Stem cell bioprocessing: fundamentals and principles

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In recent years, the potential of stem cell research for tissue engineering-based therapies and regenerative medicine clinical applications has become well established. In 2006, Chung pioneered the first entire organ transplant using adult stem cells and a scaffold for clinical evaluation. With this a new milestone was achieved, with seven patients with myelomeningocele receiving stem cell-derived bladder transplants resulting in substantial improvements in their quality of life. While a bladder is a relatively simple organ, the breakthrough highlights the incredible benefits that can be gained from the cross-disciplinary nature of tissue engineering and regenerative medicine (TERM) that encompasses stem cell research and stem cell bioprocessing. Unquestionably, the development of bioprocess technologies for the transfer of the current laboratory-based practice of stem cell tissue culture to the clinic as therapeutics necessitates the application of engineering principles and practices to achieve control, reproducibility, automation, validation and safety of the process and the product. The successful translation will require contributions from fundamental research (from developmental biology to the ‘omics’ technologies and advances in immunology) and from existing industrial practice (biologics), especially on automation, quality assurance and regulation. The timely development, integration and execution of various components will be critical—failures of the past (such as in the commercialization of skin equivalents) on marketing, pricing, production and advertising should not be repeated. This review aims to address the principles required for successful stem cell bioprocessing so that they can be applied deftly to clinical applications.

Keywords: stem cell; bioprocessing; tissue engineering

1. INTRODUCTION

The success of stem cell bioprocessing relies on robust and reproducible culture conditions and processes. For stem cell bioprocessing, this includes the scale-up of stem cells to a differentiated end product of sufficient quality and quantity for clinical and commercial goals. The considerable cost with respect to consumables, labour and time as well as the inherent variability in manual processes not only make this an unattractive option but also render it commercially unviable. Automation and the use of an efficient bioprocess paradigm are imperative for the creation of successful clinical products.

2. DESIGN PRINCIPLES

The design principles (Lim et al. 2007) pertinent to stem cell bioprocessing can be categorized into three groups: (i) process components, (ii) process requirements, and (iii) process function, as summarized in figure 1. A combination of generic, ‘off-the-shelf’ and personalized manufacturing paradigms must be considered as no single technology satisfies all requirements. The process components consist of the cell source and type, and the elucidation of appropriate signals required for cellular development, in addition to the scaffold and bioreactor design and implementation. Process requirements address practical considerations of bioprocessing, satisfying good manufacturing practices (GMPs) such as quality assurance, bioprocess monitoring control and automation, in addition to...
product transportation. Finally, the process components and process requirements need to ensure the end product’s functionality, integration and longevity, to name but a few vital factors included in the process function. This review will address only the process components and requirements.

2.1. Process components

Identification of the optimal cell source and important signals for cellular development, as well as delivering a suitable growth mimicry in terms of the scaffold used, are essential tailor-made requirements in stem cell bioprocessing and tissue engineering. In addition, provision of a controlled culture environment through the use of an appropriate bioreactor is critical. Clearly, application-specific approaches are required; however, certain common methodologies can be applied.

2.1.1. Stem cells. A stem cell is defined as a clonal precursor producing either identical stem cells (self-renew) or differentiating into other specialized cells. There are several classes of stem cells, each one presenting its own challenges and benefits for the manufacture of therapeutics for the clinic (figure 2). This review will focus only on embryonic and adult stem cells.

Embryonic stem cells (ESCs) are capable of indefinite expansion, and are pluripotent. Currently, attempts are being made to generate patient-tailored ESC lines that overcome immunological complications (Takahashi & Yamanaka 2006). However, they also are capable of teratoma formation, are difficult to control with respect to their differentiation fate, and elicit ethical considerations due to the destruction of the embryo, until recent advances (Petersen & Nilsson 2007). By contrast, adult stem cells do not elicit ethical considerations and are obtained directly from the patient or the donor. For autologous grafts, adult stem cells do not present immunogenic complications on implantation and, depending on the source, could be obtained relatively easily. Adult stem cells, however, do present their own unique challenges. Specifically, they have been found to vary in quality with respect to the age and health of the donor/patient, and differentiation is often restricted to the original lineage of the cell source (multipotent). Some adult stem cell types have been shown to be able to transdifferentiate; however, most adult stem cell therapies require cells to be harvested from specific sources (Smith et al. 2007). An important consideration with respect to bioprocessing is the expansion potential of these cells. ESCs are believed to have an almost unlimited expansion capability while adult stem cells, depending on donor and source, may be capable of only tens of population doublings.

Adult stem cells Adult stem cells, or somatic stem cells, are undifferentiated cells found throughout the adult body. They reside within specific niches in the different organs where they produce cells that regenerate damaged tissue and replace dying cells. Adult stem cells have been used clinically since the 1950s when stem cell transplantation was pioneered using bone marrow-derived stem cells. Since then, other types of adult stem cells have been identified in the body, which
have offered alternative tissue repair possibilities and originate from easier to harvest cell sources, such as fat, skin, olfactory cells and peripheral blood (PB). Haemopoietic stem cells (HSCs) can be isolated from several sources besides the adult bone marrow. These include PB, umbilical cord blood (CB) and the placenta. Isolation is generally carried out by fluorescence-activated cell sorting (FACS) or magnetic cell sorting (MACS), an immunomagnetic selection technique where paramagnetic microbeads or adsorption columns coupled with specific monoclonal antibodies (e.g. anti-CD34) are employed to capture and enrich the target cells in the presence of a magnetic field (Sandstrom et al. 1995; Roots-Weiss et al. 1997; Kekarainen et al. 2006). Isolation strategies vary from the depletion of committed lineage cells and late multipotent progenitor cells using a variety of lineage-specific markers, or by positive selection using markers such as CD34⁺, CD59⁺, CD90⁺ and CD133⁺ (Baum et al. 1992). HSCs isolated from the different sources possess roughly similar properties; however, the volumes harvested vary considerably as do the specific populations of haemopoietic stem cells (McAdams et al. 1996; Urbano-Ispizua 2007). HSCs are capable of giving rise to all blood cell types, including the myeloid and lymphoid lineages. Some transdifferentiation capability has been shown by various research groups reporting differentiation of HSCs into skeletal muscle (Ferrari et al. 1998), cardiac muscle (Bittner et al. 1999), liver (Petersen et al. 1999; Theise et al. 2000a,b), endothelial cells (Shi et al. 1998) and neurons (Eglitis & Mezey 1997; Brazelton et al. 2000; Mezey et al. 2000). Interestingly, it appears that HSCs may be capable of sustained symmetrical division, explaining how a small number of HSCs are capable of repopulating the bone marrow rapidly after depletion (Sherley 2002). The rapid and prolonged expansion capability of HSCs renders them a suitable candidate cell type for bioprocessing.

Bone marrow was originally the only source of progenitor cells used for allogeneic transplantation. In the early 1990s, mobilized [mainly by granulocyte colony-stimulating factor (G-CSF)] PB was also introduced clinically, which presented certain advantages. For instance, a greater volume of cells were able to be harvested from PB, thus increasing the number of progenitor and accessory cells in the collection. For transplantation, this leads to faster myeloid and lymphoid recovery facilitating engraftment and full T-cell chimerism (Sheridan et al. 1992; Bensinger et al. 1993; Chao et al. 1993). Additionally, irrespective of the small inherent risks involved with mobilized PB collection and the use of G-CSF (Martinez et al. 1996, 1999; Majolino et al. 1997; Tabiolo et al. 1997; de la Rubia et al. 1999), the risks and side effects involved in the BM donation are
considerably greater in comparison (Buckner et al. 1984; Stroncek et al. 1993; Pulsipher et al. 2006). For bioprocessing, however, the greater volume of cells is offset by their lower expansion capability, making PB HSCs less amenable than bone marrow-derived stem cells. In the late 1990s, CB was used as a source of HSCs and, by 2004, 3 per cent of the total allogeneic transplants in Europe and 7 per cent of the unrelated allogeneic transplants used CB (Gratwohl et al. 2005). However, the use of CB is clinically hindered, primarily due to its low harvest volume. This causes complications resulting in graft failure or relapse. The therapeutic potential of CB, however, should not be underestimated; its low human leucocyte antigen (HLA) compatibility restrictions, low-risk collection from donors and off-the-shelf availability render CB a promising source of HSCs provided sufficient numbers can be obtained (Locatelli et al. 1999; Barker et al. 2001a). Consequently, this remains an active research area with a number of different approaches currently being tested, such as double CB transplants (Barker et al. 2001b, 2005) and co-infusion of CB with either haploidentical haemopoietic progenitor cells (Fernandez et al. 2003; Magro et al. 2006) or mesenchymal stroma cells from a third party (Noort et al. 2002). In terms of ex vivo expansion of haemopoietic stem and progenitor cells, Koller et al. (1993a,b) pioneered the concepts of stem cell bioprocessing by using scalable perfusion bioreactor systems for the expansion of haemopoietic stem and progenitor cells from bone marrow, PB or CB in the early 1990s as discussed below.

Mesenchymal stem cells (MSCs) are multipotent stem cells that are primarily isolated from the bone marrow. Other populations of MSCs have been found, including from PB (Zwaïler et al. 2000), adipose tissue (Zuk et al. 2001), skin tissue (Chummeng & Tianmin 2004), trabecular bone (Sottile et al. 2002) and umbilical CB (Erices et al. 2000). MSCs have been shown to differentiate into osteoblasts, chondrocytes and adipocytes (Muraglia et al. 2000). A broader differentiation capability of human MSCs (hMSCs) has also been suggested with hMSCs being differentiated into muscle cells (Taylor & Jones 1982) and limited myogenic potential being observed when human MSCs were co-cultured with murine skeletal myocytes (Lee, J. H. et al. 2005). Cardiomyocyte differentiation has also been demonstrated (Lee, J. H. et al. 2005), resulting in cells with the proper morphology, physiological cardiomyogenic response and expression of appropriate adrenergic and muscarinic surface receptors. MSCs are obtained by either (i) direct plating, which relies on the MSC adherent properties (Luria et al. 1971), or (ii) through the use of cell surface markers. Both methodologies present certain limitations. Specifically, the homogeneity of the purified population, which is obtained through adherence to culture plastic, is not high with only 10–20 per cent of the cells capable of multipotent differentiation (Phinney et al. 1999). In addition, in the absence of a ubiquitous MSC-specific marker, marker selection varies between groups, with different protocols using a combination of CD146, CD271 (Buhring et al. 2007), CD105 (Aslan et al. 2006), CD29 (Jackson et al. 2007) and Stro-1 (Grothhos et al. 1994). Culture of MSCs has indicated that isolated cells are limited to 40 population doublings—25 for elderly donors (Stenderup et al. 2003)—which is the result of telomere shortening due to the loss of telomerase activity in tissue culture (Simonsen et al. 2002). Furthermore, of importance to clinical applications, cultured cells have demonstrated genetic instability after 250 population doublings when using this method of extended expansion (Serakinci et al. 2006), highlighting the need to develop efficient scale-up methods and effective differentiation protocols so that sufficient numbers of target cells can be delivered without prolonged ex vivo culture time.

In general, a firm correlation exists between the expansion/differentiation ability and availability of the stem cells and their clinical applicability in terms of process complexity, and ethical and regulatory restrictions. The implication of this correlation is that it directs, and potentially restricts, the manufacturing and scale-up approaches available and the associated costs. Clearly different stem cell types will require different operating conditions; however, this variability is also likely in any one stem cell type from initial expansion to differentiation. The process is transient in nature and operating conditions require tight control. Finally, stem cell standardization necessitates the identification of appropriate markers as well as the development of suitable evaluation assays.

ESCs The discovery of mouse ESCs over 25 years ago (1981) and the derivation and culture of human embryonic stem cells (hESCs) more recently (1998) represent major advances in biology with immense potential for tissue engineering and regenerative medicine (Hirai 2002; Prelle et al. 2002; Keller 2005). Culture of ESCs requires precise methodology in maintaining their undifferentiated state and directing their differentiation. The traditional ESC culture process is fragmented consisting of a maintenance/expansion phase, an embryoid body (EB) formation phase, followed by terminal differentiation to the desired cell lineage. Each phase presents obstacles that need to be overcome before widespread clinical application becomes standard practice. Specifically, during maintenance/expansion, the ESCs, especially the hESCs, are traditionally cultured on feeder cells, such as mouse embryonic fibroblast (MEF), in order to retain their undifferentiated state (Thomson & Marshall 1998, Reubinoff et al. 2000). Consequently, the quality of the feeder cells and the consistency of the culture conditions are critical elements in generating high-quality ESCs that retain their ‘stemness’. Additionally, it has been suggested that prolonged culture of hESCs can ultimately result in culture adaptation where the hESCs lose their true undifferentiated state (Draper et al. 2004). More recently, culture protocols have been developed in order to move away from the traditional MEF-dependent methods to more clinically relevant feeder-free systems. The use of human feeder cells, such as foetal muscle, foetal skin, adult fallopian tube epithelial cells (Richards et al. 2002, 2003; Amit et al. 2003), foreskin fibroblasts (Hovatta et al. 2003), adult marrow cells (Cheng et al. 2003), adult endometrial cells
Several methods of expansion and differentiation of adherent ESCs exist, in either static or bioreactor systems, including EB cultivation (Fok & Zandstra 2005; Schroeder et al. 2005; Cormier et al. 2006; zur Nieden et al. 2007), encapsulation (Dang et al. 2004; Bauwens et al. 2005; Randle et al. 2007), use of microcarriers as a substrate for cell attachment (Fok & Zandstra 2005; Abranches et al. 2007), automated tissue platforms (Narkilathi et al. 2007; Terstegge et al. 2007) and perfusion bioreactor systems (Fong et al. 2005; Oh et al. 2005). Currently, the most robust method for generating differentiated cells from ESCs is through the formation of embryoid bodies, where ESCs spontaneously differentiate and form tissue-like spheroids in suspension culture (Dang et al. 2004). EB differentiation has been shown to recapitulate aspects of early embryogenesis, including the formation of a complex three-dimensional arrangement where cell-cell and cell–matrix interactions are thought to support the development of the three embryonic germ layers and their derivatives (Keller 1995; Isakovitz-Eklor et al. 2000). The length of EB culture is dependent on the desired cell type, and EB differentiation appears to correlate temporally well with the post-implantation development of embryos (Keller 1995). Mesodermal and endodermal precursors form within a few days, whereas some endodermal cell types may benefit from more extended culture times, up to 10 days, to a stage where most EBs have cavitated and become cystic (Abe et al. 1996; Leahy et al. 1999).

The conventional methods of inducing EB formation, which include suspension culture in bacterial-grade dishes, hanging drops and methylcellulose, have been shown to produce small numbers of EBs, limiting the ability of the ESCs to differentiate into the desired cell type. Following formation of EBs, cells are returned to adherent culture conditions upon which specialized cells develop in the outgrowth area of differentiation (Kurosawa 2007). However, the conventional methods of EB formation are impractical for large-scale cell production (Fok & Zandstra 2005) and generate heterogeneous cell types not suitable for therapeutic applications. To address this, the use of bioreactors is essential to meet large-scale production needs, with the inherent benefit of amenability to process control strategies.

2.1.2. Operating conditions and signals. Successful stem cell bioprocessing, in terms of expansion and differentiation, depends on the control of key process variables: (i) the physiochemical environment, (ii) nutrients and metabolites, and (iii) growth factors (Lim et al. 2007). Physiochemical culture parameters include pH, dissolved oxygen and carbon dioxide tensions and temperature. Even though almost all expansion cultures are run at pH 7.4, 20 per cent oxygen, 5 per cent carbon dioxide and 37°C, the proliferation and differentiation of different stem cell cultures have been found to operate at different optima. Specifically, higher pH (pH 7.60) was found to enhance the differentiation and maturation of megakaryocytic progenitors and also favours erythroid differentiation (McAdams et al. 1998). By contrast, granulopoiesis is optimal at a lower pH (pH 7.21), which enhances granulocyte colony-stimulating factor receptor (G-CSFR) expression and granulocyte proliferation and differentiation (Hevehan et al. 2000b). More work has been done on the effects of pH on differentiation as opposed to optimization for the expansion of stem cells; interestingly, a pH of 7.1 has been found to increase the expansion of megakaryocyte progenitors and also favours erythroid differentiation (McAdams et al. 1998). Oxygen tension is a critical factor in haemopoiesis. In culture, oxygen tension can greatly affect the expansion of cells by modulating the production of cytokines, surface markers and transcription factors (Mostafa et al. 2001). Oxygen demand varies for different cell lineages and maturation stages—low oxygen concentration (5%) is better for progenitor cell expansion, while high oxygen (20%) promotes the growth of mature megakaryocytes and erythrocytes (McAdams et al. 1998). Low oxygen tension was also found to enhance granulocyte differentiation (Hevehan et al. 2000b). In murine embryonic stem cells (mESCs), oxygen tension has a considerable effect on differentiation. The mESCs cultured in 40 per cent oxygen retained alkaline phosphatase activity and Oct-4 gene expression for longer than at lower oxygen tensions, and normal oxygen tension (20%) allowed spontaneous differentiation of the mESCs (Kurosawa et al. 2006). These findings establish the presence of pH and oxygen gradients in vivo. Fewer investigations have been conducted on the effects of temperature, though in most haemopoietic cell cultures, the operating temperature is maintained at 37°C. Interestingly, a higher temperature of 39°C has recently been found to enhance megakaryopoiesis in CB34-enriched CB cell cultures (Proulx et al. 2004), and a lower temperature...
(32°C) has been shown to reduce oxidative damage while retaining viability and population doubling times in MSCs. MSCs are obtained in relatively low numbers and extended in vitro expansion can lead to reduced viability from oxidative damage. Therefore, optimized culture conditions are of great importance in producing suitable numbers of long-term viable MSCs for successful engraftment. This evidence highlights the fact that process variations in the culture environment could be strategically applied to direct and manipulate cellular behaviour in vitro. Nutrient and metabolite concentrations determine cell growth, differentiation and death in a culture, and therefore should be closely monitored and controlled in bioprocessing. Growth factors regulate stem cell behaviour by providing survival, proliferation and differentiation signals to the cells. They have specific functions, both positive and negative in nature, and can act on either a specific cell lineage or multiple lineages. Interactions between these growth factors and/or with other process parameters are in many cases not fully understood. It is therefore crucial to quantify and qualify these effects and their interactions with respect to one another in order to tailor the culture process for optimal production of a specific cell type population.

2.1.3. Cell adhesion and scaffolds. Organogenesis, which involves the finely regulated proliferation and differentiation of stem cells, depends, to a large extent, on cell adhesion and the provision of a three-dimensional growth environment. Cell adhesion to the extracellular matrix (ECM) is mediated by a class of heterodimeric transmembrane cell surface receptors called integrins. ECM proteins typically affect cell behaviour by binding to specific integrin cell surface receptors, thus activating intracellular signalling pathways that control gene expression, cytoskeletal organization and cell morphology (Giancotti & Ruoslahti 1999; Hynes 2002). For instance, fibroblastic, epithelial and endothelial cells must adhere to appropriate ECM components in order to survive, a phenomenon termed ‘anchorage dependence’ (Danen & Yamada 2001). Furthermore, the signal-transducing capacity of integrins has been indicated as a likely regulator of the in vitro differentiation of several types of stem cells and committed progenitor cells (Prosper & Verfaillie 2001; Schneider et al. 2001; El-Amin et al. 2002; Carvalho et al. 2003; Salasznyk et al. 2004; Klees et al. 2005; Chastain et al. 2006; Hayashi et al. 2007). Consequently, differentiation of stem and progenitor cells can be manipulated through the modification of cell culture surfaces. Indeed, numerous studies have shown that the modulation of cell adhesion properties via surface chemistry, surface micro-architecture or the types of ECM ligands present on the culturing surfaces could be applied to regulate stem cell differentiation (Prosper & Verfaillie 2001; Schneider et al. 2001; El-Amin et al. 2002; Boyan et al. 2003; Carvalho et al. 2003; Levenberg et al. 2003; Salasznyk et al. 2004; Klees et al. 2005; Chastain et al. 2006; Sun et al. 2006; Hayashi et al. 2007). Specifically, integrin activation has been implicated in the maintenance of mESCs’ self-renewal and differentiation (Hayashi et al. 2007). When the mESCs were cultured on surfaces coated with type I and IV collagen, gelatine or poly-lysine, the cells remained undifferentiated; however, the mESCs differentiated into primitive ectoderm even in the presence of LIF when cultured on surfaces coated with ECM ligands, such as laminin or fibronectin. MSC adhesion to culture surfaces coated with various ECM proteins (fibronectin, collagen I, collagen IV, vitronectin and laminin-1) has also been examined as a function of their osteogenetic potential (Salasznyk et al. 2004). It was reported that MSCs adhered with varying affinity (fibronectin>collagen I>collagen IV>vitronectin>laminin-1), which suggested that appropriate ECM contact alone may be sufficient to induce differentiation in these cells.

The majority of research on ESC differentiation in vitro has so far focused on their differentiation in adherent culture following EB formation. However, as discussed above, precisely controlled surface chemistry and ECM ligand presentation can enable regulation of gene expression with increased differentiation efficiency towards the desired target cell. Additionally, for many tissue engineering applications, the incorporation of differentiated cells into higher order structures will be essential for implants to be functional (Rippon & Bishop 2004). To this end, the use of three-dimensional scaffolds has been explored and needs to be taken into consideration. Designing a tissue-engineered scaffold requires the consideration of a large number of variables: material; porosity; pore size; mechanical stability; degradability; biocompatibility; hydrophobicity; and bioactivity (table 1). Scaffolds are typically fabricated by either natural materials, which are inherently bioactive but lack mechanical strength, or synthetic materials, which lack inherent bioactivity but are mechanically strong and can be engineered with the desirable macro- (shape) and microarchitecture (pore size and porosity), as well as being modified to possess desired bioactive properties (Safinia et al. 2006) that will facilitate cellular growth and organogenesis in a biomimetic manner (Stevens & George 2005). Polymeric scaffolds should possess a network of channels and interconnected pores to facilitate mass transport of nutrients/metabolites and to guarantee the penetration of large numbers of cells allowing the formation of cellular associations (Elema et al. 1990). Pore size and high surface area to volume ratio are also key parameters for cell spreading and expansion (Kim et al. 2005). For in vivo applications, the impact of pore size and osteogenic differentiation was studied using polyHIPE polymer; the minimum recommended pore size is suggested to be 100 μm (Hulbert et al. 1970) and the maximum to be 500 μm (Karageorgiou & Kaplan 2005) with the larger pores registering formation of fibroblastic tissue (Wake et al. 1994). Pore size affects cellular adhesion, viability, distribution and formation of an ECM by specific cell types; it has been suggested that the pore size in scaffolds can be used to preferentially support or prevent the ingrowth of specific cells. A high surface area to volume ratio is necessary for in vitro cell attachment, proliferation and subsequent matrix deposition (Zeltinger et al. 2001).
<table>
<thead>
<tr>
<th>Scaffold Material</th>
<th>Cultured Cell Type</th>
<th>Biodegradable</th>
<th>Regulatory Approval</th>
<th>Scaffold Properties</th>
<th>References</th>
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<tbody>
<tr>
<td>poly(l-lactic-co-glycolic acid) (PLGA)</td>
<td>human skin fibroblasts, porcine oesophageal smooth muscle cells (ESMCs), BM cells, chondrocytes, ligament and synovial cells</td>
<td>yes</td>
<td>yes</td>
<td>porosity/pore size: vary depending on polymer ratio (80–95%)/125–500 μm</td>
<td>Yang, H. et al. (2002), Yang, J. et al. (2002), Wu &amp; Ding (2004) and Uematsu et al. (2005)</td>
</tr>
<tr>
<td>polyglycolic acid (PGA)</td>
<td>chondrocytes, 3T3 fibroblasts, human foetal extensor tendons and BM MSCs</td>
<td>no</td>
<td>yes</td>
<td>surface area: 2 m² g⁻¹; pore size: 100–300 μm</td>
<td>Drewa et al. (2006), Moutos (2006), Hannouche et al. (2007) and Wang et al. (2008)</td>
</tr>
<tr>
<td>poly l-lactic acid (PLLA)</td>
<td>3T3 fibroblasts, neural stem cells, MSCs and mESCs</td>
<td>yes</td>
<td>yes</td>
<td>porosity/pore size: 95%/180 μm</td>
<td>Yang H. et al. (2002), Yang J. et al. (2002), Taqvi &amp; Roy (2006) and Hwang do et al. (2008)</td>
</tr>
<tr>
<td>macroporous collagen carriers</td>
<td>human fibroblasts and CB CD34⁺, CHO-K1, BHK-21, bovine endothelial and murine BM cells</td>
<td>yes</td>
<td>yes</td>
<td>pore size: large rectangular pores 100–1000 μm</td>
<td>Wang et al. (1995), Itoh et al. (2001) and Panoskaltsis et al. (2005)</td>
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<tr>
<td>cellulose porous microspheres</td>
<td>stromal cells, osteoclasts, murine BM and human BM MNCs</td>
<td>yes</td>
<td>yes</td>
<td>surface area: 2.8 m² g⁻¹; pore size: 100 μm</td>
<td>Tomimori et al. (2000) and Mantalaris et al. (2004a,b)</td>
</tr>
<tr>
<td>porous biomatrix (Cellfoam)</td>
<td>human BM CD34⁺ cells, haemopoietic progenitor cells (HPCs), 5/9 Ma3-18</td>
<td>no</td>
<td>no</td>
<td>porosity/pore size: 80%/NR</td>
<td>Banu et al. (2001)</td>
</tr>
<tr>
<td>tantalum-coated porous biomaterial (TCPB)</td>
<td>human CD34⁺ HSC and BM stromal cells</td>
<td>no</td>
<td>no</td>
<td>porosity/pore size: 90%/300 μm</td>
<td>Rosenzweig et al. (1997) and Bagley et al. (1999)</td>
</tr>
<tr>
<td>polyester non-woven fabric porous disc carriers (Fibracel)</td>
<td>mouse BM and stroma cells</td>
<td>yes</td>
<td>FDA-validatable in their original form</td>
<td>surface area: 0.12 m² g⁻¹; 6.5 cm² per disc</td>
<td>Takagi et al. (1999) and Tomimori et al. (2000)</td>
</tr>
<tr>
<td>nylon filtration screen</td>
<td>human BM mononuclear cells (MNCs)</td>
<td>yes</td>
<td>FDA-validatable in their original form</td>
<td>porosity/pore size: NR/210 μm</td>
<td>Naughton et al. (1991)</td>
</tr>
<tr>
<td>porous gelatin microspheres (CultiSpher-G)</td>
<td>human BM stroma and MNCs, human nasal chondrocytes</td>
<td>yes</td>
<td>FDA-validatable in their original form</td>
<td>porosity/pore size: NR/5–15 μm</td>
<td>Xiong et al. (2002)</td>
</tr>
<tr>
<td>non-woven polyethylene terephthalate (PET)</td>
<td>human CB MNCs and CD34⁺</td>
<td>yes</td>
<td>FDA-validatable in their original form</td>
<td>porosity/pore size: 85%/10–60 μm</td>
<td>Li et al. (2001)</td>
</tr>
<tr>
<td>porous polyvinyl format (PVF)</td>
<td>murine BM cells and hepatocytes</td>
<td>no</td>
<td>no</td>
<td>porosity/pore size: 90%/130 μm</td>
<td>Tun et al. (2002)</td>
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</table>
Biocompatibility is also an important factor in the design of synthetic scaffolds, especially for clinical applications. Furthermore, in the event of biodegradable scaffolds, the degradation rate can induce inflammation due to the localized acidity of hydrolysed products as in the case of poly(-lactic-co-glycolic acid) and needs to be taken into consideration (Ubersax et al. 2006). Finally, the lack of cell recognition signals and the low surface energy or the hydrophobicity of many synthetic polymers impose many challenges and constrains (Yang J. et al. 2002). Whereas past wisdom required bioinert scaffolds, the requirement for engineered bioactivity is essential to achieve proper organogenesis (Shim et al. 2003). Specifically, synthetic polymers have been hybridized with fibres and proteins to combine the distinct advantages of each for cell growth and interaction (Dunn et al. 1997; Chen et al. 2004; Vance et al. 2004; Battista et al. 2005). The use of cell adhesion peptides (RGDS, KQAGDV and VAPG) has also been widely spread although some studies show that peptides initially increase cell attachment; however, they affect proliferation and matrix production (Mann & West 2002).

Diverse techniques are available to produce scaffolds with a network of channels and interconnected pores, with different pore sizes and surface area to volume ratio, biocompatible and hybridized with various molecules. A novel microscale three-dimensional technique was used in an attempt to recreate the mechanical properties of a native articular cartilage where polyglycolic acid (PGA) yarns were woven into an orthotropic porous textile (Moutos 2006). The investigation showed that three-dimensional woven scaffolds can be designed to mimic the articular cartilage with anisotropic, nonlinear and viscoelastic properties. Additionally, numerous groups have also tried to mimic bone marrow using scaffolds derived from natural materials, such as collagen carriers (Wang et al. 1995), cellulose porous microspheres (Tominori et al. 2000; Mantalaris et al. 2004a) or from polymers such as porous biomatrix (Cellfoam) (Bannu et al. 2001), tantalum-coated porous biomaterial (TCPB) (Bagley et al. 1999), polyester non-woven fabric porous disc carriers (Fibra-cel) (Sasaki et al. 2003), nylon filtration screen (Naughton et al. 1991), porous gelatin microspheres (CultiSpher G) (Xiong et al. 2002), non-woven polyethylene terephthalate (PET) (Li et al. 2001) or porous polyvinyl format (PVF) (Tun et al. 2002). Other techniques used to create scaffolds include microfabrication and solid free form creation using micro-to-nanoscale features; examples include photo-patterned hydrogels or microfabricated scaffolds with large rectangular pores (Liu & Bhatia 2002). Three-dimensional scaffolds that express structural and physiological features can also be designed to resemble native cardiac muscle using a variety of materials such as foetal or neonatal rat cardiac myocytes on collagen fibres (Akins et al. 1999) or porous collagen scaffolds (Freed et al. 2006).

Supplying nutrients to the seeded cells in large constructs needs to be considered when designing scaffolds; thin scaffolds (less than 100 µm) are feasible without an internal blood/nutrient supply, but thicker scaffolds (more than 100 µm) need to be engineered to contain a ‘capillary network’ as part of the structure. Angiogenesis can be induced by the addition of growth factors such as vascular epithelial growth factor (VEGF) that promotes vascularization (Soker et al. 2000). An alternative to overcome the problem is to design materials that induce vascularization even without the use of growth factors; some examples include the use of expanded PTFE (ePTFE; Boswell & Williams 1999), porous polyvinyl alcohol (PVA; Sharkawy et al. 1998) and polyacrylate containing traces of methacrylic acid (MMA; Gorbet et al. 2003).

The hESCs have been differentiated on polymeric scaffolds designed to support complex tissue structures resulting in the formation of structures with the characteristics of neural tissues, cartilage and liver, as well as a network of blood vessel-like tubules (Levenberg et al. 2003), suggesting that the ESCs might be able to generate histologically complete tissue constructs (Rippon & Bishop 2004). In addition, the use of hydrogels represents another useful method for in vitro organogenesis. Traditionally, alginate has been extensively used as it possesses good characteristics, such as low immunogenicity and controllable biodegradability, for tissue engineering applications. Furthermore, alginate hydrogels produce low mechanical or frictional irritation to the surrounding tissue, can be physically and chemically modified for specific surgical applications, and have high permeability for transport of nutrients and metabolites (Chia et al. 2000; Uludag et al. 2000; Bienaime et al. 2003). Magyar et al. (2001) encapsulated mESCs that were capable of forming embryoid bodies, followed by smooth muscle lineage differentiation. Randle et al. (2007) demonstrated a reproducible bioprocess for the production of osteogenic cells from mESCs, integrating several laborious culture steps within a bioreactor. Chondrogenic differentiation from MSCs encapsulated in alginate beads has also been reported (Ma et al. 2003). As with other biomaterials, the chemical and physical properties of the hydrogels may be altered to improve tissue development (Elisseff et al. 2005). A combination of alginate with gelatin has been found to provide a biodegradable delivery vehicle for tissue engineering applications (Balakrishnan & Jayakrishnan 2005).

2.1.4. Mechanical stimulation. Mechanical stimuli on cells provide an additional dimension to the complexity of the microenvironment that is often overlooked in cell culture systems. Mechanical stress-induced changes in the ECM alters the cell surface directly by deforming integrin-binding sites resulting in mechanobiological responses that depend greatly on the type of mechanical loading, the type of cell and the location where the stimulus was applied. For instance, the application of mechanical stretching on myofibroblasts grown in three-dimensional porous scaffolds induced the differentiation into smooth muscle cells with a consistent cellular alignment after a certain time course of cyclic strain (Cha et al. 2006). Similarly, physiological deformational loading applied to hydrogels seeded with chondrocytes.
(Hung et al. 2004) or embryonic mesenchymal progenitor cells (Terraciano et al. 2007) demonstrated functional cartilage formation in vitro, which had mechanical properties corresponding to those of the native tissue as well as increased gene expression and ECM formation. Consequently, the ability to manipulate and control cell adhesion and function through specific ECM-integrin interactions and mechanical stimulation suggest an alternative and economical way of guiding stem cell differentiation in vitro with potentially minimal deployment of expensive growth factors (Hayashi et al. 2007).

2.1.5. Bioreactors. The successful transfer of stem cell technology and cellular products into widespread clinical applications needs to address issues of cost, automation, standardization and generation of clinically relevant cell numbers of high quality. Laboratories and industry alike have dealt with similar problems in the past through the use of bioreactors. Consequently, stem cell bioprocessing will involve the use of these specialized devices that aim to facilitate mass transport, high cell density, monitoring and feedback and tissue-specific functional specialization, thus mimicking the ultimate bioreactors, which are the tissues/organs within the human body. An optimal and universal system for stem cell culture does not exist; however, bioreactor development throughout the last 40 years has advanced the technology considerably (table 2).

As previously discussed, adult stem cells are found in relatively low numbers while large numbers are usually required for clinical applications. This highlights the need for the in vitro expansion of stem cells prior to their commitment into tissue-specific applications. The potential of bioreactors to address this is demonstrated by their capacity to support high cell densities in relatively small volumes, while the scaling up of the design, given mass transfer limitations, will depend on the type of bioreactor chosen. Traditionally, culture of stem cells is performed on flat two-dimensional surfaces that are used to support the growth of the cells (Dexter et al. 1977). Well-plates, tissue-culture flasks (T-flasks; Mellado-Damas et al. 1999; Liu et al. 2006) and gas-permeable blood bags (Collins et al. 1998) are widely used in stem cell bioprocessing due to their simplicity, ease of handling and low cost, making them the ideal choice for research screening purposes as well as for engineering simple tissues, such as skin, bone and cartilage (Bilodeau & Mantovani 2006). However, the lack of online monitoring, limitations in scaling-up due to the limited surface area per volume, as well as their inability to support complex cellular growth configurations render two-dimensional surface-limited systems inadequate for biomanufacturing and clinical applications. Three-dimensional culture systems that would closely resemble the in vivo conditions by accounting for the cell–cell, cell–matrix and cell–growth factor interactions (Mantalaris et al. 2004b) are required in many clinically relevant cases. Consequently, a variety of matrices, such as nylon screens (Naughton et al. 1991) as well as other natural or synthetic scaffolds, as described earlier, were used to provide support for cellular growth, proving to be more efficient than their two-dimensional counterparts (Mantalaris et al. 2004b) and allow the development of three-dimensional constructs (Ott et al. 2008). However, three-dimensional cultures with their increased available surface area for cellular attachment and growth, higher cell density and the ability for higher cell expansion face increased mass transport limitations.

Static bioreactors (Sardinini & Wu 1993), in which the ‘ingredients’—cells, nutrients, metabolites, oxygen and other important molecules—experience mass transport that is exclusively through the process of diffusion, result in an inhomogeneous environment that can only support low cell densities and has a low total cell output (Pansonkalsis et al. 2005). To overcome the mass transport limitations of static cultures, bioreactors that can accommodate dynamic culture conditions are used. Primarily, perfusion and stirring have been the main means for enhancing mass transport. Stirred suspension bioreactors require careful impeller design to avoid high shear stress that can damage the cells (Zandstra et al. 1994), can be operated either in batch or continuous mode, and result in at least a 10-fold increase in cell density compared with the traditional methods. The scaling-up is usually straightforward due to the very good mass transport achieved by stirring. However, the flow environment created by the impeller renders them less suitable for the support of three-dimensional constructs (Nielsen 1999), although the use of porous microcarrier beads has been considered and studied (Zandstra et al. 1994). Several perfusion bioreactors have been designed to achieve a much lower shear stress environment as well as enhanced mass transport that facilitates the supply of nutrient and the removal of metabolites—perfusion rates having to be optimized based on cell type. However, different cell types have different sensitivities/necessities in terms of the shear stress, which is an important parameter in the design of bioreactors (King & Miller 2007). Shear stress is defined as the force exerted over the cells due to the flow of the media (Chen & Hu 2006), and a low rate has been described to result in cell clumping on aggregation supporting EB cultures (leading to lower mass transport to the cells; King & Miller 2007), while high rates could be deleterious for the cells. Thus, an optimal fluid velocity promoting the proper shear stress for the cell type being cultured is crucial. As an example, mammary epithelial stem cells aggregate cultures that have an optimal shear stress of 2 dyn. cm$^{-2}$ (King & Miller 2007), while endothelial cells can support shear stresses of the order of 20–30 dyn. cm$^{-2}$ (Sarkar et al. 2007). On the other hand, it has been described that the mechanical stimuli promoted this way can be beneficial for certain cell types: shear stresses of the order of 15 dyn. cm$^{-2}$ magnitude have promoted differentiation of ESCs towards the lineage of endothelial cells, when compared with static controls (Ahsan & Nerem 2006). Several bioreactors have been designed for promoting this scenario in vitro, by promoting controlled shear stress levels: using dynamic tension for growing and developing cardiomyocytes, MSCs, skeletal muscle and macrophages; compression for chondrocytes; and...
Table 2. Cell culture systems. (n.a. not available)

<table>
<thead>
<tr>
<th>bioreactor type</th>
<th>ease of harvesting</th>
<th>ease of monitoring</th>
<th>ease of scale-up</th>
<th>shear stress (dyn. cm(^{-2}))</th>
<th>mass transfer</th>
<th>culture surface area (cm(^2) l(^{-1}))</th>
<th>cell output (cells X 10(^{6}) ml(^{-1}))</th>
<th>operation</th>
<th>standard culture environment</th>
<th>three dimensional</th>
<th>two dimensional</th>
<th>references</th>
</tr>
</thead>
<tbody>
<tr>
<td>static culture (tissue culture flask)</td>
<td>high</td>
<td>low</td>
<td>low</td>
<td>0</td>
<td>low</td>
<td>290</td>
<td>0.1</td>
<td>yes</td>
<td>no</td>
<td>no</td>
<td>yes</td>
<td>Chaudhuri &amp; Al-Rubeai (2005) and Lemoli et al. (2005)</td>
</tr>
<tr>
<td>permeable blood bag (wave bioreactor)</td>
<td>high</td>
<td>medium</td>
<td>high</td>
<td>0.1–0.5</td>
<td>medium</td>
<td>n.a.</td>
<td>10–20</td>
<td>yes</td>
<td>yes</td>
<td>no</td>
<td>yes</td>
<td>Singh (1999) and Ohashi et al. (2001)</td>
</tr>
<tr>
<td>stirred (or in suspension)</td>
<td>high</td>
<td>high</td>
<td>high</td>
<td>2–40</td>
<td>high</td>
<td>2800</td>
<td>1–10</td>
<td>yes</td>
<td>yes</td>
<td>yes</td>
<td>yes</td>
<td>Jelinek et al. (2002), Chaudhuri &amp; Al-Rubeai (2005), Portner et al. (2005) and Goodwin et al. (2007)</td>
</tr>
<tr>
<td>airlift</td>
<td>medium</td>
<td>high</td>
<td>medium</td>
<td>10–30</td>
<td>high</td>
<td>2800</td>
<td>0.5</td>
<td>yes</td>
<td>yes</td>
<td>yes</td>
<td>yes</td>
<td>Merchuk &amp; Berzin (1995) and Cabral (2001)</td>
</tr>
<tr>
<td>packed bed (Bellocell)</td>
<td>medium</td>
<td>low</td>
<td>high</td>
<td>1–5</td>
<td>medium</td>
<td>18 000</td>
<td>1.5–200</td>
<td>no</td>
<td>yes</td>
<td>yes</td>
<td>yes</td>
<td>Perry &amp; Wang (1989), Jelinek et al. (2002), Chaudhuri &amp; Al-Rubeai (2005), Portner et al. (2005) and Meuwly et al. (2007)</td>
</tr>
<tr>
<td>fluidized bed</td>
<td>medium</td>
<td>low</td>
<td>high</td>
<td>3–6</td>
<td>medium</td>
<td>25 000–75 000</td>
<td>5–100</td>
<td>no</td>
<td>yes</td>
<td>yes</td>
<td>yes</td>
<td>Reiter et al. (1991), Nakhla &amp; Suidan (2002), Chaudhuri &amp; Al-Rubeai (2005) and Portner et al. (2005)</td>
</tr>
<tr>
<td>perfusion chamber (Aastrom Replicell)</td>
<td>medium</td>
<td>medium</td>
<td>medium</td>
<td>1–5</td>
<td>medium</td>
<td>18 000</td>
<td>10–100</td>
<td>no</td>
<td>yes</td>
<td>no</td>
<td>yes</td>
<td>Palmer et al. (1997) and Chaudhuri &amp; Al-Rubeai (2005)</td>
</tr>
<tr>
<td>grooved bioreactor</td>
<td>medium</td>
<td>medium</td>
<td>medium</td>
<td>0.1–0.5</td>
<td>medium</td>
<td>18 000–20 000</td>
<td>10–100</td>
<td>no</td>
<td>yes</td>
<td>no</td>
<td>yes</td>
<td>Horner et al. (1998) and Chaudhuri &amp; Al-Rubeai (2005)</td>
</tr>
</tbody>
</table>

(Continued.)
hydrodynamic pressure for bone cartilage (Korossi et al. 2005). Thus, optimal shear stress levels will depend largely on the cell type being grown/differentiated, as each type will have different sensitivities and/or stimulation needs (Palsson et al. 1998).

When low shear stress but high fluid velocities are required, a flat bed-grooved bioreactor can be used with cells growing within the grooves being protected from the flow. Such a system has been used for the expansion and maintenance of colony-forming units granulocyte–macrophage (CFU-GM), progenitor cells and long-term culture-initiating cells (LTC-IC) in the absence of stromal cells (Cabral 2001). Perfusion bioreactors also have the advantage of simple automation providing continuous and automated feeding of the cultures. Aastrom Biosciences, Inc., have developed a design whereby the cells are injected into a disposable cassette and grown on top of a previously established layer of stromal cells with nutrients being continually perfused to the cassette, while a chamber, located just above, is filled with oxygen that diffuses to the cassette through a liquid-impervious/gas-permeable membrane (Armstrong et al. 1996, 1999, 2000; Palsson et al. 1997). This system has been used to expand bone marrow mononuclear cells and umbilical CB cells for clinical applications. Several other bioreactor designs have been implemented with varying degrees of success. Hollow-fibre bioreactors can achieve a low shear stress environment with enhanced mass transport properties. They contain a number of hollow fibres, which are responsible for carrying nutrients and oxygen to the cells, by diffusing through the selective hollow-fibre membrane, thus avoiding the shear stresses caused by perfusion. At the same time, the inclusion of membrane technology in these designs greatly increases the surface area per volume available for cell growth (over 350 times that of a normal T-flask), allowing higher cell densities while still promoting efficient mass transfer of nutrients, oxygen and other important signalling molecules. Hollow-fibre bioreactors face certain limitations, such as decrease in mass transfer through the membranes due to cells growing in their periphery, especially in hollow-fibre bioreactors that support three-dimensional growth through the use of scaffolds (Yu et al. 2003). Other designs include: (i) packed or fluidized bed bioreactors (widely used for the expansion of hepatocytes, cardiocytes, osteoblasts and others; Portner et al. 2005), (ii) the rotating wall vessel (successful in the culture of HSCs, chondrocytes, cardiac cells, various tumour cells and others), which is a suspension culture adapted for lower shear stresses (Hammond & Hammond 2001; Liu et al. 2006), and (iii) the wave bioreactor, which uses wave agitation induced by a rocking motion to provide good nutrient distribution, off-bottom suspension and excellent oxygen transfer without damaging fluid shear or gas bubbles (Singh 1999; Ohashi et al. 2001). Unlike other cell culture systems, such as spinners, hollow-fibre bioreactors and roller bottles, scale-up is simple, and has been demonstrated up to 100 l of culture volume. The main differences in bioreactor designs can be associated with mass transport (addressed by diffusion, perfusion or bubbling), shear stresses (by developing

<table>
<thead>
<tr>
<th>bioreactor type</th>
<th>operation</th>
<th>ease of harvesting</th>
<th>ease of monitoring</th>
<th>ease of scale-up</th>
<th>shear stress (dyn. cm⁻²)</th>
<th>mass transfer culture surface area (cm² l⁻¹)</th>
<th>cell output (cells × 10⁶ ml⁻¹)</th>
<th>references</th>
</tr>
</thead>
<tbody>
<tr>
<td>perfusion hollow fibre (PluriX)</td>
<td>medium</td>
<td>low</td>
<td>low</td>
<td>medium</td>
<td>0.5-2</td>
<td>medium</td>
<td>10 000-200 000</td>
<td>100-200</td>
</tr>
<tr>
<td>rotating wall vessel (Synthecon)</td>
<td>medium</td>
<td>low</td>
<td>medium</td>
<td>low</td>
<td>0.5-2</td>
<td>medium</td>
<td>18 000-22 000</td>
<td>n.a.</td>
</tr>
</tbody>
</table>
ways to enhance mass transfer without increasing flow velocity or by which mechanical stimulation can be achieved), the ability to support three-dimensional constructs or even the end purpose (research versus large scale, cell type characteristics, etc.).

With respect to industry, automated cell culture allows faster and more accurate monitoring and analysis of cell cultures than comparable traditional methods. Events such as apoptosis, cell division, cellular movement and attachment can be continuously monitored and recorded for up to several weeks (Narkilahti et al. 2007). Terstegge and colleagues have recently described a system for semiautomated cell plating, media change, growth factor addition and cell harvesting in both human and mouse ESCs (Terstegge et al. 2007). Furthermore, Narkilahti and colleagues, through monitoring dynamic growth of hESCs in both automated and conventional culture systems, reported that automated cell culture and analysis provides the optimal tool for the evaluation of hESC culture, allowing continuous monitoring of living cells that would be impossible to discover by conventional methods (Narkilahti et al. 2007). Furthermore, the expansion and/or differentiation of ESCs in a variety of bioreactors have been investigated. Stirred and perfusion bioreactors maintain significant advantages over static culture due to the homogeneous environment they provide and the control over crucial culture parameters, such as oxygen and nutrient supply, pH and metabolite removal (King & Miller 2007). In general, perfusion, or frequent feeding, enhances culture performance by replacing exhausted nutrients and removing inhibitory metabolic by-products (King & Miller 2007).

For the expansion of haemopoietic progenitor cells (CFU-GM), continuous perfusion bioreactors and automated pH and DO controllers were able to maintain culture conditions within desired ranges while minimizing nutrient step changes and physical disruptions. Compared with static cultures, these systems demonstrated faster cell growth. In addition, the expansion of primitive haemopoietic progenitors (LTC-IC) was obtained only with synergistic cytokine combinations (e.g. IL-3/IL-6 and SCF) and perfusion (Koller et al. 1993a). Culture parameters, such as gas-phase oxygen concentration, seeding density and time of cell harvest were also found to play important roles in the expansion of various types of haemopoietic progenitors in perfusion bioreactor systems (Phalsson et al. 1993). To determine the feasibility of producing a large dose of haemopoietic cells through ex vivo expansion, Koller et al. scaled up their perfusion bioreactor 10-fold by employing 10 culture chambers in the system. Their results showed that more than three billion cells containing 12 million CFU-GM progenitors (the target number of CFU-GM routinely sought to support haemopoietic engraftment following autologous bone marrow transplantation is 15 million) were reproducibly generated from the equivalent of just 10–15 ml bone marrow aspirate and, more importantly, the number of primitive LTC-IC (stem cells) consistently increased with time in all cultures resulting in a 7.5-fold expansion. Consequently, this report represented the first published quantitative evidence of human stem cell expansion in an ex vivo bone marrow culture system, and such ex vivo expansion may have direct applications in clinical and experimental bone marrow transplantation (Koller et al. 1993b). As for the ex vivo expansion of PB mononucleor cells (PB MNCs), Sandstrom et al. (2005) investigated how CD34+ selection and/or perfusion affect the performance of PB MNC cultures. Their results indicated that while perfusion supported higher LTC-IC numbers for both MNC and CD34+ cell cultures, the selection of CD34+ cells was not required to obtain extensive CFU-GM expansion from PB. In fact, unselected MNC cultures produced 1.5-, 2.6- and 2.1-fold more total cells, CFU-GM and LTC-IC, respectively, than the same sample selected and cultured as CD34+ cells (Sandstrom et al. 1995). To date, ex vivo expansion of CB cells has demonstrated their promising expansion capability, far exceeding that of PB. After three weeks in culture, CB CD34+ cell preparations continued to proliferate with the average cell dividing more than five times with a retained engraftment potential. By contrast, PB CD34+ cell preparations did not show any continued growth after just a week in culture and had lost engraftment potential (Tanavde et al. 2002). Clinical trials have highlighted the potential improvements to engraftment through ex vivo expansion of CB stem cells (McNiece et al. 2000; Elizabeth et al. 2002). However, these studies relied on the expansion of cells in static culture systems; the more recent use of bioreactors, such as rotating wall vessels, has been trialled with considerably greater success with respect to the expansion of CB stem cells (Liu et al. 2006) and highlights the significant gains possible with the advancement of bioprocessing.

The complexity and fragmentation of ESC culture has directed several groups to demonstrate that mouse EBs can be formed directly from enzymatically dissociated mESCs in stirred bioreactors (Schroeder et al. 2005) or a rotary cell-culturing system (E et al. 2006). Mouse EBs can also be formed in stirred vessels by removing LIF from mESC aggregates produced in the same vessels (Fok & Zandstra 2005) or by encapsulating mESC aggregates in agarose beads (Bauwens et al. 2005). After formation, the EBs grew in size and retained the differentiation potential, such as cardiomyocyte differentiation (Bauwens et al. 2005; Schroeder et al. 2005). Tightly controlled mixing conditions have proven critical in these stirred suspension bioreactors in producing EBs of uniform size and quality. Furthermore, more recently, an integrated bioprocess for the culture, expansion and differentiation of mESCs has been demonstrated (Randle et al. 2007). In this system, encapsulated undifferentiated mESCs are cultured in rotary cell culture systems in a single-step process that does not require the formation of EBs or the disruption of cell aggregates, resulting in the formation of three-dimensional mineralized constructs (Randle et al. 2007). Differentiation of encapsulated ESCs has been used successfully for a wide range of tissue engineering applications and into several specific lineages. Dang et al. (2004) demonstrated the controllable and scalable culture of mouse and human ESCs.

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Table 3. Estimated costs of cell culture systems. [The start-up costs producing $10^9$ cells are determined based on the published cell outputs per volume (table 2) and their respective costs. n.a.: prices were not provided by the companies.]

<table>
<thead>
<tr>
<th>bioreactor type</th>
<th>bioreactor price per volume ($ ml^{-1}$)</th>
<th>start-up costs ($ per 10^9$cells)</th>
<th>automatable operation</th>
<th>company</th>
</tr>
</thead>
<tbody>
<tr>
<td>T-flask</td>
<td>0.15</td>
<td>1500</td>
<td>no</td>
<td>Corning</td>
</tr>
<tr>
<td>permeable blood bag (Wave bioreactor)</td>
<td>0.25</td>
<td>5</td>
<td>yes</td>
<td>GE Healthcare</td>
</tr>
<tr>
<td>stirred bioreactor (Cytostir)</td>
<td>0.3</td>
<td>30</td>
<td>yes</td>
<td>Kimble/Kontes</td>
</tr>
<tr>
<td>airlift (Cytolift)</td>
<td>0.9</td>
<td>1800</td>
<td>yes</td>
<td>Kimble/Kontes</td>
</tr>
<tr>
<td>packed bed (Belloccell)</td>
<td>3.0</td>
<td>60</td>
<td>yes</td>
<td>Cesco Bioengineering Co., Ltd.</td>
</tr>
<tr>
<td>fluidized bed (System 10)</td>
<td>n.a.</td>
<td>n.a.</td>
<td>yes</td>
<td>Aastrom Biosciences Inc.</td>
</tr>
<tr>
<td>perfusion chamber (Aastrom Replicell)</td>
<td>43</td>
<td>430</td>
<td>yes</td>
<td>Baxter International</td>
</tr>
<tr>
<td>grooved bioreactor</td>
<td>40–45</td>
<td>400–450</td>
<td>yes</td>
<td>FiberCell Systems, Inc.</td>
</tr>
<tr>
<td>perfusion hollow fibre (Fibercell)</td>
<td>3.3</td>
<td>16.5</td>
<td>yes</td>
<td>Synthecom</td>
</tr>
<tr>
<td>rotating wall vessel (Synthecom)</td>
<td>25</td>
<td>250</td>
<td>yes</td>
<td>Synthecom</td>
</tr>
</tbody>
</table>

encapsulated in agarose hydrogel capsules. Encapsulation permitted the use of high cell-density culture and enabled EB formation and differentiation to haemopoietic cells in a suspension bioreactor (Dang et al. 2004).

As already mentioned, the diversity of stem cell sources and their respective culture conditions means that no one bioreactor system is suitable for all stem cells. Therefore, a comparison of bioreactor suitability for their expansion and associated cost is somewhat futile. However, with respect to industry and commercialization, cost is a consideration of great importance. Table 3 examines current stem cell output examples from literature and determines an estimated cost in achieving a target cell number following expansion. There are several important considerations that should be noted in view of the estimated costs. First, substantial variability with respect to cell output exists among the different culture systems, which is a result of differing starting materials and protocols between laboratories. For example, the stirred bioreactor can be used for either the expansion of cells that grow in suspension, or adherent cells (with the addition of microcarriers or other appropriate substrates). This changes the surface/volumetric area that can be occupied by cells and, therefore, the obtainable outputs. Additionally, for some bioreactor systems, little work has been done with stem cell cultures, and the published outputs may only represent a fraction of what may be achievable with further development.

The ultimate bioreactors, namely the organs/tissues within the body, share common operational characteristics. Specifically, mass transport in the form of circulation and diffusion to the cells is excellent—no cell is located more than 400 μm away from blood supply (size, shape and structure being determined by function). Subsequently, the cell density supported is high (can reach billions of cells ml$^{-1}$) and the spatial arrangement of cells is critical for proper organogenesis. Monitoring is performed real time, online and in situ with signal processing and feedback being executed coherently. Additionally, these in vivo bioreactors are specialized, performing different functions in a cohesive and integrated manner. Hence, the optimal bioreactors achieve production not by embracing traditional scale-up principles (larger bioreactors) but through process intensification, modularity, specialization and integration. The use of bioreactors is critical for TERM applications for control, scale-up, automation and regulatory reasons. The laboratory practices of cell/tissue culture in dishes/flasks would have to be transformed, even in the discovery phase so that bioprocesses are developed and can be directly applied to the clinic. The variability in the results and the lack in standardization represent a current obstacle. Modular, integrated and interconnected culture systems that are fabricated from standard components and are amenable to automation are also required. Specifically, modules that integrate inoculation, culture, separation and harvesting need to be developed. Furthermore, specialized culture modules that are able to provide the required mechanical stimulation and to produce the final cellular product with desired features, such as three-dimensional structure, need to be developed. Certainly, a universal bioreactor does not exist and manufacturing should be directed towards modularity and integration. The lessons learned from the biopharmaceutical industry are valuable but, in the view of the authors, do not apply to scale-up in the traditional manner (Lim et al. 2007).

2.2. Process requirements

Ultimately, the integration of the various process components will be required in order to achieve a clinically relevant product through a regulated and controlled bioprocess that is reproducible, standardized, automatable (when needed), integrated and certified. Furthermore, process requirements will also be, by the nature of the problem, application specific.

2.2.1. Bioprocess monitoring and quality control. Stem cell culture complexity, heterogeneity of cell types and the inherent variability in process performance over time and between batches render the control in bioprocess culture systems a tremendously challenging task (Lim et al. 2007). However, bioprocess control in the manufacturing of biopharmaceutical products is critical in maintaining high product quality and consistency. Efforts are continuously being made in
this area to improve process monitoring and control techniques in complex bioprocesses. Specifically, the process analytical technology (PAT) initiative was established to promote better understanding and control of manufacturing bioprocesses through the use of process and end point monitoring tools, process control tools, multivariate data acquisition and analysis tools, process analytical chemistry tools and knowledge management tools (Junker & Wang 2006). The goal is not only to gain better insight into the bioprocesses of interest, but also to implement tight process control and reduce process variability. Integration of various engineering tools is therefore necessary to achieve these goals. With advances in sensor, optical and computer technology, stronger emphasis is being placed on the integration of online, real time, in situ monitoring systems. However, certain challenges in sensor technology still remain, such as the need for novel sensors for the detection of cytokines and cell density, overall sensitivity and stability of in situ biosensors, and sensor multiplexing capabilities (Clementschitsch & Bayer 2006). Other sophisticated measurement techniques, such as spectroscopy, though powerful and comprehensive, are often expensive and do not give direct measurements of process variables or provide insights into the cell’s metabolite state (Clementschitsch & Bayer 2006). Advancements in computer technology and the availability of sophisticated data management systems capable of processing large volumes of information have made it easier for the implementation of sophisticated control systems (Junker & Wang 2006). Much of this work relies on the use of multivariate data analysis and principal component analysis (PCA) to generate models for the detection of process abnormalities and control of process variations (Gunther et al. 2007). This wide array of multivariate techniques has provided the necessary tools for enhanced process understanding through the identification of critical sources of process variation and the generation of predictive control models. Consequently, continued emphasis on improving and generating novel measurement technologies, along with the required product quality verification, will ultimately facilitate bioprocess control that generates precise, reliable and high-quality products for clinical applications.

2.2.2. Design of experiments. Design of experiments (DOE) is a powerful engineering tool mainly used for process characterization and optimization that has been widely applied in the materials, chemical, pharmaceutical and semiconductor industries. It is a systematic methodology for investigating a process using minimum effort to obtain the maximum information (Montgomery 2001). Process investigations performed via DOE are, therefore, highly efficient and informative. Specifically, one of the distinct advantages in using DOE is the ability of the design to reveal both individualistic and interactive effects for a process using the minimum number of experimental runs, reducing time and cost, and, more importantly, providing an accurate and quantitative characterization of the process of interest. Traditional experimentation methods, such as dose–response studies, often do not paint a ‘true’ picture of the story as they only examine changes due to one factor at a time, while other corresponding process factors remain unaltered, thus masking interactions. When studying a complex system, a systematic approach is required to obtain useful information and determine optimal operating conditions (Lim et al. 2007). Typically, this would involve the following three steps: screening; characterization; and optimization, using the appropriate design(s) in each stage. The objective process screening is to rapidly identify factors that have a significant influence on the process and correctly discriminate against factors that have little or no influence at all. Some of the commonly used designs for such investigations are the fractional factorial design and Plackett–Burman design (Montgomery 2001; Myers & Montgomery 2002), which enable rapid screening of a large number of factors using a very small number of experiments; in the Plackett–Burman design, for example, a 12-run experiment can screen up to 11 factors. In process characterization, the goal is to obtain a more detailed and quantitative description about the process by way of three-dimensional surface response plots. Common designs include the central composite design (CCD) and Box–Behnken design (Montgomery 2001; Myers & Montgomery 2002), which reveal process relationships that can be described using a quadratic or cubic model. In process optimization, the generation of three-dimensional plots generates optimal operating regimes and yields the best conditions for process optimization. The outcome of this systematic framework is a reproducible and statistically valid elucidation of bioprocesses that do not require a priori assumptions for the process of interest. To date, the use of DOE in stem cell bioprocessing has been somewhat limited; much of the research still uses traditional dose–response methods. However, a handful of investigators have been quite successful in using DOE for stem cell culture studies. These include the use of fractional factorial designs to perform screening experiments (Yao et al. 2003, 2004), and the use of central-composite or full factorial designs to perform process characterization (Zandstra et al. 1997a,b; Cortin et al. 2005). However, full process characterization, which considers all the process factors including physiochemical parameters, nutrient, metabolite and growth factor concentrations, has not yet been established. Owing to the complexity of these interactions and lack of sensor technology to monitor them simultaneously in culture, the investigation of these factors in combination is not straightforward. The successful application of DOE methodologies and monitoring techniques to resolve the complexity in stem cell bioprocessing will not only yield invaluable information for stem cell cultures but also provide a novel approach for other cell culture studies.

2.2.3. Bioprocess modelling. As elaborated earlier, stem cell bioprocessing will require the design and integration of different processes and systems that would be
application specific. Consequently, designing such application-specific systems or scaling up of a system to produce the necessary cell numbers requires knowledge of the momentum and mass transport characteristics of the system(s) under various operating conditions, which will also be useful for the subsequent successful operation of the system (Williams et al. 2002). Such modelling can be implemented by employing computational fluid dynamic (CFD) software packages and will facilitate the optimization of culture system specifications and operating conditions, such as porosity and perfusion flow rate, so as to achieve the best possible conditions for cell growth (Begley & Kleis 2000). A number of mathematical models have been developed to study the fluid dynamics and nutrient distribution in perfusion bioreactors (Horner et al. 1998; Begley & Kleis 2000, 2002; Williams et al. 2002; Pathi et al. 2005; Coletti et al. 2006), as well as haemopoietic cell growth dynamics (Ching-An Peng 1996; Hevehan et al. 2000c; McNiece & Bridell 2001; da Silva et al. 2003). For instance, oxygen supply in bioreactors was addressed by the comparison of convective and diffusive nutrient supply (Pathi et al. 2005). Bioreactor design operating parameters, such as the perfusion rate, liquid depth and length of culture, have been investigated to determine their effect on long-term cell culture, thus enabling optimal bioreactor design. More recently, the simulation of a realistic multilineage in vitro HSC expansion culture has also been presented (Ma et al. 2007). A culture period of 14 days yielded an increase in progenitor cells (CFUGMs) similar to experimental data, matching the spatial variation of oxygen concentrations representative of the multiple cell lineages in bone marrow in vivo to the oxygen distribution within the bioreactor sufficient to support the 14-day expansion of HSCs (Mukhopadhyay et al. 2004). These ‘realistic’ stem cell culture simulations provide relevant insights into understanding the stem cell expansion while also reducing experimental cost. Ultimately, modelling can be used to provide the guidelines for ‘designer tissues’ engineering in vitro.

2.2.4. Product/process characterization—proteomic and genomic analyses. Genomic technologies, such as microarray chromatin immunoprecipitation (ChIP-chip) and ChIP sequencing, allow for the total characterization of transcription factors and other DNA-bound proteins in a high-throughput and cost-effective fashion. Comparative genomic hybridization (CGH) can determine total genomic chromosomal losses or gain with respect to a control and test genome. Such technologies are powerful tools to help elucidate the molecular mechanisms that regulate the formation, self-renewal and differentiation of stem cells. The influence of genomics in stem cell research has been eloquently demonstrated with the induced expression of four important transcription factors. OCT4, SOX2, NANOG and LIN28 were shown to be sufficient to reprogramme mouse fibroblasts to undifferentiated, pluripotent stem cells (Takahashi & Yamanaka 2006; Maherali et al. 2007; Okita et al. 2007; Wernig et al. 2007). Subsequently, the same factors have been shown to have a comparable effect on human somatic cells, creating cells that meet all the criteria of ESCs with the exception of not being derived from embryos (Yu et al. 2007). Furthermore, comparative genomics not only helps elucidate important transcription factors during the growth and differentiation of stem cells, by observing changes in the transcriptome of the cell, but can also be used to determine the effect of drugs and growth factors on cells they are exposed to.

Similarly, proteomics is a tremendously important tool allowing proteome mapping, differential analyses and elucidation of signals and mechanisms, thus enabling the understanding of the complex biological processes and protein-regulated signalling pathways that constitute basic embryonic development and stem cell differentiation (Elliott et al. 2004; Nagano et al. 2006; Salasznyk et al. 2005; Baharvand et al. 2006; Hoffrogge et al. 2006; Van Hoof et al. 2006). Specifically, the proteome dataset of the mESC line, E14-1, has identified 1790 proteins, including nuclear proteins (Nagano et al. 2005). Similarly, the proteome profiles of three hESC lines are also being analysed (Baharvand et al. 2006), which will result in the identification of proteins involved in protein synthesis, processing and destination, probably reflecting the ability of hESCs to remain undifferentiated or to rapidly differentiate. Proteome maps for the other stem cell lines have also been established, including hippocampal neural stem cells (Maurer et al. 2003), human adipose-derived stem cells (DeLany et al. 2005) and human umbilical CB MSCs (Feldmann et al. 2005). Proteomics has facilitated the elucidation of the complex environment provided by feeder cells (Lim & Bodnar 2002; Prowse et al. 2005) leading to the development of feeder-free, defined culture conditions, which are essential for the clinical applications of cellular products. It has identified key proteins involved in the maintenance of hESC pluripotency including Wnt, BMP/TGF-β, activin/inhibin and insulin-like growth factor-1. Furthermore, proteomics is a powerful differential analysis technique. Several studies have compared differentiated and undifferentiated stem cells identifying candidate regulators of differentiation (Guo et al. 2001; DeLany et al. 2005; Wang & Gao 2005; Puente et al. 2006). Some of the proteins identified were exclusively expressed in undifferentiated ESCs suggesting possible use as ESC biomarkers, which will further facilitate standardization. Finally, stem cell regulation is also greatly influenced by culture parameters and signals, as discussed earlier. Proteomics can be used to characterize an environment that supports maintenance of undifferentiated stem cells and to help identify factors critical for their differentiation (Baharvand et al. 2007; Sze et al. 2007). In addition, more recently developed proteomic equipment enables us to optimize and control the quality of the manufacturing process for biopharmaceutical products. This can be usefully employed to monitor the bioprocess of stem cells for drug discovery and therapeutic applications in the future.
Complete integration of modular, automated and controlled systems in a fully enclosed bioprocess operation from harvest to delivery will need to be considered. Ultimately, scale-up of stem cell/tissue-engineered bioprocesses, in the view of the authors, can be achieved by small-scale modular systems operating in an ‘in-series and in-parallel’ mode where overcapacity is considered and the whole process is addressed as a supply chain model. Scale-up can be delivered by lower infrastructure cost systems where the integration, modularity and parallel operation are the keys to the problem.

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