CD25+ CD4+ T Cells Regulate the Expansion of Peripheral CD4 T Cells Through the Production of IL-10

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CD25\(^+\) CD4\(^+\) T Cells Regulate the Expansion of Peripheral CD4 T Cells Through the Production of IL-10\(^1\)

Oliver Annacker,\(^2\) Ricardo Pimenta-Araujo, Odile Burlen-Defranoux, Theolis C. Barbosa,\(^3\) Ana Cumano, and Antonio Bandeira

The mechanisms by which the immune system achieves constant T cell numbers throughout life, thereby controlling autoaggressive cell expansions, are to date not completely understood. Here, we show that the CD25\(^+\) subpopulation of naturally activated (CD45RB\(^{low}\)) CD4 T cells, but not CD25\(^-\) CD45RB\(^{low}\) CD4 T cells, inhibits the accumulation of cotransferred CD45RB\(^{high}\) CD4 T cells in lymphocyte-deficient mice. However, both CD25\(^+\) and CD25\(^-\) CD45RB\(^{low}\) CD4 T cell subpopulations contain regulatory cells, since they can prevent naive CD4 T cell-induced wasting disease. In the absence of a correlation between disease and the number of recovered CD4\(^+\) cells, we conclude that expansion control and disease prevention are largely independent processes. CD25\(^+\) CD45RB\(^{low}\) CD4 T cells from IL-10-deficient mice do not protect from disease. They accumulate to a higher cell number and cannot prevent the expansion of CD45RB\(^{high}\) CD4 T cells upon transfer compared with their wild-type counterparts. Although CD25\(^-\) CD45RB\(^{low}\) CD4 T cells are capable of expanding when transferred in vivo, they reach a homeostatic equilibrium at lower cell numbers than CD25\(^-\) CD45RB\(^{low}\) or CD45RB\(^{high}\) CD4 T cells. We conclude that CD25\(^+\) CD45RB\(^{low}\) CD4 T cells from nonmanipulated mice control the number of peripheral CD4 T cells through a mechanism involving the production of IL-10 by regulatory T cells. The Journal of Immunology, 2001, 166: 3008–3018.

The regulation of the magnitude of protective immunity to foreign Ags as well as the control of autoaggressive immune reactions are ensured by regulatory T cells. Regulatory CD4 T cells have been described in a variety of experimental systems to protect from autoimmune diseases (1–6) as well as from inflammatory bowel disease (IBD)\(^4\) (7, 8) and allograft rejection (9). Furthermore, regulatory CD4 T cells play a key role in the homeostasis of the peripheral CD4 T cell pool (10).

Useful surface markers for the discrimination between functional subsets of CD4 T cells are CD25 and CD45RB. CD25 is a component of the IL-2R and is transiently expressed on CD4 T cells after activation (11), and CD25\(^-\) T cells make up approximately 10% of the peripheral CD4 T cell pool. Transfer of CD25-depleted splenic cells into nude mice of susceptible strains leads to the development of organ-specific autoimmune diseases, which can be prevented by the cotransfer of purified CD25\(^+\) CD4 T cells (2, 3). Moreover, the lack of CD25\(^+\) cells allows for efficient clearance of tumors, demonstrating the active suppression of anti-self immune responses by regulatory T cells (12). CD25\(^-\) CD4 T cells were found to produce high levels of TGF-\(\beta\) and IL-10 compared with CD25\(^-\) CD4 T cells (3). Recently, it was shown that the protective effect of CD25\(^+\) CD4 T cells is dependent on CTLA-4 (13).

CD45RB is another marker of activation used for the discrimination of different CD4 T cell subsets. This surface molecule is up-regulated during thymic development (14), and its expression on naive CD4 T cells decreases upon activation (15). According to this marker, roughly one-third of CD4 T cells are activated in unmanipulated mice.

In an experimental system of IBD several groups showed that after transfer into immunodeficient recipients, naive CD45RB\(^{high}\) CD4 T cells cause a wasting disease characterized by intestinal inflammation (7, 8). High levels of IFN-\(\gamma\) and TNF-\(\alpha\) are found in both the spleen and the intestine of the recipients (8, 16–18), and injected T cells can expand at least 200-fold under specific pathogen-free (SPF) conditions (10). In contrast, naturally activated CD45RB\(^{low}\) CD4 T cells usually do not induce disease, expand less, and protect the recipients from naive T cell-induced IBD (7, 8, 10, 16, 17). This protective effect is dependent on TGF-\(\beta\) and IL-10 (17, 19–22). The CD45RB\(^{low}\) CD4 T cell pool contains most of the CD25\(^+\) CD4 T cells; the latter contribute one-third to the pool of CD45RB\(^{low}\) CD4 T cells. In a recent study Read et al. showed that the protection from IBD in this experimental system is enriched in the CD25\(^+\) T cell population within the CD45RB\(^{low}\) CD4 T cell pool and is also CTLA-4 dependent (23).

The general mechanisms regulating the expansion and survival of peripheral CD4 T cells are to date not well understood (for review, see Ref. 24). The peripheral T cell pool is divided into a naive and an activated/memory compartment, which are apparently independently regulated (25, 26). The size of the activated/memory CD4 T cell pool is controlled by regulatory T cells within this pool (10). The administration of recombinant murine IL-10 protected recipients reconstituted with CD45RB\(^{high}\) CD4 T cells from disease and decreased the number of recovered splenic CD4
T cells (17), suggesting a role for IL-10 in the regulation of the expansion of peripheral CD4 T cells.

Here, we investigated the survival, the dynamics of expansion, and the homeostatic equilibrium of different peripheral CD4 T cell subpopulations from normal donors upon transfer into T and B cell-deficient mice, their regulatory properties, and the role of IL-10 in the expansion process. Our results show that although both CD25+ and CD25−CD45RBloow CD4 T cell pools contain regulatory T cells, only the CD25+ population can efficiently regulate the size of the activated/memory CD4 T cell compartment via a mechanism involving the production of IL-10. Furthermore, our results show that control of CD4 T cell peripheral expansion and disease prevention are largely independent processes. Finally, the data demonstrate that although the CD25+ CD4 T cell population reaches a homeostatic equilibrium at low cell numbers, a fraction of these cells has a high potential of expansion upon transfer into lymphoid recipients.

Materials and Methods

Mice

C57BL/6-Ly5.2 mice were obtained from Janvier (Le Genest-St-Irle, France). C57BL/6-Ly5.1, C57BL/6 recombination-activating gene-2-deficient (RAG-2−) and C57.BA (Thy1.1) mice were purchased from CDTA (Orleans, France). All animals were kept under SPF conditions in the animal facilities of the Institut Pasteur (Paris, France), C57BL/6-IL-10-deficient (IL-10−) mice were purchased from The Jackson Laboratory (Bar Harbor, ME) and were bred under SPF conditions in our animal facilities. Donors and recipients were sex-matched and used at 6–12 wk of age. IL-10− donors were used at 5–8 wk of age.

Antibodies

The following mAbs were used: anti-Ly5.1-FITC or -PE (clone A20); anti-CD45RB, -FITC, -PE, -TriColor, -allophycocyanin (L3T4); anti-CD8-FITC or -PE (CT-CD8a); anti-CD45RB-PE (23G2); anti-CD25-FITC (7D4) or -PE (PC61); anti-CD38-FITC (90); anti-CD69-PE (H1.2F3); anti-CD44-PE (IM7.8.1); and anti-Thy1.1-PE (Ox-7). All Abs were purchased from PharMingen (San Diego, CA) or Caltag (Burlingame, CA).

T cell preparations

Before sorting, splenic single-cell suspensions were first enriched for CD4+ or CD25+ cells by positive selection on midIMACS columns (Miltenyi Biotec, Bergisch-Gladbach, Germany) according to the manufacturer’s instructions. In brief, cells were first incubated with biotylinated anti-CD4 Abs for 20 min on ice in PBS supplemented with 0.5% FCS, then incubated in the same buffer with streptavidin-microbeads for 15 min. The magnetically labeled positive fraction was retained on a midIMACS column. Alternatively, for the enrichment of CD25+ cells, FITC-labeled anti-CD25 Abs and anti-FITC-microbeads were used. In all cases, after enrichment the cells were washed three times in PBS and were then sorted on a FACStarFlow (Becton Dickson, Mountain View, CA). For CD45RB, the brightest 40–50% and the dimnest 20–30% of CD4+ cells were sorted as high and low, respectively. CD25+ and CD25−CD4 T cells were sorted out of the CD45RBlow population. The purity of the sorted populations was routinely >96%.

Labeling with CFSE

Labeling of cells with CFSE was performed as previously described (27). In brief, before washing FACS-sorted CD4 T cells twice in PBS, the cells were resuspended at 1–2×10⁶/mL PBS and incubated 10 min at room temperature with CFSE at a final concentration of 6 μM. This solution was incubated with an equal volume of FCS for an additional 2 min before washing twice with PBS.

Cell transfers

RAG-2− mice were injected iv. or i.p. with 3×10⁵ CD4 T cells from FACS-sorted subpopulations. When cells were coinjected (at a ratio of 1:1), Ly5.1+ and Ly5.2+ donor cells were used. For CFSE-labeled cells, 0.5–1.5×10⁶ CD4 T cells from Ly5.1+ origin were injected i.v. Here, coinjections were made with Ly5.1+ and Thy1.1+ donors at a ratio of 1:1.

Preparation of intestinal cells

Whole intestines were first flushed extensively to eliminate the lumen content, then were longitudinally opened and cut into 1- to 2-cm pieces. These were incubated twice in OptiMEM medium (Life Technologies, Gaithersburg, MD) containing 5% FCS and 450 U of collagenase (Sigma, St. Louis, MO) for 20 min at 37°C. After filtering through gauze, lymphoid cells were isolated on a 40% Percoll gradient. The cells were then washed and stained for fluorocytometric analysis.

Flow cytometric analysis

Single-cell suspensions from spleen; axillary, inguinal, and mesenteric lymph nodes; or intestine were incubated for 20 min at 4°C in microtiter plates with 50 μL of the appropriate Ab preparations in PBS supplemented with 3% FCS and 0.01% azide. When possible, one million cells were stained. Alternatively, the whole organ cell suspension was used. The Ab concentrations used were tested for optimal stainings of splenic control samples before experimental use. Dead cells were excluded from the analysis by propidium iodide. Blood samples were first stained with appropriate Abs before lysing erythrocytes with FACS Lysing Solution (Becton Dickson). Flow cytometric analysis was performed on a FACScan (Becton Dickinson) using CellQuest software (Becton Dickinson).

Statistical analysis

Unless otherwise indicated, analysis was performed using the unpaired t test. In cases where the variances between compared groups were significantly different, the unpaired t test was modified with Welch’s correction. The data were considered significantly different at p < 0.05.

Results

The incidence of CD4 T cell-induced wasting in alymphoid mice correlates with the frequency of CD25+ cells in the transferred population

Splenic CD4 T cells from normal unmanipulated mice were sorted into four subpopulations according to the expression of CD45RB and CD25 markers: 1) CD45RBhigh cells, hereafter denoted RBhigh; 2) CD45RBlow cells (of which about one-third is CD25+), denoted RBlow; 3) CD45RBlow cells, which were depleted of CD25+ cells, denoted 25−RBlow; and 4) CD45RBlow cells expressing the CD25 marker, hereafter referred to as 25+RBlow CD4 T cells (Fig. 1A).

Syngenic RAG-2− mice, kept under SPF conditions, received 3×10⁵ of either sorted CD4 T cell subset. The weight of the recipients was scored twice a week, and animals were sacrificed after 12–14 wk or when they lost at least 20% of their initial weight.

Mice injected with (naive) RBhigh T cells invariably developed signs of wasting. Only one of eight animals survived for 12 wk after transfer (Fig. 1B), and all recipients lost weight (Fig. 1C), developed diarrhea, and had a markedly enlarged colon upon analysis. Noninjected control RAG-2− mice kept under the same conditions never developed signs of wasting or diarrhea (data not shown). In the group of animals that received the total pool of RBlow cells, one of seven mice became sick (Fig. 1C) and was sacrificed 8 wk after transfer (Fig. 1B). Half of the recipients of the 25−RBlow pool remained healthy throughout the experimental period of 3.5 mo (Fig. 1C). The other half suffered from wasting, but the disease progressed more slowly compared with that in recipients of RBhigh T cells, and only two of them lost >20% of their initial body weight within 12 wk after transfer (Fig. 1B). The only experimental group in which all recipients invariably gained weight and did not develop signs of wasting was the one injected with 25+RBlow T cells (Fig. 1C). Thus, the incidence of a wasting disease in alymphoid recipients after transfer of different CD4 T cell populations appears to correlate with the frequency of CD25+ cells in the transferred population.
Peripheral expansion of CD4 T cell subsets: CD25\(^{+}\)CD45RB\(\text{low}\) CD4 T cells reach homeostatic equilibrium at low cell numbers

To assess the accumulation and the respective homeostatic equilibrium of the injected T cell populations, at the time of sacrifice, the number of CD4\(^{+}\) cells was scored in the spleen; axillary, inguinal, and mesenteric lymph nodes; as well as blood and intestine of all the recipients described in the previous section. In this series of transfers, on the average, 2.9 \(\times 10^6\) CD4 T cells were recovered from mice injected with RB\(^{\text{high}}\) T cells (Fig. 1D). In animals injected with the total pool of naturally activated/memory RB\(^{\text{low}}\) T cells, we could only score half the number of cells found in the previous group (on the average, 1.4 \(\times 10^6\); \(p < 0.03\), as shown previously (10). Interestingly, the number of cells (on the average, 2.9 \(\times 10^6\)) obtained from mice reconstituted with 25\(^{-}\) RB\(^{\text{low}}\) T cells was similar to that scored in recipients of RB\(^{\text{high}}\) T cells regardless of the state of health of the recipients (Fig. 1D) and the time point of sacrifice. Finally, recipients of 25\(^{-}\) RB\(^{\text{low}}\) T cells only yielded approximately the number of cells injected (on the average, 3.9 \(\times 10^5\)), that is, 7-fold less compared with the 25\(^{-}\) RB\(^{\text{low}}\) (\(p < 0.0001\)) or the RB\(^{\text{high}}\) (\(p < 0.001\)) population and about 4-fold less compared with recipients of unfraccionated RB\(^{\text{low}}\) T cells (\(p < 0.01\); Fig. 1D).

The organ distribution of the CD4\(^{+}\) cells in all groups of mice is shown in Table I. The majority of T cells were found in the spleen, accounting for roughly half of the recovered CD4 T cells. In the intestine, with the exception of recipients of the 25\(^{-}\) RB\(^{\text{low}}\) fraction, similar numbers of CD4 T cells were recovered in both healthy and sick animals in all other groups (Table I). However, it cannot be formally excluded that the observed differences in cell numbers between different CD4 T cell subsets are due to different expansions of these cells, but to a differential migration pattern predominantly into other organs, such as liver, lung, or bone marrow, which have not been investigated here.

In conclusion, disease is directly correlated neither with the total number of T cells recovered from the recipients nor with the number of T cells present in the intestine. In addition, the 25\(^{-}\) RB\(^{\text{low}}\) T cell population accumulates to low cell numbers after transfer into RAG-2\(^{-}\) hosts and is barely detectable in the intestine.

Both CD25\(^{+}\) and CD25\(^{-}\) CD45RB\(^{\text{low}}\) CD4 T cells contain regulatory cells capable of preventing a wasting disease induced by naive CD4 T cells

In the experiments described above (see Fig. 1), half of the recipients of the 25\(^{-}\) RB\(^{\text{low}}\) T cell population became sick, whereas the other half remained healthy for at least 3 mo. This differential...
outcome could be the result of differences in the frequency of potentially pathogenic or, alternatively, of regulatory T cells in the individual inoculums of this CD4 T cell subset.

To directly assess the presence of regulatory activity in the two CD45RB low subpopulations, RAG-2° recipients were coinjected with $3 \times 10^5$ RB high T cells and $3 \times 10^5$ CD4 T cells of either the 25$^+$ or 25$^-$ RB low T cell subset (Fig. 2A). The majority of the animals injected with these mixtures were protected from disease (Fig. 2B), and a similar fraction of animals in both groups developed a wasting disease with similar kinetics (Fig. 2C). In conclusion, the CD25$^-$ CD45RB low T cell subpopulation contained sufficient regulatory activity to prevent CD4 T cell-induced wasting in 40% of the recipients.

CD25$^+$ CD45RB low CD4 T cells control the size of the activated/memory CD4 T cell pool

The cell recovery from recipients of the total RB low T cell pool was significantly different from that of either 25$^+$ or 25$^-$ subfractions (see Fig. 1). This could be interpreted to indicate that the restricted expansion of total RB low T cells after transfer into RAG-2° hosts was due to control of the accumulation of 25$^+$ RB low T cells exerted by 25$^-$ RB low T cells. To investigate whether the

### Table I. Organ distribution of CD4$^+$ cells in RAG-2° mice reconstituted with different CD4 subpopulations

<table>
<thead>
<tr>
<th>Phenotype of Injected CD4$^+$ Subpopulations</th>
<th>No. of Animals</th>
<th>No. of Recovered CD4$^+$ Cells ($\times 10^4$ $\pm$ SEM)</th>
</tr>
</thead>
<tbody>
<tr>
<td>CD45RB low</td>
<td>7</td>
<td>Spleen: 75.7 ($\pm$ 21.5)</td>
</tr>
<tr>
<td>CD25 CD45RB low</td>
<td>8</td>
<td>Spleen: 140.3 ($\pm$ 26.7)</td>
</tr>
<tr>
<td>CD25$^+$ CD45RB low</td>
<td>6</td>
<td>Spleen: 20.9 ($\pm$ 4.4)</td>
</tr>
<tr>
<td>CD45RB high</td>
<td>8</td>
<td>Spleen: 105.1 ($\pm$ 33.4)</td>
</tr>
</tbody>
</table>

a RAG-2° recipients were injected with $3 \times 10^5$ FACS-sorted CD4 T cells. The animals were analyzed for the presence of CD4$^+$ cells in the indicated organs after 12–14 wk or when they dropped below 80% of their starting weight.

b Average ± SEM.

c Axillary, inguinal, and mesenteric lymph nodes.

d Data are expressed as cell number per milliliter of blood.

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**FIGURE 2.** CD25$^+$, but not CD25$^-$ CD45RB low, CD4 T cells inhibit the accumulation of CD45RB high CD4 T cells transferred into RAG-2° hosts. Sorted CD25$^+$ CD45RB low (CD25$^+$) or CD25$^+$ CD45RB low (CD25$^+$) CD4 T cells ($3 \times 10^5$) were coinjected with $3 \times 10^5$ CD45RB high CD4 T cells into RAG-2° hosts. A. FACS profiles of the coinjected populations. B. Recipients were sacrificed at 12–14 wk after transfer or when they dropped below 80% of their starting weight as indicated by the survival curve. C. Weight of the recipients at sacrifice (time points of sacrifice as indicated in B). Sick animals were defined as described in Fig. 1. D. Sum of the total CD4$^+$ cell numbers scored in spleen; axillary, inguinal, and mesenteric lymph nodes; blood (assuming 3 ml of blood per animal); and intestine. The mean ($\pm$ SEM) is shown. *, The difference is statistically significant ($p < 0.04$, by unpaired t test with Welch’s correction). E. CD4$^+$ cell number for each donor population in the two groups of recipients. CD4$^+$ cells from different origins were identified by the expression of different Ly5 isoforms. The data are pooled from three independent experiments. The difference between recovered CD45RB high T cell numbers in the two groups is statistically significant ($p < 0.05$, by unpaired t test with Welch’s correction; see also text).
CD25+ T cell pool is indeed responsible for control of the size of the activated/memory CD4 T cell compartment, we analyzed the coinjected recipients (see Fig. 2) for the level of T cell reconstitution either 12–14 wk after transfer or when the recipients dropped to <80% of their initial weight. The identification of the origin of each donor population was based on the expression of the Ly5.1 and Ly5.2 markers.

The total CD4 T cell recovery from mice injected with mixed 25+ RBlow and RBhigh T cells was >4-fold higher compared with that in animals that received mixed 25+ RBlow and RBhigh cells (p < 0.04, by unpaired t test with Welch’s correction; Fig. 2D). In the group cotransferred with 25+ RBlow and RBhigh T cells both populations expanded to similar numbers as those scored in animals injected with either population alone (Figs. 2E and 1D), with a similar distribution of each subset in all organs (Table II). Again, no significant difference was observed between healthy and sick animals (Fig. 2E).

In the group of recipients injected with 25+ RBlow and RBhigh T cells, similar numbers of 25+ RBlow T cells were recovered as from animals injected with these cells alone regardless of the presence of RBhigh T cells in the injected animals (Fig. 2E). However, approximately 4-fold less T cells of originally RBhigh T cells were scored in these animals compared with recipients of RBhigh T cells injected alone (p < 0.004) or coinjected with 25+ RBlow T cells (p < 0.05, by unpaired t test with Welch’s correction). T cell numbers were mostly reduced in spleen, blood, and intestine (Table II). Interestingly, the relative frequency of originally 25+ RBlow to RBhigh T cells in the intestine was 1:10 to 1:20, since 25+ RBlow T cells were again barely detected in the intestine (Table II). This ratio was independent of the health state of the mice, and the presence of 25+ RBlow T cells did not inhibit the activation of the original RBhigh T cells, as assessed by the low density of the CD45RB marker (Fig. 3).

Taken together, these results show that the 25+, but not the 25-, RBlow T cell population has the potential to efficiently inhibit the accumulation of other CD4 T cells. Here again, no direct correlation was found between the magnitude of T cell accumulation and the incidence of wasting disease in any of the experimental groups.

**CD25+ CD45RBlow CD4 T cells can expand in vivo**

CD25+ T cells do not expand upon stimulation with anti-CD3 Abs in vitro (28, 29), which was interpreted to reflect an anergic state of these cells (28). The reconstitution of RAG-2− mice with 25+ RBlow cells yielded approximately the number of cells injected (see above). However, because in transfer experiments only a minority of the injected cells survives in the host 24–48 h after transfer (10), this suggested that the CD25+ T cell population could nevertheless expand to a certain extent in the recipients.

To more accurately address this issue, CD4 T cells from normal Ly5.1+ donors were sorted into three subpopulations according to the expression of CD25 and CD45RB and were labeled with CFSE. Then, 0.5−1.5 × 106 cells of each subset were separately injected into congenic Ly5.2− RAG-2− hosts. Thirty-six to 48 h after transfer, donor-derived CD4 T cells were analyzed in the peripheral lymphoid organs, the blood, and the gut. While no donor cells were recovered from the intestine, the bulk of the populations were found in the spleen. Naive RBhigh T cells survived much better than naturally activated 25+ or 25+ RBlow T cells (Fig. 4C). On the average 2.6% of the RBhigh T cell population could be recovered at this early time point, whereas only 1.1 and 0.3% of injected 25+ and 25+ RBlow T cells, respectively, were recovered. At this time point, very few of the injected cells had divided, as assessed by CFSE staining (data not shown).

In the following days all populations expanded, and 12 days after transfer very few cells from any of the injected populations remained CFSE positive, indicating that in all cases the bulk of the cells present at this time point went through a minimum of six rounds of division. As shown in Fig. 4C, 4- and 6.8-fold the number of injected cells were recovered from recipients of 25− RBlow populations.

<table>
<thead>
<tr>
<th>Conjoined Populations</th>
<th>No. of Animals</th>
<th>Phenotype of Injected CD4+ Subpopulations</th>
<th>No. of Recovered CD4+ Cells (×10^6)</th>
</tr>
</thead>
<tbody>
<tr>
<td>CD25−</td>
<td>10</td>
<td>CD25−</td>
<td>139.0 (±68.1)</td>
</tr>
<tr>
<td>+ CD45RBhigh</td>
<td></td>
<td>CD45RBhigh</td>
<td>129.0 (±54.5)</td>
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<td></td>
<td></td>
<td></td>
<td>16.7 (±2.7)</td>
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<tr>
<td></td>
<td></td>
<td></td>
<td>32.1 (±5.4)</td>
</tr>
<tr>
<td>CD25+</td>
<td>6</td>
<td>CD25+</td>
<td>28.2 (±7.7)</td>
</tr>
<tr>
<td>+ CD45RBhigh</td>
<td></td>
<td>CD45RBhigh</td>
<td>18.3 (±4.2)</td>
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<td></td>
<td></td>
<td></td>
<td>8.5 (±1.9)</td>
</tr>
<tr>
<td></td>
<td></td>
<td></td>
<td>16.4 (±2.9)</td>
</tr>
</tbody>
</table>

* RAG-2− recipients were injected with 3 × 10^6 FAC-stained CD4 T cells of either population as indicated. Differentiation of the origin of the cells was achieved by using Ly5-congenic donors. The animals were analyzed for the presence of CD4+ cells in the indicated organs after 12–14 wk or when they dropped below 80% of their starting weight.

* Average ± SEM.

* Axillary, inguinal, and mesenteric lymph nodes.

* Data are expressed as cell number per milliliter of blood.
and of RB high T cells, respectively. In contrast, recipients of 25^1 RB low T cells contained 20% of the number of injected cells. However, taking into account the differential survival of the injected populations in the three groups of recipients, the increase in cell numbers over the surviving populations on day 2 was 60-fold for the 25^1 RB low T cell population (corresponding to five rounds of division of the surviving T cells), compared with 360- and 260-fold for the 25^2 RB low and the RB high T cell populations (equivalent to seven or eight rounds of division), respectively (Fig. 4C).

In conclusion, these results show that a fraction of 25^1 RB low cells has an expansion capacity not very different from that of the 25^2 RB low or RB high T cells. The lower reconstitution capacity of 25^1 cells 3.5 mo after transfer probably reflects a difference in the homeostatic regulation of steady state numbers in these populations.

CD25^+ CD45RB^low CD4 T cells regulate the size of the CD45RB^high CD4 T cell compartment early after transfer

In these series of experiments CFSE-labeled 25^+ RB^low T cells contained 20% of the number of injected cells. However, taking into account the differential survival of the injected populations in the three groups of recipients, the increase in cell numbers over the surviving populations on day 2 was 60-fold for the 25^+ RB^low T cell population (corresponding to five rounds of division of the surviving T cells), compared with 360- and 260-fold for the 25^+ RB^low and the RB^high T cell populations (equivalent to seven or eight rounds of division), respectively (Fig. 4C).

In conclusion, these results show that a fraction of 25^+ RB^low T cells has an expansion capacity not very different from that of the 25^+ RB^low or RB^high T cells. The lower reconstitution capacity of 25^+ cells 3.5 mo after transfer probably reflects a difference in the homeostatic regulation of steady state numbers in these populations.
cells constitutively express this molecule (29, 31). Since T cells express high levels of CD25 upon activation (11), it was also suggested that CD25⁺ T cells are continuously activated in vivo (32). Here we analyzed whether the reconstitution of the CD25 compartment is dependent on the origin of the injected cells.

At sacrifice, the peripheral lymphoid organs of RAG-2° animals injected with different CD4 T cell subsets, as shown in Figs. 1 and 2, were analyzed for the expression of CD25 on the recovered CD4 T cells as well. Interestingly, the frequency of lymph node T cells that stained positively for CD25 was similar in the recipients of 25⁺ and 25⁻ RBlow cells (Fig. 5). In the spleen, the frequency of CD25⁺ cells was 13.9% (SEM = 3.0; n = 8) within the group that received 25⁻ RBlow cells compared with 19.4% (±1.1; n = 4) in the recipients of 25⁻ RBlow cells. The values observed were independent of the state of health of the mice. In contrast, transfers of naive RBhigh cells reconstituted the CD25 compartment to a lower extent (in the spleen 5.6% (±2.0); n = 5).

To assess whether the expression of CD25 on originally CD25⁺ cells is autonomously regulated within the population or requires the presence of other T cells in the recipient, we also analyzed the recipients coinjected with mixed CD4 T cell subsets, described in Fig. 2, for expression of the CD25 marker. Originally RBhigh T cells showed similar frequencies of CD25-expressing cells in mesenteric lymph nodes regardless of the presence of other T cells in the injected recipients (5.8% (±1.2; n = 7) in the presence of 25⁺ RBlow cells and 8.5% (±3.0; n = 6) in the presence of 25⁻ RBlow T cells). The frequency of CD25-expressing cells in the originally 25⁻ RBlow population was modestly increased by the presence of RBhigh cells in mesenteric lymph nodes, but was within the same range as in animals injected with 25⁻ RBlow cells alone (25⁻ RBlow alone, 18.9% (±2.0; n = 8); 25⁻ RBlow in coinjections, 22.5% (±3.2; n = 7); Fig. 5). In the presence of RBhigh cells, the donor CD25⁺ population consisted mostly of CD25⁺ cells in the mesenteric lymph nodes (70.3% (±3.6; n = 6) compared with 21.9% (±4.1; n = 4) in recipients of CD4 T cells alone). These findings were similar in spleen and pooled axillary and inguinal lymph nodes.

We conclude that for the majority of transferred CD25⁺ CD4 T cells, the expression of the CD25 molecule requires the presence of other CD4 T cells. In addition, the 25⁻ RBlow population can generate higher frequencies of CD25⁺ cells than RBhigh T cells upon transfer.

CD25⁺ CD45RBlow CD4 T cells from IL-10° mice cannot efficiently regulate peripheral CD4 T cell numbers

Administration of rIL-10 leads to decreased numbers of splenic T cells recovered from mice injected with RBhigh cells (17), and IL-10° mice develop, apart from IBD, splenomegaly (33). Given these data, we hypothesized that IL-10 plays a role not only in the protection from disease, but also in regulation of the expansion of CD4 T cells. Earlier studies have shown that IL-10° mice at 6 wk of age contain normal numbers of thymocytes and splenic T cells (19). To confirm and extend these findings, we analyzed the thymus, spleen, lymph nodes, blood, and intestine of 6-wk-old IL-10° and wild-type (wt) mice for CD4 and CD8 T cells. Indeed, IL-10° T cells were indistinguishable from wt mice with regard to numbers and expression of CD45RB, CD25, CD38, CD69, and CD44, including the presence of CD25⁺ CD4⁺ CD8⁻ thymocytes (data not shown), suggesting that the development of both CD4 and CD8 T cells is not strongly affected by the lack of IL-10.

To address the question of whether IL-10 is necessary for efficient control of the size of the activated T cell pool, we performed the same transfer experiments described above with donor cells from healthy IL-10° mice bred onto the C57BL/6 background. Transfer of 3 × 10⁵ sorted RBhigh or 25⁻ RBlow cells from IL-10° donors induced wasting in all RAG-2° recipients, suggesting that the regulatory T cells in the 25⁻ RBlow population are IL-10 dependent. The wasting in mice injected with IL-10° 25⁻ RBlow cells developed somewhat faster than that in hosts of IL-10° RBhigh T cells (Fig. 6A). Both populations expanded to a similar level (Figs. 6C and 1D), with a comparable organ distribution of the recovered T cells (Table III) as the corresponding populations from wt animals.
IL-10°CD25+CD45RBlow CD4 T cells do not efficiently regulate peripheral CD4 T cell numbers. C57BL/6 RAG-2° recipients were the CD45RB low population. Five of seven animals remained healthy for 12–14 wk (Fig. 6C and 1D). Interestingly, the expression of CD25 as well as the organ distribution of the cojected populations were very similar to those in the recipients of wt T cells (data not shown and Table III).

As shown above, regulation of the peripheral expansion of CD4 T cells in RAG-2° hosts (Fig. 6, A and B). The numbers of cells recovered from mice coinjected with IL-10° 25+ CD4 T cells and wt CD45RBhigh cells were not significantly different from those recovered from recipients of either population alone (Figs. 6C and 1D). Our data showing the capacity of the CD25+CD4 T cells to the prevention of wasting, do not contribute significantly to the regulation of CD4 T cell homeostasis.

Our results also provide the first description of the population dynamics of CD25+ T cells upon in vivo transfer. They establish that although CD25+ T cells reach a homeostatic equilibrium at low cell numbers, a fraction of these cells has a high potential of expansion. In contrast, CD25− CD45RBlow CD4 T cells, which also prevent wasting, do not contribute significantly to the regulation of CD4 T cell homeostasis.

Development of regulatory CD4 T cells protect from autoimmune diseases and IBD. Regulatory T cells prevent T cell-induced wasting and control peripheral CD4 T cell homeostasis. We show, first, that CD25+ CD45RBlow CD4 T cells contain potentially harmful T cells in the CD45RBlow population (19, 35, 36). Our data showing the capacity of the CD25− CD4 T cells to prevent wasting, do not contribute significantly to the regulation of CD4 T cell homeostasis.

We conclude that control of CD4 T cell peripheral expansion and disease prevention are largely independent processes. Furthermore, we show that the mechanism underlying the regulation of the size of the peripheral T cell compartment is IL-10 dependent. In the present study we characterize two different subpopulations of CD4 T cells in their capacity to both prevent T cell-induced wasting disease and control peripheral CD4 T cell homeostasis.

In the group of mice transferred with 3 × 10^5 IL-10° 25+ RBlow cells, five of seven animals remained healthy for 12–14 wk (Fig. 6B). These mice, on the average, 1.1 × 10^6 CD4 T cells could be recovered, representing 3-fold more than from animals that received wt 25+ T cells (p < 0.02; Figs. 6C and 1D). This was largely due to increased cell numbers in spleen and blood of the recipients (Table III), and IL-10° 25+ RBlow cells were found in the intestine only in very low numbers. The difference in cell recovery was not due to an increased survival of IL-10°CD4 T cells, since similar numbers were obtained 2 days after transfer into RAG-2° recipients compared with the corresponding wt population (data not shown). Furthermore, the frequency of CD25+ expressing cells among the recovered IL-10°CD4 T cells was for all populations indistinguishable from that of the corresponding wt CD4 T cells (data not shown).

Consistent with previous results (22), when 3 × 10^5 IL-10° 25+ RBlow cells were injected together with 3 × 10^5 wt RBhigh cells, the IL-10°CD25+ T cells could not prevent wasting in the RAG-2° hosts (Fig. 6, A and B). The numbers of cells recovered from mice coinjected with IL-10° 25+ RBlow T cells and wt RBhigh cells were not significantly different from those recovered from recipients of either population alone (Figs. 6C and 1D). Interestingly, the expression of CD25 as well as the organ distribution of the cojected populations were very similar to those in the recipients of wt T cells (data not shown and Table III).

As shown above, regulation of the peripheral expansion of RBhigh by 25+ RBlow cells was already effective by 12 days after transfer. We ascertained the lack of regulatory activity of IL-10° T cells by coinjecting normal CD25+ cells with IL-10°CD25+ T cells at different ratios into RAG-2° recipients. As shown in Fig. 7, IL-10°, but not wt CD25+ cells showed a complete absence of regulatory effect on the expansion of CD25-depleted CD4 T cells. Even at a ratio of six IL-10°CD25+ T cells to one wt CD25+ cells, no signs of inhibitory activity were detected. Taken together, the data demonstrate that 25+ RBlow cells from IL-10° mice are able to control the accumulation of peripheral CD4 T cells. Moreover, it was shown previously that normal animals contain potentially harmful T cells in the CD45RBlow population (10). Thus, although it is possible that IL-10° animals contain higher frequencies of potentially disease-inducing activated CD4 T cells, the data allow the possibility that the disease inhibitory activity of 25− RBlow CD4 T cells is also IL-10 dependent.

Discussion

Regulatory T cells protect from autoimmune diseases and IBD. Also, T cells contained in the CD45RBlow CD4 population control the size of the peripheral CD4 T cell compartment (10). In the present study we characterize two different subpopulations of CD4 T cells in their capacity to both prevent T cell-induced wasting disease and control peripheral CD4 T cell homeostasis. We show, first, that CD25+ CD45RBlow CD4 T cells, which prevent the onset of wasting, contribute to the regulation of peripheral T cell numbers. In contrast, CD25− CD45RBlow CD4 T cells, which also prevent wasting, do not contribute significantly to the regulation of CD4 T cell homeostasis.
CD45RB<sub>low</sub> CD4 T cells to protect from wasting disease are well in line with a recent report (23) in which a similar reduction in the incidence of CD4 T cell-induced IBD was observed at a comparable CD25<sup>−/+</sup> T cell ratio.

This observation raises the question of the relationship between CD25<sup>+</sup> and CD25<sup>−</sup> regulatory CD4 T cells. At this point we cannot exclude that the two subsets differ in their development, function, and/or specificity, although this is not very likely. First, the expression of CD25 on different CD4 T cell subsets is not stable after transfer into alymphoid hosts, and in the case of CD25<sup>+</sup> cells it is dependent on the presence of other CD4 T cells. It is thus possible that in normal animals CD25 expression is dynamic and, therefore, the marker is not identifying the entire pool of regulatory T cells. Secondly, both CD25<sup>+</sup> and CD25<sup>−</sup> regulatory T cells depend on IL-10 for efficient disease protection. Furthermore, CD25<sup>−</sup> regulatory T cells do not compensate for a block of the function of CD25<sup>+</sup> regulatory T cells when the protective activity of total CD45RB<sub>low</sub> CD4 T cells is inhibited by anti-TGF-β or anti-CTLA-4 Ab treatment (20, 23).

The dissociation between protection of disease and systemic (and local) regulation of CD4 T cell numbers observed in our studies indicates that both processes are to a large extent independently regulated. This may be the result of a quantitative difference in the number of regulatory CD4 T cells required to control both processes. Disease protection may require lower numbers of regulatory T cells and/or rely on the presence of appropriate TCR specificities in the pool of regulatory T cells. Thus, it can be effective even in the absence of efficient growth control. The lack of appropriate specificities would also explain why the CD25<sup>+</sup> CD4 population did not confer protection from wasting in some cases, while showing a quite efficient growth inhibitory activity on other CD4 T cells in the sick recipients. Similar observations were described in other systems: tolerance can be ensured when T cells expand (37), and differentiation can take place in the absence of overt proliferation of T cells (38, 39).

Taken together, it is possible that regulatory CD25<sup>−</sup> T cells are descendants of thymic regulatory CD25<sup>+</sup> T cells (34, 35) and represent an alternative state of the same functional pool of peripheral regulatory T cells. CD25<sup>−</sup> CD4 T cells might be enriched for regulatory T cells simply because they are activated (effector state), but regulatory T cells might become CD25<sup>−</sup> T cells in the absence of the appropriate stimuli (memory state).

In contrast to our previous report (10), we could now recover sizable numbers of CD4 T cells from the intestines of healthy recipients. Moreover, similar numbers of intestinal T cells were observed between healthy and sick recipients within the same experimental group. However, this appears to represent a different organ distribution of the cells rather than a higher level of T cell expansion, because the differences in total cell numbers from recipients of CD45RB<sub>low</sub> T cells compared with recipients of CD45RB<sub>high</sub> T cells was in this study very similar to what we reported previously. This argues for a systemic regulation of peripheral CD4 T cell numbers and not for a compartmentalized control in individual organs. The increased frequency of intestinal CD4 T cells reported here could perhaps reflect a subclinical state of inflammation in these overall healthy mice due to an unbalanced ratio of regulatory to target CD4 T cells. Nevertheless, a >3-fold reduction in the number of intestinal T cells belonging to the transferred CD45RB<sub>high</sub> CD4 T cell population was observed in conjunction with the CD25<sup>−</sup> CD45RB<sub>low</sub> T cells. This is consistent with our previous report, namely that regulatory CD4 T cells inhibit the accumulation of CD4 T cells in the intestine. The observation that similar T cell numbers are scored in the intestines of sick and healthy animals reinforces the conclusion that T cell expansion and incidence of disease are not directly linked.

<table>
<thead>
<tr>
<th>Coinjected Populations</th>
<th>Phenotype of Injected CD4&lt;sup&gt;+&lt;/sup&gt; Subpopulations</th>
<th>No. of Animals</th>
<th>Spleen</th>
<th>Lymph Nodes&lt;sup&gt;a&lt;/sup&gt;</th>
<th>Blood&lt;sup&gt;b&lt;/sup&gt;</th>
<th>Intestine</th>
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<tr>
<td>IL-10&lt;sup&gt;−&lt;/sup&gt; CD25&lt;sup&gt;+&lt;/sup&gt; CD45RB&lt;sub&gt;low&lt;/sub&gt;</td>
<td>7</td>
<td>138.0 ± (49.6)</td>
<td>24.5 ± (6.3)</td>
<td>27.4 ± (9.8)</td>
<td>79.6 ± (36.2)</td>
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<tr>
<td>IL-10&lt;sup&gt;−&lt;/sup&gt; CD25&lt;sup&gt;+&lt;/sup&gt; CD45RB&lt;sub&gt;low&lt;/sub&gt;</td>
<td>7</td>
<td>78.0 ± (23.5)</td>
<td>8.8 ± (2.5)</td>
<td>8.1 ± (2.2)</td>
<td>1.5 ± (0.3)</td>
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<tr>
<td>IL-10&lt;sup&gt;−&lt;/sup&gt; CD45RB&lt;sub&gt;high&lt;/sub&gt;</td>
<td>7</td>
<td>102.2 ± (13.5)</td>
<td>39.7 ± (8.3)</td>
<td>34.4 ± (4.7)</td>
<td>60.3 ± (14.1)&lt;sup&gt;f&lt;/sup&gt;</td>
<td></td>
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<tr>
<td>IL-10&lt;sup&gt;−&lt;/sup&gt; CD25&lt;sup&gt;+&lt;/sup&gt; + wt CD45RB&lt;sub&gt;high&lt;/sub&gt;</td>
<td>7</td>
<td>41.0 ± (12.8)</td>
<td>19.8 ± (6.3)</td>
<td>1.3 ± (0.2)</td>
<td>1.9 ± (1.4)</td>
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<tr>
<td>wt CD45RB&lt;sub&gt;high&lt;/sub&gt;</td>
<td>7</td>
<td>85.3 ± (36.8)</td>
<td>30.4 ± (7.3)</td>
<td>20.9 ± (5.4)</td>
<td>48.1 ± (21.7)</td>
<td></td>
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</table>

* RAG-2° recipients were injected with 3 × 10<sup>5</sup> FACS-sorted CD4 T cells from IL-10<sup>−</sup> donors. In addition, 3 × 10<sup>5</sup> IL-10<sup>−</sup> CD25<sup>−</sup> CD4 T cells were also injected with 3 × 10<sup>5</sup> wt CD45RB<sub>high</sub> CD4 T cells. Differentiation of the origin of the cells was achieved by using Ly5<sup>−</sup>-congenic donors. The animals were analyzed for the presence of CD4<sup>+</sup> cells in the indicated organs after 12–14 wk or when they dropped below 80% of their starting weight.

<sup>a</sup> Average ± SEM.
<sup>b</sup> Axillary, inguinal, and mesenteric lymph nodes.
<sup>c</sup> n = 6; one animal, excluded from these data, contained more than 2000 × 10<sup>4</sup> CD4<sup>+</sup> cells in the intestine. Including this animal results in an average CD4<sup>+</sup> cell number of 352.1 (±29.2) × 10<sup>4</sup> in the intestine in this group.

FIGURE 7. High numbers of IL-10<sup>−</sup>CD25<sup>+</sup> CD4 T cells do not show growth inhibitory activity. CD4 T cells from wt or IL-10<sup>−</sup> donors were separated by two consecutive rounds of magnetic bead purification into CD25<sup>+</sup> and CD25<sup>−</sup> subsets. CD25<sup>−</sup> CD4 T cells (9 × 10<sup>5</sup>; <1% CD25<sup>+</sup> cells) from wt animals were then injected into RAG-2° hosts, either alone or together with wt CD25<sup>−</sup> (<93% pure) or IL-10<sup>−</sup>CD25<sup>+</sup> CD4 T cells (<92% pure) at the indicated ratios. Eleven days after transfer the recipients were analyzed for CD4<sup>+</sup> cells in spleen; axillary, inguinal, and mesenteric lymph nodes; blood (assuming 3 ml of blood per animal); and intestine. The resulting numbers are expressed as a percentage of the injected cell numbers. The symbols show individual mice from the same experiment. ■, wt CD25<sup>−</sup> CD4 T cells injected alone; ▲, CD25<sup>−</sup> CD4 T cells injected with the indicated CD25<sup>+</sup> CD4 T cell population. Differentiation of the origin of the cells was achieved using Thy1.1 and/or Ly5.1 congenic markers.
Our studies also assessed the proliferative potential and the homeostatic equilibrium of peripheral CD25+ CD4 T cells. The idea has been that regulatory T cells have a limited capacity of expansion, perhaps as a result of their own growth inhibitory activity. This is in line with the inability of these cells to proliferate in vitro upon stimulation unless exogenous IL-2 is added. Here we provide evidence that a fraction of CD25+ CD45RBlow cells is capable of considerable in vivo proliferation despite the fact that the population reaches a homeostatic equilibrium at low cell numbers. The present data do not provide information on the rate of apoptosis occurring after each round of division, but extensive apoptosis during the expansion process will only increase the number of cell divisions required to account for the observed cell numbers.

The reasons why the homeostatic equilibrium of CD25+ CD45RBlow T cells is reached at low cell numbers are nevertheless unclear at this point. It could be that these cells are driven and/or regulated by different growth factors or have limited functional niches compared with the other CD4 T cell populations.

Asseman et al. (22) demonstrated that the IBD protective function of regulatory CD4 T cells is IL-10 dependent. The lack of efficient growth inhibitory activity of CD25+ T cells from IL-10 mice reveals a role for this IL in peripheral T cell homeostasis. In the results presented here CD25+ T cells from IL-10 mice showed many characteristics of wt CD25+ CD4 T cells, and most recipients of IL-10/CD25+ T cells remained healthy, although this subset contained potentially aggressive T cells. This suggests that the CD25+ pool of IL-10/CD4 T cells, although not homogenous, is highly enriched for cells of the regulatory lineage, which, in the absence of IL-10, have a higher potential of expansion.

Other groups reported a linkage between the susceptibility to autoimmune diseases and the balance between IL-12 and IL-10 as well as a role for IL-12 in CD4 T cell expansion (40, 41). It is thus possible to envisage that IL-10 produced by regulatory T cells leads to down-regulation of IL-12 production by APC, resulting in decreased levels of IL-2 and, in turn, restricted CD4 T cell expansion. Our data show that regulatory CD25+ T cells prevent extensive T cell activation and do not seem to interfere significantly with the activation of naive T cells in the recipient.

The onset of IBD and splenomegaly in IL-10 mice occurs relatively late in life compared with other situations in which deregulation of peripheral T cell homeostasis is already apparent 3–4 wk after birth. This strongly suggests that factors other than IL-10 are involved in the regulation of peripheral T cell numbers. Indeed, spontaneous autoimmune disease and disruption of T cell homeostasis were recently described in mice transgenic for a T cell-targeted dominant negative TGF-β receptor (42, 43). It is worth pointing out, however, that whatever the cellular interactions or mechanisms that delay the development of disease in IL-10 mice, they are disrupted in the transfer experiments presented here. Thus, further studies are needed to dissect the dependence of T cell homeostasis from cytokines produced by regulatory T cells.

Acknowledgments

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