Optimization of cell disruption in Raphidocelis subcapitata and Chlorella vulgaris for biomarker evaluation
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INTRODUCTION

The use of the unicellular microalgae, *Raphidocelis subcapitata* and *Chlorella vulgaris* as bioassay test organisms is significant as they play a vital role in the structure and functioning of the aquatic ecosystems and any adverse effect on them will affect the upper trophic levels (Li et al., 2006). In ecotoxicological studies, biomolecules such as the antioxidative enzymes and heat stress proteins are often used as biomarkers in assessing the adverse effects of various environmental toxicants on aquatic organisms (Dewez et al., 2005; Bierkens et al., 1998). Protocols for determining the activities of some of these biomolecules in test organisms usually involve an initial cell disruption or homogenization step. In microalgae, cell disintegration is often necessary for the recovery of intracellular products (Mendes-Pinto et al., 2002; Molina Grima et al., 2003). However, the cell walls of certain phytoplankton such as *C. vulgaris* and *R. subcapitata* are composed predominantly of hemicellulose and saccharides which hinder the release of intracellular biological molecules (Abo-Shady et al., 1993; Doucha and Livansky, 2008). Since membrane and/or cell wall species-specific composition could have an effect on disruption efficiency in different organisms, it is expected that the extent of cell disintegration will depend on the algal species (Lavoie et al., 2009).

A range of treatment methods have been suggested and used for breaking algal cells. These include freezing, alkali and organic solvents, sonication, high pressure homogenization (French press), bead milling, microwaving, enzymatic lysis and manual grinding (Wimpenny, 1967; Chisti and Moo-young, 1986; Molina Grima et al., 2004; Zheng et al., 2011). *Chlorococcum sp.*, *Botryococcus sp.*, *C. vulgaris*, *R. subcapitata* and *Chlamydomonas reinhardtii* have been lysed for various purposes employing different techniques such as high pressure homogenization, sulphuric acid treatment, bead beating, ultrasonics, microwaves, liquid nitrogen grinding, rotor stator homogenizer and vortex agitator (Halim et al., 2012; Lee et al., 2010; McMillan et al., 2013; Zheng et al., 2011; Lavoie et al., 2009). The French press, sonication and bead beating are considered the most effective and widely used methods for the homogenization of single cells, cell suspensions and microorganisms for the recovery of intracellular products (Burden, 2012). During ultrasonication process, high frequency sonic waves created by the expansion and contraction of a crystalized probe or bath, bring about implosive collapse of the gas-filled cavitation bubbles and generate intense microscopic shock waves, which ultimately cause cell wall disruption (Zheng et al., 2011).

Bead beating is a simple disruption technique that breaks cells by shaking a closed container filled with the target cells and beads made up of quartz or metal (Kim et al., 2013). It uses not only beads but other grinding media such as balls and satellites, while non-spherical media such as garnet has been effectively used in slicing resilient samples when used in conjunction with larger grinding balls (Burden, 2012). Although, some of these homogenization techniques are relatively old and are frequently used (Lavoie et al., 2009), information in literature about their efficiency in breaking the resilient cell wall of *R. subcapitata* and *C. vulgaris* as well as the endpoint of cell disruption is scarce. The aim of this study was to assess the efficacy of four mechanical treatment methods in breaking *R. subcapitata* and *C. vulgaris* cells for antioxidant enzymological study. These methods were ultrasonication bath, ultrasonication probe, vortex agitation and bead milling. The degree of algal cell disruption was evaluated and quantified using direct optical microscopy techniques.

MATERIAL AND METHODS

Strain and Cultivation

The microalgae, *R. subcapitata* formerly known as *Selenastrum capricornutum* (strain CCAP 278/4) and *C. vulgaris* (strain CCAP 211/12) were obtained from Culture Collection of Algae and Protozoa (CCAP), UK, and grown in 500-ml Erlenmeyer flasks containing sterile Jaworski’s medium (JM). The cultures were maintained on a shaker (100 rpm) in a culturing chamber at 20°C ± 1°C under continuous irradiance in the range 2000-3000 lux. Algal cells (10⁵ cells/ml) were transferred after 5 days to fresh JM in 50-ml conical flasks and cultivated for 5 days under the same environmental conditions. Algal cells were then harvested in sodium phosphate buffer (pH 7) and treated with either: ultrasonic bath, ultrasonic probe, vortex agitation and bead beating for varying times before being microscopically analysed for cell rupture. Control samples were without treatment and each treatment was done in triplicate.

Methods for cell disruption of algal cells

Ultrasonic bath

Sonication was applied indirectly to algae suspension (5.0 ± 5.4 x 10⁷ cells/ml) inside glass tubes in a 75 W ultrasonic bath (S 80, D-78224 Singen/Htw, Elma)
Lid, Germany) with an ultrasonic wave frequency of 50/60 KHz. The samples were treated for 60 min and 30μl samples were taken for analysis at 15 min intervals. Since the distribution of ultrasonic intensity in an ultrasonic bath is not homogeneous, the aluminum foil test (Mason, 2000) was performed prior to the sample treatment to locate the position with the highest sonication intensity.

Ultrasonic probe

Direct sonication was applied to cell suspensions (5.0 – 5.5 x 10^6 cells/ml) in eppendorf tubes with an ultrasonic processor (Soniprep 150, MPE, UK) equipped with 3 mm exponential tip, at 100% amplitude giving 20 KHz ultrasonic wave frequency for 2, 5, and 10 min. The samples were kept on ice during the sonication process to avoid overheating.

Sonication/Repeatead freezing and thawing (RFT)

Cell suspensions sonicated using the above methods underwent slow freezing at -20 °C after which they were flash frozen in liquid nitrogen followed by slow thawing in ice bath. Three separate trials of freezing/thawing cycles were conducted. 30µl samples of the algae were taken after each freezing-thawing cycle for microscopic analysis.

Vortex agitation

Samples (6.4 – 6.7 x 10^8 cells/ml) were agitated in eppendorf tubes using 0.56-0.7 mm garnet sand in combination with two 3.5 mm zirconium balls (lyzing matrix A, QBiogene, USA) using a vortex mixer (VB3B011, SciQuip Ltd., UK) at 2500 rpm for 5 and 5 min processing time.

Bead mill homogenizer

Algal samples (6.4 – 7.0 x 10^8 cells/ml) were ground in 1.5 ml tubes containing 0.42-0.60 mm glass beads (Sigma) or 1.0 mm silica spheres (lyzing matrix C, MP Biomedicals) or 3.15 mm stainless steel balls (lyzing matrix S, MP Biomedicals) with a beads/solution ratio of 50% vol/vol and 70% vol/vol for 1, 3 and 5 min using a shaking type multirubbe bead beater (FastPrep®-24, MP Biomedicals, France) at 4.5 and 6.5 ms⁻¹.

Quantifying and evaluating cell disruption

In order to quantify the effectiveness of each treatment, the fraction of physically disrupted cells were measured. This was achieved by counting the number of intact cells treated after a specific time interval against those initially determined from the control values (McMillan et al., 2013). A Leica DM5000 microscope was employed in the bright field configuration moving between 10 × and 40 × objectives. The microscope was equipped with a digital camera (Leica DFC50, Leica UK Ltd) linked to a desktop computer using Leica software (LAS EZ, Switzerland) where images were grabbed and stored. 30 μl of the treated algae sample or control was pipetted onto a slide for examination under the microscope. 10 μl of the sample was also pipetted onto the loading area of a haemocytometer and covered with a cover slip, for counting under the microscope (40 x). At each position, the focus was adjusted and the intact cells counted. Dₜ, the total cell disruption at time t, was quantified in terms of the ratio of the average number of intact cells counted after a treatment time (L₁) to the initial counts of intact cells at t = 0, denoted I₀, and then, is given by

\[
Dₜ = \left( \frac{I₁}{I₀} \right) \times 100
\]

where the ratio I₁/I₀ indicates the cell survival probability (McMillan et al., 2013).

RESULTS AND DISCUSSION

Microscopy and Disruption Methods

The effects of the different treatments on cell disruption efficiency are shown in Figure 1 while a view of the suspensions of Raphidocelis and Chlorella cells before and after treatment is given in Figure 2. The algae species were observed to be intact before treatment (Fig 2a and 2b) while damaged or disrupted R. subcapitata and C. vulgaris appeared as empty or ghost cells following bead milling (Fig 2c and 2d respectively). Ghost cells are dead cells with visible outlines devoid of nucleus and cytoplasmic components (Stedman, 1995). The observation of damaged microalgae, Isochrysis galbana, Pavlova sp., Tetraselmis striata, Nanochloropsis sp. and Chlorella sp. as ghost cells following long periods of refrigerated storage has been reported in literature (Espinosa and Allam, 2006). There was no significant difference in the degree of cell disintegration induced by the disruption methods between the phytoplankton species (p > 0.05). The disruption efficiency of the treatments in R. subcapitata and C. vulgaris are as follows: ultrasonic bath (0, 0%); ultrasonic probe (2.3, 0.6%); vortex agitation (0.9, 0.5%) and bead beating (99.6, 99.2%) respectively. Indirect sonication using a bath had no disruptive effects on the microalgae with the cells remaining intact after an hour of treatment. This may be attributed to the heating produced in ultrasonic baths which can be transmitted to the samples, interfering with sonication intensity or cavitation (Patricio et al., 2006).

Surprisingly, liquid shearing with a sonic probe was hardly effective in breaking the algal cells, resulting in less than 3 % disruption of the algae species. Ultrastrong beams delivered about 50 times the ultrasound intensity of an efficient ultrasonic bath (Santos et al., 2008) and its use for rapid algal cell homogenization has been reported in literature (Bierkens et al., 1998; Zargar et al., 2006). Lavoie et al. (2009) achieved 26.8 % and 98 % disruption efficiency in R. subcapitata and C. reinhardtii respectively using sonication and it was the least effective method in breaking Botryococcus cells at 8.8 % (Lee et al., 2010). Its inefficacy is likely due to the differences in the equipment and size of the tip used. The probe or detachable horn is the most important part of a sonicator and its shape or design has an influence on sonication intensity or performance (Santos et al., 2008). The stepped probe is more suitable for micro-applications and transmits a much higher ultrasonic energy efficiency than its flat counterpart. Slow freezing followed by rapid freezing of biological samples is known to induce intracellular ice crystals, and this can lead to the osmotic rupturing of cells on thawing due to the presence of water (Nord and Bier, 1952). However, RFT had no effects on disruption or sonication efficiency in this study, in consonance with previous report by Lavoie et al. (2009). Vortex agitation yielded very poor results with over 99% of cells presenting intact walls after 10 min of agitation. Vortexers are readily available and as a result, have been adapted to bead beat biological samples. However, they lack the power of the true bead beaters, thus they are less effective at cell disruption (Burden, 2012). About 18 % and 99 % disruption of R. subcapitata and C. reinhardtii respectively were obtained using a vortex mixer with cooling using 0.5 mm zirconia-silica beads with beads/solution ratio of 90 % vol/vol and an agitation time of 6 min (Lavoie et al., 2009). Vortexing with quartz sand for 3 min was also found to be adequate in the disruption of D. subplicata (Tukaj and Tukaj, 2010). Its inefficiency in this study possibly lies in the differences in grinding media as well as the mechanical strength of algal cell walls.

Bead Milling

Bead beating with 0.42-0.6 mm glass beads was the most effective treatment method in this study disagreeing with some previous studies that showed bead beating was not as efficient as other approaches (Cheng et al., 2010; Prabakaran and Ravindran, 2011; Sheng et al., 2012; Zheng et al., 2011). It resulted in 83–99.6 % disruption of the algal cells as evidenced by the ghost cells, developing ghosts, and cell debris produced within 1-1.5 min of treatment. Lee et al. (2010) found bead beating to be the most effective method of disintegrating Botryococcus sp. for lipid extraction (28.1 %) while more than 90 % of Chlorella cells were disrupted when passed through different bead mills using glass and zirconium beads in the range 0.3-0.7 mm (Doucha and Livansky, 2008). Bead beating was also found to extract the highest lipid content from wet pellets of Botryococcus braunii when compared to sonication and high pressure homogenization (Lee et al., 1998). The effects of the beads diameter and the agitation speed on the degree of algal cell breakage were studied (Tables 1 & 2). Using beads/solution ratio of 50%vol/vol and maximum speed of 6.5 ms⁻¹, the glass beads (0.42–0.6 mm) caused the highest disruption (99.3 % and 99 %) of Raphidocelis and Chlorella cells respectively. Disruption of the algae increased as beads diameter and the agitation speed increased in beads diameter with the larger 1.1 mm silica beads and 3.15 mm stainless steel balls inducing 56 %, 49 % and 16 %, 19 % disruption respectively in R. subcapitata and C. vulgaris. Less disruption (87 %, 84 %) of Raphidocelis and Chlorella cells respectively was obtained with the glass beads at lower agitation speed of 4.5 ms⁻¹. The impact of disintegration time and the volume of beads to cell suspension ratio on the extent of cell disruption in the algal cells was also investigated (Tables 3 & 4 respectively). The disruption efficacy of the algal cells using a bead size 0.42-0.6 mm was found to increase with beads rolling and exposure time reaching a peak after 5 min. The lowest disruption (83 %, 85.5 %) was obtained using 50% vol/vol bead-solution ratio within 60 seconds exposure time for Chlorella and Raphidocelis cells respectively. The highest disruption resulted after 5 min of processing time with 50 %vol/vol and 70% vol/vol beads loading causing (99.3 %, 99 %) and (99.6 %, 99.2 %) disruption of Raphidocelis and Chlorella cells respectively.

According to Hopkîm (2014), the beads size have the volume ratio of beads to cell suspension are influential on the degree of cell disintegration. About 90 % of Scenedesmus obliquus were disrupted in a bead mill using 0.35-0.50 mm glass beads as compared to 0.5-0.7 mm beads (80 % disruption) and 1.1-1.2 mm beads (50 % disruption) (Hedenskog et al., 1969). More Raphidocelis and Chlorella cells were disrupted when the speed of agitation was increased in this study. The influence of agitation time on the degree of cell disintegration of bead mills were earlier investigated by Doucha and Livansky (2008), achieving 99% disintegration of Chlorella cells with optimum glass bead diameter of 0.42-0.58 mm, 82 % beads rolling and agitator speed of 14ms⁻¹ using a Dyno-Mill KDL-Pilot.

The discrepancies among some of the published reports on the efficiency of bead mill in algal cell disintegration may be attributed to various factors such as variations in container shape, agitator speed, bead size, bead composition and...
Effects of bead diameter and agitation speed on the degree of cell disruption of *R. subcapitata* and *C. vulgaris*. 1 Sonication bath, 2 vortex agitation, 3 sonication probe, 4 bead mill (6.5 m/s, 5 min and 70 % beads filling). Error bars represent standard error of mean.

![Figure 1](image1.png)

**Figure 1** Effects of different methods on cell disruption of *R. subcapitata* and *C. vulgaris*. 1 Sonication bath, 2 vortex agitation, 3 sonication probe, 4 bead mill (6.5 m/s, 5 min and 70 % beads filling). Error bars represent standard error of mean.

**Table 1** Effects of bead diameter and agitation speed on the degree of *R. subcapitata* cell disintegration in the bead mill after 5 min

<table>
<thead>
<tr>
<th>Trial no</th>
<th>Beads filling (%)</th>
<th>Agitation speed (ms⁻¹)</th>
<th>Degree of cell disruption (%)*</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>50</td>
<td>4.5</td>
<td>87 ± 0.86</td>
</tr>
<tr>
<td>2</td>
<td>50</td>
<td>6.5</td>
<td>99.3 ± 0.45</td>
</tr>
<tr>
<td>4</td>
<td>50</td>
<td>6.5</td>
<td>56 ± 4.1</td>
</tr>
<tr>
<td>5</td>
<td>50</td>
<td>6.5</td>
<td>16 ± 2.0</td>
</tr>
</tbody>
</table>

*Values represent mean of three replicate assays ± standard error of mean (SEM)

**Table 2** Effects of bead diameter and agitation speed on the degree of *C. vulgaris* cell disintegration in the bead mill after 5 min

<table>
<thead>
<tr>
<th>Trial no</th>
<th>Beads filling (%)</th>
<th>Agitation speed (ms⁻¹)</th>
<th>Degree of cell disruption (%)*</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>50</td>
<td>4.5</td>
<td>84 ± 1.5</td>
</tr>
<tr>
<td>2</td>
<td>50</td>
<td>6.5</td>
<td>99 ± 0.2</td>
</tr>
<tr>
<td>4</td>
<td>50</td>
<td>6.5</td>
<td>49 ± 4.3</td>
</tr>
<tr>
<td>5</td>
<td>50</td>
<td>6.5</td>
<td>19 ± 1.5</td>
</tr>
</tbody>
</table>

*Values represent mean of three replicate assays ± standard error of mean (SEM)

**Figure 2** Suspension of *Raphidocelis* cells before (a) and after (c) treatment (96 % disruption efficiency) and *Chlorella* cells before (b) and after (d) treatment (93 % disruption efficiency), (FastPrep®-24 homogenizer, glass beads 0.42-0.6 mm, agitation speed 6.5 ms⁻¹, 3 min processing time, arrows showing ghost cells).

**Table 3** Influence of disintegration time and beads filling on the degree of *R. subcapitata* cell disintegration in the bead mill at maximum speed of 6.5 ms⁻¹

<table>
<thead>
<tr>
<th>Trial no</th>
<th>Beads filling (%)</th>
<th>Disruption time (min)</th>
<th>Degree of cell disruption (%)*</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>50</td>
<td>1</td>
<td>85.5 ± 4.5</td>
</tr>
<tr>
<td>2</td>
<td>50</td>
<td>3</td>
<td>96 ± 2.5</td>
</tr>
<tr>
<td>4</td>
<td>50</td>
<td>5</td>
<td>99.3 ± 0.45</td>
</tr>
<tr>
<td>6</td>
<td>70</td>
<td>5</td>
<td>99.6 ± 0.1</td>
</tr>
</tbody>
</table>

*Values represent mean of three replicate assays ± standard error of mean (SEM)

**Table 4** Influence of disintegration time and beads filling on the degree of *C. vulgaris* cell disintegration in the bead mill at maximum speed of 6.5 ms⁻¹

<table>
<thead>
<tr>
<th>Trial no</th>
<th>Beads filling (%)</th>
<th>Disruption time (min)</th>
<th>Degree of cell disruption (%)*</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>50</td>
<td>1</td>
<td>83 ± 2.1</td>
</tr>
<tr>
<td>2</td>
<td>50</td>
<td>3</td>
<td>93 ± 2.6</td>
</tr>
<tr>
<td>4</td>
<td>50</td>
<td>5</td>
<td>99 ± 0.2</td>
</tr>
<tr>
<td>6</td>
<td>70</td>
<td>5</td>
<td>99.2 ± 0.1</td>
</tr>
</tbody>
</table>

*Values represent mean of three replicate assays ± standard error of mean (SEM)
CONCLUSION
In addition to identifying ghost cells as indicators of cell disruption, the findings from this study clearly revealed that bead miling, using the appropriate parameters, is one of the most effective methods in disrupting microalgal species including the resilient C. vulgaris and R. subcapitata cells for the recovery of intracellular products.

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