	\$74.95
Algae Culture	\$75.00
Total	\$212.45

Cost of 1st Unit	Cost of nth Unit
\$568.94	\$356.49

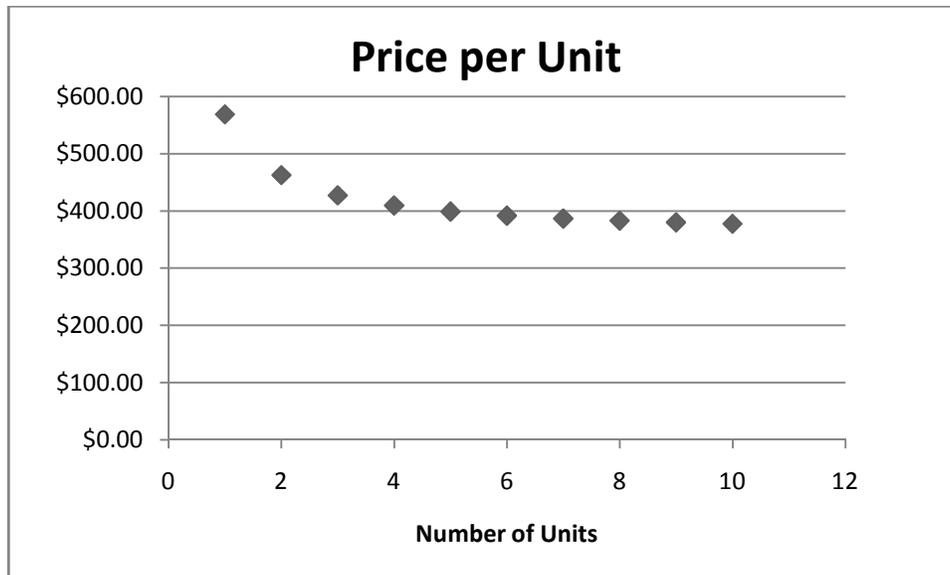


Figure 22: Price per unit

This could be maintained to a maximum of approximately 10 units, at which point the efficiency of the pump and the holding capacity of the carbon dioxide tank would have to be re-

evaluated, and the components possibly replaced. In the case of many units, replacing the carbon dioxide tank with a larger model might be desirable because the 5lb tank will have to be refilled frequently. With so many units, it would likely also be most cost effective to buy one larger pump than to continue purchasing multiple smaller pumps.

2) Operating Cost

The operating cost arises due to the cost of nutrient and energy inputs to the system. It varies linearly with the number of units being operated. The operating cost was calculated according to two climatic scenarios.

a) *Warm Climate*

The first is a warm climate, where temperatures are consistently high (25° C) year-round. Such a climate would allow the photobioreactor system to be stationed outside year-round. There are two consequences to such a scenario. Being outside would expose the photobioreactor to intense direct sunlight. Therefore, the algae would be approximately three times more productive. Yields would thus increase threefold. However, the algae would accordingly require three times more nutrient inputs, tripling the cost of nutrients and carbon dioxide.

Table 9: Operating cost for warm climate

WARM CLIMATE			
	Unit	Price per Unit	Price per Growing Cycle
Fertilizer (liquid, 20-2-3)	4 gallon container	\$135.35	\$9.16
CO ₂	5 lbs	\$12.00	\$5.44
electricity for light (32 W) kWh/day	kWh	\$0.07	\$0.78
electricity for pump	kWh	\$0.07	\$0.15
Total			\$15.53

According to the expected yield of 68g of oil, the operating cost of producing one liter of oil is \$228.38.

b) Cool climate

The second scenario is a cool climate, or a climate where the temperature is periodically too cold for algae to flourish, such as Canada's own climate. In this climate, the photobioreactor system would have to be kept indoors. Though the weather may be suitable for the photobioreactor system for extended stretches of time in such a climate, if there are many units, the system may be too unwieldy and difficult to transport, even when empty, to make moving it outdoors worthwhile. As there is no monetary cost associated with such an operation as moving the system from indoors to outdoors, it would be purely at the owner's discretion whether or not the task is worthwhile.

While the photobioreactor system resides indoors, fluorescent lighting, which provides much less intensity than direct sunlight, is used to simulate natural sunlight. This is a disadvantage in two ways. Not only is there a cost associated with the fluorescent lighting, whereas sunlight is free, there is a loss in algae productivity. Although the operating cost is much lower when the system is indoors, the operating cost per liter of oil yielded is actually higher because productivity is so negatively affected.

Table 10: Operating cost for cold climate

COOL CLIMATE			
	Unit	Price per Unit	Price per Growing Cycle
Fertilizer (liquid, 20-2-3)	4 gallon container	\$135.35	\$3.05
CO ₂	5 lbs	\$12.00	\$5.44
electricity for light (32 W) kWh/day	kWh	\$0.07	\$3.14
electricity for pump	kWh	\$0.07	\$0.15
		Total	\$11.78

According to the expected yield of 22g of oil per batch, the cost of producing one liter of biodiesel is \$535.45. Therefore, in terms of operating cost, the efficiency of the system is highest in a warm climate where the photobioreactor can be housed outdoors.

c) *Comparison with Gasoline*

The price of gasoline was 95.2 cents per liter on January 27, 2009. When the price peaked last summer, it was at \$1.43 a liter (May 27, 2008) (CBC News, 2009).

Therefore, the cost of producing biodiesel using this design system is not comparable to the current price of fossil fuels. However, below is a projection of the cost of gasoline prices in the United States over the coming years.

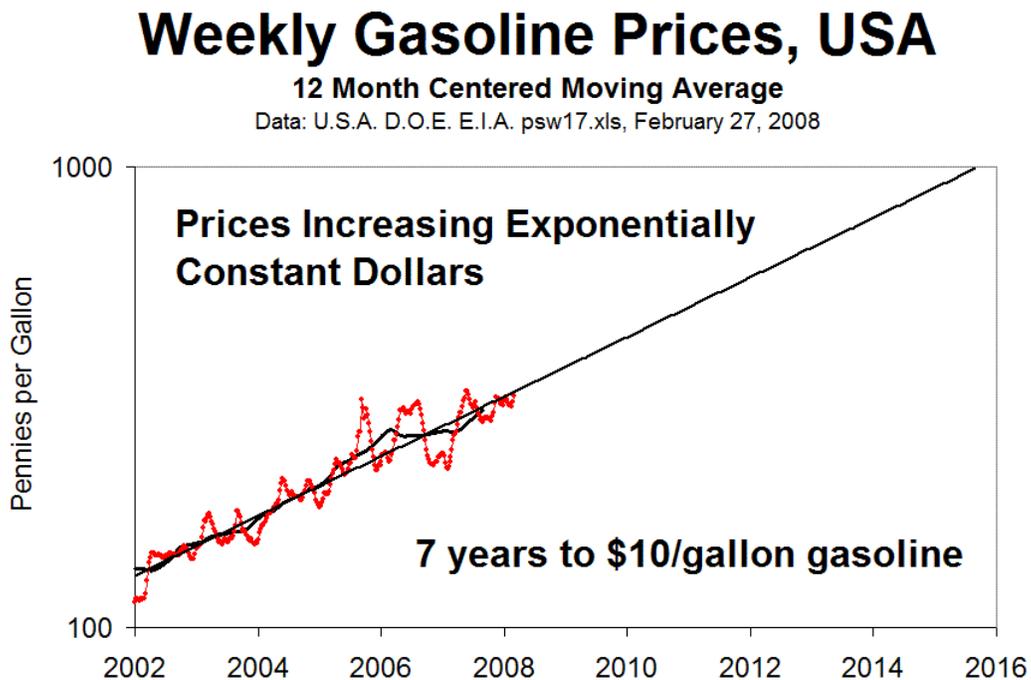


Figure 23: Gasoline prices
([Live Journal](#), 2008)

Though this projection applies to American gasoline prices, and was calculated before the current economic crisis led to a substantial drop in gasoline prices, it demonstrates that, on a large time scale, the price of gasoline rises exponentially. Therefore, as time progresses, this system will begin to look more and more attractive from an economic perspective.

d) Comparison with Similar Systems

Though the system is far from comparable with current gasoline prices, literature values for the cost of building and operating a similar unit demonstrate that the algae cultivation system designed is in fact considerably more affordable. The following table summarizes a comparison of this design and standard literature values for the annual cost of the system:

Table 11: Annual cost of a 200L system

Annual Cost of a 200L System		
	This Design	
Standard Cost	Warm Climate	Cool Climate
\$1,415.80	\$429.41	\$403.86

This cost includes the annual operating cost of 10 photobioreactors, as well as the initial cost of 10 photobioreactors, depreciated over the expected 20 year lifespan of the design. 10 photobioreactors were required to match the 200L system it was compared to. The comparative affordability of this design can largely be attributed to two factors. Firstly, all labor for both the construction and operation of the system are provided free of charge by the owner. Secondly, great effort was made to use affordable, readily available materials without compromising the productivity of the system. Therefore, in every aspect, productivity was maintained at a high level while using a less costly structure.

3) Improving the Economic Viability of the Designed System

The economic viability could be greatly improved if two measures were adopted. Firstly, carbon credits are a key component to the current attempts to reduce or stabilize the concentration of greenhouse gases in the atmosphere. Because biodiesel produces less harmful emissions than traditional gasoline, carbon credits would help offset the discrepancy in price between this system and gasoline. Secondly, a large portion of the operating cost is incurred from the purchasing of carbon dioxide. If a free or inexpensive source of carbon dioxide were available, this would greatly reduce the operating cost. Such a source would arise for example, by capturing the waste exhaust from an industrial process. Though this is currently practiced, the practice is not widespread, and the captured carbon dioxide is usually injected into the ground rather than given away or sold. Finally, there is currently an abundance of research being conducted on the genetic engineering of algae strains. Should a new, more productive strain emerge from this research, the productivity of the design system would increase, lowering the cost per unit of oil produced.

Conclusion

Algae is an alternative fuel source of great potential. This design enables any person to cultivate algae in their own residence. Though the design is simple to execute, it utilizes the same principles and techniques as the most advanced research institutes.

Expected yields are notably lower than those achieved in research centers, but both initial and operating costs are a mere fraction of those quoted in literature. The design compares well with others in terms of performance per cost, but is far simpler to construct and operate.

Building a physical model of the system allowed us to observe the design in action and improve it in significant ways. The result is a refined design for a smoothly-running system whose productivity will continue to increase as new algae strains are developed.

Future development of the project would include extending the design to include the harvesting and oil extraction processes. The end result could be a system that would allow an individual to produce his own renewable transport fuel in a completely sustainable manner.

Bibliography

Abedon, S. T. (2003). "Important words and concepts from Chapter 6, Black, 1999 ", from <http://www.mansfield.ohio-state.edu/~sabedon/black06.htm>.

Acién Fernández, F .G., J .M. Fernández Sevilla, J.A. Sánchez Pérez, E. Molina Grima and Y. Chisti. 2001. Airlift-driven external-loop tubular photobioreactors for outdoor production of microalgae: assessment of design and performance. *Chemical Engineering Science*. 56: 2721-2732.

AlgaeLink. "Algae Growing Systems."The Netherlands. 20 Nov. 2008

<http://www.algaefuels.org/algaepond.htm>.

Amos Richmond, Z. C.-W., Yair Zarmi (2003). "Efficient use of strong light for high photosynthetic productivity: interrelationship between the optical path, the optimal population density and cell-growth inhibition." *Biomolecular Engineering* **20**: 229-236.

Augenstein, D.C., J.R. Benemann, R.P. Goebel and J.C. Weissman. 1982. "Microalgae as a source of liquid fuels." U.S. Department of Energy, Office of Energy Research.

Augero, E. J., I. G. Borlongan and O. M. Millamena. "Techniques on Algae Harvesting and Preservation for Use in Culture and as Larval Food." *Aquacultural Engineering*. 9 (1990): 295-304.

Assaf Sukenik, O. Z., Yael Carmeli (1993). "Biochemical quality of marine unicellular algae with special emphasis on lipid composition. II. *Nannochloropsis* sp " *Aquaculture* **117**: 313-326.

Asterio Sánchez Mirón a, M.-C. C. G. i. a. and E. M. G. a. Francisco Garc'ia Camachoa, Yusuf Chisti b, (2002). "Growth and biochemical characterization of microalgal biomass produced in bubble column and airlift photobioreactors: studies in fed-batch culture." Enzyme and Microbial Technology **31** 1015-1023.

Becker, E. W. (1994). Microalgae: Biotechnology and Microbiology, Cambridge University Press.

Bitaubé Pérez, E., I. Carp Pina, L. Pérez Rodriguez (2008). " Kinetic Model for Growth of *Phaeodactylum tricornutum* in Intensive Culture Photobioreactor" Biochemical Engineering Journal **40**: 520-525.

Borowitzka, Michael. "Culturing Microalgae in Outdoor Ponds." Algal Culturing Techniques. Burlington, Mass. : Elsevier/Academic Press, 2005.

Chisti, Y. (2007). "Biodiesel from microalgae." Biotechnology Advances **25**: 294-306.

Dempster, T. A., K. M. McGinnis and M. R. Sommerfeld. "Characterization of the growth and lipid content of the diatom *Chaetoceros muelleri*." Journal of Applied Phycology 9.1 (1997): 19-24.

Erikson, N.T. 2008. The technology of microalgae culturing. Biotechnol Lett. 30: 1525-1536.

G. Chini Zittelli, F. L., A. Bastianini, L. Rodolfi, M Vincenzini, M.R. Tredici (1999). "Production of eicosapentaenoic acid by *Nannochloropsis* sp. cultures in outdoor tubular photobioreactors " Journal of Biotechnology **70**: 299-312.

Huesemann, Michael and Chad Weldy. "Lipid Production by *Dunaliella Salina* in Batch Culture: Effects of Nitrogen limitation and light intensity." U.S. Department of Energy

Incropera, F. P. and J. F. Thomas. "A model for solar radiation conversion to algae in a shallow pond." Solar Energy. 20 (1978): 157-165.

Journal of Undergraduate Research. U.S. Department of Energy. 20 Nov. 2008

http://www.scied.science.doe.gov/SciEd/JUR_v7/pdfs/Lipid%20Production%20by%20Dunaliella.pdf.

James C. Ogonna, T. S., Hideo Takana (1998) (1998). "An illuminated solar and artificial light system for internal illumination of photobioreactors." Journal of Biotechnology **70**(1999): 289-297.

Janssen, M., J. Tramper, L.R. Mur and R.H. Wijffels. 2003. Enclosed Outdoor Photobioreactors: Light Regime, Photosynthetic Efficiency, Scale-Up, and Future Prospects. Biotechnology and Bioengineering. 81: 193-208.

John L. Ingraham, C. A. I. (2000). Introduction to Microbiology Pacific Grove, CA 93950 USA.

John Sheehan, T. D., John Benemann, Paul Roessler (1998). A Look Back at the U.S. Department of Energy's Aquatic Species Program—Biodiesel from Algae. U. S. D. o. E. s. O. o. F. Development, National Renewable Energy Laboratory.

K. Miyamoto, O. W. a. J. R. B. (1988). "Verical column reactor for microalgae cultivation " Biotechnology Letters **10**: 703-708.

Laliberte, G., J. de la Noue and D. Proulx. "Algae and waste water." Journal of Applied Phycology. 4 (1992): 247-254.

Liu, Z., Wang, G., and Zhou, B. (2007). "Effect of iron on growth and lipid accumulation in *Chlorella vulgaris*". Bioresource Technology 99:4717-4722.

Michael A Borowitzka, L. J. B. (1988). Micro- algal biotechnology Cambridge University Press.

Miyamoto, K., O. Wable and J.R. Benemann. 1988. Vertical Tubular Reactor For Microalgae Cultivation. *Biotechnology Letters*. 10: 703-708.

Molina Grima, E., A. Fernández, F. García Camacho and Y. Chisti. 1999. Photobioreactors: light regime, mass transfer, and scaleup. *Journal of Biotechnology*. 70: 231-247.

Oceanography, T. A. S. o. L. a. (1999). "The nutrient stoichiometry of benthic microalgal growth: Redfield proportions are optimal." *Limnology and Oceanography* 44 (2): 440-446.

Oswald, W. J. "Large-scale algal culture systems (engineering aspects)." *Micro-Algal Biotechnology*. Cambridge University Press. (1988) 357-394.

Pienkos, Philip. "The Potential for Biofuels from Algae." *Algae Biomass Summit*. 15 Nov. 2007. National Renewable Energy Laboratory.

Raines, Maribeth. (1993). "Methods of Sanitation and Sterilization". *Brewing Techniques*, 1(2).

Richmond, A., S. Boussiba, A. Vonshak and R. Kopel. 1993. A new tubular reactor for mass production of microalgae outdoors. *Journal of Applied Phycology*. 5: 327-332.

Rodolfi L, B. N., Parente I, Padovani G, Bonini G, Chini Zittelli G, Biondi and T. N (2008). "Lipid production from marine microalgae: Strain selection, induction of lipid synthesis and outdoor cultivation in a Low-Cost photobioreactors." *Biotechnology and Bioengineering*.

Tawfiq Institute for Scientific Research , M., Jaber Al-Shimmari & Peter Dias (1999). "Optimum production conditions for different high-quality marine algae " Hydrobiologia **403**: 97-107.

"The Changing Cost of Gasoline: Pump Prices Across the Country". (March 3, 2009). CBC.ca <<http://www.cbc.ca/consumer/story/2008/11/17/f-map-pumprices.html>>.

Live Journal. (2008). "The Story Behind Gas Prices". (March, 2009). <<http://valuesystem.livejournal.com/tag/gasoline+prices>>.

Tredici, M.R. and G.C. Zittelli. 1998. Efficiency of Sunlight Utilization: Tubular Versus Flat Photobioreactors. *Biotechnology and Bioengineering*. 57: 187-197.

Ugwu, C.U., H. Aoyagi and H. Uchiyama. 2008. Photobioreactors for mass cultivation of algae. *Bioresource Technology*. 99: 4021-4028.

Science, B. (2004). *Handbook of Microalgal Culture Biotechnology and Applied Phycology*. A. Richmond, Blackwell publishing Company.

S. John Pirt, Y.-K., Amos Richmond and Margaret Watts Pirt (1980). "The Photosynthetic Efficiency of Chlorella Biomass Growth with Reference to Solar Energy Utilisation " J. Chem. Tech Biotechnolo. **30**: 25-34.

Vasquez, V. and P. Heussler. "Carbon dioxide balance in open air mass culture of algae." Arch. Hydrobiol. Ergeb. Limnol. 20 (1985): 95-113.

Zhi-Yuan Liu, G.-C. W., Bai-Cheng Zhou (2008). "Effect of iron on growth and lipid accumulation in Chlorella vulgaris " Bioresource Technology 99: 4117-4722.

APPENDIX I: Complementary Literature Review

APPENDIX II: Growth Media Used

APPENDIX III: Calculation Spreadsheets