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Microalgae for CO_2 Capture and Its Biomass: A Mini Review

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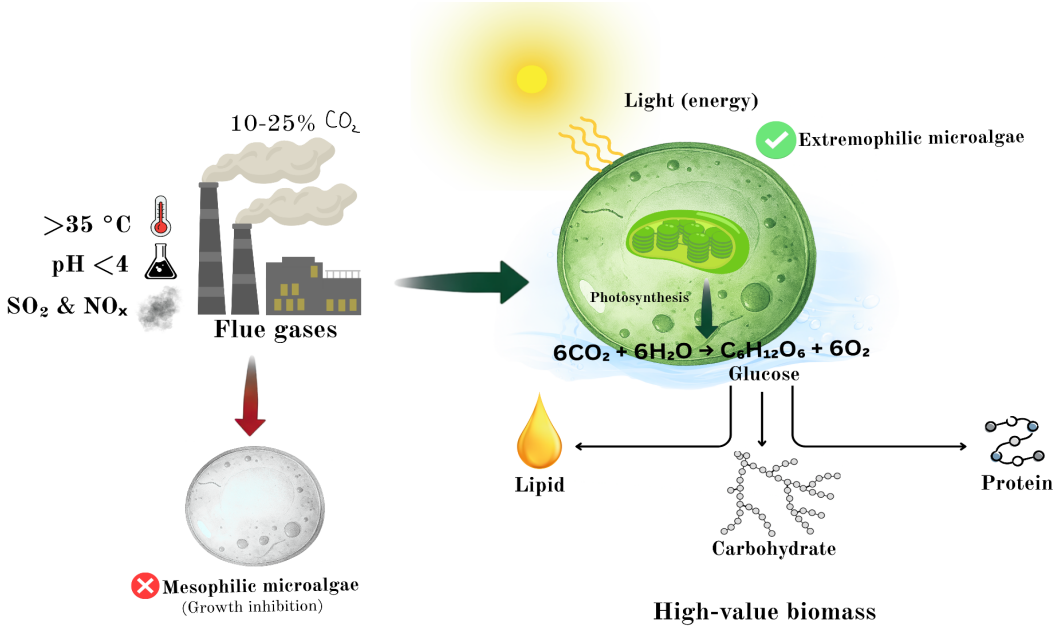
GRAPHICAL ABSTRACT



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ABSTRACT

The continuous increase in carbon dioxide (CO_2) emissions has raised atmospheric CO_2 concentrations to over 420 ppm. This has become one of the main drivers of global climate change, underscoring the need for efficient, sustainable mitigation strategies. Microalgae offer great potential for CO_2 capture. As photosynthetic organisms, microalgae use CO_2 for growth. They are reported to have photosynthetic efficiencies 10–50 times higher than those of terrestrial plants and to grow in various media, making them superior for carbon utilization. However, most conventional microalgae cannot thrive under extreme conditions, such as low pH (<4), high temperatures (>35 °C), high CO_2 levels (>10%), or the presence of pollutants like sulfur dioxide (SO_2) and nitrogen oxides (NO_x). Extremophilic microalgae are species capable of surviving in highly acidic conditions, elevated temperatures, and environments contaminated with SO_2 and NO_x. This makes them highly promising candidates for CO_2 capture under extreme conditions. Furthermore, microalgae capture CO_2 and convert it into high-value biomass, thereby enhancing their value as CO_2 capture agents.

1. INTRODUCTION

Global warming has numerous negative impacts on human life, including rising sea levels, extreme weather, the spread of disease, and threats to food security and the economy [1]. The primary cause of global warming is the high concentration of greenhouse gases in the atmosphere. One of the dominant greenhouse gases is carbon dioxide (CO_2), which accounts for 73.5%. Furthermore, in 2024, the power industry was reported as the largest contributor to greenhouse gas emissions, accounting for 38.1%. In addition to high CO_2 levels, flue gas from power plants, such as coal-fired power plants, is acidic, hot, and contains harmful SO_2 and NO_x gases [2].

Increased emissions from various sectors have significantly increased atmospheric CO_2 levels over the past few decades [3]. Currently, global CO_2 concentrations have exceeded 420 ppm, a level significantly higher than pre-industrial levels, indicating an imbalance in Earth's carbon cycle. This imbalance is a major driver of global climate change, with a broad impact on the stability of terrestrial and aquatic ecosystems [4]. Therefore, CO_2 capture mitigation requires an integrated, efficient, scalable, and sustainable strategy to reduce the rate of greenhouse gas emissions into the atmosphere. One promising approach to carbon emission mitigation is the utilisation of photosynthetic organisms, such as microalgae, which can efficiently fix CO_2 through photosynthesis.

Microalgae are microorganisms capable of utilising CO_2 as a carbon source in photosynthesis. Microalgae have been reported to potentially achieve higher photosynthetic efficiency than land plants [5]. Furthermore, microalgae can be cultivated in various media, such as freshwater, seawater, and wastewater [6], and have a promising biomass profile. Each gram of microalgae biomass is reported to fix approximately 1.83 grams of CO_2, making it an effective and sustainable mitigation agent [7]. The CO_2 capture capacity of microalgae depends on how environmental conditions affect their growth.

The pH and temperature are important factors influencing microalgal growth. Most microalgae grow optimally at neutral pH and room temperature. At pH less than 4, most conventional mesophilic microalgae cannot survive due to ionic stress and cell wall damage [8]. The optimal temperature for microalgae growth generally ranges from 25 to 30 °C. Temperatures above 35 °C can denature photosystem proteins and reduce photosynthetic efficiency [9]. Together, these stressors represent major limiting factors for microalgae cultivation under extreme environmental conditions.

Coal-fired power plants produce exhaust gases with high CO_2 content (10–25%) [10-12], acidic condensate (pH <4) [13, 14], high temperatures (>120 °C) [15], and the presence of sulphur dioxide (SO_2) and nitrogen oxides (NO_x) [16] reflects extreme conditions. In general, microalgae grow optimally at CO_2 concentrations between 5–10%, such as *Spirulina* sp., *Spirulina platensis*, and *Chlorella sorokiniana*. At higher CO_2 concentrations, the growth slows, even decreasing drastically [17-19]. On the other hand, SO_2 is toxic to most microalgae, even at low levels (50 ppm), such as *Chlorella sp*. ABC [20]. At concentrations of 100 ppm, most microalgae can barely grow [21]. Meanwhile, the inhibitory effect of NO_x on microalgae growth is both concentration and strain-dependent. NO_x can be absorbed by the cultivation medium, where it is oxidized to NO_2^-, and can thus be further utilized as a nitrogen source [22-24]. The acidic conditions, high temperatures, and presence of SO_2 in coal-fired power plant exhaust flue gases necessitate pre-treatment steps to adjust the pH, lower the temperature, and remove SO_2 before the CO_2 capture process by microalgae.

Microalgae's intolerance to CO_2 concentrations exceeding 10%, acidic pH, high temperatures, and the presence of SO_2 and NO_x are limiting factors in their use as CO_2 capture agents. Therefore, microalgae capable of effectively capturing CO_2 under these extreme conditions are needed to support the implementation of more efficient microalgae-based mitigation technologies.

2. EXPERIMENTAL METHODS

The literature used in this mini review was collected from several scientific databases, including Scopus, Google Scholar, and SciFinder. The search was conducted using keywords such as “microalgae for CO_2 capture”, “extremophilic microalgae as CO_2 capture agent”, “flue gas”, and “extremophilic microalgae biomass”. Studies focusing on microalgae-based CO_2 capture, the potential of extremophilic microalgae under flue gas or high-CO_2 conditions, and the promise of their biomass were included. The retrieved articles were screened based on their titles, abstracts, and full

texts. Relevant information from the selected studies was comparatively analyzed to identify challenges and research gaps in the application of microalgae for CO_2 mitigation.

3. MEASUREMENT APPROACHES IN MICROALGAE AS CO_2 CAPTURE RESEARCH

3.1 Microalgae growth determination

Microalgae growth can be determined by counting cells using a haemocytometer or a flow cytometer, measuring optical density (OD) at a specific wavelength (670–690 nm), or weighing dry biomass [17, 18]. A haemocytometer counts cells directly under a microscope, providing a more accurate estimation of cell density and allowing observation of cell morphology. However, this method is relatively time-consuming, requires high precision, and is less efficient when many samples need to be analyzed. In contrast, the OD method uses a spectrophotometer to measure the absorbance or turbidity of a cell suspension, making it faster, more practical, and suitable for routine monitoring of culture growth. Nevertheless, this method does not directly measure cell numbers, and its results depend on a calibration curve derived from the relationship between OD values and cell numbers previously determined using a haemocytometer. In addition, OD measurements can be influenced by other factors such as cell size, pigments, and cell aggregation. Meanwhile, dry weight can accurately measure biomass and be used to validate other methods. However, this method requires large sample volumes (3–10 mL of culture), is destructive and time-consuming (centrifugation, washing, and drying at 80°C), and is less effective for low-density cultures [25, 26]. In the reviewed studies, biomass was commonly calculated using Equation 1, and biomass productivity was calculated using Equation 2.

Specific growth rate (Equation 1):

$$\mu\ (\text{day}^{-1}) = \frac{\ln(X_2) - \ln(X_1)}{\Delta t}$$

μ : specific growth rate
X_1 : initial biomass concentration at the initial time (t_1)
X_2 : final biomass concentration at the final time (t_2)
Δt : time interval between measurements ($t_2 - t_1$)

Biomass productivity (Equation 2):

$$P_{\text{Biomass}} = \frac{X_2 - X_1}{t_2 - t_1}$$

P_{biomass} : biomass productivity
t_1 : initial time
t_2 : final time
X_2 : biomass concentrations measured at t_2
X_1 : biomass concentrations measured at t_1

3.2 CO_2 capture quantification

Two primary approaches commonly used to quantify CO_2 fixation in microalgae cultivation are the biomass-based (indirect) method and direct gas measurement. The indirect method estimates CO_2 fixation by multiplying biomass productivity by the biomass carbon fraction, which is either assumed to be approximately 50% of dry weight or determined experimentally using an elemental analyzer (Equation 3) [17, 27]. The direct method estimates CO_2 fixation by measuring changes in gas concentration within the chamber [28]. CO_2 fixation rate by the indirect method can be calculated using Equation 3.

CO_2 fixation rate (Equation 3):

$$R_{CO_2} = C_C \cdot P_{bio} \cdot \frac{M_{CO_2}}{M_c}$$

C_C : carbon content of the microalgal biomass (% w/w)
P_{bio} : the average biomass productivity
M_{CO2} : CO_2 molecular weights
M_C : carbon molecular weights

Measuring the change in CO_2 concentration in the chamber directly can also be done using the FL23 Algal CO_2 Package (Qubit). The FL23 Algal CO_2 Package measures CO_2 exchange by enclosing liquid biomass in a sealed chamber and monitoring changes in headspace CO_2 concentration over time. The rate of CO_2 consumption by microalgae can be determined using linear regression in LoggerPro, yielding the initial reaction rate (V_0) [29].

The reliability of indirect estimates of CO_2 fixation is constrained by the variability in carbon content across different microalgal species and growth conditions, making fixed assumptions potentially misleading [27]. To improve the accuracy of biomass-based calculations, the carbon content of microalgal biomass can be directly determined using an elemental analyzer, enabling CO_2 fixation to be estimated from the actual carbon fraction rather than assumed values. However, indirect measurement still does not fully represent the total carbon flow in the system because the supplied carbon can undergo other transformation pathways, for example, into inorganic carbon forms such as carbonates or other carbon-containing compounds, and therefore is not entirely accounted for as microalgal biomass. When direct measurement is employed, CO_2 uptake is instead inferred from real-time monitoring of gas-phase CO_2 dynamics within the cultivation system, offering a more condition-responsive quantification approach [27].

3.3 Microalgae biomass characterization

Protein content in microalgal biomass can be estimated using several methods. One commonly used method is the Kjeldahl method, which measures total nitrogen and converts it to protein using a nitrogen-to-protein conversion factor. However, the commonly used factor of 6.25, originally developed for animal and terrestrial plant proteins, may not be suitable for microalgae because not all nitrogen in microalgal biomass is derived from protein [30]. Alternatively, colorimetric methods such as the Bradford, Lowry, and BCA assays quantify soluble proteins by interpolation from calibration curves prepared with protein standards, typically bovine serum albumin (BSA). However, the measured protein content can vary depending on the protein standard used. It may also be affected by the presence of interfering substances in the sample extract, which can modify absorbance readings and lead to inaccurate estimations [31]. Carbohydrate content can be quantified using two approaches, depending on the target fraction. Total carbohydrate is commonly measured using the phenol–sulfuric acid method, in which freeze-dried samples are first hydrolyzed with hydrochloric acid (0.2 M) at 85°C for 1 hour, followed by colorimetric detection. Reducing sugars can be determined separately using the Fehling test after neutralization with sodium hydroxide, or quantified using a biochemical analyzer (YSI 2700, YSI Inc., USA). Lipid content was typically extracted using the chloroform-methanol mixture (Folch method) or the chloroform-methanol-water mixture (Bligh and Dyer method) [32].

4. DISCUSSION

4.1. Effect of CO_2 Concentration on Microalgal Growth

Microalgae commonly used as CO_2 capture agents are Chlorella (green microalgae) and Spirulina/Arthrospira (blue-green microalgae) due to their high growth rates and CO_2 fixation capabilities [33, 34]. The following section discusses studies on these two microalgae genera.

4.1.1. *Chlorella sp.* ABC

Chlorella sp. ABC cultivated at atmospheric CO_2 concentration (0.04%) produced a relatively low final biomass compared to cultivation at higher CO_2 concentrations. Variations in CO_2 flow rates (0.1 and 0.6 vvm) significantly affected its growth (0.17 and 0.93 g L^{-1}). At 1% CO_2 addition, the biomass increased significantly to 2.33 g L^{-1}. However, increasing CO_2 levels from 1 to 10% with the same gas flow rate (0.1 vvm) did not show significant differences (1%: 2.33 g L^{-1}; 5%: 2.32 g L^{-1}; and 10%: 2.20 g L^{-1}). When 15% CO_2 (0.1 vvm) was added, the lag phase lasted slightly longer, with a final biomass yield of 2.73 g L^{-1} on day 7, lower than the final biomass yield at 10% CO_2 (0.6 vvm) of 2.9 g L^{-1} [20]. Thus, the best growth condition for *Chlorella sp*. ABC was achieved with the addition of 10% CO_2 at a flow rate of 0.6 vvm. The effect of CO_2 concentration on the dry biomass yield of *Chlorella sp.* ABC is shown in Table 1.

TABLE I. Dry biomass yield of *Chlorella sp*. ABC was cultivated at different CO_2 concentrations (harvested on the 7th day) [20].

CO_2 concentration (%)	Flow rate (vvm)	Dry biomass (g L^{-1})
0.04 (atmosphere)	0.1	0.17
0.04 (atmosphere)	0.6	0.93
1	0.1	2.33
5	0.1	2.32
10	0.1	2.2
10	0.6	2.9
15	0.1	2.73

4.1.2. *Spirulina platensis* and a mixture of local microalgae from Doha (MIMA)

Spirulina platensis and a mixture of local microalgae from Doha, Qatar (Mixed Indigenous Microalgae, or MIMA) were cultivated for 20 months. To capture seasonal variations, the cultivation was divided into four temperature periods: 1st period (January–March: 18.1–24.3 °C), 2nd period (April–June: 28.7–36 °C), 3rd period (July–September: 32.5–41.2 °C), and 4th period (October–December, 22.7–31.3 °C) experienced a 42% increase in biomass productivity ($P_{(bio)}$) in each season as CO_2 concentration increased from 2.5% to 10% (v/v) [17].

4.1.3. *Spirulina sp.*

Spirulina sp. has been reported to have higher $P_{(bio)}$ values when added to CO_2 (2–15%) compared to CO_2 from air (0.04%). LAMB 171 and 172 strains had the highest $P_{(bio)}$ values when added to 10% CO_2, while LAMB 220 had the highest $P_{(bio)}$ values when added to 5 and 10% CO_2. At 15% CO_2, the $P_{(bio)}$ values were lower in all three strains [19]. The $P_{(bio)}$ values of *Spirulina sp*. for various strains and CO_2 concentrations can be seen in Table 2.

TABLE II. Biomass productivity of *Spirulina sp*. at various CO_2 concentrations [19].

Strain	CO_2 concentration	Biomass productivity ($P_{(bio)}$) (mg L^{-1} day^{-1})
LAMB171	Control (air)	145.71 ± 0.006^{c}
	2%	218.33 ± 0.012^{b}
	5%	234.55 ± 0.012^{b}
	10%	272.12 ± 0.019^{a}
	15%	228.75 ± 0.009^{b}
LAMB172	Control (air)	161.43 ± 0.007^{c}
	2%	207.50 ± 0.017^{b}
	5%	246.67 ± 0.019^{ab}
	10%	265.45 ± 0.008^{a}

LAMB220	15%	237.50 ± 0.012^{ab}
	Control (air)	123.33 ± 0.007^{b}
	2%	162.08 ± 0.017^{b}
	5%	262.42 ± 0.011^{a}
	10%	260.91 ± 0.007^{a}
	15%	233.75 ± 0.010^{a}

*a, b, c, d, e indicate statistically significant differences (a > b > c > d > e). Different letters: significant differences.

4.1.4. *Chlorella sorokiniana* TH01

Chlorella sorokiniana TH01 was reported to have optimal growth at 5–10% CO_2, with total biomass yields of 2.53 and 2.20 g L^{-1} on the 7th day of cultivation. At higher CO_2 concentrations, the growth rate decreased, with biomass yields of 2.06 g L^{-1} (15% CO_2) and 0.85 g L^{-1} (20% CO_2). The addition of higher CO_2 levels also decreased the culture pH from 7.74 to 6.25 (15% CO_2) and from 7.74 to 5.36 (20% CO_2). This pH decreases inhibited carbonic anhydrase activity and reduced photosynthetic rate, thereby inhibiting the growth of *Chlorella sorokiniana* TH01 cells. The biomass decrease in cultures with the addition of 5–10% CO_2 after the 7th day was due to reduced nutrient levels in the medium, whereas the decrease in cultures with 15–20% CO_2 was due to a decrease in pH [18].

4.1.5. *Arthrospira platensis*

Arthrospira platensis is the official name of *Spirulina platensis*, a more popular trade name. *Arthrospira platensis* has been reported to grow well at 6% CO_2, with a maximum growth rate of 0.140 g L^{-1} day^{-1}. During the 14th day experiment, the CO_2 concentration decreased from 6.01% to 4.21%, corresponding to an average reduction of approximately 0.13% per day, with a CO_2 capture rate of 0.220 (based on biomass productivity) until 0.235 g L^{-1} day^{-1} (based on direct CO_2 measurements). However, higher CO_2 concentrations were not tested [28].

Based on this description, adding CO2 within a certain range (5–10%) is beneficial for microalgal growth. However, adding higher CO_2 (≥15%) inhibits it. The addition of CO_2 can lower the culture's pH by reacting with water to form carbonic acid (H_2CO_3). Carbonic acid can form two different salts when dissolved, namely bicarbonate ions (HCO_3^-) and carbonate ions (CO_3^{2-}) [35]. Therefore, the addition of CO_2 to the culture medium has also been reported to increase the dissolved inorganic carbon (DIC) concentration in the form of these two ions [19]. The reaction between dissolved CO_2 and water is expressed in Equation 4.

$$CO_{2(aq)} + H_2O_{(l)} \rightleftharpoons H_2CO_{3(aq)} \rightleftharpoons HCO_3^-{}_{(aq)} + H^+_{(aq)} \rightleftharpoons CO_3^{2-}{}_{(aq)} + 2H^+_{(aq)} \quad (4)$$

In *Spirulina sp*., the addition of CO_2 lowers the culture pH from 9.8–10.5 to 8.5–9.0. Within this pH range, HCO_3^- is the dominant form of DIC [19], which is beneficial for microalgae. However, excess free CO_2 not only lowers the pH of the medium but also inhibits mass transfer (including diffusion and dissolution) of CO_2 itself, thereby reducing the efficiency of photosynthesis and carbon fixation [17] and inhibiting the activity of the carbonic anhydrase, which plays a crucial role in the carbon concentrating mechanism (CCM). Consequently, the algal growth rate slows [19]. This explains why the $P_{(bio)}$ of microalgae decreases at 15% CO_2. Besides lowering the culture's pH, CO_2

solubility is also highly dependent on it. The molar fractions (%) of CO_2, HCO_3^-, and CO_3^{2-} at various pH values are shown in Figure 1.

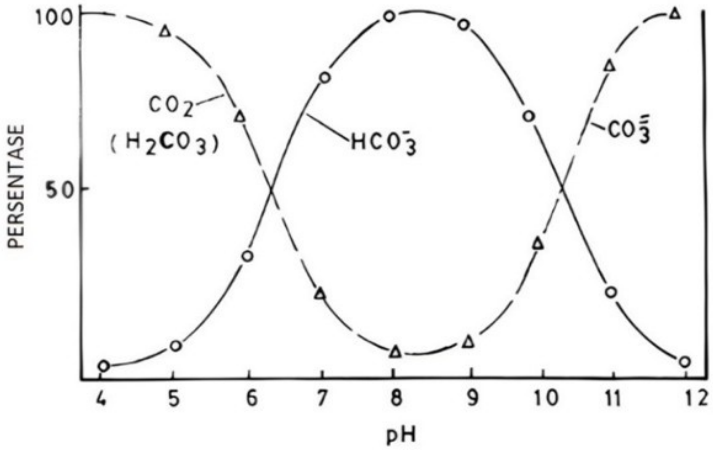

Figure 1. Molar fractions (%) of CO_2, HCO_3^-, and CO_3^{2-} at various pH values *(https://www.fao.org/4/ac183e/ac183e06.htm).*

Microalgae, which generally live in aquatic habitats face limitations in CO_2 supply due to the gas's low solubility and its easily fluctuating concentration in water. This condition poses a challenge for photosynthesis because RuBisCO has a relatively low affinity for CO_2. In addition, RuBisCO can catalyze oxygenation reactions, thereby reducing photosynthetic efficiency [36]. To overcome the low efficiency of RuBisCO, microalgae have evolved a carbon concentrating mechanism (CCM).

CCM is a physiological mechanism in microalgae, cyanobacteria, and certain photosynthetic bacteria that enables them to accumulate DIC inside the cell, particularly under conditions of low atmospheric CO_2 availability [36]. CCM has been widely studied in the green microalga *Chlamydomonas reinhardtii* as a model organism [37, 38]. When CO_2 levels are low, transporter proteins located in the plasma membrane of *C. reinhardtii* (HLA3 and LCI1) [39, 40] and in the chloroplast envelope (LCIA) [41] actively take up inorganic carbon from the environment and generate a high accumulation of CO_2 in the chloroplast stroma. The accumulated inorganic carbon, predominantly in the form of HCO_3^-, is then transported into the thylakoid lumen through transporters (BST1–3) [42]. Subsequently, HCO_3^- is converted into CO_2 by carbonic anhydrase (CAH3), providing an abundant supply of CO_2 for RuBisCO [39]. Furthermore, to prevent CO_2 leakage from the pyrenoid, the protein complex LCIB/LCIC functions to recapture CO_2 escaping from the pyrenoid [43, 44]. These components operate in a coordinated manner within the CCM system, enabling *C. reinhardtii* to grow and survive under low CO_2 concentrations.

4.2 Effect of pH on Microalgal Growth

Singh Nikita & Rathilal Sudesh emphasised that microalgae are highly sensitive to small changes in pH environmental conditions [45]. Microalgal growth and biomass productivity can increase or decrease significantly depending on the stability of the pH culture. *Scenedesmus sp*. was reported to grow optimally at pH 8 with a $P_{(bio)}$ of 39×10^4 cells mL^{-1} day^{-1} and a growth rate of 0.86 day^{-1}. Meanwhile, *Chlorella vulgaris* showed the best growth at pH 9 with a $P_{(bio)}$ of 24.38×10^4 cells mL^{-1} day^{-1} and a growth rate of 0.434 day^{-1}. *Spirulina platensis* grew very well at pH 9 with a $P_{(bio)}$ of 23.36×10^4 cells mL^{-1} day^{-1}. The $P_{(bio)}$ values of *Scenedesmus sp*. and *Chlorella vulgaris* decreased at both lower and higher pH values. Moreover, *Scenedesmus sp*., *Chlorella vulgaris*, and *Spirulina platensis* do not grow well at a slightly acidic pH (6.5). Even some microalgae species, such as *Bracteacoccus minor*, *Pseudococcomyxa simplex*, and *Chlorococcum infusionum*, have been reported to tolerate pH as low as 4. However, at pH levels below 4, these microalgae experience discoloration (loss of green colour), protoplast damage, and even complete cell destruction [8].

pH is a significant factor influencing microalgal growth. In addition to affecting the availability of inorganic carbon in solution, at very low pH, microalgal cells are under stress because they must maintain a neutral cytoplasmic pH. At the same time, H^+ ions continuously permeate the plasma membrane. Furthermore, low pH can damage cell walls by weakening hydrogen bonds within molecules, leading to uncontrolled changes in cell size and altering membrane permeability [8]. Medium pH also plays a crucial role in determining the activity of metabolic enzymes and the efficiency of ion uptake by microalgae, thereby directly influencing their growth rate and carbon fixation capacity. Each microalgae species has a different optimum pH range, but generally, the best growth is achieved at near-neutral conditions. Aeration using flue gas containing CO_2, NO_x, and SO_2 can significantly lower the medium's pH. Flue gas containing 10–30% CO_2 can lower the pH to around 5.5, while SO_2 at 100–250 ppm can further lower it to 2.5–3.5 [46]. Although some microalgal species have been reported to tolerate pH 4 or lower, their tolerance to such low pH remains very limited. Furthermore, most of these studies were conducted at room temperature (20–25°C), without considering the effects of high temperatures.

4.3. Effect of Temperature on Microalgal Growth

Chlorella protothecoides exhibited optimum growth in the temperature range of 25–30 °C, with a specific growth rate of 0.54 day^{-1} and $P_{(bio)}$ reaching 70 mg L^{-1} day^{-1}. Increasing the temperature above this range causes a gradual decrease in growth rate, 0.49 day^{-1} (35 °C), 0.41 day^{-1} (40 °C), and 0.25 day^{-1} (45 °C), respectively. At 40 °C, $P_{(bio)}$ decreased sharply to only about 6 mg L^{-1} day^{-1} [47]. *Thalassiosira weissflogii* has also been reported to grow optimally at room temperature. At 10 °C, its growth rate is 0.34 day^{-1}, increasing to 1.24 day^{-1} at 25 °C, and increasing by approximately 3% at 30 °C. The optimum temperature for this species was recorded at 28.24 °C, with a maximum growth rate of 1.28 day^{-1}. Meanwhile, *Pyramimonas sp*. showed a similar pattern up to 25 °C, but exhibited a sharp decline at 30 °C, with a growth rate of only 0.52 day^{-1}, nearly the same as at 10 °C. The optimum temperature for *Pyramimonas sp*. was estimated to be around 23.96 °C with a maximum growth rate of 1.11 day^{-1} [48]. Furthermore, López Muñoz & Bernard used Light Oxygen Temperature (LOT) modelling calibrated with experimental data on *Chlorella minutissima*. The LOT model is a mathematical model that describes the combined effects of light, oxygen concentration, and temperature on microalgae growth. This model incorporates oxygen concentration as a parameter to represent oxidative stress in the culture by adding a toxicity term to the net growth rate equation. Therefore, the LOT model can be used to predict microalgae growth and to evaluate the sensitivity of different species to oxidative stress, thereby helping optimize culture conditions to improve productivity. Using this model, they reported that *Chlorella minutissima* grows optimally at 29–30 °C. The growth decreases sharply above 35 °C and stops completely at the higher temperatures [9].

In general, temperature affects cellular enzymatic activity. In non-thermophilic microalgae, low temperatures (<20°C) slow enzymatic reactions, thereby reducing photosynthesis and cell division rates. As temperatures approach the optimum (25–35 °C), enzyme activity, such as RuBisCO and ATP synthase, increases, resulting in higher growth rates and biomass accumulation. However, above a critical threshold (>35–40 °C), proteins in photosystem II (PSII) denature, disrupting the electron transport chain in chloroplasts and reducing photosynthetic efficiency [49]. Flue gas from coal-fired power plants (CFPP) generally has a high temperature, exceeding 120 °C, requiring cooling before introduction into the culture medium to prevent damage to microalgal cells [46]. The ability of microalgae to survive and photosynthesize at high temperatures has the potential to increase CO_2 capture efficiency by saving time and energy required for flue gas cooling.

4.4. The Effect of SO_2 and NO_x Gases on Microalgal Growth

In addition to high CO_2 levels, low pH, and high temperatures, coal-fired power plant exhaust gas also contains SO_2 and NO_x gases. SO_2 gas is highly acidic and toxic to most microalgae. *Chlorella sp*. ABC, which can grow well at 10% CO_2, cannot survive at 50 ppm SO_2, and its growth ceases completely on the second day [20]. At concentrations higher than 100 ppm, growth is nearly impossible for most microalgae. Although certain microalgae can still grow at lower SO_2 levels, their

growth rates decrease sharply, and their adaptation phase is longer than in cultures without SO_2 exposure. Increasing SO_2 concentrations decrease carbon fixation efficiency and biomass productivity by increasing medium acidity, resulting from SO_2 hydrolysis, which produces additional oxidation products, including hydrogen ions (H^+), bisulfate (HSO_4^-), and sulfate (SO_4^{2-}). When the medium pH drops below 3.0, microalgal cells generally die [21, 50]. However, unlike CO_2, SO_2 toxicity is not primarily due to its acidic nature, but rather to its chemical mechanisms that damage cells. Consequently, pH control at 50 ppm SO_2 only slightly increases growth, while toxicity remains high. Meanwhile, at 100 ppm SO_2, controlling the pH has no significant effect [20]. HSO_4^- and SO_4^{2-} ions also inhibit the RuBisCO enzyme and disrupt energy (ATP) production in mitochondria, resulting in decreased photosynthetic activity, reduced cellular energy efficiency, overall growth, and even cell death [20, 51].

Meanwhile, the effects of NO_x on microalgae growth vary. Several studies have shown that nitrogen monoxide (NO), one of the NO_x, can serve as an alternative nitrogen source for microalgae cultivation. There are two main pathways for NO uptake by microalgae. The first pathway involves the oxidation of dissolved NO to nitrate (NO_3^-) in the medium through a reaction with dissolved oxygen, allowing the resulting nitrate to be absorbed by the cells and utilized in nitrogen biosynthesis. The second pathway indicates that NO molecules can diffuse directly through the microalgae cell membrane and then undergo oxidation within the cells. However, this direct diffusion mechanism has been reported in only one study and is not yet widely recognized as a common pathway of nitrogen assimilation in microalgae [24]. However, at high levels, nitrogen oxides (NO_x) have been reported to inhibit microalgal growth. At high levels, nitrogen oxides are reported to be toxic to microalgae because they generate reactive radicals, such as peroxynitrite ($ONOO^-$), which disrupts photosynthetic electron transport [52, 53]. The effect of NO concentration on microalgal growth is shown in Table 3.

These findings indicate that most microalgae exhibit growth retardation at high CO_2 concentrations (>10%) and are unable to survive at low pH (<4), high temperatures (>35 °C), and high levels of SO_2 and NO_x. Should a microalgal species exhibit thermotolerance, acidotolerance, and tolerance to SO_2 and NO_x, the costs and time associated with pre-treatment processes, including cooling, pH adjustment, and flue gas desulfurization, could be substantially reduced.

TABLE III. Effect of nitric oxide (NO) on microalgal growth [24].

Microalgae	NO concentration (ppm)	Reactor type	Growth inhibition
Dunaliella terliolecta	100	Air-lift reactor	✗
Dunaliella terliolecta	20	Bubble column	✗
Chlorella KR1	100	Glass bottle (50 mL)	✗
Chlorella sp.	38	Bubble column	✗
Dunaliella parva	20	Bubble column	✗
Nannochloris sp.	300	Tubular reactor	✓
Nannochloris sp.	300	Glass bottle (1L)	✓
Nannochloropsis sp.	300	Glass bottle (1L)	✓
Chlorococcum littorale	100	Glass bottle (50 mL)	✓
Chlorella KR1	300	Glass bottle (50 mL)	✓
Chlorella sp.	300	Glass bottle (1L)	✓
Scenedesmus sp.	300	Cylindrical glass bioreactor	✓
Micrasterias denticulata	300	Erlenmeyer	✓

4.5. Extremophilic Microalgae

Extremophilic microalgae are microalgae that thrive in extreme natural habitats, such as volcanic hot-spring environments, solfatara soils (soils that emit hot sulfur gases), and extreme anthropogenic environments (environments created by human activity) [54]. Some extremophilic microalgae, such as *Galdieria sulphuraria*, has high metabolic flexibility because it can grow in various metabolic modes, namely autotrophic (using light and CO_2 as a carbon source), mixotrophic (using light and sugar as a carbon source), and heterotrophic (growing without light) [55] and utilizes a variety of inexpensive substrates, including waste [56, 57].

Extremophilic microalgae can grow in extreme conditions due to several factors, such as their ability to produce extremozymes, enzymes capable of catalyzing chemical reactions at very high or very low pH, temperature, or pressure, and extremolyte compounds, small organic molecules produced by extremophilic microorganisms to protect cells from environmental stress. One of these compounds is floridoside, characteristic of red algae, which stabilizes cells and proteins in very harsh environments [58]. The extremophilic abilities of these microalgae are also reported to stem from a combination of their cell structure and composition:

a) a protein-rich cell wall (up to 55%) in the form of a more stable and relatively acid-resistant glycoprotein [59, 60],
b) a saturated and branched lipid membrane that is stable at high temperatures [61], and
c) a genome that shows evidence of horizontal gene transfer from various extremophilic bacteria and archaea that live in sulfur-rich environments [54]. It is suspected that extremophilic microalgae acquired key genes for sulfur metabolism, such as ATP sulfurylase [62].

4.5.1. *Quadrigula closterioides*

Quadrigula closterioides LCM607 is an extremophilic green microalgae that was successfully isolated from a coal mine environment within a 2 km radius (pH 7.2 ± 0.3, temperature 30 ± 2 °C, and high CO_2 (15–20% v/v). *Quadrigula closterioides* LCM607 was reported to grow at 15% CO_2. At this concentration, *Quadrigula closterioides* LCM607 had the highest carbon uptake rate (RCO_2) of 182 ± 0.009 mg L^{-1} day^{-1} (calculated from elemental analysis data) and the highest P_{bio} of 60 ± 0.003 mg L^{-1} day^{-1}. This value is higher than lower CO_2 concentrations (RCO_2 10%: 123±0.006; 5%: 104±0.005; 0.03%: 70±0.003 mg L^{-1} day^{-1}) and (P_{bio}: 10%: 39±0.001; 5%: 32±0.001; 0.03%: 17±0.003 mg L^{-1} day^{-1}). Meanwhile, the optimum pH for growth is 7.5, and the optimum temperature is 28 °C [3].

4.5.2. *Galdieria sulphuraria*

Galdieria sulphuraria is a model extremophilic red microalga. *Galdieria sulphuraria* CCME 5587.1 has been reported to absorb 50 ppm SO_2 while still growing well and absorbing some NO gas [20]. Unlike most microalgae, *Galdieria sulphuraria* showed a higher growth rate at 40 °C (4.90 ± 0.588) than at lower temperatures, 35 °C (1.40 ± 0.098), 30 °C (0.87 ± 0.069), and 25°C (0.16 ± 0.022). This microalgae can also grow at pH 1–4, with maximum dry biomass achieved at pH 3 (autotrophic: 600 µg/mL; mixotrophic: 1000 µg/mL) [63]. Mixotrophic cultivation of *Galdieria sulphuraria* SAG 21.92 at pH 2, 42 °C, and 30 µmol photons m^{-2} s^{-1} (25 mM D-sorbitol) was reported to release 76.6 ± 2 µM O_2. Under autotrophic conditions with atmospheric CO_2 as the carbon source (0.04%, equivalent to approximately 10 µM dissolved CO_2), O_2 release only reached 32 ± 7.5 µM. Meanwhile, under heterotrophic conditions (25 mM D-sorbitol, in the dark), no O_2 release was detected (0 µM). The amount of O_2 released was assumed to be equivalent to the amount of carbon captured [64].

4.5.3. *Pumiliosphaera acidophila*

Pumiliosphaera acidophila (NCBI ID: 190058) is an extremophilic microalgae formerly known as *Chlorella protothecoides* var. *acidicola* and some also as *Auxenochlorella protothecoides* var. *acidicola*, but has now been taxonomically revised based on modern phylogenetic studies (SSU

& ITS rDNA). It is a very small, single-celled green alga (±3–6 µm), with thin cell walls, dumbbell-shaped chloroplasts, no pyrenoids, and reproduces by 2–4 autospores (sometimes also by budding) (Figure 5). This microalga is a unique and rarely reported species due to its unusual habitat conditions, including acidic volcanic lava flows and surrounding soil with a pH of 2–3 [65]. Green microalgae, particularly terrestrial species, have been reported to exhibit greater tolerance to CO_2 concentration than other microalgal groups [66]. As a green microalga, *Pumiliosphaera acidophila* may potentially share this characteristic, though further investigation is needed.

4.6. Microalgae Biomass

Microalgae are not only capable of capturing CO_2 and converting it into carbohydrates and lipids but also produce other nutritionally valuable metabolites such as proteins [6, 20, 67-69]. Therefore, microalgae are often referred to as green gold. Thus, the advantages of extremophile microalgae not only make them highly promising for CO_2 mitigation under extreme conditions but also as a source of high-value biomass. Unfortunately, data on the biomass profile of extremophile microalgae remain very limited and are mostly derived from *Galdieria sulphuraria*, a model extremophile microalga widely used for physiological and metabolic studies under acidic conditions and high temperatures. This extremophile microalgae model has been reported to accumulate proteins up to 68.86% w/w, carbohydrates up to 36.2% w/w, and lipids up to 39% w/w, depending on the strain and cultivation conditions [70, 71]. The biomass profile of *Galdieria sulphuraria* is shown in Table 4.

TABLE IV. Biomass profile of *Galdieria sulphuraria*.

Strain	Cultivation condition		Protein (% w/w)	Carbohydrate (% w/w)	Lipid (% b/b)	Reference
064/309	Autotroph 36±1 °C, pH 1.5, 150 µE m^{-2} s^{-1}		25	48	1	[32]
SAG 21.92	Autotroph 42 °C, pH 2, 100 µmol m^{-2} s^{-1}, 16:8 (light:dark), 150 rpm, CO_2 (0.04%)		42.58 ± 2.72	8.01 ± 0.58	8.89 ± 1.58	[68]
SAG 107.79			39.31 ± 3.2	8.89 ± 0.08	6.78 ± 0.77	
SAG 108.79			43.44 ± 1.73	8.15 ± 0.85	9.56 ± 1.35	
UTEX 2919			46.86 ± 1.35	3.94 ± 0.31	7.31 ± 0.97	
CCMEE 5587.1			43.95 ± 3.15	5.91 ± 0.35	14.11 ± 0.84	
CCMEE 5587.1	Heterotroph, pH 2, 42 °C, varied C/N ratio, initial stationary phase harvested	Glucose C/N (5)	58.29 ± 0.27	6.09 ± 0.11	6.03 ± 0.09	[70]
		Glucose C/N (10)	41.89 ± 0.19	11.24 ± 0.33	6.34 ± 0.14	
		Glucose C/N (20)	29.70 ± 0.13	16.82 ± 0.38	5.04 ± 0.19	
		Glucose C/N (30)	25.77 ± 0.12	20.46 ± 0.29	6.10 ± 0.08	
		Glycerol C/N (5)	51.51 ± 0.24	6.99 ± 0.32	7.05 ± 0.42	
		Glycerol C/N (10)	37.69 ± 0.17	6.95 ± 0.27	7.31 ± 0.22	
		Glycerol C/N (20)	29.70 ± 0.13	10.35 ± 0.44	7.28 ± 0.26	
		Glycerol C/N (30)	26.18 ± 0.12	10.99 ± 0.46	39 ± 0.30	
	Mixotroph, 100 µmol/m²s, initial stationary phase harvested	Glucose C/N (5)	68.86 ± 0.32	5.61 ± 0.22	7.71 ± 0.21	
		Glucose C/N (10)	37.29 ± 0.17	12.88 ± 0.31	5.16 ± 0.20	
		Glucose C/N (20)	29.17 ± 0.13	17.15 ± 0.31	4.99 ± 0.17	
		Glucose C/N (30)	27.50 ± 0.12	15.12 ± 0.19	7.07 ± 0.24	
		Glycerol C/N (5)	59.92 ± 0.28	4.92 ± 0.12	5.92 ± 0.38	
		Glycerol C/N (10)	38.10 ± 0.18	9.38 ± 0.29	6.28 ± 0.21	
		Glycerol C/N (20)	28.51 ± 0.13	10.69 ± 0.32	6.89 ± 0.28	
		Glycerol C/N (30)	27.94 ± 0.13	11.16 ± 0.20	6.40 ± 0.27	
074 G	Heterotroph pH 1.8, 45 °C, 130 rpm	Defined medium (glycerol)	25.0 ± 1.7%	36.2 ± 7.4%	2.4 ± 0.9%	[71]
		Complex medium I (brewer's spent grain+ molasses)	37.4 ± 5.8%	3.9 ± 0.1%	5.3±0.6%	
		Complex medium II (molasses + glycerol)	41.7 ± 4.7%	13.0 ± 2.3%	3.6 ± 0.4%	

TABLE V. Several studies on microalgae as CO_2 capturing agents.

No	Reference	Microalgae	Experimental condition	Results		Conclusion
1	[17]	*Spirulina platensis* UTEX, local microalgae mixture from Qatar/*Mixed Indigenous Microalgae* (MIMA)	2.5, 5, 10, 15, 20% CO_2, pH ±9.5 (Spirulina) and 7.5 (MIMA), controlling the medium pH, 16–38 °C (according to natural seasons), 180 $\mu E \cdot m^{-2} s^{-1}$, waste water, bacterial-free culture, duration: 20 months	RCO_2 max. (mg/L/d) 1–2 period • *S. platensis:* ~250 (period 1) • MIMA: ~400 (period 2) 3–4 period • *S. platensis:* ~250 (period 4) • MIMA: ~600 (period 3)	The optimal conditions for both microalgae were: temperature: 20–25°C, CO_2 concentration: 10% v/v. The $P_{(bio)}$ of both microalgae decreased at 15% CO_2 and sharply decreased at 20% CO_2. Meanwhile, the microalgae mixture (MIMA) showed a higher P_{bio} than the single Spirulina strain. In summer, the $P_{(bio)}$ of both microalgae decreased by 30–60%. $P_{(bio)}$ increased by 42% from the addition of 2.5% to 10% CO_2.	Secondary wastewater from local industries in Doha. *Spirulina platensis* and MIMA do not grow well at temperatures above 30 °C. MIMA is more sensitive to temperature and CO_2 fluctuations than *Spirulina platensis*. Acidic pH is not reported because both microalgae were cultivated at neutral (MIMA) and slightly alkaline (9.5, Spirulina) pH.
2	[18]	*Chlorella sorokiniana* TH01	0.04, 5, 10, 15, CO_2 20%, initial pH ~ 7.7, 28 °C, indoor sterile culture.	RCO_2 max. (mg/L/d) 757	Increasing the CO_2 concentration from 0.04% to 5% significantly increased growth, with a maximum biomass reaching 2.53 g/L and a maximum Pbio of 413 mg/L/day. At higher CO_2 concentrations, the concentration and Pbio decreased to 2.06 g/L and 322 mg/L/day (15%) and 0.85 g/L and 230 mg/L/day (20%). Optimal growth was achieved at 5% CO_2, with the pH decreasing from 7.74 to 6.25 (at 15% CO_2) and 7.74 to 5.36 (at 20% CO_2). The CO_2 fixation rate was 757 mg/L/day (5% CO_2), and the culture pH remained relatively stable in the range of 6.7–7.7. These values differ significantly from	The decrease in biomass concentration and productivity due to increased CO_2 was related to a decrease in culture pH. The addition of CO_2 increased biomass concentration and productivity, provided the culture pH did not change significantly. The effect of temperature was not reported because the experiment was only conducted at 28 °C.

					those of 460 (0.04%), 576 (10%), 591 (15%), and 457 mg/L/day (20%). The 0.04% CO_2 (control) condition showed nearly 100% fixation efficiency, but very low biomass and productivity (1.09 ± 0.02 g L^{-1}; 226 ± 5 mg L^{-1} day^{-1}), indicating significant carbon limitation.	
3	[2]	*Spirulina sp. LAMB171, LAMB172, & LAMB220*	0.03, 2, 5, 10, 15% CO_2, pH 8.5–9.0 (Zarrouk media), 28±1 °C.	RCO_2 max. (mg/L/d) LAMB171 • 0.03%: 210.29 • 2%: 316.02 • 5%: 363.64 • 10%: 424.12 • 15%: 389.34 LAMB172 • 0.03%: 244.26 • 2%: 325.3 • 5%: 386.4 • 10%: 457.77 • 15%: 408.4 LAMB173 • 0.03%: 171.63 • 2% : 240.53 • 5% : 397.32 • 10% : 461.52 • 15% : 400.23	Increasing CO_2 concentration from 0.03% (control) to 10% consistently increased the specific growth rate from approximately 0.20 per day to 0.26–0.31 per day, decreasing to 0.279 per day at 15%. The average CO_2 fixation rate increased from 208.7 mg L^{-1} day^{-1} at 0.03% CO_2 to 447.8 mg L^{-1} day^{-1} at 10% CO_2, then decreased to 399.3 mg L^{-1} day^{-1} (approximately 10.8%) when the CO_2 concentration was increased to 15%.	Adding CO_2 (1–10%) accelerated biomass productivity, reaching optimal conditions. A 15% CO_2 concentration reduced growth and fixation rates, indicating an inhibitory effect due to excess dissolved CO_2. The effect of temperature was not reported because the experiments were conducted at approximately room temperature.
4	[20]	• *Chlorella sp. ABC* • *Galdieria sulphuraria*	0.04 (atmosphere), 1, 5, 10, and 15% CO_2,	RCO_2 max. (mg/L/d) • *Chlorella sp.*	The highest biomass of *Chlorella sp.* ABC was at 10% CO_2 (~3 g/L). In a single Chlorella culture, the absorption efficiency was 5% (only 0.5% absorbed)	*Chlorella sp.* ABC is a green microalgae reported to have a higher CO_2 fixation rate than red microalgae, but is

			50 and 100 ppm NO or SO_2, ~6 pH controlling, 30 °C, 170 µmol photons m^{-2} s^{-1}.	ABC single culture: 760 • *Chlorella sp.* at dual cultivation: 526 • *G. Sulphuraria*: 267 Total dual strain: 793	for CO_2 and 4.6% for NO_2 (50 ppm), and the culture was unable to absorb SO_2 (dead) even after pH control. Meanwhile, a single Galdieria absorbed 4% CO_2, 8.8% NO_2, and 100% SO_2. In a dual-strain system, where gas was passed to Galdieria and then to Chlorella, the efficiency increased to 9% for CO_2 and 13.4% for NO_2, with SO_2 remaining perfectly absorbed (100%).	sensitive to acidic pH and the presence of SO_2. *Galdieria sulphuraria* is a red extremophile microalgae capable of surviving at acidic pH and in the presence of SO_2 gas, but with a low CO_2 fixation rate. The effect of temperature was not reported because the experiments were only conducted at 30 °C.
5	[28]	*Arthrospira platensis*	6% CO_2 (indoor size 12 m^3 = 12.000 L → 1296 g CO_2), pH 8.5–11.5, 27±1°C. Light intensity: 14.0–14.4 klx & 12.1–13.4 klx 24-hour, Zarrouk media. 4 experiments: 1st experiment (tap air+media, 0.12 g/dm^3), 2nd experiment (tap water+media), 3rd experiment (2nd experiment filtrate+media at day+8),	RCO_2 max. (mg/L/d) 235	Grows well at 6% CO_2 with a maximum growth rate of 0.140 g/(dm^3·day), CO_2 capture effectiveness of 0.235 g/(dm^3·day) or 25% (absorbed 1.5%) on day 14 (end of experiment), and a decrease in bicarbonate, nitrate, and phosphate nutrients of 25–50%. Experiment 2, the maximum growth rate was 0.139 g/L·day (highest), experiment 3, the growth rate decreased by 0.08 g/L·day, and experiment 4, the lowest rate was 0.013 g/(dm^3·day) due to reduced nutrients and metabolite accumulation.	Mimicking emissions from gas-fired power plants. *Arthrospira platensis* is a blue-green microalgae/cyano-bacteria reported to have weak CO_2 capture capacity. No experiments were conducted on the effects of SO_2 and high temperatures.

			4th experiment (3rd experiment filtrate). Initial culture: Exp. 1–4: 0.12, 0.16, 0.16, & 0.26 g/dm^3			

5. CONCLUSION

Based on the reviewed studies, CO_2 can enhance microalgal growth at concentrations up to 10%, whereas concentrations exceeding this threshold inhibit growth due to culture acidification. The presence of SO_2 additionally complicates CO_2 capture applications, as SO_2 exerts toxicity through its chemical compounds rather than solely through pH reduction, rendering pH control alone insufficient. Extremophilic microalgae such as *Galdieria sulphuraria*, *Quadrigula closterioides*, and *Pumiliosphaera acidophila* show strong potential for CO_2 capture under these harsh conditions (high CO_2, elevated temperature, and acidic pH) where conventional mesophilic species cannot survive. Despite this promise, standardizing CO_2 fixation quantification methods and scaling up beyond laboratory systems remain key challenges. Future research should focus on isolating novel extremophilic strains and integrating biomass valorisation to further improve the economic viability of this approach.

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